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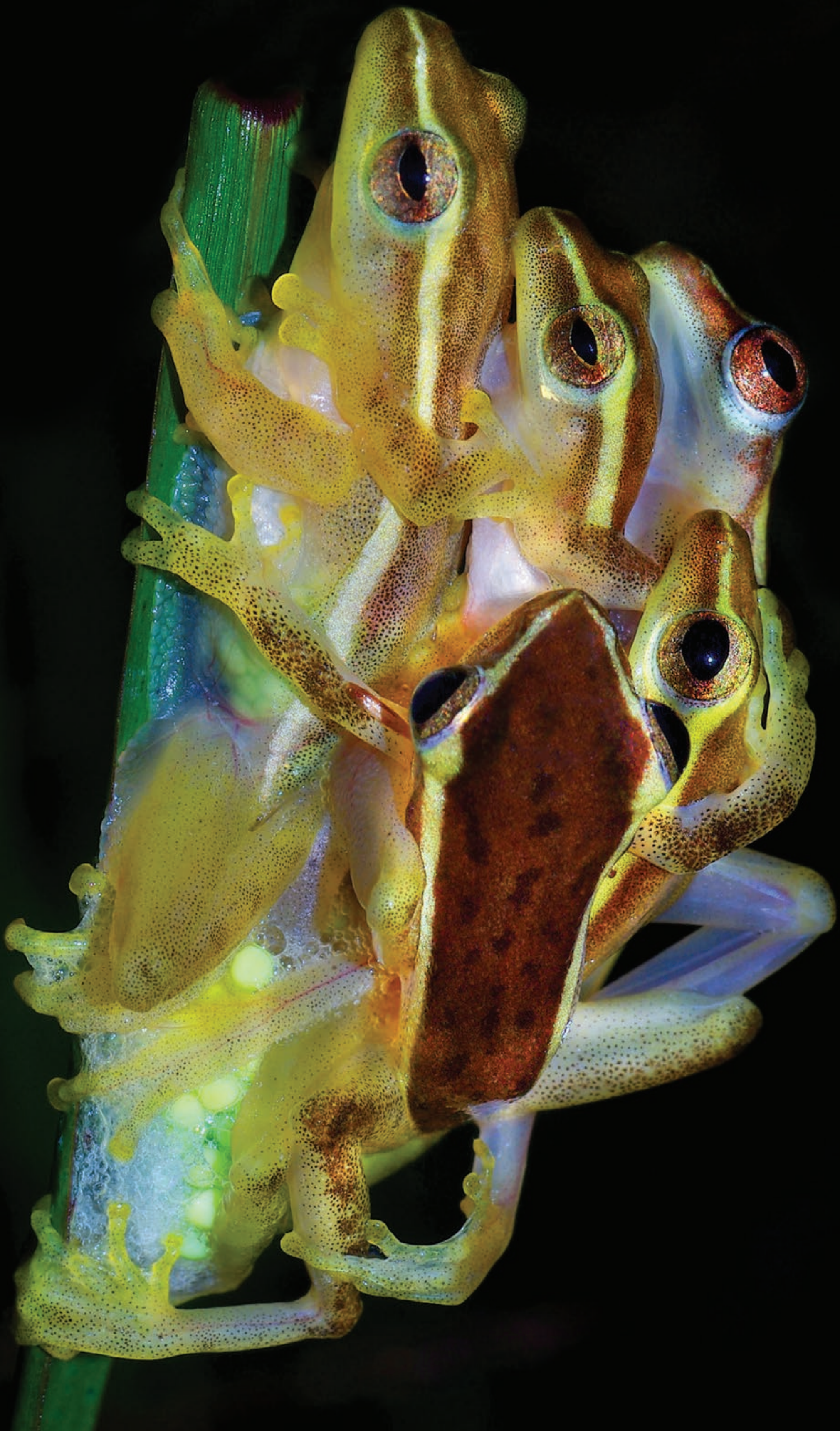

HERPETOLOGICAL REVIEW

THE QUARTERLY BULLETIN OF THE

SOCIETY FOR THE STUDY OF AMPHIBIANS AND REPTILES

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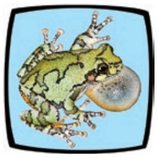

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Ecological, Morphological, and Reproductive Aspects of a Diverse Assemblage of Hyperoliid Frogs (Family: Hyperoliidae) Surrounding Mt. Kupe, Cameroon

Detailed natural history information is lacking for a large number of African amphibians, and observations on general aspects of ecology and reproductive biology of species have often been made in the context of regional amphibian summaries (Wager 1965; Schiøtz 1967; Wager 1986; Schiøtz 1999; Channing 2001; Channing and Howell 2006; du Preez and Carruthers 2009; Amiet 2012) and embedded in species accounts supplied in survey reports. Hyperoliidae is the largest frog family in Sub-Saharan Africa with more than 230 species occurring in 17 genera (AmphibiaWeb 2018), however a majority of these species occurs within two genera: *Hyperolius* Rapp, 1842 (150+ species) and *Afrixalus* Laurent, 1944 (30+ species). Focused studies on hyperoliid reproductive behaviors have contributed greatly to our broader understanding of anuran mate-choice and sexual selection, though these are restricted to the South African *H. marmoratus* species complex (Dyson and Passmore 1988; Telford and Dyson 1988; Telford et al. 1989; Dyson et al. 1992; Bishop et al. 1995; Docherty et al. 1995; Jennions et al. 1995a, 1995b; Polakow et al. 1995; Grafe 1996, 1997; Dyson et al. 1998; Docherty et al. 2000; but see Starnberger et al. 2018). Studies of West African savannah-dwelling *Hyperolius* have revealed novel ecological and reproductive adaptations related to survival in highly seasonal environments (Linsenmair 1998; Lampert and Linsenmair 2002; Rödel et al. 2006). Despite this progress, the general ecology and reproductive biology of most hyperoliids remains poorly known (but see Amiet 2012; Kouamé et al. 2015), and still less information is available pertaining to community ecology and species interactions (Telford and Passmore 1981; Lötters et al. 2004; Ernst and Rödel 2008; Lawson and Moyer 2008). A growing number of field surveys coupled with DNA-barcoding methods have improved our ability to distinguish African amphibian species and accumulate relevant data (e.g., Rockney et al. 2015; Portik et al. 2016; Deichmann et al. 2017), yet reports on natural history are still scarce.

Approximately 50 hyperoliid species are known from Cameroon (Schiøtz 1967, 1999; Amiet 2012), including 29 species of *Hyperolius* and 8 species of *Afrixalus*. A recent survey in the low elevation forest surrounding Mt. Kupe revealed exceptional hyperoliid diversity, and twelve species were documented and identified based on morphological (body size, coloration) and genetic (16S mtDNA) evidence, including *A. dorsalis*, *A. laevis*, *A. paradorsalis*, *H. bolifambae*, *H. camerunensis*, *H. concolor*, *H. dintelmanni*, *H. fusciventris*, *H. ocellatus*, *H.* sp., *Kassina decorata*, and *Phlyctimantis leonardi* (Portik et al. 2016). When combined with results of previous surveys, a total of 18 species are known from Mt. Kupe, making it one of the most species-rich locations for hyperoliid frogs (Amiet 1975; Hofer et al. 1999; Schmitz et al. 1999; Portik et al. 2016). The presence of seven species of *Hyperolius* and three species of *Afrixalus* in syntopy make this region particularly appealing for studying species interactions, spatial and temporal partitioning of reproductive resources, and community ecology. Natural history information remains scarce for many of these species, and therefore we characterize the habitat use, breeding behavior, and reproductive biology for this diverse hyperoliid community, which supplements the species accounts provided by Portik et al. (2016). We also generate sex-specific body size distributions for all taxa to quantify the levels of sexual size dimorphism and the degree of body size overlap among species in the community. Our goal is to provide baseline natural history information that can be used to guide future ecological and evolutionary biology studies.

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Fig. 1. Sampling locality next to banana plantation consisting of water-filled drainages.

Materials and Methods

Field Surveys.—Surveys at multiple sites in the foothills of Mt. Kupe, accessed through the nearby town of Manjo, were conducted 11–14 July 2013 and 24–29 September 2014, with details provided in Portik et al. (2016). Hyperoliid frogs were found at a variety of sites, including permanent ponds in semi-disturbed forest, fish ponds near a large stream, small streams in secondary forest, and anthropogenic habitats such as drainage ditches. During nocturnal visual searches, specimens were hand-captured and natural history observations were made. Sex was determined by the presence of male secondary sexual characters including the vocal sac and gular gland. Individuals found in amplexus were collected together and isolated from others to allow identification of particular male and female pairs. Captured animals were euthanized using MS-222, tissue samples were preserved in RNA Later (Ambion, Inc.), and whole specimens were preserved with 10% buffered formalin.

Body Size Comparisons.—Measurements of snout–urostyle length (SUL) were taken for 244 preserved specimens from Mt. Kupe representing 10 species: *A. dorsalis* (25 males, 5 females), *A. laevis* (3 males, 1 female), *A. paradorsalis* (19 males, 5 females), *H. bolifambae* (25 males, 8 females), *H. camerunensis* (13 males, 6 females), *H. concolor* (45 males, 14 females), *H. dintelmanni* (3 males, 4 females), *H. fusciventris* (26 males, 11 females), *H. ocellatus* (11 males, 11 females), and *H.* sp. (12 males). All measurements were made by DMP with a Mitutoyo Series 500 Digimatic Caliper (Mitutoyo U.S.A., Illinois) and recorded to the nearest 0.1 mm. We calculated a sexual size dimorphism index (SSDi) to quantify the level of sexual size dimorphism across species, where SSDi = ((larger sex / smaller sex) – 1), set negative if males are the larger sex and positive if females are the larger sex (Lovich and Gibbons 1992). This particular SSDi has been widely used, is properly scaled and symmetric around zero, and has high intuitive value because positive values indicate female-biased dimorphism and negative values indicate male-biased dimorphism (Lovich and Gibbons 1992). We tested for homogeneity of variance of body size distributions using the Fligner-Killeen test and examined potential differences in the mean body size across species using a one-way ANOVA, for males and females separately. We performed these analyses first by pooling all species of *Afrixalus* and *Hyperolius*, and subsequently for the genus *Hyperolius* independently, to examine potential differences within and between genera. For significant one-way ANOVA tests, we performed post-hoc Tukey multiple pairwise-comparisons to identify each species pair that displayed a statistically significant difference in mean body size. Analyses were carried out using the ‘fligner.test’, ‘aov’ and ‘TukeyHSD’ functions in R (R Core Team 2015).

Fig. 2. Sampling locality consisting of a fish pond with little emergent vegetation.

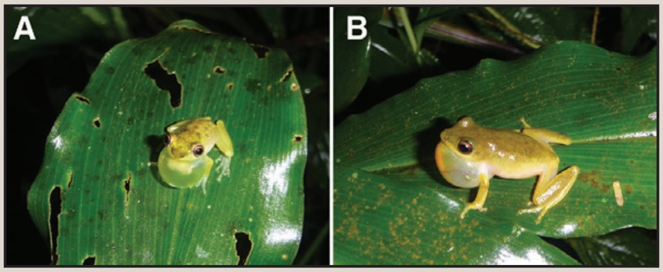

Fig. 3. Hyperoliid males actively calling at the fish ponds with emergent vegetation, including A) *H. fusciventris* and B) *H. concolor*.

Results

Breeding Habitat.—We observed differences in the quantity of species that were present across breeding habitat types. The only species not found at pond sites was *A. laevis*, which was found exclusively at slowly moving streams in secondary

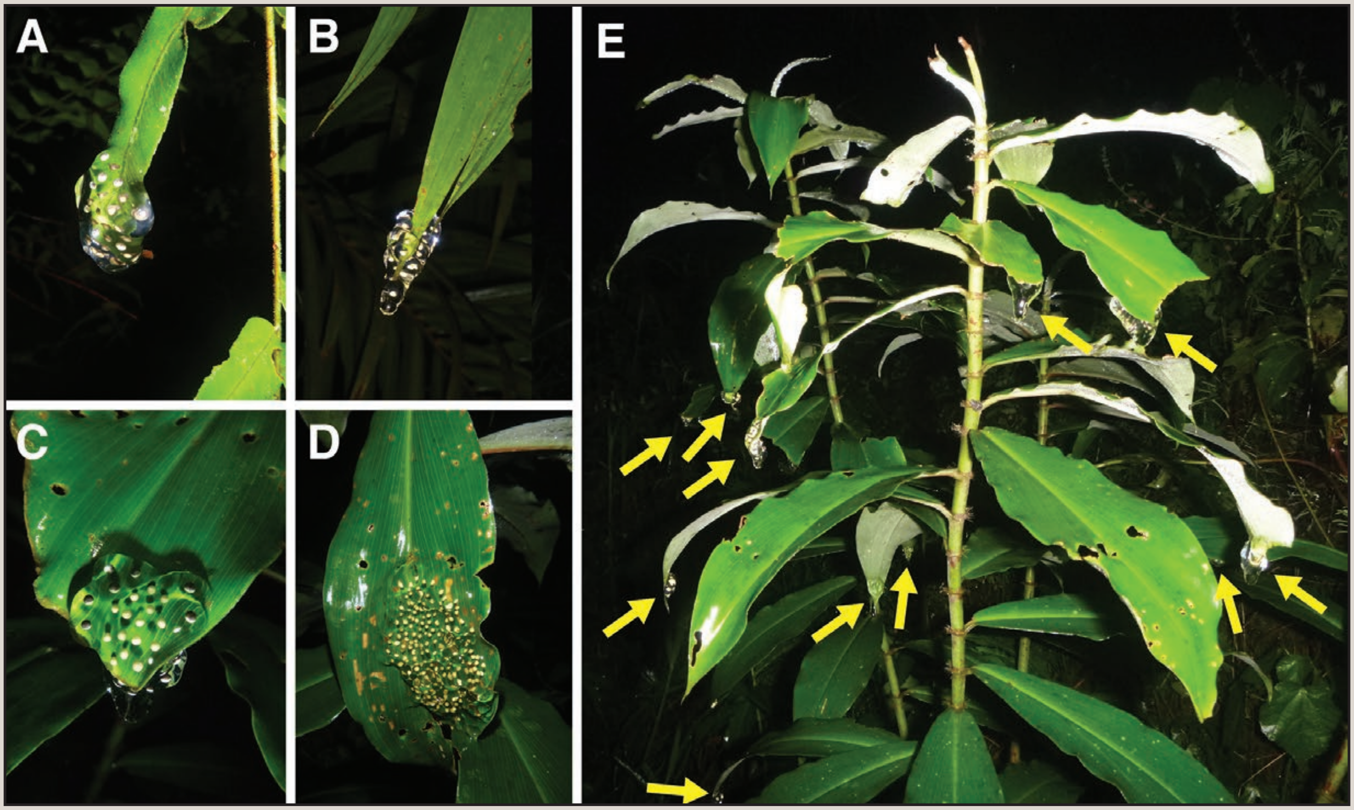

Fig. 4. Arboreal egg masses found at the fish ponds, including on the ends of raffia leaves, fern leaves, and on the surface and underside of emergent plant leaves (A–E). The yellow arrows in panel E show locations of arboreal egg nests.

Fig. 5. A large permanent pond located in disturbed forest, which had the highest density of emergent plants and was largely surrounded by trees, raffia, and shrubs.

forest. Males were heard calling on leaves approximately 1–3 m in height, and a single pair was found in amplexus. No other hyperoliid species were found along these streams. In the most disturbed habitats, including roadside ditches along flooded agricultural plots (4.9553°N, 9.8678°E) and grass-filled ditches within a large banana plantation (4.8498°N, 9.7718°E) (Fig. 1), we found large numbers of *A. dorsalis* and *H. concolor*, and only rarely *H. fusciventris* and *H. camerunensis*.

We sampled a series of fish ponds adjacent to a large river (4.8247°N, 9.7702°E; 2014) which differed in the amount of surrounding and emergent vegetation. At a fish pond surrounded mainly by reeds and with little emergent vegetation (Fig. 2), we collected males and females of *H. dintelmanni*. Additional ponds a short distance away were surrounded by trees, raffia, and other plants and contained a higher abundance of emergent vegetation, and here we found active breeding choruses and females of *H. concolor*, *H. fusciventris*, and *H. ocellatus* (Fig. 3A, B). We discovered an abundance of arboreal egg masses at these locations, which were located on the ends of raffia leaves, fern leaves, and on the surface and underside of emergent plant leaves in the fish ponds (Fig. 4A–E). These egg masses could not be field identified because multiple species of *Hyperolius* perform arboreal oviposition, and unambiguous species assignment at these diverse sites is only possible through direct observation of oviposition or through DNA barcoding of egg material.

Our main sampling site was a large permanent pond located in disturbed forest (4.8498°N, 9.7718°E), which had the highest density of emergent plants and was partially surrounded by trees, raffia, and shrubs (Fig. 5). This location, which was visited multiple times during each survey period, had the greatest number of species and highest abundance of any breeding site. At this pond we collected *A. dorsalis*, *A. paradorsalis*, *H. bolifambae*, *H. camerunensis*, *H. concolor*, *H. dintelmanni*, *H. fusciventris*, *H. ocellatus*, *H.* sp., and *P. leonardi*. In July 2013, the species with the highest relative abundances (based on specimen collection) were *H. bolifambae* and *H. concolor*, followed by *H. camerunensis*, *H. fusciventris*, and *A. paradorsalis*. In September 2014, the species with the highest relative abundance was *H. fusciventris*, followed by *H. bolifambae*, *H. concolor*, and *A. paradorsalis*. We observed males of all species of *Hyperolius* in close proximity using similar leaf perches as calling sites, and choruses were active during all surveys (Fig. 6A–D). Interspecific aggression and combat was documented, and we observed a male *H. concolor* displace a

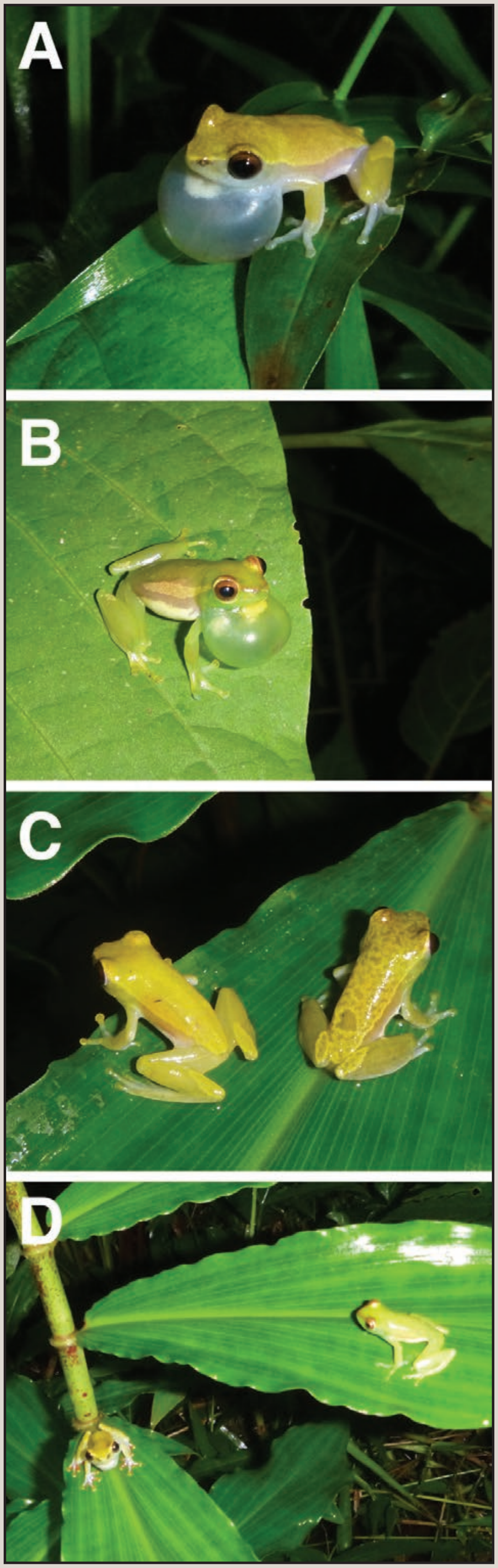

Fig. 6. Males of species of *Hyperolius* found in syntopy at the pond in Fig. 5, including A) *H. bolifambae*, B) *H. camerunensis*, C) two male *H. bolifambae*, and D) a male *H. bolifambae* (left) and *H.* sp. (right).

Fig. 7. *Afrixalus paradorsalis* in the process of egg deposition and leaf-folding.

male *H. fusciventris* from a calling site, and likewise observed aggressive behavior between a male *H. bolifambae* and male *H. camerunensis*. We observed amplexus in several species, and most of our observations of mating pairs occurred at this location (see below). Oviposition was observed in *A. paradorsalis*, and the breeding pair was found in the process of egg deposition and leaf-folding, which was overhanging a puddle located at the edge of the pond (Fig. 7). Male *A. paradorsalis* were encountered at similar calling sites as the species of *Hyperolius*, however *A. dorsalis* was only found calling near the ground in clumps of grasses and reeds surrounding a dirt track filled with water that ran parallel to the pond.

Amplexus.—In total we found 34 pairs in amplexus during our surveys (Fig. 8A–G), including the following species: *A. dorsalis* (2 pairs), *A. laevis* (1 pair), *A. paradorsalis* (4 pairs), *H. bolifambae* (4 pairs), *H. camerunensis* (5 pairs), *H. concolor* (3 pairs), *H. fusciventris* (10 pairs), and *H. ocellatus* (6 pairs). The species identities of males and females found in amplexus were confirmed using morphological and genetic data and in all instances represent conspecific mating, and we found no evidence of heterospecific amplexus. We provide information for all pairs in amplexus in Table 1. Portik et al. (2016) provided details on the occurrence of sexual dichromatism in *H. bolifambae, H. camerunensis, H. concolor, H. dintelmanni,* and *H. fusciventris*, along with frequency of Phase F males (sensu Schiøtz 1999) if they occurred. The highest prevalence of Phase F males occurred in *H. concolor* (27%), and of the three mating *H. concolor* pairs found in amplexus, two males were Phase J (CAS 253929, CAS 253932) and one male exhibited Phase F (CAS 256902). The frequency of Phase F males in *H. camerunensis* was considerably lower (7%), and all males found in amplexus were Phase J.

Predation.—At pond sites, we encountered several large spiders that were actively moving across the emergent vegetation on which male hyperoliids were calling. We witnessed a predation event, in which an adult female *H. fusciventris* had recently been killed by a spider and was in the process of being eaten (Fig. 9A). The spider and partially consumed frog were both collected and preserved (CAS 254087), and the spider has been putatively identified as *Nilus* cf. *curtus* (Family: Pisauridae). Predation events involving spiders and African frogs have also been reported by Barej et al. (2009; Family: Ctenidae) and Rödel (1996;

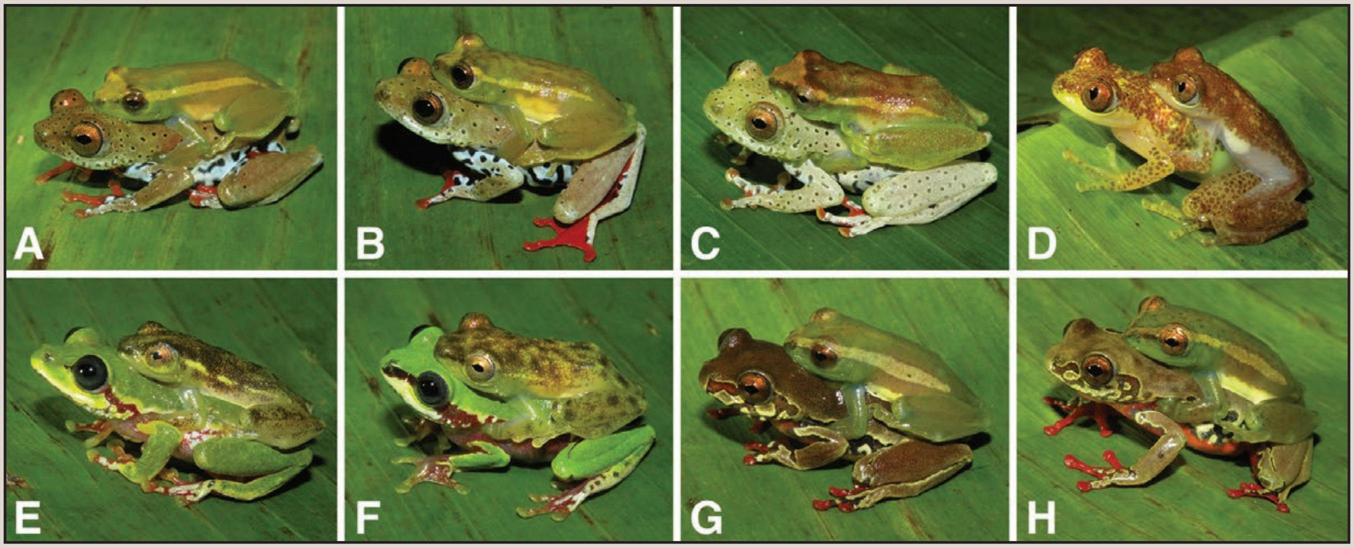

FIG. 8. Amplecting pairs of hyperoliid species encountered during surveys: A–C) *Hyperolius ocellatus*, D) *A. laevis*, E–F) *H. fusciventris*, and G–H) *H. camerunensis*.

Family: Pisauridae). At one pond site we observed and collected a Boulenger's Brown Tree Snake, *Dipsadoboa duchesnii*, actively searching on the emergent vegetation (CAS 254086) (Fig. 9B). Although we did not make a direct observation of *D. duchesnii* preying on hyperoliid species, this elongated, arboreal snake is known to consume frogs (Rasmussen 1989) and is likely an important nocturnal predator at pond sites.

Breeding Phenology.—We sampled a combination of breeding sites in July 2013 and September 2014, with a majority of sites visited during both years. We did not perform comprehensive population counts during our exploratory fieldwork, and for all results we acknowledge the potential confounding effects of sampling effort and between-year variance on our population estimates. However, our specimen collection was a consequence of the species abundances encountered during our surveys, and in this sense our specimen counts reflect the community structure across breeding sites and can provide at least preliminary insight on breeding phenology. Our data suggest a potential decrease in breeding activity for *A. dorsalis, H. bolifambae,* and *H. camerunensis*, with potential increases in breeding activity for *H. concolor*, *H. fusciventris*, *H. ocellatus*, and *H.* sp., from July to September (Fig. 10). These results complement the temporal patterns of amplexus, where in July

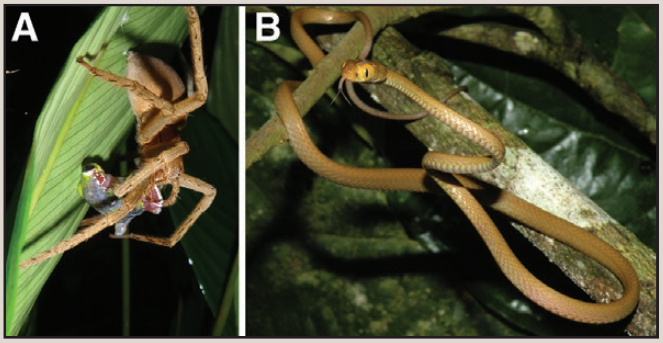

FIG. 9. Examples of anuran predators, including A) predation event encountered involving a large spider (*Nilus* cf. *curtus*) and an adult female *Hyperolius fusciventris*, and B) *Dipsadoboa duchesnii* found traversing breeding hyperoliid habitat.

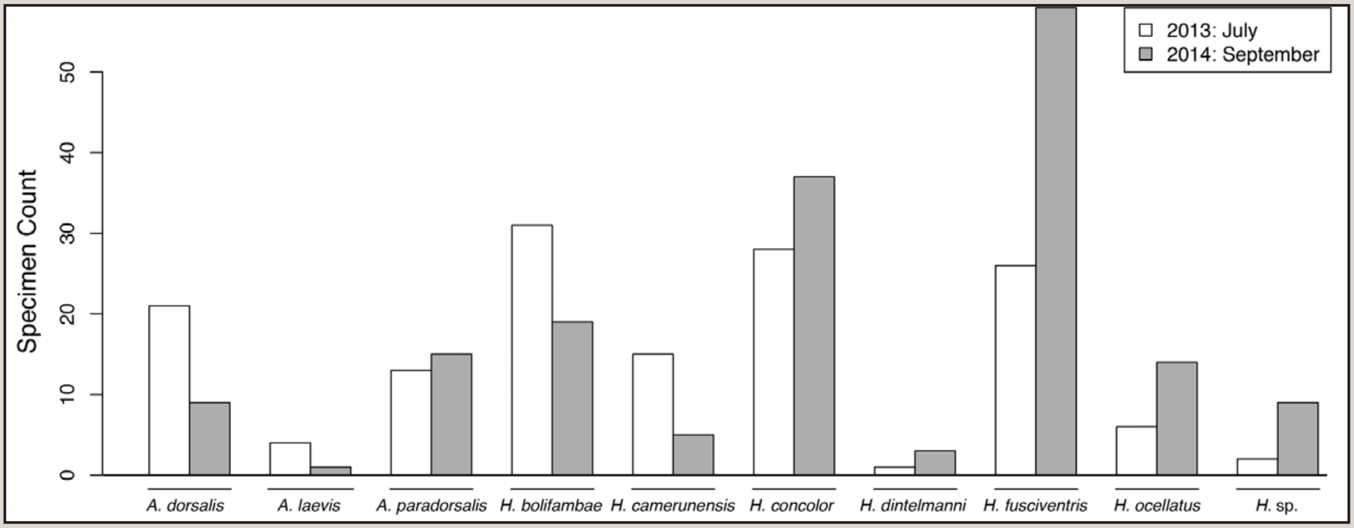

FIG. 10. Species abundances estimated from specimen collections made during July 2013 and September 2014.

Table 1. Instances of amplexus for eight species of hyperoliid frogs encountered during surveys.

Species	Mating pair	Female voucher	Male voucher	Year
Afrixalus dorsalis	1	CAS 253855	CAS 253854	2013
	2	CAS 253955	CAS 253956	2013
Afrixalus laevis	1	CAS 254072	CAS 254073	2013
Afrixalus paradorsalis	1	CAS 253947	CAS 253948	2013
	2	CAS 253949	CAS 253950	2013
	3	CAS 253951	CAS 253952	2013
	4	CAS 253953	CAS 253954	2013
Hyperolius bolifambae	1	CAS 253895	CAS 253896	2013
	2	CAS 253898	CAS 253899	2013
	3	CAS 253900	CAS 253901	2013
	4	CAS 253904	CAS 253905	2013
Hyperolius camerunensis	1	CAS 253935	CAS 253936	2013
	2	CAS 253937	CAS 253938	2013
	3	CAS 254059	CAS 254060	2013
	4	CAS 254078	CAS 254079	2013
	5	CAS 256729	CAS 256730	2014
Hyperolius concolor	1	CAS 253928	CAS 253929	2013
	2	CAS 253931	CAS 253932	2013
	3	CAS 256901	CAS 256902	2014
Hyperolius fusciventris	1	CAS 254005	CAS 254006	2013
	2	CAS 254007	CAS 254008	2013
	3	CAS 254009	CAS 254010	2013
	4	CAS 254011	CAS 254012	2013
	5	CAS 256720	CAS 256721	2014
	6	CAS 256722	CAS 256723	2014
	7	CAS 256811	CAS 256812	2014
	8	CAS 256813	CAS 256814	2014
	9	CAS 256833	CAS 256834	2014
	10	CAS 256835	CAS 256836	2014
Hyperolius ocellatus	1	CAS 254057	CAS 254058	2013
	2	CAS 254074	CAS 254075	2013
	3	CAS 254076	CAS 254077	2013
	4	CAS 256701	CAS 256702	2014
	5	CAS 256725	CAS 256726	2014
	6	CAS 256727	CAS 256728	2014

Table 2. Counts of female hyperoliid specimens collected during surveys.

Species	July 2013	September 2014
Afrixalus dorsalis	3	1
Afrixalus laevis	1	0
Afrixalus paradorsalis	4	1
Hyperolius bolifambae	7	6
Hyperolius camerunensis	5	1
Hyperolius concolor	3	10
Hyperolius fusciventris	6	8
Hyperolius ocellatus	3	6

Table 3. Mean body size [mm] of each sex and sexual size dimorphism index for hyperoliid species.

Species	Male Size	Female Size	SSDi
Afrixalus dorsalis	25.6	25.9	0.01
Afrixalus laevis	20.9	24.2	0.16
Afrixalus paradorsalis	30.1	32.3	0.07
Hyperolius bolifambae	24.4	29.8	0.22
Hyperolius camerunensis	23.4	29.2	0.25
Hyperolius concolor	27.8	34.3	0.23
Hyperolius dintelmanni	31.4	33.6	0.07
Hyperolius fusciventris	19.6	25.1	0.28
Hyperolius ocellatus	22.2	28.5	0.28
Hyperolius sp.	25.9	—	—

TABLE 4. Multiple pairwise-comparisons of the mean body size of males or females across species.

	H. bolifambae		*H. camerunensis*		*H. concolor*		*H. dintelmanni*		*H. fusciventris*		*H. ocellatus*	
	Male	Female	Male	Female	Male	Female	Male	Female	Male	Female	Male	Female
H. bolifambae												
H. camerunensis	0.24	0.98										
H. concolor	**<0.01**	**<0.01**	**<0.01**	**<0.01**								
H. dintelmanni	**<0.01**	0.22	**<0.01**	0.08	**<0.01**	0.41						
H. fusciventris	**<0.01**	**<0.01**	**<0.01**	**<0.01**	**<0.01**	**<0.01**	**<0.01**	**<0.01**				
H. ocellatus	**<0.01**	0.62	0.32	0.98	**<0.01**	**<0.01**	**<0.01**	**<0.01**	**<0.01**	**<0.01**		
H. sp.	**<0.01**	NA	**<0.01**	NA	**<0.01**	NA	**<0.01**	NA	**<0.01**	NA	**<0.01**	

Significant p-values (<0.05) of the Tukey HSD test are bolded, indicating a difference in mean body size for a given sex and species comparison. NA indicates data were not available for a particular comparison.

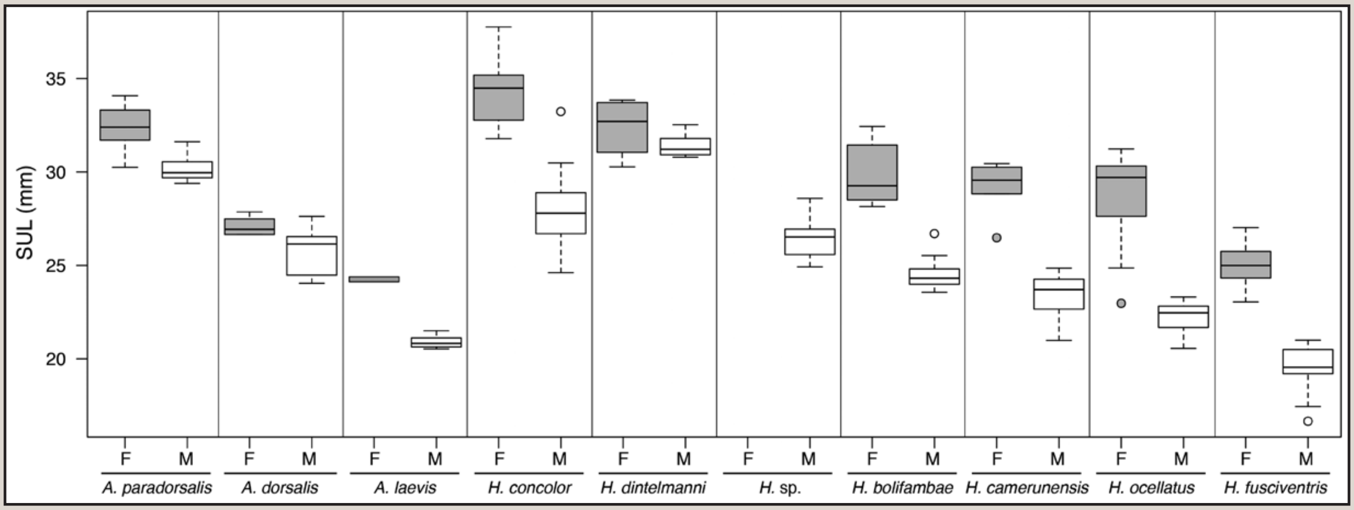

FIG. 11. Body size measurements for males and females of hyperoliid species, where boxplots depict the median, upper and lower quartiles, and range of body sizes.

we observed mating pairs in eight species (*A. dorsalis*: 2;, *A. laevis*: 1; *A. paradorsalis*: 4; *H. bolifambae*: 4; *H. camerunensis*: 4; *H. concolor*: 2; *H. fusciventris*: 4; *H. ocellatus*: 3) and in September we observed mating pairs in four species (*H. camerunensis*: 1; *H. concolor*: 1; *H. fusciventris*: 6; *H. ocellatus*: 3). We also considered the total number of females present (in amplexus or solitary) to be a relevant indicator of breeding activity (Table 2). In general our data indicate the abundance of females tracked their respective species abundance across seasons, and the number of active females decreased in *A. dorsalis*, *A paradorsalis*, and *H. camerunensis* from July to September, and increased in *H. concolor*, *H. fusciventris*, and *H. ocellatus*.

Body size patterns.—We present a summary of the sex-specific body size distributions for all 10 hyperoliid taxa in Figure 11. In all species, we observed female-biased sexual size dimorphism (Table 3). Strong levels of size dimorphism (SSDi > 0.15) were detected in all species except *A. dorsalis*, *A. paradorsalis*, and *H. dintelmanni*, which exhibited less prominent female-biased size dimorphism.

Our investigation of the similarity of mean body sizes across species produced several key findings. In the pooled analyses of both genera, there were statistically significant differences between species body size means as determined by one-way ANOVA for males ($F = 124.7$, $p < 0.001$) and for females ($F = 24.1$, $p < 0.001$). The Tukey's honest significant difference (HSD) post hoc test revealed 39 of 45 pairwise species comparisons had statistically significant differences in mean body size for males, whereas in females only 17 of 36 pairwise species comparisons had statistically significant differences in mean body size. We recovered similar results for the analyses containing only *Hyperolius* species, and statistically significant differences between species body size means were again detected in males ($F = 123.1$, $p < 0.001$) and females ($F = 35.2$, $p < 0.001$). The Tukey HSD post-hoc test revealed 19 of 21 pairwise species comparisons had statistically significant differences in mean body size for males, whereas in females only 9 of 15 pairwise species comparisons had statistically significant differences in mean body size (Table 4). The two nonsignificant results for the mean body size comparisons in males are attributed to *H. camerunensis*, as males of this species appear to overlap in body size with males of *H. bolifambae* and *H. ocellatus* (Table 4, Fig. 11). However, the average male body sizes of *H. concolor, H. dintelmanni, H.* sp., *H. bolifambae, H. ocellatus*, and *H. fusciventris* are all statistically significantly different from one another. In contrast, significant body size differences in females were generally only found when comparisons included the largest (*H. concolor*) or smallest (*H. fusciventris*) species. Unlike the patterns for males, we found average female body sizes were not statistically different between *H. dintelmanni*, *H. bolifambae*, *H. camerunensis*, and *H. ocellatus*. Taken together, these results indicate relatively little overlap occurs in the body sizes of males of different species, whereas the body sizes of females tend to be much more similar.

We observed some outliers in our body size data, and these represent either small females or large males. For large outlier males in species with size dimorphism, we would not predict their body size to be equivalent to female size, especially if a high degree of female-biased size dimorphism is present (Table 3). However, we found this to be the case for *H. concolor*, where the size of one male (33.2 mm) falls well outside the range of other males (24.6–30.4 mm; mean 27.8 mm) but within the range of female size (31.8–37.7 mm; mean 34.3). We note this extraordinarily large male (CAS 256899) is Phase J and exhibits a gular gland. The other male outlier in our data set occurs in *H. bolifambae* (26.7 mm; CAS 253883), and although it is larger than typical males (23.5–25.5 mm; mean 24.4 mm) it does not reach female size (28.1–32.4 mm; mean 29.8 mm).

The collection of a large series of male *H. concolor* in Phase J (N = 32) and in Phase F (N = 10) provided an opportunity to investigate if the phases differed in body size. Although the average body size for Phase F is larger (mean 28.2 mm; range 26.2–30.4 mm) than for Phase J (mean 27.6 mm; range 24.6–30.1 mm), based on a Welch two-sample t-test we were unable to reject the null hypothesis that the mean body sizes are equal ($t = -1.57$, $df = 24.6$, $p = 0.12$). Therefore, we found no statistically significant difference in the body sizes of Phase J and Phase F males in *H. concolor*.

Discussion

In this study, we characterized various aspects of natural history and reproductive biology for the hyperoliid assemblage found at Mt. Kupe in an effort to generate baseline information for future work. In addition, we quantified sex-specific body size patterns to explore sexual size dimorphism and body size overlap among syntopic species. Below, we discuss several of our key findings related to reproductive biology, species interactions, and body size evolution, and draw comparisons between the levels of hyperoliid diversity found at Mt. Kupe and other regions in sub-Saharan Africa.

Nearly all the hyperoliid species we collected at Mt. Kupe rely on lentic water systems for breeding, with the exception of *A. laevis*, which breeds in forest streams. At disturbed habitat with ephemeral pools, we mainly found *A. dorsalis* and *H. concolor*. However, *H. concolor* was also present in less degraded environments, including secondary forest ponds, suggesting its habitat requirements are more generalized. Other species, such as *H. dintelmanni*, *H. fusciventris*, and *H. ocellatus*, were present at ponds in both disturbed and forest sites indicating a reliance on permanent water sources for breeding activity, but no preference for cover type could be detected. Finally, *A. paradorsalis*, *H. bolifambae*, *H. camerunensis*, *H.* sp., and *P. leonardi* were only found breeding at the main pond that was partially surrounded by secondary forest, suggesting different requirements in their selection of breeding site. The amount of forest cover and the prevalence and type of emergent vegetation could play an important role in determining the suitability of reproductive habitat, and the leaves of emergent plants served not only as calling sites for males but also as a location for egg deposition and fertilization. Systematic studies of habitat structure, taxon-specific calling sites, and oviposition sites could begin to disentangle the factors relevant for determining suitable breeding habitat for these species (Kouamé et al. 2015). In their study of a Kenyan hyperoliid community, Lötters et al. (2004) found three species had general habitat requirements and occurred across sites, whereas two species were more restricted and occurred primarily at either ephemeral pools or permanent ponds. Our preliminary data suggest spatial partitioning is occurring to a limited extent, and a majority of taxa appear to be restricted to ponds in secondary forest whereas a handful of generalists are also capable of exploiting more disturbed habitats.

Most hyperoliids are thought to be prolonged breeders (Schiøtz 1999; Lötters et al. 2004; Rödel et al. 2006; Kouamé et al. 2015), but little is known about the breeding phenology of hyperoliid communities. Lötters et al. (2004) found seasonal variation in a Kenyan hyperoliid community, where several species persisted almost year-round at breeding sites while others exhibited a much narrower window of breeding activity. We performed surveys in July and September, and based on total species abundances, the number of females present, and occurrences of amplexus we found that *A. dorsalis*, *H. bolifambae,* and *H. camerunensis* had the highest reproductive activity in July, whereas *H. concolor*, *H. fusciventris*, *H. ocellatus*, and *H.* sp. were more reproductively active in September (Fig. 10, Tables 1, 2). The rainy season in the Bakossi Mountains (including Mt. Kupe) begins in April and peaks between late August and the end of October, indicating shifts in abundance may be related to increased rainfall. We recognize breeding phenology is best studied using systematic sampling techniques (Rödel et al. 2006; Lawson and Moyer 2008) and across regular time intervals in a single season (Kouamé et al. 2015), but the marked differences we observed in species composition and breeding activity in the ponds during our two surveys indicate that temporal partitioning is likely occurring at this site. The breeding phenology of this hyperoliid community can be further studied using traditional sampling strategies or more automated techniques, such as acoustic monitoring.

The dense multispecies breeding aggregations observed during our surveys raise interesting questions about inter- and intraspecific signaling, mate recognition, and mate-choice. For example, how do males interact with conspecific versus heterospecific males? With only general observations of males, we are unable to determine if species are spatially partitioned within breeding sites. However, Rödel et al. (2006) found differences in calling sites selected by five syntopic hyperoliid species, including height, substrate, and distance to water source, indicating species differed in their microhabitat use. The use of different microhabitats could mitigate certain aspects of resource competition and quantifying male calling sites within and between species will be a first step towards understanding their interspecific interactions. In many species, male hyperoliids establish a small territory around a calling site and subsequently defend it against invaders (Telford 1985; Backwell and Passmore 1990; Rödel et al. 2006). Territory invasion by a conspecific may be resolved through encounter calling, submissive behavior, or physical combat (Telford 1985; Backwell and Passmore 1990). The response of males to invasion of their territory by a heterospecific male is largely unknown, although we did observe instances of interspecific aggression between males. The prevalence of these interactions is not known, but the spatial partitioning of microhabitats among species may serve to reduce this type of heterospecific aggression. Another important question to consider is, what signals are important for intersexual signaling and mate recognition? We found a total of 34 amplectant pairs from eight species, each consisting of a conspecific male and female. Studies focused on the reproductive biology of other species of *Hyperolius* have only recorded instances of conspecific amplexus, despite the presence of multiple congeners at breeding sites (Telford and Dyson 1988; Rödel et al. 2006; Kouamé et al. 2015). The lack of heterospecific mating pairs suggests a high degree of mate recognition, mechanisms causing release during heterospecific amplexus, or both. Hyperoliid mating behavior is based primarily on observations of *H. marmoratus*: females slowly and deliberately approach males, males increase calling rate, and the females commence amplexus through physical contact (Telford and Dyson 1988). Additionally, female *H. marmoratus* prefer to initiate amplexus and tend to flee when being actively pursued by males. In other species, it has been demonstrated that females have a high ability to discriminate between conspecific advertisement calls and heterospecific calls of syntopic taxa (Telford and Passmore 1981). Mate recognition and mating behaviors can be examined for the hyperoliid community at Mt. Kupe, by recording and characterizing advertisement calls and performing cross-species phonotaxis experiments. In addition to acoustic cues, chemical signaling may play a role in hyperoliids, and the production and emittance of a variety of chemical compounds from the gular gland of males may facilitate mate localization (Starnberger et al. 2013). It is also possible that sexual dichromatism, which is present in

TABLE 5. Hyperoliid species richness estimates currently available for locations throughout Central, East, and West Africa.

Region	Country	Location	Species richness	Sources
Central Africa	Cameroon	Korup National Park	14	Lawson 1993
		Mt. Kupe	18	Amiet 1975; Hofer et al. 1999; Portik et al. 2016; Schmitz et al. 1999
		Mt. Nlonako	17	Herrmann et al. 2005
	Gabon	Batéké Plateau National Park	8	Zimkus and Larson 2013
		Crystal National Park	7	Pauwels and Rödel 2007
		Gamba	8	Burger et al. 2006
		Ivindo National Park	9	Pauwels and Rödel 2007
		Lekoko	13	Jongsma et al. 2017
		Loanga Park	8	Burger et al. 2006; Pauwels and Rödel, 2007
		Mokalaba-Doudou National Park	16	Burger et al. 2006
		Rabi-Toucan	14	Burger et al. 2006
East Africa	Kenya	Kakamega Forest	8	Schick et al. 2005
		Shimba Hills National Reserve	12	Bwong et al. 2017
	Tanzania	East Usambara Mountains	13	Lawson and Collett 2011
		Udzungwa Mountains	14	Lawson and Collett 2011
		Uluguru Mountains	8	Lawson and Collett 2011
	Rwanda	Butare	7	Sinsch et al. 2011
	Uganda	Impenetrable Forest Reserve	13	Drewes and Vindum 1994
West Africa	Côte d'Ivoire	Banco National Park	7	Assemian et al. 2006
		Comoé	10	Rödel and Spieler 2000
		Lamto	14	Adeba et al. 2010
		Marahoué National Park	9	Rödel and Ernst 2003
		Mt. Péko National Park	10	Rödel and Ernst 2003
		Mt. Sangbé	14	Rödel 2003
		Taï National Park	11	Ernst and Rödel 2006, 2008
		Tanoé-Ehy Forest	10	Kpan et al. 2014
		Yakassé-Mé Forest	6	Kouamé et al. 2014
	Ghana	Ankasa National Park	8	Hillers et al. 2009
		Atewa Range Forest Reserve	10	Kouamé et al. 2013
		Togo Hills	10	Leaché et al. 2006
	Guinea	Déré Forest Reserve	9	Rödel et al. 2004
		Diécké Forest Reserve	12	Rödel et al. 2004
		Mt. Béro Forest Reserve	9	Rödel et al. 2004
		Mt. Nimba	15	Rödel et al. 2004
		Pic de Fon	10	Rödel et al. 2004
		Ziama Forest Reserve	11	Rödel et al. 2004
	Liberia	Gola National Forest	7	Hillers and Rödel 2007
		Grebo National Forest	7	Hillers and Rödel 2007
	Nigeria	Oyo	8	Onadeko and Rödel 2009

all seven species of *Hyperolius* at Mt. Kupe, may enhance mate recognition. Although males tend to be similar in coloration, females differ drastically across these species (Portik et al. 2016), and female color may reduce reproductive interference by serving as a visual cue to males. Although historically overlooked, recent studies have demonstrated the importance of nocturnal visual signals for anuran mate choice (Gomez et al. 2009, 2010; Jacobs et al. 2016; Akopyan et al. 2018). Future work can help clarify the role of natural and sexual selection in the evolution of acoustic and chemical signals and determine whether visual signals are involved in communication in hyperoliids.

Anurans exhibit a wide range of body sizes and degree of sexual size dimorphism, yet close to 90% of species evaluated exhibit female-biased sexual size-dimorphism (Shine 1979; Han and Fu 2013). We found a pattern of female-biased size dimorphism in all the hyperoliid species present at Mt. Kupe, though the degree of dimorphism was variable (Table 3). The most common explanation for female-biased SSD in frogs is a fecundity advantage (Shine 1989; Andersson 1994), as fecundity increases more rapidly with body size for females than for males (Trivers 1972; Crump 1974). Interestingly, we found the body sizes of females are similar across most *Hyperolius* species, whereas male body sizes are significantly different across nearly all species comparisons (Table 4). This pattern may reflect niche partitioning in males, where a range of body sizes may allow differential access to prey items, shelters, or calling sites. The consistency in female body size is a curious pattern, as females body size should track male body size isometrically or increase based on sexual selection theory (Han and Fu 2013). A vast majority of forest-dwelling species of *Hyperolius* utilize arboreal sites for oviposition, often on the ends of leaves on emergent vegetation. This reproductive trait may constrain the maximum body size of females insofar as these leaves may not support the weight of a larger female in amplexus. Other equivalent-sized or larger arboreal species including *Leptopelis* and *Chiromantis* may occur on these microhabitats, but they do not use these sites for oviposition (terrestrial and foam nest oviposition, respectively). Though currently speculative, the functional relationship between hyperoliid female body size and arboreal egg laying may oppose selection for increased fecundity. This relationship could be tested by characterizing the properties of vegetation selected by females for arboreal oviposition, determining the body sizes and masses of amplecting pairs, and subsequently examining the weight limitations of those arboreal oviposition sites.

The forests of Mt. Kupe, Cameroon have the highest documented hyperoliid diversity in Sub-Saharan Africa with a total of 18 species (Amiet 1975; Hofer et al. 1999; Portik et al. 2016) (Table 5). This richness estimate is higher than locations in other West, Central, and East African countries based on published survey results (36 sites, Table 5). The species richness at Mt. Kupe is most comparable to other regions in Cameroon, including Korup National Park (14 species; Lawson 1993) and the neighboring Mt. Nlonako, which has a total of 17 hyperoliid species (Herrmann et al. 2005). Together, Mt. Kupe, Mt. Nlonako, and Mt. Manengouba highlight the greater Cameroonian Volcanic Line as an important center of hyperoliid species diversity. Beyond overall measures of species richness, another important assessment of diversity consists of identifying species composition across sampling sites, or community structure. The presence of 10 syntopic hyperoliid species breeding at Mt. Kupe is truly exceptional and is similar to the number of syntopic species from hotspots like the Atewa Hills, Ghana (9 species, DMP, pers. obs.; Kouamé et al. 2013) and Omubiyanja Swamp, Uganda (8 species, Drewes and Vindum 1994). These numbers are even more impressive considering the high morphological and reproductive mode similarity within hyperoliid genera (particularly *Afrixalus* and *Hyperolius*). These sites should prove to be ideal settings for a variety of ecological and evolutionary biology studies of hyperoliid frogs in the future.

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Herpetological Survey of Cangandala National Park, with a Synoptic List of the Amphibians and Reptiles of Malanje Province, Central Angola

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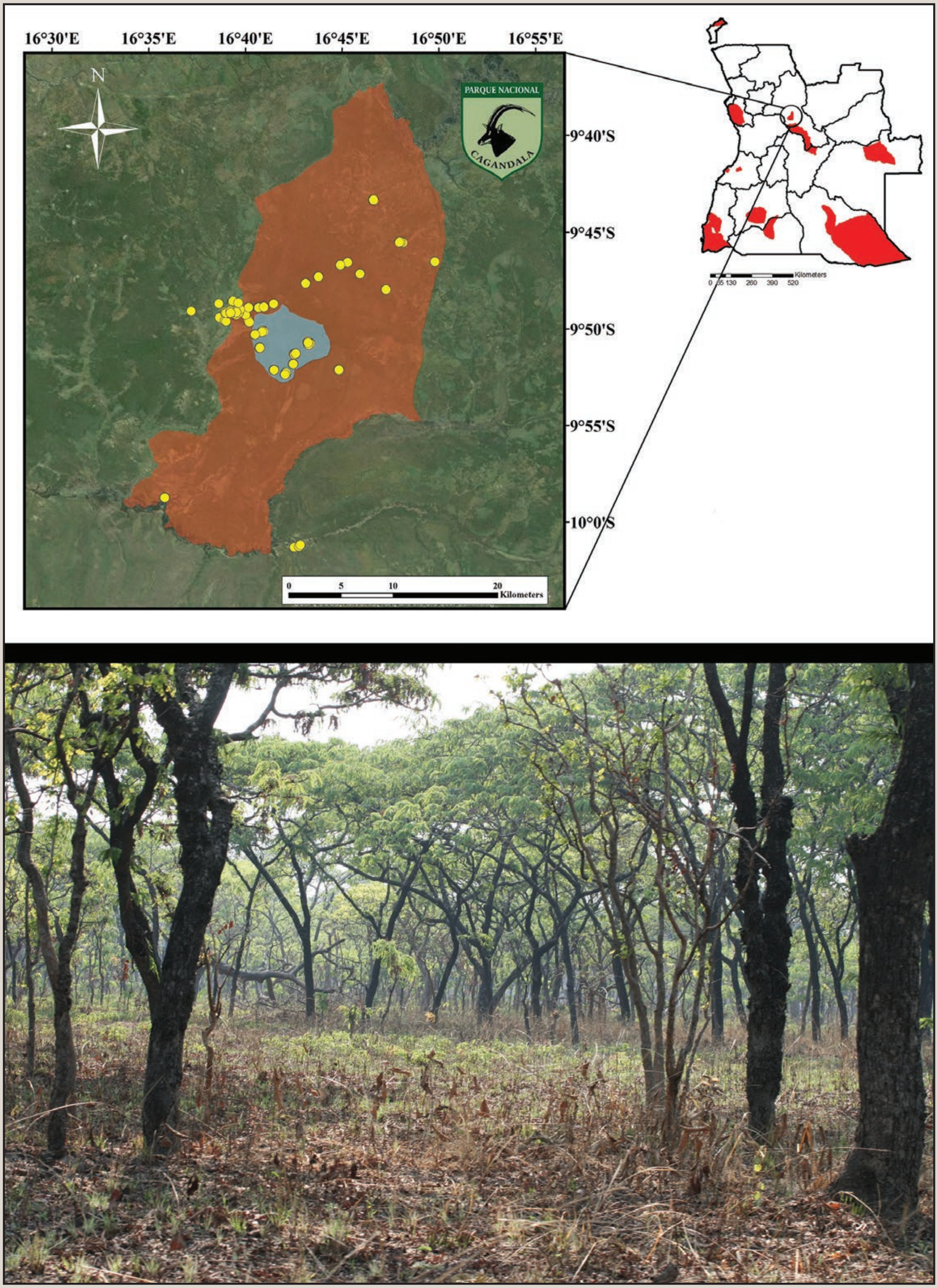
Angola is one of the most poorly known sub-Saharan African countries in terms of its biodiversity, in large part due the violent armed conflicts that afflicted the country from 1961 to 2002. However, in recent years, several expeditions and ongoing studies have been carried out, uncovering previously unknown cryptic diversity, new country records, and expanding known distributions of species considerably (Conradie et al. 2012a,b; Conradie et al. 2013; Ceríaco et al. 2014a; Ernst et al. 2014; Branch and Conradie 2015; Ernst et al. 2015; Ceríaco et al. 2016a; Conradie et al. 2016; Stanley et al. 2016; Branch et al. 2017). Despite this new wave of studies on Angolan herpetology, data are limited, even within national conservation areas. Angola currently has 16 protected areas, scattered across 11 of its 18 provinces, and covering an area of 145,859 km^2, approximately 12% of the Angolan territory. Conservation areas in the country are divided into four major categories: National Parks, Regional Parks, Nature Reserves, and Coutadas (Game Parks). The majority of these areas were created during the first half of the 20th century and into the 1970s, during Portuguese colonial times. Many of these areas were mostly dedicated to hunting and tourism, and their delimitation was almost exclusively based on the presence of game species and iconic megafauna to the neglect of other biological groups (see Frade 1959a, b).

Located in Malanje Province, Cangandala National Park (CNP) is the smallest of all Angolan National Parks, at roughly 630 km^2. Situated about 50 km south of Malanje city in the Cangandala Municipality, the park is limited to the north by the Cuije River, to the west by the Maúbi River, and to the south by the Cuque River, all part of the Kwanza Basin (Fig. 1). The park lies



PHOTOS BY L. M. P. CERÍACO

Fig. 1. Cangandala National Park map and collecting localities (yellow dots). The orange area represents the limits of the park, while the grey area represent the Giant Sable Special protection area, known as "Santuário." Below is a view of the Miombo Woodlands habitat, the dominant habitat in Cangandala National Park.

at an approximate elevation of 1000 m and its climate is similar to that of the Angolan central plateau. Its vegetation is mostly dominated by miombo woodlands (Fig. 1), with *Brachystegia wangermeeana*, *B. floribunda*, *Julbernardia paniculata*, *Erythrophleum africanum*, *Combretum* spp., and *Rhus* spp. growing on the red clay soils of the park (Grandvaux-Barbosa 1970). Some small riverine gallery forests also occur. The park was originally founded in 1963 as an "Integral Reserve," and only later, in June 1970, was reclassified as a National Park. The main objective of CNP since its foundation has been the protection to the Giant Sable Antelope, *Hippotragus niger variani* Thomas, 1916, one of Angola's most endangered endemic species, and now a national symbol. Despite CNP being one of the best-managed and funded conservation areas in the country, with the exception of reports on the Giant Sable and associated large mammals, the available published data on its biodiversity is limited to one checklist of the avifauna of the park (Mills et al. 2008). There are currently no available data regarding the diversity of amphibians and reptiles for CNP. However, Malanje Province is historically one of the most important provinces in terms of the development of current knowledge on Angolan herpetofauna (see section below). Approximately 35 species of amphibians and 68 species of reptiles are known from Malanje Province (Table 1), which represents approximately 30% and 25%, respectively, of the currently known numbers of these groups in Angola.

Biogeographically, the province represents one of the most obvious areas of faunal and habitat turnovers in the country (Crawford Cabral 1991; Marques et al., *in press*). Due to its boundaries and geographical position, the Province exhibits a north-south gradient of vegetation types, being dominated by Western Congolian Forest-Savannah Mosaic in the northern half, and by Angolan Miombo woodlands in the southern half (Romeiras et al. 2014; Rodrigues et al. 2015). This division is roughly associated with elevation, as the northern areas of the Province lie between 500–1000 m elevation, surrounded by higher ground (1000–1500 m) except along the Democratic Republic of Congo (DRC) border (Baixa de Cassanje), whereas the southern, Miombo-dominated half is on the Angolan plateau above 1000 m. This division also roughly corresponds to two different river drainage systems – the Congo-Casai in the north, and the Kwanza in the south. This contact zone is reflected in the associated faunal assemblies, particularly that of the herpetofauna.

This paper presents the results of an expedition conducted by a team from the California Academy of Sciences (CAS), San Francisco (USA), Villanova University (VU), Villanova (USA), Museu Nacional de História Natural e da Ciência (MUHNAC), Lisbon (Portugal) and the Instituto Nacional da Biodiversidade e Áreas de Conservação (INBAC), Kilamba-Kiaxi (Angola). A total of 33 herpetological taxa were collected, including putative new species, eight new provincial records, and new records for taxa rarely cited for the country. We provide a brief discussion of the present status and future prospects for the study of the herpetofauna of the province and the country. This was the second expedition to Angola under the memorandum of understanding signed by INBAC and international partners and has been included in the national biodiversity plan. The results of the first expedition to southwest Angola were published by Ceríaco et al. (2016a). Ceríaco et al. (2016b) published a booklet on the herpetofauna of CNP, written in Portuguese, chiefly for educational purposes and distributed almost exclusively in Angola, which presented preliminary results of the expedition but without taxonomic or specimen details. Thus, the current paper more fully presents the results of this expedition.

History of the Herpetological Exploration of the Province

Malanje Province can be considered the "birthplace" of Angolan herpetology. The Austrian naturalist Friedrich M. J. Welwitsch (1806–1872), appointed by the Portuguese Government to conduct a botanical expedition to Angola between 1853 and 1860, was the first to collect some herpetological material in the province. This material was sent to the British Museum where it was studied by Albert Günther (1830–1914), John Edward Gray (1800–1875), and George Albert Boulenger (1858–1937). One of the first herpetological species to be described from the country, *Dalophia* [currently *Monopeltis*] *welwitschii* Gray, 1865, was from Pungo Andongo in Malanje Province. Later, Günther (1888) described *Psammophis* [currently *Psammophylax*] *acutus* based on Welwitsch's material from this locality, and Boulenger described *Mabouia* [currently *Trachylepis*] *bocagii* (Boulenger, 1887) based on two specimens, one collected by Pinheiro Bayão in Duque de Brangança (currently Kalandula) and the other by Welwitsch collected at Pungo Andongo.

The Portuguese zoologist and director of the zoological section of the Natural History Museum in Lisbon, José Vicente Barbosa du Bocage (1823–1907), worked extensively with collections from the Portuguese colonial officer Captain Francisco António Pinheiro Bayão (birth and death dates unknown), who was based in Malanje Province between 1863 and 1866. Pinheiro Bayão sent Bocage some of the first shipments of Angola specimen received in the Lisbon Museum, mostly from Duque de Bragança (Malanje), Dondo (Kwanza-Norte Province) and Luanda (Luanda Province). Initially Bocage allowed foreign naturalists to study and help identify the Angolan herpetofauna, as in the case of Günther (1865a, b) who described *Hylambates Bocagii* (currently *Leptopelis bocagii*), *Rappia microps, Rappia nasuta* (currently *Hyperolius nasutus*) and *Limnophis bicolor*, and the Austrian zoologist Franz Steindachner (1834–1919), who named *Hyperolius angolensis* (described as *Hyperolius marmoratus* var. *angolensis*) and *Hyperolius bocagei* (Steindachner 1867). The first Angolan species described by Bocage himself was *Rana bragantina* (currently a synonym of *Hoplobatrachus occipitalis* (Günther 1858)) based on a specimen sent by Bayão from Duque de Bragança (Bocage 1864), and eventually he described 11 new amphibian taxa and 12 new reptile taxa based on Bayão's material, as well as adding several previously described taxa to the list of Angolan herpetofauna.

Wilhelm Peters (1815–1883), curator and director of the Berlin Museum also contributed to the study of the Malanje Province herpetofauna. This was largely based on the material collected during two German expeditions to Angola—one from 1873 to 1876 to the "Kingdom of Loango" Chinchoxo (Cabinda Province) and Loango (Malanje Province)—made by the *Afrikanischen Gesellschaft*, led by Paul Gussfeldt (1840–1920), in the company of Dr. Julius Falkenstein (1842–1917), Max Buchner (1846–1921) and Major Friedrich Wilhelm A. von Mechow (1831–1904); and the expedition of von Mechow and Major A. V. Homeyer to Malanje (Kwango River) and Pungo Andongo between 1879 and 1882. From the first expedition he described *Euprepes notabilis* Peters, 1879, currently considered as a synonym of *Trachylepis maculilabris* (Gray, 1845), based in part on a specimen from Pungo Andongo and, from the Malanje collection of Mechow,

Table 1. Amphibians and reptiles known to occur in Malanje Province. Only published records are included. Additional species known from the Province based on unpublished museum records are not included unless also supported by published records. For details on the specific localities of the records check the original reference or Marques et al. (*in press*).

Taxon	References
AMPHIBIANS	
ANURA	
Pipidae	
Genus *Xenopus* Wagler, 1827	
Xenopus petersii Bocage, 1895	Boulenger (1905); Monard (1938); Schmidt and Inger (1959); Loumont (1983); Ruas (1996); Ruas (2002); Ceríaco et al. (2014a); Ceríaco et al. (2014a, 2016a,b); This study.
Bufonidae	
Genus *Sclerophrys* Tschudi, 1838	
Sclerophrys funerea (Bocage, 1866)	Bocage (1866a, 1866b, 1882b, 1895, 1897); Loveridge (1957); Perret (1976a); Frost (1985, 2017); Ruas (1996, 2002).
Sclerophrys gutturalis (Power, 1927)	Ruas (1996, 2002).
Sclerophrys pusilla (Mertens, 1937)	Bocage (1866a, 1895); Boulenger (1882, 1905); Poynton and Haacke (1993); Ruas (1996, 2002); Ceríaco et al. (2014a, 2016b); this study.
Hyperoliidae	
Genus *Afrixalus* Laurent, 1944	
Afrixalus fulvovittatus (Cope, 1861)	Boulenger (1882).
Afrixalus wittei (Laurent, 1941)	Bocage (1866a, 1895); Perret (1976b); Ceríaco et al. (2016b); This study.
Genus *Hyperolius* Rapp, 1842	
Hyperolius angolensis Steindachner, 1867	Bocage (1866a, 1866b, 1893, 1895, 1897); Peters (1882b); Boulenger (1882, 1905); Laurent (1961); Ceríaco et al. (2014a, 2016b); This study.
Hyperolius bocagei Steindachner, 1867	Bocage (1873, 1895, 1897); Boulenger (1905); Ferreira (1906); Ceríaco et al. (2014b).
Hyperolius cinnamomeoventris Bocage, 1866	Bocage (1866a, 1866b, 1895, 1897); Laurent (1961); Schiøtz (1975), Perret (1976a); Frost (1985, 2017); Conradie et al. (2013); Ceríaco et al. (2016b); This study.
Hyperolius concolor (Hallowell, 1844)	Bocage (1866a, 1866b).
Hyperolius fuscigula Bocage, 1866	Bocage (1866a, 1866b, 1895, 1897); Perret (1976a).
Hyperolius glandicolor Peters, 1878	Bocage (1866a).
Hyperolius nasutus Günther 1865	Günther (1865b); Bocage (1866a, 1895, 1897); Boulenger (1882); Loveridge (1936a, 1936b, 1953, 1957); Schiøtz (1975); Frost (1985, 2017); Amiet (2005); Channing et al. (2013); Channing et al. (2013); Ceríaco et al. (2016b); This study.
Hyperolius pusillus (Cope, 1862)	Bocage (1866a, 1866b, 1895, 1897a); Boulenger (1905).
Hyperolius quinquevittatus Bocage, 1866	Bocage (1866a, 1866b, 1895); Schiøtz (1975); Perret (1976a); Frost (1985, 2014); Poynton and Broadley (1987); Pickersgill (2007a).
Hyperolius steindachneri Bocage, 1866	Bocage (1866a, 1866b, 1895, 1897); Loveridge (1936a); Perret (1976a); Frost (1985, 2017); Poynton and Haacke (1993).
Genus *Kassina* Girard, 1853	
Kassina cf. *maculosa* (Sternfeld, 1917)	Ceríaco et al. (2014a).
Arthroleptidae	
Genus *Arthroleptis* Smith, 1849	
Arthroleptis xenochirus Boulenger, 1905	Boulenger (1905); Laurent (1954b); Frost (1985, 2017); Ceríaco et al. 2016b); This study.
Genus *Leptopelis* Günther, 1859	
Leptopelis bocagii (Günther, 1864)	Günther (1865b); Bocage (1866a, 1895, 1897); Loveridge (1933, 1953, 1957); Schiøtz (1975); Perret (1976a); Frost (1985, 2017); Poynton and Broadley (1987); Largen (2001); Ceríaco et al. (2014b, 2016b); This study.
Leptopelis viridis (Günther, 1869)	Bocage (1873, 1895, 1897).
Ptychadenidae	
Genus *Ptychadena* Boulenger, 1917	
Ptychadena anchietae (Bocage, 1867)	Ceríaco et al. (2016b); This study.

TABLE 1. Continued.

Taxon	References
Ptychadena guibei Laurent, 1954	Ceríaco et al. (2016b); This study.
Ptychadena oxyrhynchus (Smith, 1849)	Bocage (1866a, 1895); Boulenger (1882, 1905); Ruas (1996); Ceríaco et al. (2016b); This study.
*Ptychadena porossisim*a (Steindachner 1867)	Bocage (1866a, 1895); Boulenger (1882); Ruas (1996).
Ptychadena subpunctata (Bocage 1866)	Bocage (1886a, 1886b, 1895, 1897); Schmidt and Inger (1959); Perret (1976a); Poynton and Broadley (1985b); Frost (1985, 2017); Ruas (1996).
Ptychadena taenioscelis Laurent, 1954	Poynton and Haacke (1993); Ruas (1996).
Ptychadena uzungwensis (Loveridge, 1932)	Poynton and Haacke (1993).
Phrynobatrachidae	
Genus *Phrynobatrachus* Günther, 1862	
Phrynobatrachus parvulus (Boulenger, 1905)	Boulenger (1905); Loveridge (1933); Marx (1958); Schmidt and Inger (1959); Poynton and Broadley (1985b); Frost (1985, 2017); Ruas (1996).
Phrynobatrachus natalensis (Smith,1849)	Bocage (1866a, 1895); Günther (1865a); Boulenger (1882, 1905); De Witte (1919); Poynton and Haacke (1993); Ruas (1996, 2002); Ceríaco et al. (2014b, 2016b); This study.
Pyxicephalidae	
Genus *Amietia* Dubois, 1987	
Amietia angolensis (Bocage, 1866)	Bocage (1866a, 1866b, 1895, 1897); Loveridge (1933, 1936b; 1953, 1957); Perret (1976a); Poynton and Broadley (1985b); Frost (1985, 2017); Ruas (1996); Largen (2001); Pickersgill (2007), Channing and Baptista (2013); Channing et al. (2016); Bocage (1895); Boulenger (1905); Ruas (1996).
Genus *Tomopterna* Duméril and Bibron, 1841	
Tomopterna tuberculosa (Boulenger, 1882)	Günther (1865a, 1869); Boulenger (1882); Bocage (1895); Loveridge (1957); Schmidt and Inger (1959); Poynton and Broadley (1985b); Ruas (1996).
Dicroglossidae	
Genus *Hoplobatrachus* Peters, 1863	
Hoplobatrachus occipitalis (Günther, 1858)	Bocage (1864, 1866a, 1895); Perret (1976a); Ruas (1996).
Ranidae	
Genus *Amnirana* Dubois, 1992	
Amnirana cf. *darlingi* (Boulenger, 1902)	Ceríaco et al. (2016b); This study.
REPTILES	
TESTUDINES	
Testudinidae	
Genus *Pelomedusa* Cope, 1868	
Pelomedusa subrufa (Bonnaterre, 1789)	Bocage (1895); Monard (1937b); Loveridge (1941a).
Genus *Pelusios* Wagler, 1830	
Pelusios nanus Laurent, 1956	Ceríaco et al. (2004a).
Pelusios rhodesianus Hewitt, 1927	Bocage (1866a, 1866b, 1895); Loveridge (1941b); Broadley (1981); Ceríaco et al. (2014a).
Genus *Kinixys* Bell, 1827	
Kinixys belliana Gray, 1831	Bocage (1866a, 1895); Loveridge and Williams (1957); Ceríaco et al. (2014a, 2016b); This study.
CROCODYLIA	
Crocodylidae	
Genus *Crocodylus* Laurenti, 1768	
Crocodylus niloticus Laurenti, 1768	Günther (1865a); Ferreira (1903); Ceríaco et al. (2014a).
SQUAMATA	
Gekkonidae	
Genus *Chondrodactylus* Peters, 1870	
Chondrodactylus pulitzerae (Schmidt, 1933)	Ceríaco et al. (2014a).
Genus *Hemidactylus* Oken, 1817	
Hemidactylus longicephalus Bocage, 1873	Bocage (1895, 1897); Loveridge (1947); Boulenger (1885); Loveridge (1947); Ceríaco et al. (2014a).
Hemidactylus mabouia (Moreau De Jonnés, 1818)	Bocage (1895); Boulenger (1905); Loveridge (1947); Ceríaco et al. (2016b); This study.

Table 1. Continued.

Taxon	References
Hemidactylus cf. *muriceus* Peters, 1870	Ceríaco et al. (2016b); This study.
Genus *Lygodactylus* Gray, 1864	
Lygodactylus angolensis Bocage, 1896	Ceríaco et al. (2016b); This study.
Amphisbaenidae	
Genus *Monopeltis* A. Smith, 1848	
Monopeltis welwitschii (Gray, 1865)	Gray (1865a, 1865b); Bocage (1895, 1897); Loveridge (1941b); Gans (1967, 2005); Broadley et al. (1976).
Lacertidae	
Genus *Ichnotropis* Peters, 1854	
Ichnotropis bivittata Bocage, 1866	Bocage (1866a, 1895), Boulenger (1887, 1905, 1921); Ferreira (1903); Loveridge (1933, 1957); Bauer et al. (1995); Ceríaco et al. (2016b); This study.
Gerrhosauridae	
Genus *Gerrhosaurus* Wiegmann, 1828	
Gerrhosaurus multilineatus Bocage, 1866	Bocage (1866a, 1866b); Peters (1881); Ceríaco et al. (2016b); This study.
Gerrhosaurus cf. *nigrolineatus* Hallowell, 1857	Bocage (1895); Ferreira (1903); Boulenger (1905); Bocage (1895); Ceríaco et al. (2014b).
Scincidae	
Genus *Lubuya* Horton, 1972	
Lubuya ivensii (Bocage, 1879)	Bocage (1879a); Laurent (1964a); Branch and Haagner (1993); Wagner et al. (2012).
Genus *Panaspis* Cope, 1868	
Panaspis cabindae (Bocage, 1866)	Ceríaco et al. (2016b); This study.
Genus *Sepsina* Bocage, 1866	
Sepsina angolensis Bocage, 1866	Bocage (1866a,b, 1867, 1895, 1897); Boulenger (1905).
Genus *Trachylepis* Fitzinger, 1843	
Trachylepis acutilabris (Peters,1862)	Bocage (1895).
Trachylepis affinis (Gray, 1838)	Ferreira (1903).
Trachylepis bayonii (Bocage, 1872)	Bocage (1866a, 1872, 1895, 1897); Boulenger (1887, 1905); Bauer et al. (2003); This study.
Trachylepis bocagii (Boulenger, 1887)	Bocage (1866a, 1872, 1895); Boulenger (1887); Brygoo (1895); Bauer et al. (2003).
Trachylepis maculilabris (Gray, 1845)	Peters (1879); Bocage (1895); Loveridge (1957); Bauer et al. (2003); Ceríaco et al. (2014a).
Trachylepis cf. *megalura* (Peters, 1878)	Ceríaco et al. (2016b); This study.
Trachylepis quinquetaeniata (Lichtenstein, 1823)	Boulenger (1905).
Trachylepis wahlbergii (Peters, 1870)	Bocage (1866a, 1895); Ceríaco et al. (2016b); This study.
Trachylepis cf. *varia* (Peters, 1867)	Boulenger (1905); Ceríaco et al. (2016b); This study.
Varanidae	
Genus *Varanus* Merrem, 1820	
Varanus niloticus (Linnaeus, 1758)	Bocage (Bocage 1879b, 1895); Mertens (1942); Manaças (1955); Bayles (2002); Ceríaco et al. 2014a, 2016b); This study.
Chamaeleonidae	
Genus *Chamaeleo* Laurenti, 1768	
Chamaeleo dilepis Leach, 1819	Bocage (1895); Ceríaco et al. (2016b); This study.
Chamaeleo gracilis etiennei Schmidt, 1919	Bocage (1866a, 1895), Boulenger (1887, 1905); Ferreira (1904); Parker (1936); Ceríaco et al. (2014b).
Agamidae	
Genus *Agama* Daudin, 1802	
Agama aculeata Merrem, 1820	Bocage (1866a; 1895; 1896).
Agama congica Peters, 1877	Bocage (1895); Hellmich (1957a); Ceríaco et al. (2014a).
Agama planiceps schacki Mertens, 1938	Boulenger (1885); Ferreira (1903); Boulenger (1905); Manaças (1963).

Table 1. Continued.

Taxon	References
Genus *Acanthocercus* Fitzinger, 1843	
Acanthocercus cyanocephalus (Falk, 1925)	Peters (1881); Bocage (1866a, 1895); Boulenger (1885; 1905); Ferreira (1903); Monard (1937b); Ceríaco et al. (2014a, 2016b); This study.
Typhlopidae	
Genus *Afrotyphlops* Broadley and Wallach, 2009	
Afrotyphlops angolensis (Bocage, 1866)	Bocage (1866a, 1866b, 1873, 1879b); Loveridge (1957); Laurent (1964b); Roux-Estève (1974a,b); Broadley and Wallach (2009); Wallach et al. (2014).
Afrotyphlops lineolatus (Jan, 1864)	Bocage (1873, 1895); Peters (1881); Monard (1937b); Bocage (1895); Monard (1937b).
Leptotyphlopidae	
Genus *Leptotyphlops* Fitzinger, 1843	
Leptotyphlops scutifrons (Peters, 1854)	Bocage (1866a, 1873, 1895); Monard (1937b).
Genus *Namibiana* Hedges, Adalsteinsson and Branch, 2009	
Namibiana rostrata (Bocage, 1886)	Broadley and Broadley (1999).
Viperidae	
Genus *Bitis* Gray, 1842	
Bitis arietans (Merrem, 1820)	Bocage (1866a, 1895); Monard (1937b); Manaças (1981); Ceríaco et al. (2016b).
Genus *Causus* Wagler, 1830	
Causus bilineatus Boulenger, 1905	Bocage (1866a, 1895); Monard (1937b); Manaças (1981); Rasmussen (2005).
Causus rhombeatus (Lichtenstein, 1823)	Günther (1865b); Peters (1881); Bocage (1895); Boulenger 1905; Monard (1937b); Manaças (1982); Rasmussen (2005); Ceríaco et al. (2016b).
Pythonidae	
Genus *Python* Daudin, 1803	
Python sebae (Gmelin, 1789)	Ceríaco et al. (2016b).
Atractaspidae	
Genus *Atractaspis* Smith, 1849	
Atractaspis congica Peters, 1877	Boulenger (1905); Laurent (1950b); Manaças (1982).
Atractaspis irregularis (Reinhardt, 1834)	Günther (1865b).
Genus *Polemon* Jan, 1858	
Polemon collaris (Peters, 1881)	Peters (1881); De Witte and Laurent (1947); Hellmich (1957a); Chippaux (2006); Chirio and LeBreton (2007); Wallach et al. (2014).
Genus *Xenocalamus* Günther, 1868	
Xenocalamus mechowii Peters, 1881	Peters (1881); De Witte and Laurent (1947); Chippaux (2006).
Lamprophiidae	
Genus *Boaedon* Duméril, Bibron and Duméril, 1854	
Boaedon cf. *angolensis*	Bocage (1866a, 1895); Monard (1937b); Günther (1865b); Boulenger (1893, 1905); Monard (1937b); Ceríaco et al. (2016); This study.
Genus *Lycophidion* Fitzinger, 1843	
Lycophidion multimaculatum Boettger, 1888	Bocage (1866a, 1895); Boulenger (1893); Broadley (1996b).
Genus *Psammophis* Boie, 1825	
Psammophis angolensis (Bocage, 1872)	Peters (1877, 1881); Bocage (1895, 1897); Loveridge (1940, 1957);; Broadley (1977b, 2002).
Psammophis mossambicus Peters, 1882	Boulenger (1905); Loveridge (1940); Broadley (2002); Ceríaco et al. (2016b).
Genus *Psammophylax* Fitzinger, 1843	
Psammophylax acutus (Günther, 1888)	Günther (1865b; 1888, 1895); Bocage (1895); Boulenger (1896); Loveridge (1933); Monard (1937b); Broadley (1971); Chirio and Ineich (1991); Chippaux (2006); Wallach et al. (2014).
Genus *Prosymna* Gray, 1849	
Prosymna ambigua (Bocage, 1873)	Bocage (1866a, 1873, 1895); Loveridge (1933, 1958); Monard (1937b); Broadley (1980); Chippaux (2006); Wallach et al. (2014); Ceríaco et al. (2016b); This study.
Elapidae	
Genus *Dendroaspis* Schlegel, 1848	
Dendroaspis jamesoni (Traill, 1843)	Bocage (1895).
Dendroaspis polylepis Günther, 1864	Peters (1881); Ceríaco et al (2016b).

Table 1. Continued.

Taxon	References
Genus *Naja* Laurenti, 1768	
Naja (*Afronaja*) *nigricollis* Reinhardt, 1843	Peters (1881); Bocage (1895); Manaças (1981).
Naja (*Boulengerina*) *melanoleuca* Hallowell, 1857	Bocage (1866a, 1895); Ferreira (1900); Boulenger (1905); Manaças (1982).
Naja (*Uraeus*) *anchietae* Bocage, 1879	Ceríaco et al. (2014a, 2016b).
Colubridae	
Genus *Crotaphopeltis* Fitzinger, 1843	
Crotaphopeltis hotamboeia (Laurenti, 1768)	Bocage (1866a); Boulenger (1906); Ceríaco et al. (2016b); This study.
Genus *Dasypeltis* Wagler, 1830	
Dasypeltis palmarum (Leach, 1818)	Günther (1865b); Boulenger (1905); Monard (1937b); Gans (1959); Manaças (1973).
Dasypeltis scabra (Linnaeus, 1758)	Ceríaco et al. (2016b); This study.
Genus *Dispholidus* Duvernoy, 1832	
Dispholidus typus (Smith, 1828)	Bocage (1866a, 1895); Peters (1881); Monard (1937b); Ceríaco et al. (2016b); This study.
Genus *Grayia* Günther, 1858	
Grayia ornata (Bocage, 1866)	Bocage (1866a, 1866b, 1895, 1897); Loveridge (1936a); Broadley (1983); Chippaux (2006); Chirio and LeBreton (2007); Wallach et al. (2014).
Genus *Philothamnus* Smith, 1840	
Philothamnus dorsalis (Bocage, 1866)	Bocage (1866b, 1882, 1895); Loveridge (1933)..
Philothamnus heterolepidotus (Günther, 1863)	Bocage (1866a, 1866b, 1882a, 1895); Boulenger (1905); Monard (1937b).
Philothamnus irregularis (Leach, 1819)	Günther (1865b); Bocage (1866a, 1882, 1895); Ferreira (1906); Monard (1937b).
Genus *Thelotornis* A. Smith, 1849	
Thelotornis kirtlandii (Hallowell, 1844)	Bocage (1866a, 1895); Loveridge (1944).
Natricidae	
Genus *Limnophis* Günther, 1865	
Limnophis bicolor Günther, 1865	Günther (1865a); Bocage (1866a, 1866b, 1895, 1897a, 1879a); Mertens (1963).
Genus *Natriciteres* Loveridge, 1953	
Natriciteres bipostocularis Broadley, 1962	Peters (1882); Peters (1895); Broadley (1966).
Natriciteres olivacea (Peters, 1854)	Bocage (1895); Boulenger (1905); Broadley (1966).

Peters (1881) described *Xenocalamus mechowii* and *Microsoma collare,* and later (Peters 1882a) *Hyperolius vermiculatus* (currently considered a member of the *Hyperolius angolensis* species complex) from Malanje.

At the beginning of the 20th century, the Portuguese explorer Francisco Newton (1864–1909) was hired by the Polytechnic Academy of Porto to lead an expedition to Angola, and from 1903 to 1905 he explored several provinces of Angola, including certain areas in Malanje. His collections were studied and partially published by Portuguese naturalist José Júlio Bettencourt Ferreira (1866–1948) in two papers (Ferreira 1904, 1906) resulting in the description of nine amphibians and one reptile, some of them particularly problematic (Ceríaco et al. 2014b). At about the same time, William John Ansorge (1850–1913), who explored the Congo Basin, also collected extensively in Angola, including in Malanje Province, from 1903 to 1905. Ansorge's collections were sent to the British Museum (Natural History) where they were studied and published on by Boulenger (Boulenger 1905, 1907a,b, 1915), although only the 1905 paper mentioned Malanje specimens.

After the discovery and description of the endemic Giant Sable (Thomas 1916), the Province became of great interest to foreign museums and institutions that planned expeditions to Malanje to collect specimens of the sable. Surprisingly, however, almost no published herpetological records exist for the province during the rest of the 20th century. In 1925, Arthur S. Vernay (1877–1960) organized an expedition to Angola to collect zoological specimens for the AMNH, with the special aim of collecting specimens of Giant Sable. A large collection of amphibians and reptiles resulted from this expedition, of which only the snakes were studied and published on by Charles M. Bogert (1908–1992) (Bogert 1940). Although the expedition explored Malanje, no data regarding amphibians or reptiles of the province were ever published, and Bogert's (1940) paper on the snakes did not include any material from Malanje. In August 1930, Harold T. Green (1896–1967) led a three-month expedition to Malanje Province promoted by the Academy of Natural Sciences of Philadelphia (ANSP), again to collect Giant Sable, and some reptiles and amphibians were collected. This small

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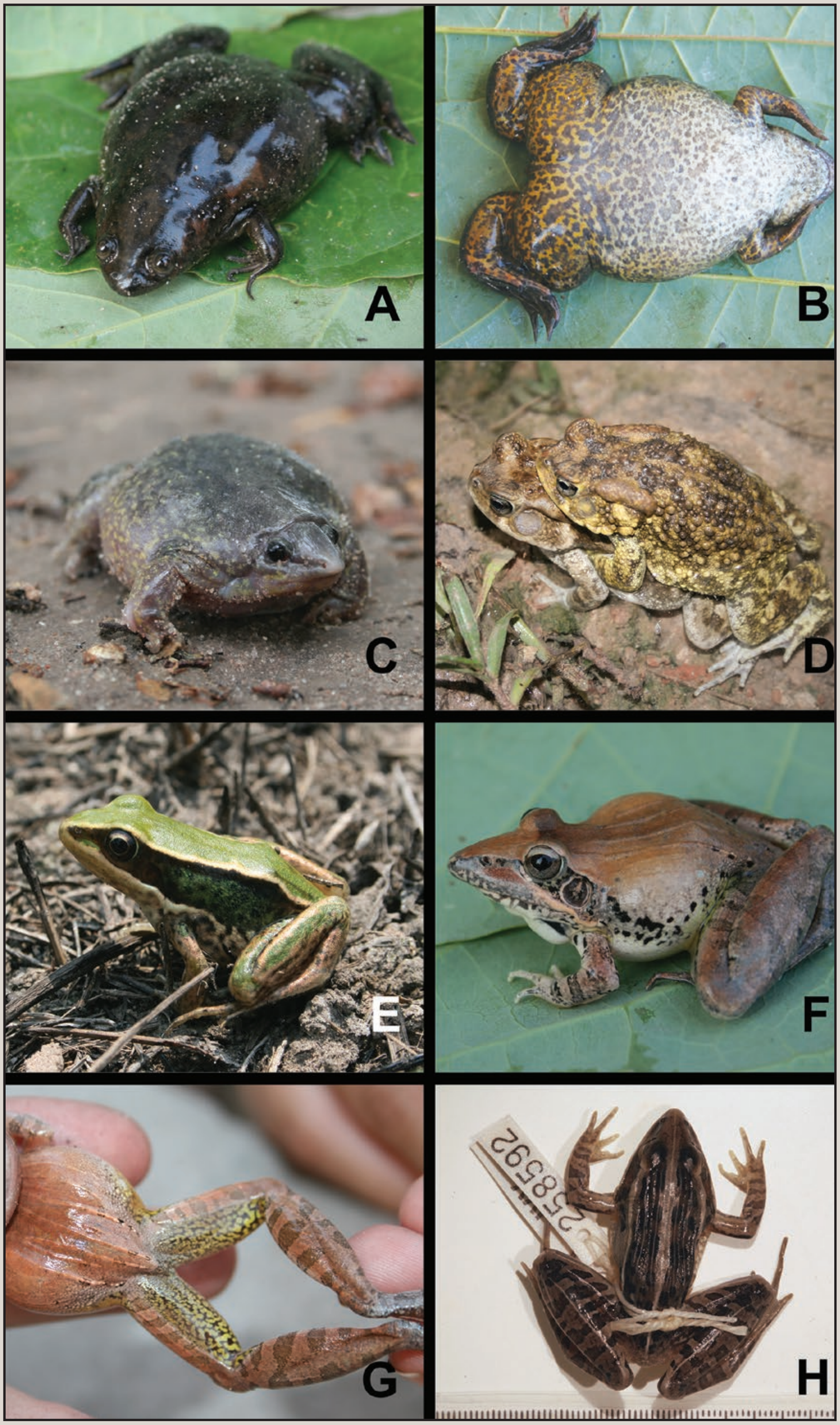

Fig. 2. A) Peters' Platanna, *Xenopus petersii* Bocage, 1895; B) Ventral pattern of Peters' Platanna, *Xenopus petersii* Bocage, 1895; C) Guinean Snout-Burrower, *Hemisus guineensis* Cope, 1865; D) Mertens' Striped Toad, *Sclerophrys pusilla* (Mertens, 1937); E) Darling's Frog, *Amnirana* cf. *darlingi* (Boulenger, 1902); F) Anchieta's Ridged Frog, *Ptychadena anchietae* (Bocage, 1868); G) Thigh coloration and pattern of Anchieta's Ridged Frog, *Ptychadena anchietae* (Bocage, 1868); H) Guibe's Ridged Frog, *Ptychadena guibei* Laurent, 1954. All photos from Cangandala National Park.

collection, still extant in the ANSP, was likewise never published upon. Gert Hermann Heinrich (1896–1984), led the Conover Angola Expedition sponsored by the Chicago Natural History Museum (now Field Museum of Natural History, FMNH), which explored different areas in Angola, including Malanje, from 1953 to 1955. A total of 560 herpetological specimens were collected but, with the exception of some lacertids studied by Marx (1956), the collection was never properly studied.

Possibly the first herpetological collection made in Malanje following the end of the Angolan Civil War was that based on an Environmental Impact Assessment (EIA) associated with the construction of the Capanda Dam. From January to April 2003, a team comprising researchers from the Museu Nacional de História Natural of Luanda (MNHNL), the Gabinete de Aproveitamento do Médio Kwanza (GAMEK), and private EIA contractors, collected material in the area to be flooded by the dam. Those specimens were deposited in the collections of the MNHNL and studied by Ceríaco et al. (2014a), resulting in considerable range extensions for several species. Recent surveys (July 2016, March 2017) in the area influenced by the Laúca Hydroelectric Dam, on the Kwanza River, in the southwest of the Province, have also resulted in important new collections, with additions of species new to the Province, the country and for science (Marques et al., unpubl.).

Materials and Methods

We conducted herpetological surveys in Malanje Province from 12–30 September 2015 in Cangandala National Park and nearby areas. In each area, we attempted to opportunistically sample a diversity of habitat types, during both day and night. We captured specimens using nooses and rubber bands, or by hand during both diurnal and nocturnal surveys. All specimens were euthanized following an approved IACUC protocol (CAS #2014-2), preserved in 10% buffered formalin in the field, and then transferred to 70% ethanol for storage. Liver tissues were preserved in 95% ethanol and RNALater. Voucher specimens and tissue samples are deposited in the herpetological collection of the California Academy of Sciences, with a subset of specimens housed at INBAC in Kilamba, Angola (pending cataloging and accession numbers). In some cases, we further confirmed species identifications by sequencing the mitochondrial 16S ribosomal RNA gene. A complete list of all amphibian and reptile species reported from Malanje Province was also assembled (Table 1). This list, including localities and associated bibliographic references was based on the ongoing project for the first atlas of the Angolan amphibians and reptiles (Marques et al., *in press.* These, however, include only published records and do not take into account specimens housed in different museums that have not been published upon.

Results

A total of 460 specimens were collected during the expedition, representing 10 amphibian genera and 14 reptile genera. Many of the species collected likely represent complexes of species, including undescribed taxa. Molecular phylogenetic and systematic studies that are in progress will likely result in taxonomic revisions and description of new species. In the following species accounts, we provide information on CAS voucher specimens, localities, and natural history. Latitude, longitude (in decimal degrees) and elevation (in meters) of the collection site are provided in each species account. In addition, when appropriate, we provide brief taxonomic or geographic notes.

SPECIES ACCOUNTS

Amphibia
Anura
Pipidae
***Xenopus petersii* Bocage, 1895**
PETERS' PLATANNA – Fig. 2A,B

Material.—**53 specimens:** CNP, Park Headquarters [-9.81942°, 16.65386°, 1089 m]: CAS 258438–40; CNP**,** along park entrance road, 200 m W of park headquarters [-9.81978°, 16.65197°, 1085 m]: CAS 258441; CNP**,** along park entrance road [-9.81894°, 16.64906°, 1088 m]: CAS 258442–52; 1.4 km NW (by air) of CPN, Park Headquarters [-9.81161°, 16.64381°, 1082 m]: CAS 258453–57, 258477; CNP, pond along entrance road [-9.81944°, 16.64942°, 1100 m]: CAS 258458–76; CNP [9.82689°, 16.65003°, 1100 m]: CAS 258477–83, [-9.81986°, 16.65267°, 1079 m]: CAS 258484–88, 260965.

Comments.—A recent molecular phylogeny by Furman et al. (2015) clarified the distribution of the various lineages presented within the *X. laevis* group. This led to the recognition of *X. laevis* sensu stricto, *X. petersii*, *X. victorianus* Ahl, 1924 and *X. poweri* Hewitt, 1927 as full species, and the western Central African populations (including the central and western Angolan populations) as *X. petersii.* All of our specimens were collected in natural and artificial ponds, where they occurred in high densities. The species is already known from several localities in Malanje Province (see Marques et al., *in press*).

Hemisotidae
***Hemisus guineensis* Cope, 1865**
GUINEAN SNOUT-BURROWER – Fig. 2C

Material.—**17 specimens:** CNP, Park Headquarters [-9.81942°, 16.65386°, 1089 m]: CAS 258489–12; CNP [-9.81867°, 16.65511°, 1091 m]: CAS 258513–28, [-9.81783°, 16.65747°, 1097 m]: CAS 258529–32, [-9.81864°, 16.65536°, 1082 m]: CAS 258533–36, [-9.81914°, 16.65453°, 1084 m]: CAS 258537-39, [-9.81914°, 16.65453°, 1084 m]: CAS 258540–41.

Comments.—*Hemisus guineensis* is part of a larger complex of cryptic species and the patterns of variation across Africa remain poorly documented (Onadeko and Rödel 2009; Rödel and Ernst 2003). We identify CNP material as *H. guineensis* based on the speckled dorsal pattern (marbled in *marmoratus*, see Channing 2001). This is the first report of the genus for Malanje Province, although records of both *H. guineensis* and *H. marmoratus* exist for the neighboring provinces and generally across Angola (Marques et al., *in press*).

Bufonidae
***Sclerophrys pusilla* (Mertens, 1937)**
MERTENS' STRIPED TOAD – Fig. 2D

Material.—**18 specimens:** CNP [-9.82828°, 16.64381°, 1082 m]: CAS 258571, [-9.81936°, 16.65439°, 1089 m]: CAS 258572–74, [-9.86900°, 16.69131°, 1092 m]: CAS 258575, [-9.81758°, 16.65878°, 1099 m]: CAS 258576, [-9.81914°, 16.65453°, 1084 m]: CAS 258577, [-9.81961°, 16.64908°, 1075 m]: CAS 258578, [-9.81865°, 16.65536°, 1082 m]: CAS 258579, [-9.81933°, 16.65403°, 1097 m]: CAS 258580-82, [-9.73281°, 16.78794°, 1143 m]: CAS 258583, [-9.72261°, 16.77703°, 1117 m]: CAS 258584; CNP, Vicinity of

Park Headquarters [-9.81917°, 16.65436°, 1089 m]: CAS 258585; Right bank of Kwanza River [-9.88478°, 16.28628°, 1013 m]: CAS 258586; Camassa west side of the highway, near the road to Kwanza bridge [-9.87622°, 16.28694°, 1058 m]: CAS 258587.

Comments.—Poynton et al. (2016) restricted *S. maculata* (Hallowell, 1854) to western African from Senegal to western Cameroon, and suggested that *S. pusilla* represents other populations previously assigned to *S. maculata* found in eastern and southern Africa, including Angola. The species is already known from Malanje Province (see Table 1) and has a wide distribution across the central-south regions of the country (Marques et al., *in press*).

Ranidae

***Amnirana* cf. *darlingi* (Boulenger, 1902)**

DARLING'S FROG – Fig. 2E

Material.—**4 specimens:** CNP [-9.72133°, 16.77725°, 1013 m]: CAS 258627, [-9.72161°, 16.77719°, 1126 m]: CAS 258630–32.

Comments.—*Amnirana darlingi* was originally described by Boulenger (1902) based on material from Zimbabwe. The taxonomy of this species has been mostly stable since its description (apart from generic allocation, see Olivier et al. 2015), but Channing (2001) suggested that specimens from northeast Zambia, morphologically similar to our material, may represent a different species based on differences in coloration and size. While molecular studies are currently assessing the specific identity of the Angolan populations of *A. darlingi* (G. Jongsma, pers. comm.), it is possible that the name *Rana albolabris adiscifera* may be available for it. This taxon was erected by Schmidt and Inger (1959) based on specimens from Chitau (Bié Province, less than 200 km south of Cangandala) and later synonymized with *Rana darlingi* (Boulenger, 1902) by Laurent (1964a). This is the first record of this species for Malanje Province.

Ptychadenidae

***Ptychadena anchietae* (Bocage, 1868 "1867")**

ANCHIETA'S RIDGED FROG – Fig. 2F,G

Material.—**5 specimens:** CNP, along park entrance road, ca 200 m W of park headquarters [-9.81978°, 16.65197°, 1085 m]: CAS 258588; road from Cangandala to CNP [-9.81814°, 16.61989°, 1077 m]: CAS 258591; CNP, pond along entrance road [-9.81944°, 16.64942°, 1100 m]: CAS 258597–99.

Comments.—Originally described based on specimens from "Benguella," and sometimes confused with *P. oxyrhyncus* and *P. mascarienensis*, its validity was established by Perret (1976a) and Poynton and Broadley (1985b). As currently recognized, the species extends through central and southern Africa to Kenya and Somalia and may contain cryptic species diversity (Bwong et al. 2009; Dehling and Sinsch 2013). The specimens collected in Cangandala National Park, represent the first record for Malanje Province, although specimens are known from other localities in Angola (see Marques et al., *in press*). Our material was found in syntopy with *P. oxyrhynchus* (Smith, 1849) (see below) in small natural and artificial ponds.

***Ptychadena guibei* Laurent, 1954**

GUIBE'S RIDGED FROG – Fig. 2H

Material.—**2 specimens:** CNP, Cuqui River [-10.02128°, 16.71292°, 1047 m]: CAS 258592; CNP [-9.72133°, 16.71058°, 1013 m]: CAS 258626.

Comments.—Originally described as *Ptychadena chrysogaster guibei* by Laurent (1954a) from "Muita (Luembe E)" in Lunda Norte Province, Angola, it was later elevated to full species by Poynton and Broadley (1985b). The two specimens collected in this survey represent the first record for the province and the westernmost records of the species in the country. To date, the species is otherwise only known from two localities in Lunda Norte Province, and one locality in Moxico Province (Laurent 1950a, 1954a, 1964a; Poynton and Broadley 1985b; Ruas 1996; Schmidt and Inger 1959; and see also Marques et al., *in press*).

***Ptychadena oxyrhynchus* (Smith, 1849)**

SHARP-NOSED RIDGED FROG – Fig. 3A

Material.—**8 specimens:** Road from Cangandala to CNP [-9.81814°, 16.61989°, 1077 m]: CAS 258589; CNP, pond along entrance road [-9.81944°, 16.64942°, 1100 m]: CAS 258600–02; CNP [-9.82689°, 16.65003°, 1089 m]: CAS 2586005, [-9.81956°, 16.64925°, 1013 m]: CAS 2586023–25.

Comments.—This species is widely distributed across sub-Saharan Africa in areas of moist savanna, circum-forest savana, in secondary vegetation with tall herbaceous vegetation, and in marshy and agricultural areas (Ruas 1996; Schmidt and Inger 1959). It is relatively common in Angola, except in the arid and semiarid areas in southern regions of the country, and it is known from Pungo Andongo and Duque de Bragança (currently Kalandula) in Malanje Province (see Marques et al., *in press.*). The species strongly resembles *P. anchietae* and *P. mascarienensis* (Duméril and Bibron, 1841) with which it has been confused in the past but can be distinguished from those species by its distinct coloration and the lack of markings along the jaw and thigh (Perret 1976a).

Hyperoliidae

***Afrixalus wittei* (Laurent, 1941)**

DE WITTE'S SPINY REED FROG

Material.—**1 specimen:** CNP, Cuqui River [-10.02183°, 16.70869°, 1055 m]: CAS 258644.

Comments.—A single specimen collected in a floodplain near the Cuqui River represents the second record for Angola. The only previous record is based on a specimen from "Duque de Bragança" (currently Kalandula) in Malanje Province that was originally identified as *Hyperolius fulvovittatus* by Bocage (1866a) and later corrected to *Afrixalus wittei* by Perret (1976b). The species is known from tropical lowland savannas from the southern DRC east to Zambia and is also expected to occur in eastern Angola (Channing 2001).

***Hyperolius angolensis* Steindachner, 1867**

ANGOLAN REED FROG – Fig. 3B

Material.—**4 specimens:** CNP, vicinity of park headquarters [-9.81810°, 16.65535°, 1092 m]: CAS 258645–48.

Comments.—This species was initially described by Steindachner (1867) based on a single specimen from "Angola," restricted to "Duque de Bragança" (currently Kalandula), in Malanje Province by Ceríaco et al. (2014a). The Angolan Reed Frogs have a complex taxonomic and nomenclatural history, being identified by dozens of names and combinations during the last 150 years (see Marques et al., *in press* for a complete list of the chrysonyms used for Angolan material). Although *Hyperolius parallelus* Günther, 1858 appears to be an earlier name for this taxon, the type series contains material from both South Africa and Angola (see Günther 1859 "1858"). It is unlikely that the material from South Africa is conspecific with that from Angola. Instead, it may belong to *Hyperolius marmoratus* Rapp,

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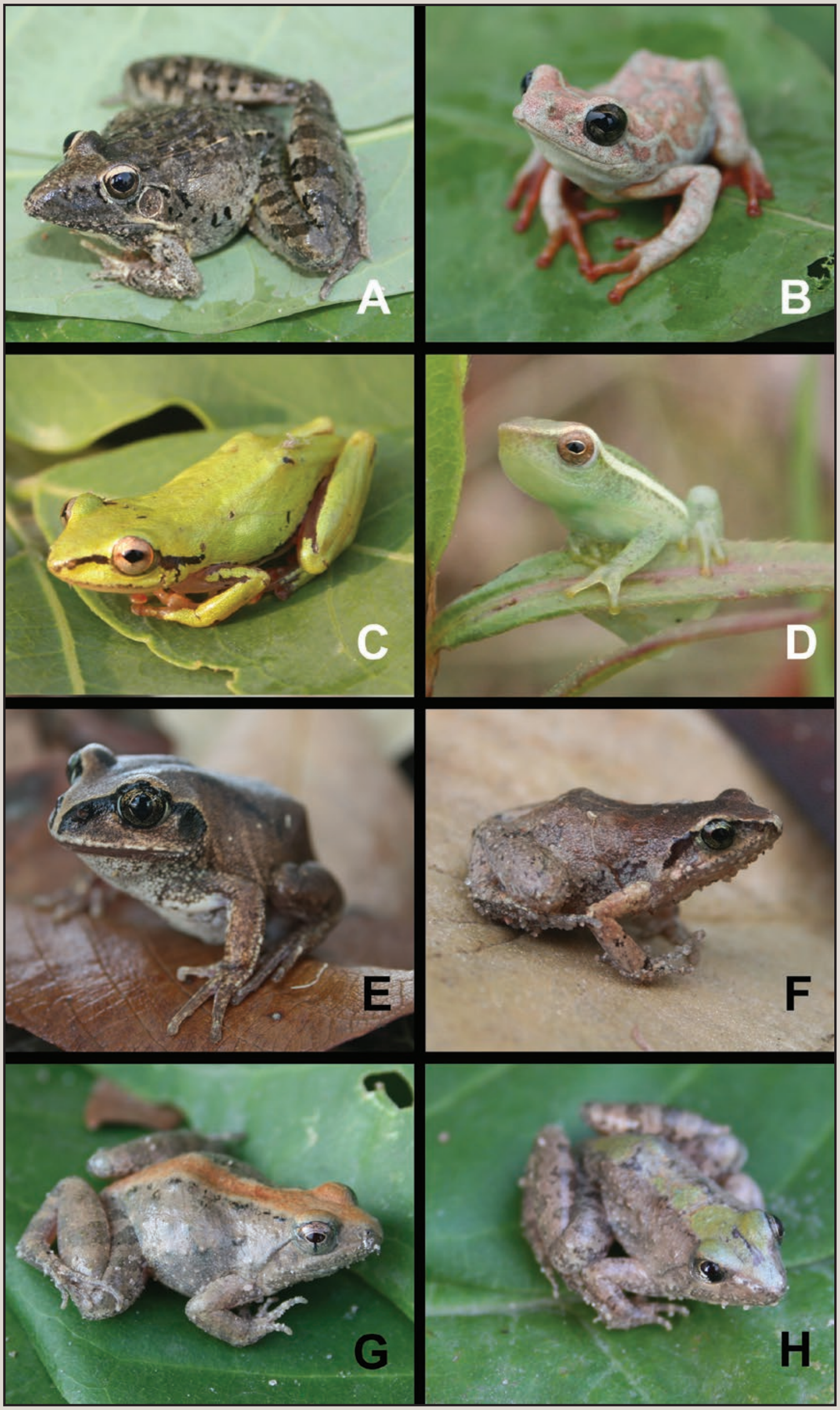

FIG. 3. A) Sharp-nosed Ridged Frog, *Ptychadena oxyrhynchus* (Smith, 1849); B) Angolan Reed Frog, *Hyperolius angolensis* Steindachner, 1867; C) Cinnamon-bellied Reed Frog, *Hyperolius cinnamomeoventris* Bocage, 1866; D) Long-Nosed Reed Frog, *Hyperolius nasutus* Günther, 1865; E) Bocage's Tree Frog, *Leptopelis bocagii* (Günther, 1865); F) Plain Squeaker, *Arthroleptis xenochirus* Boulenger, 1905; G) Orange morph of Natal Dwarf Puddle Frog, *Phrynobatrachus natalensis* (Smith, 1849); H) Green morph of Natal Dwarf Puddle Frog, *Phrynobatrachus natalensis* (Smith, 1849). All photos from Cangandala National Park.

1842, which suggests that a review of the original type material is necessary. We follow our previous interpretations (Ceríaco et al. 2014a, Marques et al., *in press*) by continuing to recognize *H. angolensis*.

***Hyperolius cinnamomeoventris* Bocage, 1866**
CINNAMON-BELLIED REED FROG – Fig. 3C

Material.—**2 specimens:** CNP [-9.72242°, 16.77778°, 1140 m]: CAS 258631, [-9.81161°, 16.64381°, 1082 m]: CAS 258636.

Comments.—Long known to refer to a species complex (Bell et al. 2015, 2017; Lötters et al. 2004; Schick et al. 2010), *H. cinnamomeoventris* was originally described from "Duque de Bragança," (currently Kalandula) in Malanje Province (Bocage 1866b). This is the same locality from which Bocage (1866b) described *Rappia* [=*Hyperolius*] *tristis,* considered a synonym by Laurent (1943, 1947) and Perret (1976). Our material agrees entirely with the original description. The species is widespread throughout the neighboring provinces of Lunda Norte, Kwanza-Norte, Kwanza-Sul, and Benguela, and other records for the province exist (see Marques et al., *in press*). Recent work by Bell et al. (2017) suggests that further revision of this species complex may be necessary.

***Hyperolius nasutus* Günther, 1865**
LONG-NOSED REED FROG – Fig. 3D

Material.—**20 specimens:** Cangandala National Park, along park entrance road, ca 200 m W of park headquarters,[-9.81978°, 16.65197°, 1084 m]: CAS 258649–67; Cangandala National Park, along park entrance road [-9.81894°, 16.66558°, 1088 m]: CAS 258669.

Comments.—Originally described by Günther (1865a) based on one specimen from "Duque de Bragança" (currently Kalandula) in Malanje Province, these small frogs form a species complex with several associated names (see Amiet 2005 and Channing et al. 2002). A recent revision of the complex by Channing et al. (2013) based on molecular and morphological data and advertisement calls, included specimens of the nominotypical form collected previously in the CNP, and suggested that nominotypical *nasutus* is mostly confined to northern Botswana and northern Angola. Some of the specimens collected were young juveniles and were mostly collected in swampy areas. The species is known from the surrounding provinces, but also from Huambo, Benguela, Huíla, Cunene, and Cuando-Cubango (see Marques et al., *in press*).

Arthroleptidae
***Leptopelis bocagii* (Günther, 1865)**
BOCAGE'S TREE FROG – Fig. 3E

Material.—**43 specimens:** CNP [-9.81850°, 16.65556°, 1105 m]: CAS 258542–47, [-9.81783°, 16.65747°, 1097 m] – CAS 258543–44, [-9.81956°, 16.66594°, 1073 m]:CAS 258548–52, [-9.81928°, 16.65403°, 1101 m]: CAS 258553–54, 258558, [-9.81864°, 16.65536°, 1082 m]:CAS 258555–57, [-9.81889°, 16.65369°, 1084 m]: CAS 258559–61, [-9.81942°, 16.65492°, 1101 m]: CAS 258562–64, [-9.81933°, 16.65403°, 1097 m]: CAS 258565–67, [-9.81969°, 16.65206°, 1092 m]: CAS 25856–70.

Comments.—*Leptopelis bocagii* was described by Günther (1865a) based on a specimen from "Duque de Bragance" [= Duque de Bragança] (currently Kalandula) in Malanje Province. This species is widespread in southern and central Africa (Channing 2001), but may comprise several cryptic species (Amiet 2012; Largen 1977). Poynton and Broadley (1987) described a similar species, *Leptopelis parbocagii* based on five specimens collected at Mabwe on the eastern shore of Lake Upemba, Zaire, currently the DRC. The distribution of *L. parbocagii* overlaps that of *L. bocagii,* which extends east from Angola and DRC to Malawi, Zambia, and Mozambique, although doubts remain as to differentiating these species (Schiøtz 1999; Schiøtz and Van Daele 2003).

Ceríaco et al. (2014b) reviewed the complex history regarding the original description of *Leptopelis bocagii* and the uncertainties regarding the number and whereabouts of the type material. We subsequently located a specimen of *Leptopelis bocagii* from "Angola" in the collection of the Museé d'Histoire Naturelle de la Ville de Genéve (MHNG), Switzerland, that might be one of the three syntypes of the species, or at least, the second specimen mentioned by Günther in his letters to Bocage (see Ceríaco et al. 2014b). This specimen is morphologically similar to our material from CNP. As already noted by Parker (1936) and Poynton and Broadley (1987), the dorsal pattern is highly variable, even within the specimens collected in Cangandala, and this trait is not sufficient for diagnosis on its own. The species was abundant just after rain on the ground and in vegetation.

***Arthroleptis xenochirus* Boulenger, 1905**
PLAIN SQUEAKER – Fig. 3F

Material.—**12 specimens:** CNP, Park Headquarters [-9.81942°, 16.65386°, 1089 m]: CAS 258612–16; CNP [-9.81942°, 16.65386°, 1089 m]: CAS 258617–22, [-9.81894°, 16.65489°, 1088 m]: CAS 258643.

Comments.—This species is well documented from northeastern Angola (Channing 2001). It is possible that more than one species may occur within the currently recognized range (see Marques et al., *in press*). This species was commonly found on the ground during rains.

Phrynobatrachidae
***Phrynobatrachus natalensis* (Smith, 1849)**
NATAL DWARF PUDDLE FROG – Fig. 3G,H

Material.—**20 specimens:** CNP, Cuqui River [-10.02128°, 16.71292°, 1047 m]: CAS 258593, 258637, [-9.97906°, 16.59694°, 1065 m]: CAS 258594–96; Right bank of Kwanza River [-9.88478°, 16.28628°, 1013 m]: CAS 258606–10; CNP [-9.72133°, 16.71058°, 1013 m]: CAS 258628–29, [-9.81814°, 16.61989°, 1077 m]: CAS 258633–35, [-9.82517°, 16.79942°, 1102 m]: CAS 258640, [-9.72261°, 16.77703°, 1117 m]: CAS 258641–42.

Comments.—*Phrynobatrachus natalensis* is a widespread species in the savanna and grassland regions of sub-Saharan Africa extending across much of southern Africa. Zimkus et al. (2010) and Zikmus and Schick (2010) discussed the nature of nominal *P. natalensis* as a species complex and indicated that this taxon contains multiple undescribed species (Lara 2016). The species was commonly found near the margins of small ponds and river margins, as well on the moist ground after rain.

Reptilia
Testudinidae
***Kinixys belliana* Gray, 1830**
BELL'S HINGE-BACK TORTOISE – Fig. 4A

Material.—**1 specimen:** 13.67 km SW (by road) of Cangandala [-9.84233°, 16.31733°, 1086 m]: CAS 258437.

Comments.—The precise delineation of the distribution of *Kinixys* species in Angola remains unclear, and genetic data will be necessary to establish species boundaries (U. Fritz, pers.

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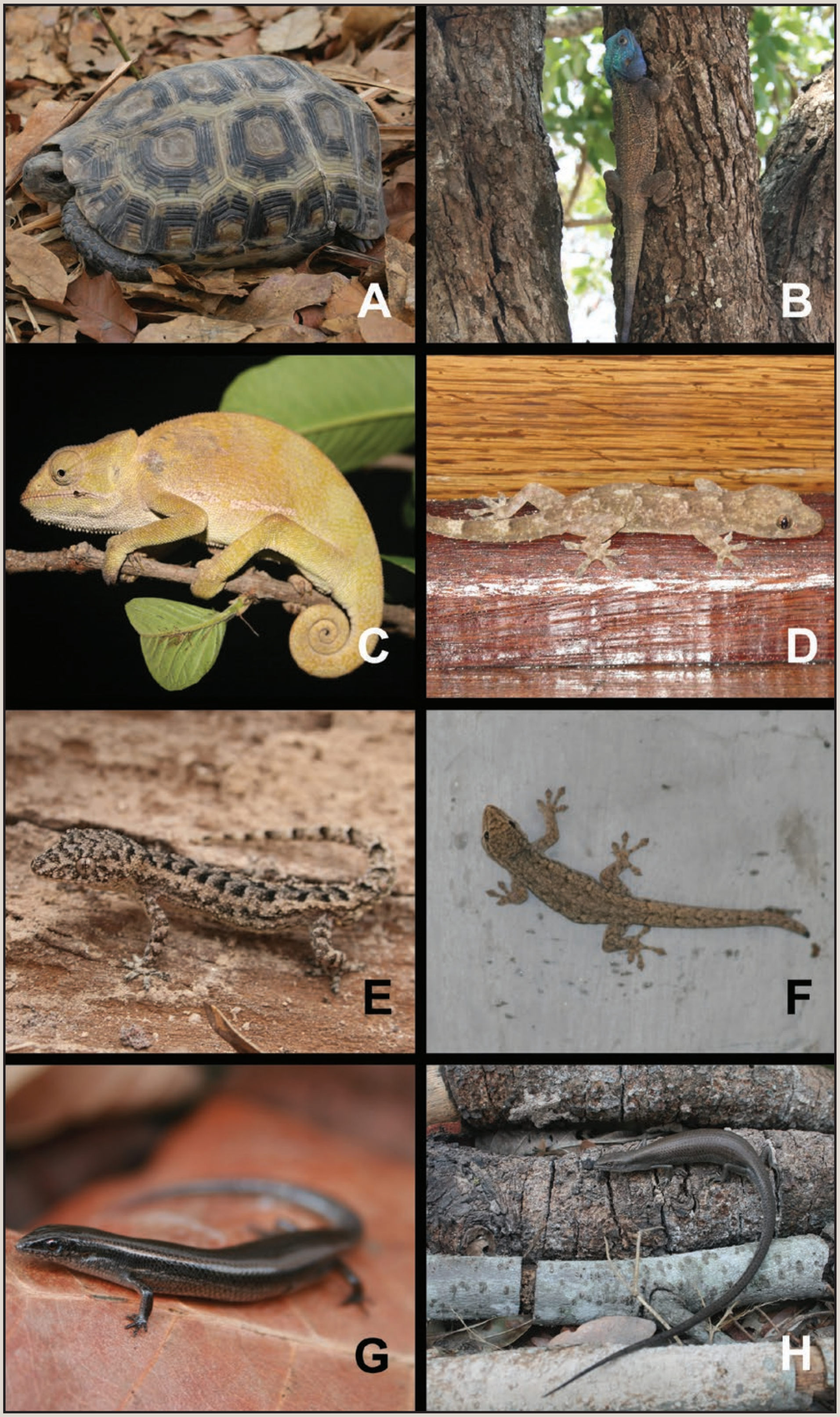

FIG. 4. A) Bell's Hinge-back Tortoise, *Kinixys belliana* Gray, 1830; B) Male Blue-headed Ridgeback Agama, *Acanthocercus cyanocephalus* (Falk, 1925); C) Common African Flap-necked Chameleon, *Chamaeleo dilepis* (Leach, 1819); D) Tropical House Gecko, *Hemidactylus mabouia* (Moreau de Jonnés, 1818); E) Guinea Split-toed Gecko, *Hemidactylus* cf. *muriceus* Peters, 1870; F) Angolan Dwarf Gecko, *Lygodactylus angolensis* Bocage, 1896; G) Cabinda Snake-eyed Skink, *Panaspis cabindae* (Bocage, 1866); H) Long Tailed Skink, *Trachylepis* cf. *megalura* (Peters, 1878). All photos from Cangandala National Park.

comm.). Doubts have existed regarding how many species of this genus occur in the country and which is their distribution within Angolan borders, with several authors presenting contradictory interpretations (Branch 2008; Fritz and Havaš 2007; Mifsud and Stapleton 2014; Turtle Taxonomy Working Group 2017; Vetter 2011); for a more detailed review on the topic, see Marques et al., *in press*). However, Kindler et al. (2012) provided molecular evidence that *K. belliana* comprises at least three deeply divergent lineages and considered the Angolan populations to represent the nominotypic form.

Ceríaco et al. (2016a) considered the Cangandala *Kinixys* to belong to *K. spekii*. However, we now tentatively assign the Cangandala population to *K. belliana* (U. Fritz, pers. comm.). Genetic analyses will facilitate comparison of this material to other known lineages (Fritz et al., unpubl.). During our field survey, we encountered poachers with a live specimen intended for the local bush meat market.

Squamata
Agamidae
***Acanthocercus cyanocephalus* (Falk, 1925)**
BLUE-HEADED RIDGEBACK AGAMA – Fig. 4B

Material.—**9 specimens:** CNP, Park Headquarters [-9.81942°, 16.65386°, 1089 m]: CAS 258430–31; CNP, vicinity of park headquarters [-9.81887°, 16.65415°, 1096 m]: CAS 258428, [-9.81892°, 16.65410°, 1106 m]: CAS 258429, [-9.81858°, 16.65403°, 1089 m]: CAS 258433–34, [-9.81922°, 16.65414°, 1094 m]: CAS 258435; CNP [-9.86869°, 16.74736°, 1094 m]: CAS 258432; 13.67 km SW (by road) of Cangandala [-9.84233°, 16.31733°, 1086 m]: CAS 258436.

Comments.—Wagner et al. (2018) revised the Angolan *Acanthocercus* and presented evidence that most Angolan populations may be referable to the forgotten name *A. cyanocephalus* proposed by Falk (1925) based on Angolan material. This arboreal species occurs throughout most of the country with exception of the arid southwest and the northwestern regions, as well as in portions of neighboring countries (Marques et al., *in press*).

Chamaeleonidae
***Chamaeleo dilepis* (Leach, 1819)**
COMMON AFRICAN FLAP-NECKED CHAMELEON – Fig. 4C

Material.—**12 specimens:** CNP, Park Headquarters [-9.81942°, 16.65386°, 1089 m]: CAS 258363–65; CPN, 0.25 km ENE (by rd) of park headquarters [-9.81836°, 16.65606°, 1116 m]: CAS 258371; CNP, Park Headquarters [-9.81942°, 16.65386°, 1089 m]: CAS 258366–67, [-9.81942°, 16.65386°, 1089 m]: CAS 258368, [-9.81942°, 16.65386°, 1089 m]: CAS 258369–70; CNP [-9.82358°, 16.64431°, 1085 m]: CAS 258372, [-9.81450°, 16.68267°, 1093 m]: CAS 258373, [-9.78831°, 16.72969°, 1106 m]: CAS 258374.

Comments.—In Angola, *C. dilepis* is widely distributed across most of the country. Despite its wide range this is only the second recorded locality for the species in the Malanje Province (Table 1), but localities are known from many other provinces (Marques et al., *in press*). The species is locally feared and erroneously believed to be highly venomous by the locals.

Gekkonidae
***Hemidactylus mabouia* (Moreau de Jonnès, 1818)**
TROPICAL HOUSE GECKO – Fig. 4D

Material.—**1 specimen:** CNP, vicinity of park headquarters [-9.81810°, 16.65535°, 1092 m]: CAS 258427.

Comments.—As currently recognized, the widespread and human commensal *Hemidactylus mabouia* is part of a complex of many species (Vences et al. 2004). Several names are currently in the synonymy of *H. mabouia*, but ongoing work will likely elevate some of these to full species (I. Agarwal et al., unpubl.). Cangandala specimens, similar to Angola populations in general, appear to belong to *H. mabouia* sensu stricto (I. Agarwal, pers. comm.). The species is widespread in Africa and is commonly found around human settlements in both natural and altered habitats and its range is known to have expanded accordingly (Kluge 1969). In Angola, it occurs mainly in the north of the country, but also along the coast, including the Cabinda enclave. It is broadly sympatric with *Hemidactylus longicephalus* Bocage, 1873 throughout much of its range (see Marques et al., *in press*).

***Hemidactylus* cf. *muriceus* Peters, 1870**
GUINEA SPLIT-TOED GECKO – Fig. 4E

Material.—**17 specimens:** CNP, Park Headquarters [-9.81942°, 16.65386°, 1089 m]: CAS 258410; CNP [-9.85464°, 16.70978°, 1096 m]: CAS 258411; [-9.85547°, 16.70892°, 1101 m]: CAS 258412, [-9.81161°, 16.64381°, 1085 m]: CAS 258413, [-9.84581°, 16.72036°, 1106 m]: CAS 258414, [-9.84703°, 16.72125°, 1121 m]: CAS 258415, [-9.83558°, 16.72125°, 1067 m]: CAS 258416, [-9.83600°, 16.68108°, 1102 m]: CAS 258417, [-09° 50’ 09.6” S, 16° 40’ 51.9” E, 1102 m]: CAS 258418, [-9.83831°, 16.67494°, 1113 m]: CAS 258419, [-9.78608°, 16.73211°, 1122 m]: CAS 258420, [-9.80961°, 16.65533°, 1082 m]: CAS 258421, [-9.81011°, 16.65564°, 1059 m]: CAS 258422, [-9.81653°, 16.66208°, 1108 m]: CAS 258423, [-9.80972°, 16.65631°, 1090 m]: CAS 258424; CNP, Park Headquarters [-9.81942°, 16.65386°, 1089 m]: CAS 258425–25826.

Comments.—The identity of *H. muriceus* and its taxonomic relationships with other members of the genus have long been problematic (Bauer et al. 2006; Henle and Böhme 2003; Perret 1975). Preliminary molecular data reveals that within the *muriceus* group there are several deeply divergent lineages, and that the Angolan specimens do not belong to the nominotypic form that occurs in Guinea. The only other known Angolan records of this "*muriceus*-like" species are from Malanje Province, from the "Cuango = Quango river" (Bocage 1895; Peters 1881). Our specimens were collected in and under fallen logs, under bark and on the ground, which suggests a chiefly terrestrial lifestyle for the species.

***Lygodactylus angolensis* Bocage, 1896**
ANGOLAN DWARF GECKO – Fig. 4F

Material.—**9 specimens:** CNP, Park Headquarters [-9.81942°, 16.65386°, 1089 m]: CAS 258354–56, [-9.81942°, 16.65386°, 1089 m]: CAS 258357, [-9.81942°, 16.65386°, 1089 m]: CAS 258358, [-9.81942°, 16.65386°, 1089 m]: CAS 258359, [-9.81942°, 16.65386°, 1089 m]: CAS 258360, [-9.81942°, 16.65386°, 1089 m]: CAS 258361; CNP [-9.81011°, 16.65564°, 1059 m]: CAS 258362.

Comments.—The distribution of *Lygodactylus angolensis* is poorly known in much of its range. Pasteur (1964) provided a map indicating that *L. angolensis* occupies a wide range from eastern to south-central regions of Angola where it might be sympatric with *Lygodactylus capensis* (Smith, 1849). The specimens collected in Cangandala National Park represent the first record of the species in Malanje Province and a northern range extension for the species in Angola. The species was relatively common basking in human dwellings and individuals were observed preying on stingless bees of the genus *Trigona*, waiting close to the tube-shaped hive and quickly attacking when the bees entered or left.

Scincidae

Panaspis cabindae (Bocage, 1866)

CABINDA SNAKE-EYED SKINK – Fig. 4G

Material.—**5 specimens:** CNP [-9.85464°, 16.71003°, 1104 m]: CAS 258403–04, [-9.83497°, 16.68150°, 1052 m]: CAS 258405, [-9.75842°, 16.80061°, 1102 m]: CAS 258406, [-9.81500°, 16.67800°, 1122 m]: CAS 258407.

Comments.—A recent molecular phylogeny by Medina et al. (2016) shows that *P. cabindae* has an extensive distribution from the DRC to southern Angola. However, our preliminary molecular analyses based on the mitochondrial 16S and ND2 genes on several populations of *P.* cf. *cabindae* in Angola show a considerable degree of structure, potentially comparable to that found within the *P. wahlbergi* species complex by Medina et al. (2016). Further investigation is needed to assess the existence of putative cryptic species within *P. cabindae*. The newly collected material represents the first record of the species for Malanje Province, although the species has been recorded from several locations in the neighboring provinces of Bengo, Kwanza-Norte, and Kwanza-Sul (Marques et al., *in press*). Elsewhere in Angola, this species occurs along the coastal provinces from Cabinda to Namibe Province. The species was commonly found on the ground under leaves or logs.

Trachylepis bayonii (Bocage, 1872)

BAYÃO'S SKINK

Material.—**3 specimens:** CNP, Park Headquarters [-9.81942°, 16.65386°, 1089 m]: CAS 258357; CNP [-9.82689°, 16.65003°, 1089 m]: CAS 258387; right bank of the Kwanza River [-9.88525°, 16.28722°, 1005 m]: CAS 258388.

Comments.—This species, originally described by Bocage (1870) from "Duque de Bragança" (currently Kalandula) in Malanje Province, was later (Bocage 1895) split into two different varieties – variety *A*, from around the type locality, and variety *B*, from the highlands of Huíla in southwestern Angola. Variety *B* was later described by Laurent (1964a), as a subspecies, *Mabuya bayoni huilensis*. A third extralimital subspecies – *Trachylepis bayoni keniensis* (Loveridge, 1956) – is recognized by some authors (Menegon and Spawls 2013; Spawls et al. 2004). Our material is referable to the nominotypical form, and our records lie approximately 120 km southeast of the type locality. In Angola, this species mainly occurs in the central and southwestern regions of the country. This is the second locality for this species in Malanje Province, but localities are known from many other provinces (Marques et al., *in press*). Our specimens were initially confounded with *Trachylepis striata* and thus not included in Ceríaco et al. (2016b).

Trachylepis cf. *megalura* (Peters, 1878)

LONG-TAILED SKINK – Fig. 4H

Material.—**1 specimen:** CNP [-9.84606°, 16.72233°, 1101 m]: CAS 258401.

Comments.—This slender, long-tailed skink is apparently related to the morphologically similar *Trachylepis megalura*. The presence of a *megalura*-group skink in Angola was previously reported by Laurent (1964a). According to Laurent (1964a), the specimens from "Alto Cuílo" differed from typical *T. megalura* by lacking the distinctive lateral line and having separated supranasal scales. The author assumed that the specimen from Alto Cuílo, together with some specimens from the Upemba National Park cited by de Witte (1953, not 1933, as wrongly cited by Laurent) belonged to a new and undescribed "angolo-katangaise" form. Based on a combination of study of Laurent's (1964a) and De Witte's (1953) specimens, a review of available museum specimens including the *megalura* holotype, as well as new analyses of mitochondrial 16S and ND2 genes, we support Laurent's (1964a) suggestion that the Angolan and Katanga specimens represent different taxon. These are closely related to topotypical *T. megalura*, and this species will be formally described elsewhere (Marques et al., unpubl.). The specimen was collected foraging under a shrub and contained 10 well-developed embryos.

Trachylepis wahlbergii (Peters, 1870)

WAHLBERG'S STRIPPED SKINK – Fig. 5A

Material.—**7 specimens:** CNP, Park Headquarters [-9.81942°, 16.65386°, 1089 m]: CAS 258394; CNP, vicinity of park headquarters [-9.81810°, 16.65535°, 1092 m]: CAS 258395, [-9.81810°, 16.65535°, 1092 m]: CAS 258396, [-9.81939°, 16.65383°, 1097 m]: CAS 258397, [-9.81772°, 16.65464°, 1088 m]: CAS 258398–99, [-9.81908°, 16.65469°, 1102 m]: CAS 258400.

Comments.—The validity of *T. wahlbergii* as a full species (as opposed to a synonym or subspecies of *T. striata*) is disputed (Branch 1998; Broadley 1977a, 2000; Castigilia et al. 2006; see Marques et al., *in press*). In their booklet, Ceríaco et al. (2016b) referred to this material as *T. striata*. These animals are widely distributed across Angola, although apparently absent from the forested far north and the arid far southwest. All records in the *T. striata* complex from across Namibia are potentially assignable to *T. wahlbergii* as are all records from western Zambia. It would seem likely, therefore that members in this group in Angola would also be assignable to this taxon. Color patterns of Angolan members of the group are variable and some specimens do at least superficially resemble true *T. striata*. However, it is probable on biogeographic grounds that most or all older Angolan literature records of "*T. striata*" are, in fact, referable to *T. wahlbergii*, or perhaps to another species, but not to *T. striata* sensu stricto. The species was commonly seen basking near human settlements and in areas with sparse vegetation.

Trachylepis cf. *varia* (Peters, 1867)

VARIABLE SKINK – Fig. 5B

Material.—**58 specimens:** CNP, Park Headquarters [-9.81858°, 16.65403°, 1089 m]: CAS 258376; CNP [-9.86900°, 16.69131°, 1092 m]: CAS 258377, [-9.85464°, 16.71003°, 1104 m]: CAS 258378–79, [-9.84489°, 16.72103°, 1102 m]: CAS 25838–81, [-9.84461°, 16.72058°, 1118 m]: CAS 258382, [-9.86383°, 16.70797°, 1103 m]: CAS 258383, [-9.84539°, 16.72061°, 1096 m]: CAS 258384–85, [-9.84969°, 16.67908°, 1108 m]: CAS 258386, [-9.82756°, 16.66997°, 1097 m]: CAS 258389, [-9.75881°, 16.80233°, 1163 m]: CAS 258390–91, [-9.81175°, 16.69094°, 1084 m]: CAS 258392, [-9.77611°, 16.75472°, 1197 m]: CAS 258393.

Comments.—There are several cryptic species within the taxon currently known as *Trachylepis varia*, and the Angolan material is not referable to true *varia* (Weinell and Bauer, 2018). The name *albopunctata* may represent the Angolan population, although confusion with other available names and limited data on the distribution of different lineages precludes definitive conclusions at this time (see Marques et al., *in press*). In Angola, the species has a large distribution (Marques et al., *in press*), being known in Malanje from Pungo-Andongo (Boulenger 1905) and in the Kwanza River (Marques et al., *in press*).

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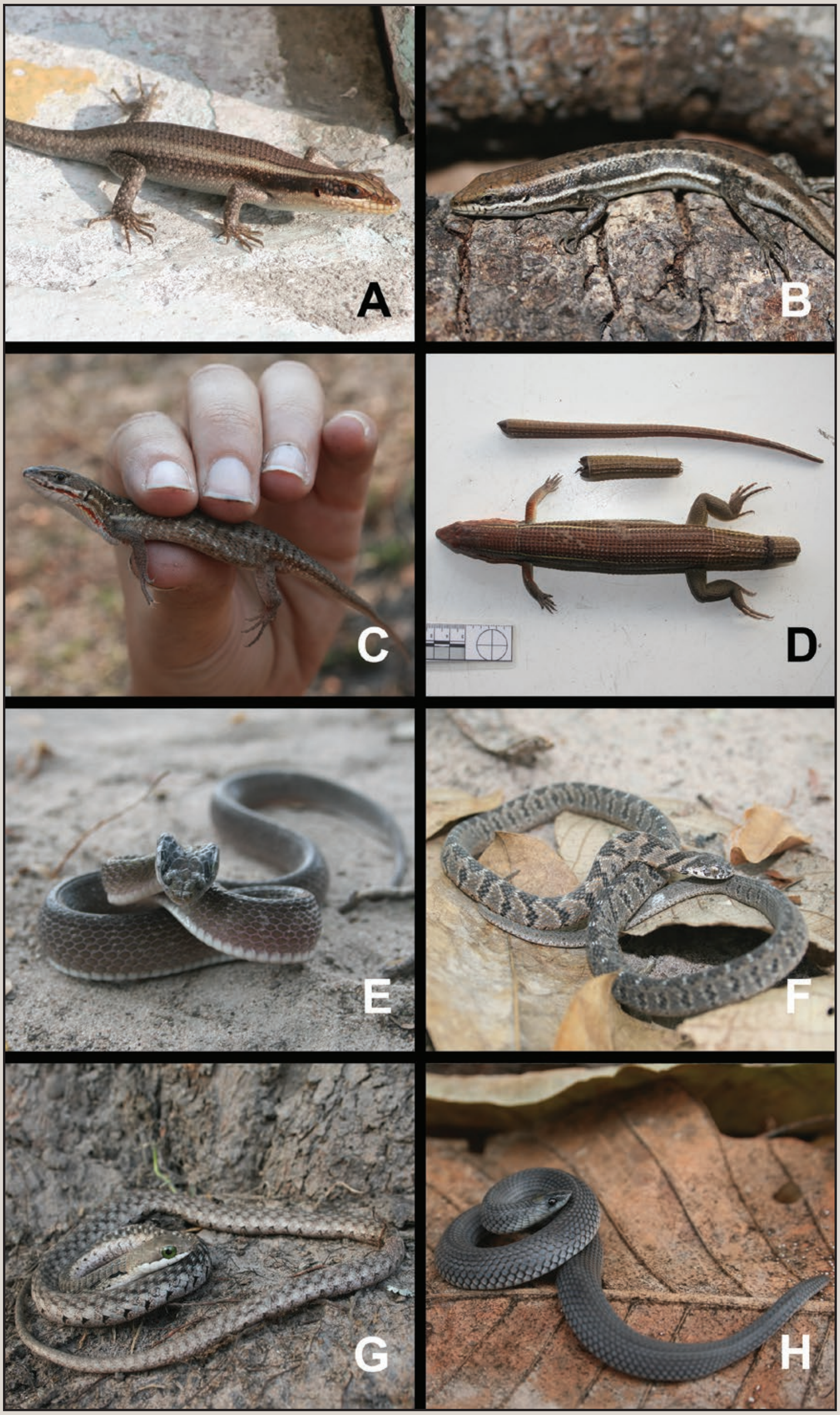

Fig. 5. A) Wahlberg's Striped Skink, *Trachylepis wahlbergii* (Peters, 1870); B) Variable Skink, *Trachylepis* cf. *varia* (Peters, 1867); C) Angolan Rough-scaled Lizard, *Ichnotropis bivittata* (Bocage, 1866); D) Keeled Plated Lizard, *Gerrhosaurus multilineatus* (Bocage, 1866); E) Red-lipped Snake, *Crotaphopeltis hotamboeia* (Laurenti, 1768); F) Common Egg Eater, *Dasypeltis scabra* (Linnaeus, 1758); G) Juvenile Boomslang, *Dispholidus typus typus* (Smith, 1829); H) Angolan Shovel Snout, *Prosymna ambigua* Bocage, 1873. All photos from Cangandala National Park.

Lacertidae

Ichnotropis bivittata (Bocage, 1866)
ANGOLAN ROUGH-SCALED LIZARD – Fig. 5C

Material.—**2 specimens:** CNP [-9.85464°, 16.71003°, 1104 m]: CAS 258402; right bank of Kwanza River [-9.88439°, 16.28583°, 1041 m]: CAS 258409.

Comments.—Described from "Duque de Bragança" (currently Kalandula) in Malanje Province, *I. bivittata* was for some time misidentified with *Ichnotropis capensis* (A. Smith, 1838) (see Marques et al., *in press*). No modern revisions of the genus are available (Edwards et al. 2013). Our material agrees with the original description and is from close to the type locality. Records for the species are known for many provinces of Angola (see Marques et al., *in press*), and a subspecies, *I. b. pallida,* is recognized from Huíla Province. Recently, Ineich and Le Garff (2015) reported the species for Gabon, a northern record for the species.

Gerrhosauridae

Gerrhosaurus multilineatus (Bocage, 1866)
KEELED PLATED LIZARD – Fig. 5D

Material.—**1 specimen:** CNP, park headquarters [-9.82078°, 16.60625°, 1101 m]: CAS 262312.

Comments.—The taxonomic and nomenclaturally problematic *G. multilineatus* was described based on a juvenile specimen from "Duque de Brangaça" (currently Kalandula) in Malanje Province, 120km northwest of CNP. Bates et al. (2013) described in great detail the issues surrounding the identity of *G. multilineatus* and type material which is now lost. For a long time *G. multilineatus* was considered a synonym of *G. nigrolineatus*, and it is likely that some Angolan records referred to *nigrolineatus* represent *multilineatus* (see Marques et al., *in press*). A new revision of the *Gerrhosaurus nigrolineatus* group (which includes *G. multilineatus, G. nigrolineatus*, *G. bulsi*, *G. flavigularis* and *G. intermedius*) is being prepared (M. Bates et al., unpubl.). Based on preliminary genetic analyses, our specimens, together with other specimens from Kissama National Park and the Kwanza River Basin, form a well-supported clade that likely represents nominotypical *multilineatus*. The species is abundant in CNP but extremely difficult to catch, as they rapidly flee into deep holes in the ground at first approach.

Varanidae

Varanus niloticus (Linnaeus, 1758)
NILE MONITOR

Material.—**Observation:** CNP [-9.75889°, 16.79914°, 1142 m]: Specimen not collected.

Comments.—Angolan populations appear to fit within the southern group of nominotypical *V. niloticus* (Dowell et al. 2016). A single individual was observed in a flooded area in CNP.

Colubridae

Crotaphopeltis hotamboeia (Laurenti, 1768)
RED-LIPPED SNAKE – Fig. 5E

Material.—**5 specimens:** CNP [-9.81942°, 16.65386°, 1089 m]: CAS 258671, [-9.81894°, 16.65489°, 1088 m]: CAS 258672, [-9.81810°, 16.65535°, 1092 m]: CAS 258673–74; CNP, park headquarters [-9.81942°, 16.65386°, 1089 m]: CAS 258675.

Comments.—This species has a wide distribution in sub-Saharan Africa (Broadley and Cotterill 2004) and a phylogeographic analysis is therefore desirable to investigate the possibility of cryptic species (Bates et al. 2014). It is widespread throughout Angola, with exception of the desert of the far southwestern regions. It mainly occurs in savanna but also in wooded areas including miombo forest. Several specimens were found after rain feeding on *Sclerophrys pusilla*, *Hemisus guineensis*, and *Leptopelis bocagii*.

Dasypeltis scabra (Linnaeus, 1758)
COMMON EGG EATER – Fig. 5F

Material.—**1 specimen:** CNP [-9.75889°, 16.79914°, 1142 m]: CAS 258669.

Comments.—The taxonomy of this species in southern Africa is being investigated, and the presence of cryptic taxa has been suggested (Bates and Broadley 2018). The species occurs in the entire country (including Cabinda Enclave), with exception of the desert regions of the far southwestern Angola. The collected material represents the first record of the species for Malanje Province, although the species has been recorded from several locations in the neighboring provinces of Kwanza-Norte, Lunda-Norte, and Lunda-Sul (Marques et al., *in press*).

Dispholidus typus typus (Smith, 1829)
BOOMSLANG – Fig. 5G

Material.—**1 specimen:** CNP, Park Headquarters [-9.81942°, 16.65386°, 1089 m]: CAS 262313.

Comments.—An extensive review of the genus *Dispholidus* is currently in preparation by Eimermacher and Broadley (in prep.), after initial works suggesting the existence of multiple distinct lineages with potential taxonomic and nomenclatural implications (Eimermacher 2012). Of the four recognized subspecies, only two have been cited for Angola, the nominotypical form and *D. t. punctatus*, which Laurent (1955) reported for Dundo in northeastern Angola. However, despite limited sampling, Eimermacher (2012) suggested that the available name for specimens from northern Namibia is *D. t. viridis*, which might imply that the Angolan population not referable to the conspicuous *punctatus* (and currently identified as *typus*) may belong to this taxon. While *D. punctatus* is only known from northeastern Angola, the animals currently identified as *D. typus* are recorded across the country (see Marques et al., *in press*). For now, we regard the Angolan non-*punctatus* boomslang as the nominotypical form. Our juvenile specimen was collected after falling from a tree while preying on an adult *Chamaeleo dilepis*. Known locally as being venomous, locals believe that boomslangs derive their venom through predation on chamaeleons, through which they "receive" the venom (see *Chamaeleo dilepis* account above).

Lamprophiidae

Boaedon cf. _angolensis_ Bocage, 1895
ANGOLAN BROWN HOUSE SNAKE

Material.—**1 specimen:** CNP, village Bola-Cassaxi [-9.77363°, 16.82283°, 1089 m]: CAS 262315.

Comments.—Recent molecular genetic analyses conducted by Kelly et al. (2011) demonstrated extensive genetic variation in the widespread *B.* "*fuliginosus*," suggesting several cryptic species are present. Wallach et al. (2014) assigned the Angolan records to *B. fuliginosus*. However, Marques et al. (*in press*) noted that some literature references to the *B. fuliginosus* complex may belong to *B. angolensis* or *B. variegatus*. Hallerman et al. (unpubl.) are evaluating species boundaries in this group.

Prosymna ambigua **Bocage, 1873**
ANGOLAN SHOVEL-SNOUT – Fig. 5H

Material.—**1 specimen:** CNP [-9.81800°, 16.65697°, 1091 m]: CAS 258670.

Comments.—Described by Bocage (1873) based on one specimen from "Duque de Bragança" (currently Kalandula) in Malanje Province, 120 northwest of CNP. The specimen was collected during the night on a sandy road. All earlier records from Malanje are from the type locality in Duque de Bragança, thus this specimen is only the second locality for the province.

Discussion

Malanje Province is topographically and ecologically diverse and is dominated by two different river basins – the Congo-Casai, reaching Angola from the Republic of the Congo and the DRC, and the entirely Angolan Kwanza. Because of this hetereogeneity, Malanje is one of the provinces in which the turnover between Central and Southern African faunas is most evident. Bocage (1895) was the first to propose two main biogeographic zones in Angola—a northern region and a southern region—with the Kwanza River being the main divider between these. Within these regions, Bocage (1895) proposed a subsequent division—the coastal zone, the intermediate zone, and finally the high plateaus zone. Recent studies show that this biogeographic pattern is more complex than a simple north vs. south regionalization (Rodrigues et al. 2015). While it is evident that current knowledge regarding the zoogeography of Angola is incomplete, it is possible to make some biogeographic inferences. Malanje Province, and especially the area of CNP, corresponds to the border between the north-south regions proposed by Bocage (1895) and Frade (1963), and the intergradation between the northern "Zaire-Lunda-Cuanza" region and the more central "Central Plateau" region, and some of the taxa presented here support this pattern.

Additional faunal surveys are needed for Malanje Province. Currently, the northern half of the Province is almost virgin territory in terms of herpetological knowledge. Given its similarity in habitat and proximity to the DRC, we expect to discover many new records of species unknown to occur in Angola (and possibly even some that are new to science). The discovery of several frog species in neighboring Uíge Province by Ernst in Lautenschläger and Neinhuis (2014) suggests that species diversity of the northern areas of Malanje and Angola, in general, are underestimated. Besides these new areas of exploration, Malanje Province includes some of the most important type localities for many taxa, such as Duque de Bragança (currently Kalandula) and Pungo Andongo. Duque de Bragança alone is the type locality of 16 taxa of amphibians and 12 taxa of reptiles, whereas five taxa were described based on material for Pungo Andongo. The Duque de Bragança type material (with all of Bocage's Angolan collections housed in Lisbon) were destroyed in the 1978 fire that destroyed Museu Bocage. Although topotypical material was offered by Bocage to other museums across the world, the age and preservation of these specimens may preclude extraction of DNA from these. Fresh topotypical material for these taxa will be important to address outstanding taxonomic and nomenclatural uncertainties of different groups known to represent species complexes, including *Hyperolius angolensis*, *H. cinnamomeoventris*, *Leptopelis bocagii* and *Gerrhosaurus multilineatus*. Some specimens reported here are representatives of taxa for which Duque de Brangaça and Pungo Andongo are type localities, including *Hyperolius angolensis*, *H. cinnamomeoventris*, *H. nasutus*, *Leptopelis bocagii*, *Ichnotropis bivittata*, *Trachylepis bayonii*, *Gerrhosaurus multilineatus*, and *Prosymna ambigua*. Despite not being truly topotypical, the close proximity of CNP to these type localities provides an initial approximation to solve some of the taxonomic and nomenclatural uncertainties regarding these taxa and are likely to be of importance to several ongoing reviews and phylogenetic and phylogeographic studies.

Fig. 6. A) General view of a recently burned area in Cangandala National Park; B) Live specimens of Bell's Hinge-back Tortoise, *Kinixys* cf. *belliana* Gray, 1830, together with other bush meat, collected by poachers in the park premises. Both photos from Cangandala National Park.

PHOTOS BY L. M. P. CERÍACO

We have identified a total of 14 and 19 species of amphibians and reptiles, respectively, from CNP. Of these, *Hemisus guineensis*, *Ptychadena anchietae*, *Ptychadena guibei*, *Amnirana* cf. *darlingi*, *Lygodactylus angolensis*, *Hemidactylus* cf. *muriceus*, *Panaspis cabindae*, *Trachylepis* cf. *megalura* and *Dasypeltis scabra* are recorded for the first time for Malanje Province, whereas several taxa (*Amnirana* cf. *darlingi*, *Hemidactylus* cf. *muriceus* and *Trachylepis* cf. *megalura*) may represent species new to science. It is likely that many additional species will still be found in CNP. We were fortuitous in finding several snake species during our survey of the park. CNP rangers attributed the relatively small number of snakes found to the climatic conditions, noting that snakes were generally more abundant during and after the rains. In fact, a good number of the snakes collected (*Crotapholpeltis hotamboeia* and *Boaedon* cf. *angolensis*) were collected during the last days of work, after the rains had started. Other snake species, e.g., *Python sebae*, *Causus rhombeatus*, *Bitis arietans*, *Naja anchietae* and *Dendroaspis polylepis*, are known to occur in the park but were not encountered in our survey (P. Vaz Pinto, pers. comm.; see Ceríaco et al. 2016b). Other species of snakes, including *Afrotyphlops angolensis, A. lineolatus, Letheobia scutifrons, Lycophidion multimaculatum, Philothamnus dorsalis, P. heterodermus, P. heterolepidotus, P. irregualris, Psammophis angolensis, Thelotornnis kirtlandi*, *Naja subfulva*, *Naja nigricollis*, and *Causus bilineatus* occur in nearby regions and in similar habitats, and their presence in the park may also be expected. Further field surveys in CNP as well as opportunistic observations will certainly find some of the above-mentioned taxa within the borders of the park, and possibly new species for Angola and to science.

Due to its limited size and the presence of the iconic Giant Sable, CNP is probably the most heavily guarded conservation area in the country, with a considerable number of local rangers, continuous monitoring and a large fenced area. However, these facilities do not entirely prevent events like wildfires or bush meat hunting. The presence of human communities in close proximity to the park results in regular wild fires in the context of traditional slash and burn agriculture, as well as the harvest of specific reptiles for human consumption. We observed both of these events during the short field survey made in the park. On the night of our arrival a large wildfire engulfed the east side of the park, and other burned areas were located in the interior of the park in the following days (Fig. 6A). Fires are known to have a detrimental effect on herpetological communities in Africa (Kennedy et al. 2012; Masterson et al. 2008), and a low number of specimens were indeed located and collected in these post-burn areas.

During our stay CNP guards retrieved eight live *Kinixys* cf. *belliana* from poachers inside the park limits, apparently intended to the local bush meat market (Fig. 6B). This is the first recorded event of *Kinixys* poaching in the country, but although bush meat is recognized as a conservation problem by the authorities and already mentioned by Bersacola et al. (2014), data regarding this practice in the country are lacking. Local persecution and killing of some reptiles, especially chamaeleons and snakes, may also be a potential threat to CNP reptile populations.

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Activity Pattern and Behavior of an Endemic Bromeliad Frog Observed through Camera Trapping

Camera trapping has been widely used to assess the occurrence, abundance, distribution and behavior of species and communities (O'Connell et al. 2011; Meek et al. 2014). Although the use of camera traps has expanded considerably to investigate a diverse range of mammal species, the effectiveness of this method to evaluate other faunal groups is relatively poorly investigated (Ariefiandy et al. 2013; Welbourne et al. 2015; Adams et al. 2017; Laughlin et al. 2017). In fact, just a small proportion of studies (< 2%) cover any ecological aspects of amphibians and reptiles (Burton et al. 2015; Welbourne et al. 2017). Most digital camera traps are triggered by a passive infrared (PIR) sensor that detect differences in the surface temperature of objects in the detection zone; consequently, they are regarded as less reliable for ectotherms (Ariefiandy et al. 2013). However, improvements in camera technology to detect small animals (Welbourne 2013; Hobbs and Brehme 2017) and use of time-lapse mode can improve the detection of ectothermic vertebrates (Welbourne et al. 2017).

Camera traps have been used to characterize reptile communities (Welbourne et al. 2015; Adams et al. 2017) and were previously applied to collect data on the vulnerable Komodo Dragon (Ariefiandy et al. 2013); to monitor activity patterns of the endangered Grassland Earless Dragon in Australia (McGrath et al. 2012); and to identify individuals in reptile assemblages (Welbourne 2013; Bennett and Clements 2014). For amphibians, camera traps have been successfully used to assess movements (Pagnucco et al. 2011; Crosby 2014) and oviposition behavior (Ramsdell 2013); identify potential predators (Velo-Antón and Cordero-Rivera 2017); and describe activity patterns (Hoffman et al. 2010; Engbrecht and Lannoo 2012). Most camera trap implementations are species-specific (Bennett and Clements 2014), and for amphibians they have focused on both newts (Pagnucco et al. 2011; Crosby 2014; Velo-Antón and Cordero-Rivera 2017) and frogs (Hoffman et al. 2010; Engbrecht and Lannoo 2012; Ramsdell 2013; Laughlin et al. 2017). Although most studies using camera traps come from Asia and the Americas (Burton et al. 2015), the majority of studies on amphibians have been conducted in North America (Hoffman et al. 2010; Pagnucco et al. 2011; Engbrecht and Lannoo 2012; Ramsdell 2013; Crosby 2014; Laughlin et al. 2017).

In this study we describe activity patterns and behavior of the rare and elusive *Crossodactylodes itambe*—a micro-endemic frog strictly dependent on bromeliads, where they spend their entire life cycle (Barata et al. 2013; Santos et al. 2017). There are five species in the genus—each restricted to a single location and occurring in high elevation areas of the Atlantic Rainforest in Brazil—and there is still very little information on the ecology and natural history of this group. Using camera trapping and video recording, we investigate activity patterns of this poorly known *Crossodactylodes* species and report periods of activity with descriptions of behavior. We also discuss the application of camera traps to study bromeliad-dwelling frogs and how this technique can be used to enhance our understanding of the group's ecology and natural history.

METHODS

Crossodactylodes itambe is a small frog species (average 15.7 mm; Fig. 1A–B) only known from the type locality in an area of < 0.5 km^2 at 1700 m above sea level (Barata et al. 2013). Adults and tadpoles are reported to exclusively use a single species of bromeliad, *Vriesea medusa* (Barata et al. 2013; Santos et al. 2017), where they spend their entire life cycle without leaving the plant. Bromeliads are flowering terrestrial or arboreal plants characterized by multiple sized interlocking leaves forming a circular central tank (hereby, rosette) that collects and holds water, leaf litter and detritus. *Vriesea medusa* is a night-blooming flowering bromeliad (Fig. 1B–C) with a funnel-form rosette c. 70 cm high (Versieux 2008) that can hold a large amount of water even during periods of low rainfall. Both the bromeliad and frog are known to occur at the summit of Pico do Itambé State Park; a protected area with 4700 ha located in the state of Minas Gerais, southeastern Brazil (18°23'S, 43°20'W). This system provides an opportunity to explore the use of camera traps to study amphibians, especially because the sampling unit (each

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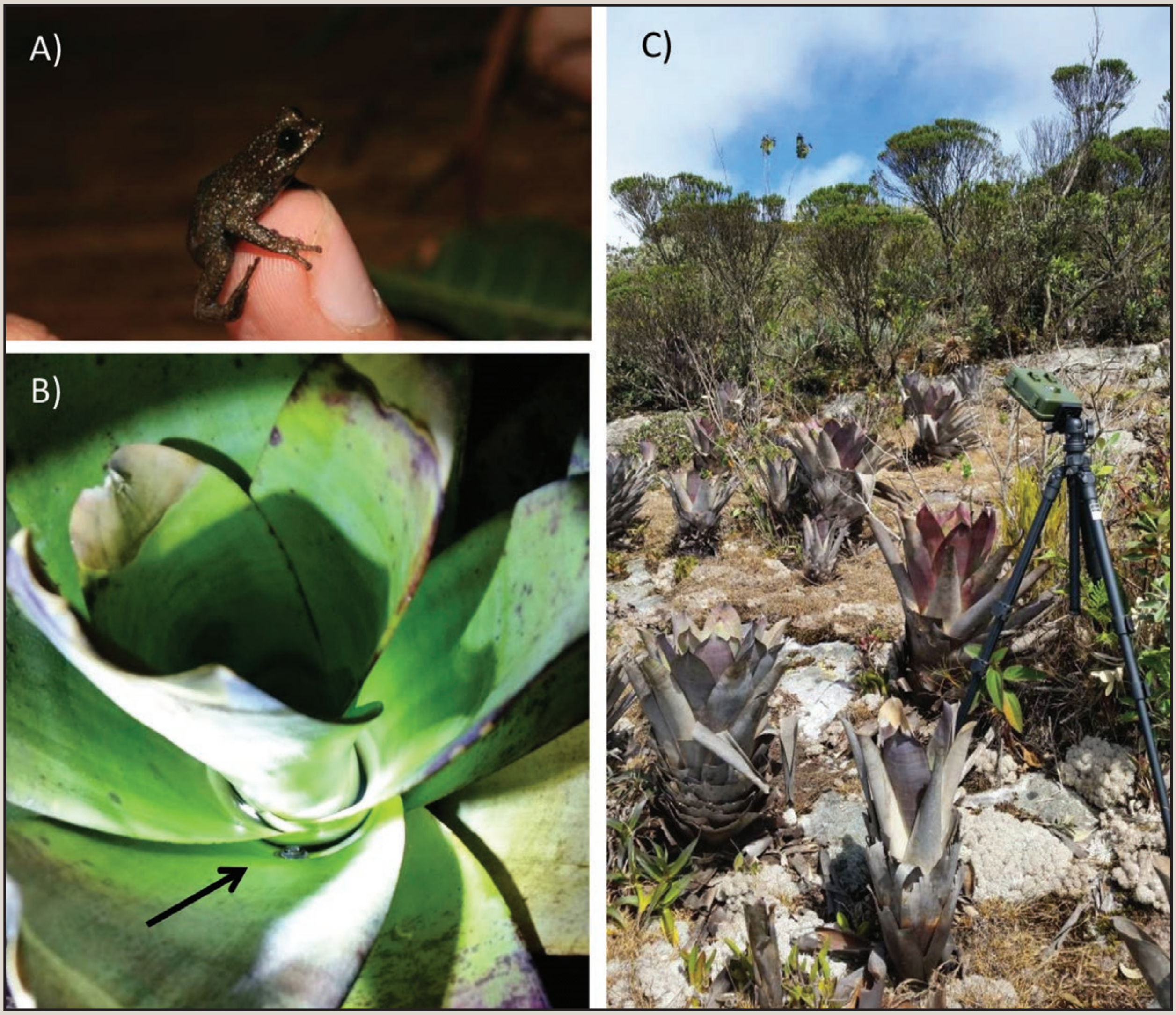

PHOTOS BY I. M. BARATA

Fig. 1. Adult male of *Crossodactylodes itambe* (A), occupying the axil leaf of a bromeliad *Vriesea medusa*, as indicated by the black arrow (B). Camera trap deployment is shown in detail (C) with the tripod and camera facing the bromeliad at a focal distance of at least 30 cm.

bromeliad) is self-contained and frog movement is restricted to the plant.

To describe activity patterns and how frogs use the bromeliad we installed a camera trap (Bushnell Nature View HD) that allowed observations of frogs for the whole night. We used an interchangeable lens, allowing a focal distance of approximately 45 cm (i.e., from the bromeliad and frogs). We mounted the camera trap onto a tripod (Induro AKB0 AT014), which was positioned close to the bromeliad with its flexible head adjusted so the camera could face down (Fig. 1C). Tripod height was about 1 m and the camera trap was at least 30 cm distant from the focal object. At each bromeliad, we tested the camera and adjusted the tripod to ensure the images would frame the largest view as possible.

In May and June 2016, we deployed one camera trap in four different bromeliads known to be occupied by *C. itambe* for one to three consecutive nights. Sampled bromeliads were at least 25 m apart from each other. We set the camera trap in time-lapse mode taking one picture per minute for 12 hours on each night (from 1700 h to 0500 h), apart from our first survey night when we set up the camera to take one picture every five minutes (but kept all other settings equal). In addition to setting the camera in time-lapse function, we also allowed it to be triggered by movements through its PIR sensor (sensitivity was set to high). Because the camera trap was too close to the focal object, we set up the infrared flash control to low and used a thin piece of cloth covering the camera flash to avoid overexposure. The camera operated with 12 batteries and was equipped with a 32 gigabyte SD card to store the images.

We considered each photograph taken as a record that could be either positive (i.e., a photo with presence of our target species) or negative (i.e., species was not recorded). For all positive records, we defined whether the camera had been triggered due to the time-lapse mode or by its PIR sensor. We did this by looking at the time of the record, as time-lapse pictures were taken on the first second of each minute (e.g. 22h 10min 01sec, 22h 11min 01sec, etc.), while pictures triggered by the PIR sensor could have been taken at any time during the survey period. We classified positive records as active or inactive, based on the sequence of movements taken by the camera. While we considered inactivity as a sequence of records where frogs remained immobile, activity was considered when any

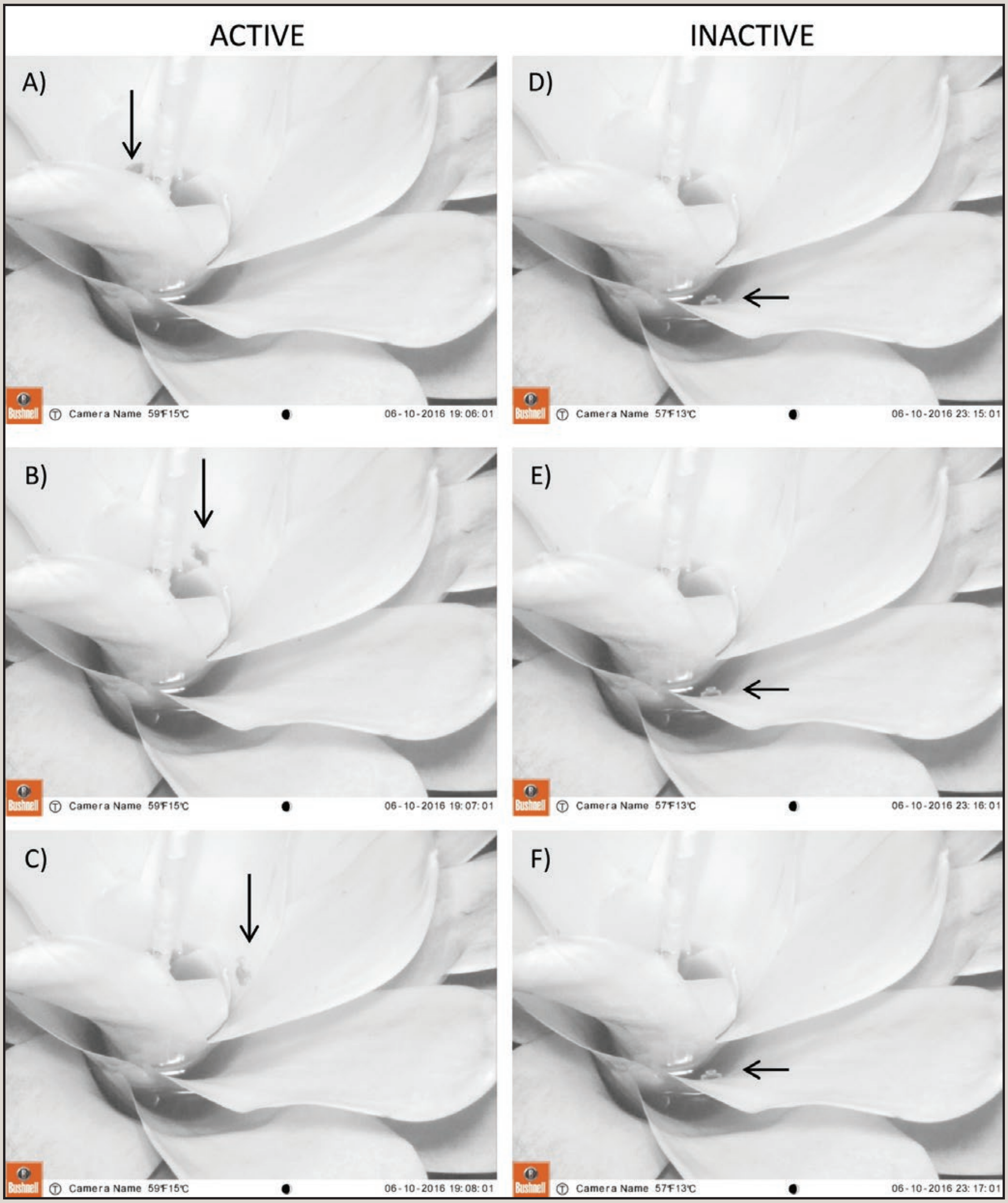

Fig. 2. Sequence of positive records taken with a camera trap on the same night of survey, showing when the species is active (A–C) and inactive (D–F). Time-lapse was set for one picture every minute, as shown by the pictures.

movement was detected between consecutive records (Fig. 2). Adults and tadpoles of *C. itambe* are rarely observed during the day (Barata et al. 2013; Santos et al. 2017) and, for that reason, we assumed frogs were inactive during the day and we only analyzed nocturnal activity patterns.

We used the R package 'activity' to fit a flexible circular distribution to time-of-detection data extracted from camera trap photos (Rowcliffe et al. 2014) to describe the target species' activity pattern and its overall activity level. Activity level was estimated as the percentage of time the species was active, with standard errors obtained through nonparametric bootstrapping (Rowcliffe et al. 2014). For this analysis, we considered only the positive records classified as active and excluded data from the first survey night because the interval between photographs was distinct from all other nights. We set the number of bootstrap iterations to 10,000 and defined 'data' as the sampling method for bootstrapping errors.

We performed monthly visual surveys at the study area, between Feb–May 2015 and Feb–June 2016, and bromeliads were visited on four to six consecutive nights as part of a population monitoring project (Barata et al. 2017). Based on occasional encounters, we described three behaviors made during our surveys: 1) locomotion: movement (usually walking) inside the bromeliad; 2) escaping: sudden movement (going

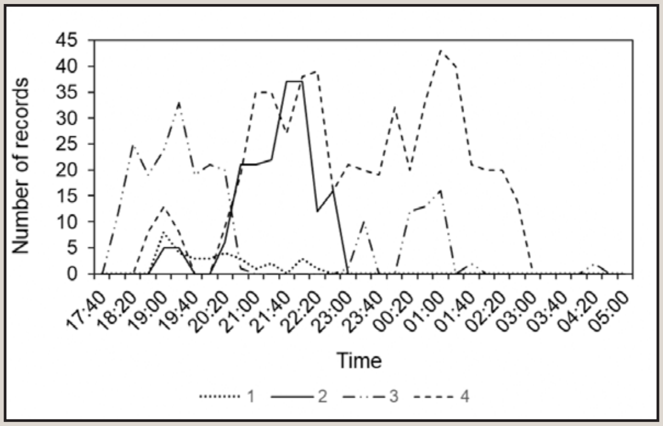

Fig. 3. Total number of records (i.e., total number of photographs taken) over time at four different bromeliads (numbered from 1 to 4) surveyed in this study, starting at 1700 h and finishing at 0500 h.

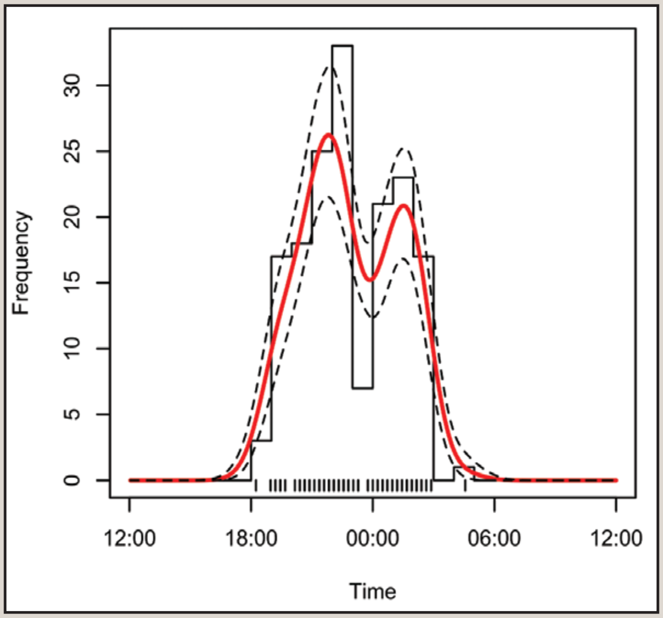

Fig. 4. Overall activity pattern of *Crossodactylodes itambe* given by the frequency of positive records (N = 993) in all surveyed bromeliads (bars), with estimated levels of activity (red line) and 95% confidence interval (dotted lines). Dashes represent a positive record at a specific time.

quickly from inactive to active) whenever frogs were disturbed; and 3) vocalizing: frog calling behavior. Notes on behaviors were only taken by one trained observer. Observations were made opportunistically (i.e., whenever these behaviors were detected during our survey) and continued until the behavior ceased. Finally, in April 2016, we video recorded the frogs in their natural habitat using a digital video camera Sony HDR-CX110E to illustrate our descriptions of behavior.

Results

In 2016, four bromeliads were surveyed using camera traps for eight nights, resulting in more than 4000 records. Despite using the same settings (except on the first survey night), the number of records over time, as well as the total records per night varied among bromeliads (Fig. 3). We had a total of 993 frog detections with the camera trap (i.e., positive records), corresponding to 24% of our records. Most of our positive records were made through time-lapse mode (83.3%). Our first positive record was at 1815 h and the last one at 0427 h, with no detections at 1700 h, 0300 h or 0500 h. Most of our positive records were made between 1900–2200 h (57.5%, N = 571), and less than 30% were made after midnight (Fig. 4). We detected two activity peaks, at 2300 h and 0100 h (Fig. 4); however, frogs were mostly inactive throughout the sampling period (Activity level = 0.26, SE = 0.02, CI = 0.22–0.30).

Through our occasional encounters we were able to record three different behaviors (illustrated in supplemental material, Fig. S1–S4). When inactive, frogs remain immobile and occupy the inner axil of the bromeliad close to the water line: the body is usually flattened, with front legs tucked under the body and head low. In most of our positive records, frogs were inactive in the leaf axils of the bromeliad, in a resting posture. When active, frogs were usually walking towards the rosette or moving to another leaf in the bromeliad. Frogs moved slowly in the bromeliad leaf, with the body raised and front legs held apart. In the camera trap, whenever active, frogs were detected moving short distances on the bromeliad leaf (Fig. 2). The only occasions where individuals moved fast were during escaping. In these cases, they jumped back into the water and did a fast dive. When diving into the bromeliad rosette, individuals usually floated in the water with stretched flat body and wide-open front and rear legs or submerged and hid within suspended sediment. We recorded a total of 34 sequences (40 min 55 sec) demonstrating these behaviors.

Within our survey period, calling behavior was rarely observed, with 10 individuals calling in 2015 and five individuals calling in 2016. The earliest calls were made at 1630 h and the latest at 2200 h. We recorded only one male calling at each site, but occasionally the same bromeliad was occupied by other individuals (usually up to two, non-calling individuals). Although we detected the species calling 15 times, we were able to directly observe this behavior on only three occasions. When calling, individuals remained close to the water line (but never submerged), with half of their body inside the water, front legs spread, and head held low at the surface of the leaf axil. Sound was produced with discrete subgular movements, with small pulses during the entire call. The call was continuous, volume was low, and maximum duration observed was 04 min 03 sec (average call duration was 02 min based on a sample size of seven records).

Discussion

Activity pattern, bromeliad use and vocalization.—We successfully investigated the activity pattern of *C. itambe* using a camera trap and our records suggest the species has an activity peak between 2100–2300 h. Although bromeliads might be used as shelter by anuran species during the day (Pertel et al. 2010; Silva, Carvalho and Bittencourt-Silva 2011), bromeligenous frogs (i.e., amphibians strictly restricted to bromeliads, where they complete their entire life cycle, *sensu* Peixoto 1995) are usually nocturnal (Cunha and Napoli 2016; Eterovick 1999; Mageski et al. 2014; Oliveira and Navas 2004). Due to our survey design, we have no records of diurnal activity; however, on only rare occasions (twice, in a very cloudy afternoon before a thunderstorm) did we observe active individuals (engaged in vocal activity) around 1600 h. Based on our data and available literature, we believe *C. itambe* is inactive during the day.

Frogs moved very short distances when active. This corroborates our observations on locomotion, which showed that frogs moved slowly inside the bromeliad. During visual encounters, individuals were never seen outside the bromeliad. From camera trapping, frogs were rarely recorded on the top of the leaf or distant from the rosette – a pattern also observed during our direct visual surveys. Although Santos et al. (2017) recorded up to four adults sharing the same bromeliad with tadpoles, using camera traps we had only one bromeliad where individuals were seen in pairs, sharing the same plant. Most of our photographs captured only one individual in the bromeliad, which seems to be the pattern for this species (Barata et al. 2018) and most bromeligenous frogs (Schineider and Teixeira 2001; Ferreira et al. 2012; Motta-Tavares et al. 2016; Santos et al. 2017).

All observations of calling males of *C. itambe* were made in the leaf axil, during occasional encounters. Cunha and Napoli (2016) observed that most calling males of the bromeligenous frog *Phyllodytes melanomystax* were preferentially positioned in the bromeliad rosette, a behavior attributed to the structure of the plant and the reduced space between leaf axils. In our case, calling males were never seen in the central tank, which was usually used only when the frog was escaping. We did not record calling activity by camera traps, but we consider this would be impractical given the subtle movements and postures associated with this behavior. In our study, only one male was recorded vocalizing in a plant. Other bromeligenous frogs were previously observed calling from the same bromeliad (Cunha and Napoli 2016) and individuals did not seem to be disturbed by other calling males (Eterovick 1999). However, for *C. itambe*, calling ceased whenever individuals were disturbed by our torches or movement.

Benefits, potential and limitations of camera traps to study amphibians.—Despite it being relatively easy for a trained observer to detect *C. itambe* during night visual surveys (Barata et al. 2017), once the species is detected it will usually quickly escape and hide among bromeliad leaves, avoiding flashlight. Camera traps were considered to have low disturbance when investigating the behavior of lizards (Bennett and Clements 2014), and we saw no evidence of camera trap affecting species behavior in bromeliads. Apart from removing bias caused by the presence of the observer, camera traps also eliminate bias in detectability caused by the difference in expertise between observers (Barata et al. 2017) because records are stored and available for independent validation.

The use of more camera trap units combined with existing analysis frameworks for time-of-detection data (e.g., Ridout and Linkie 2009; Rowcliffe et al. 2014) would allow for the collection of more robust data and the formal testing of ecological hypotheses related to activity levels. For instance, it would be possible to investigate the effect of temperature and rainfall on activity patterns, or compare levels of activity in relation to distance from hiking trails. The initial financial costs would be relatively high, but because a large amount of data on *C. itambe* can be gathered with only a few weeks of sampling, this cost could be offset by joint research projects where camera trap units are shared with other researchers. Some large mammal surveys using camera traps take place during part of the year only (for example, during dry season when sampling is more effective), potentially leaving the equipment free for other types of use (e.g., Ahumada et al. 2011; Ferreira et al. 2017).

Camera trapping provided insights into the activity patterns of *C. itambe*, but we acknowledge some limitations with the method. Firstly, the quality of some photographs was impaired by weather conditions: on some occasions the amount of mist blocked any visualization of our target—a condition that is hard to predict and difficult to mitigate. The area framed by the camera was also limited by the position of the tripod and the angle in which the camera was facing the object. In this case, observations were restricted to either a side view (showing a limited number of leaves but reaching the leaf axils close to the water), or a top view (enabling observation of the central tank and the end edge of many leaves), but never the entire plant. This resulted in a large amount of negative records (i.e., without the target species), which means frogs might have been active or inactive, but outside the camera field of view. A possible solution is to use two camera trap units surveying different parts of the same bromeliad, but this would increase the costs per plant assessed causing a trade-off between thoroughly surveying a single bromeliad or increasing the number of bromeliads surveyed in the population.

Camera traps can detect ectothermic animals if they have different temperatures from the background (Welbourne et al. 2016); however, in our study the proportion of positive records triggered by the PIR sensor is much smaller than the ones obtained through time-lapse mode. This suggests the difference in electromagnetic radiation between our target species and the bromeliad leaf (background) in most situations is below the threshold for triggering the system. Laughlin et al. (2017) suspected that positive records of arboreal frogs using camera traps were caused by the presence of an active mammal in the canopy, which triggered the PIR sensor. Even for the records triggered by the PIR sensor we cannot be completely confident that in all of them the frog actually activated the sensor, because camera traps in the field can be triggered without the presence of an animal (false triggers). Considering the current technology and available equipment, the use of camera traps to study very small ectothermic species will very likely be restricted to time-lapse mode.

Although limiting the possibilities of use, time-lapse cameras have been effectively used to study herpetofauna (Adams et al. 2017; Welbourne et al. 2017) and should be useful in self-contained habitats with high probability of use by a target species. Finally, although we captured frogs moving inside the bromeliads, we were unable to observe behaviors such as calling, reproduction or feeding with camera trapping. In a longer survey, camera traps may aid in the investigation of these behaviors but given the current technology they cannot replace direct observations in the field.

Conclusions

Camera traps were a useful tool for investigating the activity patterns of *C. itambe* without observer interference or disturbance, especially considering the escape behavior observed during direct visual surveys. Because the bromeliad is a self-contained environment, the method was successfully applied, and we recommend its use for habitat-specific species, such as bromeligenous frogs. Camera traps are now being produced with two important features: a time-lapsed trigger and a shorter focal distance. Most camera traps have a fixed focal distance restricted to a few meters, but in our case, the equipment with an interchangeable lens provided a close focal distance, which was enough to detect a small-sized species and provide good quality images. The PIR trigger system was not able to capture all events even for larger reptiles (Bennett and Clements 2014), and the use of camera traps to study very small ectothermic species will be limited to time-lapse mode. Although cameras have a high initial cost, they are considered less expensive in the long term (Welbourne et al. 2015) and might also be beneficial to obtain data from a longer time series. Although the use of camera traps to study herpetofauna has mainly been restricted to larger species of lizards and snakes (Meek et al. 2014), we have shown that within self-contained micro-habitats, using the correct equipment and the right settings will permit the study of very small frogs.

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Supplemental Material

Notes on the behavior of *Crossodactylodes itambe* made though occasional encounters. All images were extracted from video recording, which can be made available upon request.

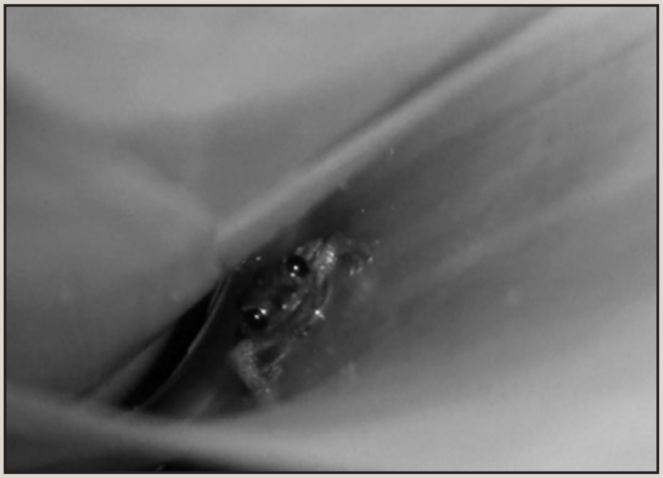

PHOTO BY M. BECHELENI

Fig. S1. Resting posture of *Crossodactylodes itambe* showing an inactive frog at the leave axil with flattened body, front legs tucked under the body and head low.

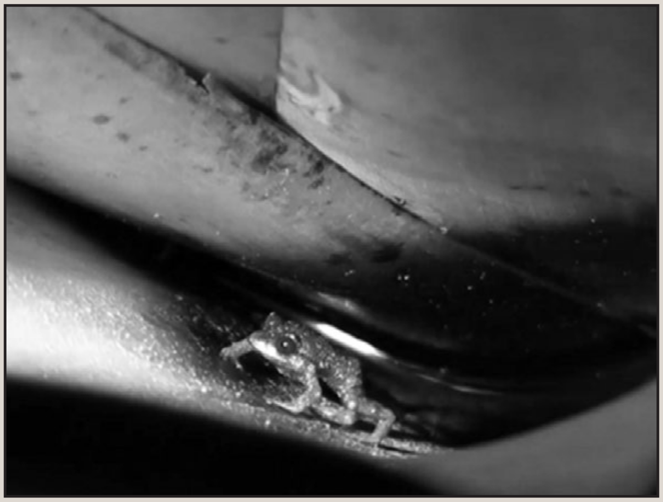

PHOTO BY M. BECHELENI

Fig. S2. Locomotion of an active *Crossodactylodes itambe* showing slow movements in the bromeliad leaf, with raised body.

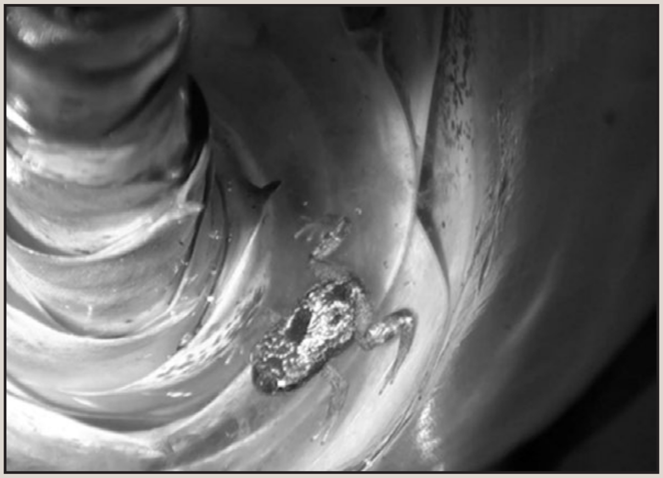

PHOTO BY M. BECHELENI

Fig. S3. Escaping behavior of *Crossodactylodes itambe* showing one individual floating in the water accumulated in the rosette, with stretched flat body and wide-open front and rear legs.

PHOTO BY M. BECHELENI

Fig. S4. Escaping behavior of *Crossodactylodes itambe* showing a submerged individual, after escaping, hiding within suspended sediment in the bromeliad rosette.

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Habitat Mapping of the Saltwater Crocodile (*Crocodylus porosus*) in Timor-Leste

Human-Crocodile Conflict (HCC) is increasing worldwide (Fukuda et al. 2014; Amarasinghe et al. 2015; Pooley 2015; CrocBITE 2018), but limited financial and technical resources in many developing countries impede scientifically sound management to mitigate such conflicts. Analysis of crocodilian habitats is an important step for effective HCC management and crocodilian conservation (Thorbjanarson et al. 2006; Leach et al. 2009; Ihlow et al. 2015). Using Timor-Leste as a case study, here we demonstrate how potential habitats of Saltwater Crocodiles (*Crocodylus porosus)* can be mapped using cost-free tools and data from the World Wide Web, and local knowledge. One of the least developed countries in the World, Timor-Leste struggles to manage HCC with *C. porosus*, the largest and one of the most aggressive crocodilian species (Britton et al. 2012; Brien et al. 2013), the population of which has increased significantly in Timor-Leste since its independence from Indonesian occupation in 2002. Since 1996 at least 130 people have been attacked by crocodiles in Timor-Leste, yet the information available is insufficient to identify priority areas for a conservation scheme on a national scale (NBWG 2015; Sideleau et al. 2016; Brackhane et al. 2018). Thus, we suggest that crocodile habitat mapping is an important first step towards the development of the first crocodile management plan for the country.

We performed habitat analysis based on Geographic Information Systems (GIS) to identify: 1) core habitats (including perennial waterbodies such as lakes, swamps, billabongs and rivers providing possible breeding sites for *C. porosus*); 2) coastal marine habitats, *inter alia*, *C. porosus* perennial range for hunting; and, 3) seasonal range, namely potential habitat for *C. porosus* during the wet season.

Timor-Leste is dominated by a mountain ridge ranging from the westerly Mount Ramelau (2963 m elev.) to the Fuiloro Plateau in the east, which includes Timor-Leste's largest lake, Ira Lalaro (318 m elev.) (GERTIL 2002). These elevated areas divide the country into northern and southern parts with distinct seasonal variability affected by the West Pacific Monsoon (PCCSP 2011). Maximum precipitation is only reached in the high elevations of the central mountain ridge. The relatively dry northern coast is characterized by a rainy season from December to March with 50–350 mm precipitation/month, a transition period in November, April and May with 50–150 mm precipitation/month; and a distinctive dry season from June to October with little or no rainfall (0–50 mm precipitation/month) (Seeds of Life 2013). In contrast, the climate along the southern coast is characterized by a rainy season from December to June with 100–400 mm precipitation/month, a transition period in July and November with 50–150 mm precipitation/month; and a dry season from August to October (0–50 mm precipitation/month).

Almost all creeks and rivers originate in the central mountain ridge, with many of them running dry during the dry season. The number of perennial rivers, which potentially can be inhabited by crocodiles throughout the year, is limited to three rivers in the northern part (Northern Lacló, Seiçal, Loes) and eight in the southern part (Irabere, Bebui, Dilor, Tafara, Belulik, Caraulun, Southern Lacló, and Clerec). Potential habitat for *C. porosus* is limited to a narrow plain between the coast and the mountain ridges, and includes mangroves, mainly along the northern coast (Alonghi and Carvalho 2008), and various billabongs, lagoons, estuaries, floodplains, and swamps, especially in the southern part as a result of the higher precipitation patterns (Fox 2003).

Core crocodile habitats were identified based on knowledge of members of the Crocodile Task Force (CTF) who were familiar with the areas through regular sightings of crocodiles, and traditional ecological knowledge of local stakeholders from eight communities (Vessuro, Mehara, Uani Uma, Malahara, Com, Baucau, Hera, Irabin de Baixo) affected by HCC. Usually, local knowledge holders, Xefe Suku (Village headmen), Dato Lulik/Lia Na`in (Traditional elders) or local fishermen were contacted during surveys (2007–2017) and asked to identify areas where saltwater crocodiles reside throughout the year. The identified areas were then inspected by the CTF to determine numbers of crocodiles and to measure GPS coordinates of control points of the relevant waterbodies using (Garmin) hand devices. The measured control points were transferred to a GIS where the extent of crocodile core habitat could be identified and measured based on Google satellite imagery (https://www.google.de/maps). As a sea-going species, with the potential for long-distance movement (Read et al. 2007; Campbell et al. 2010; Webb et al. 2010), *C. porosus* may be found along the entire coastline of Timor-Leste throughout the year. We applied a 1-km buffer to coastlines and core (buffer only inwards) habitats digitized by GIS to account for seasonal variations in the habitat extent as well as adjacent coastal habitats. These areas, accounting for 341.6 km² of core habitat (126.3 km² excluding the 1-km buffer) and approximately 766 km of coastline, represent the habitat where *C. porosus* can be found during the whole year.

We followed Brackhane et al. (2018) in defining the areas of potential crocodile range as temporary waterbodies with a 3-km buffer to include seasonally possible, but unusual habitat for saltwater crocodiles. These include rice paddies and the canals

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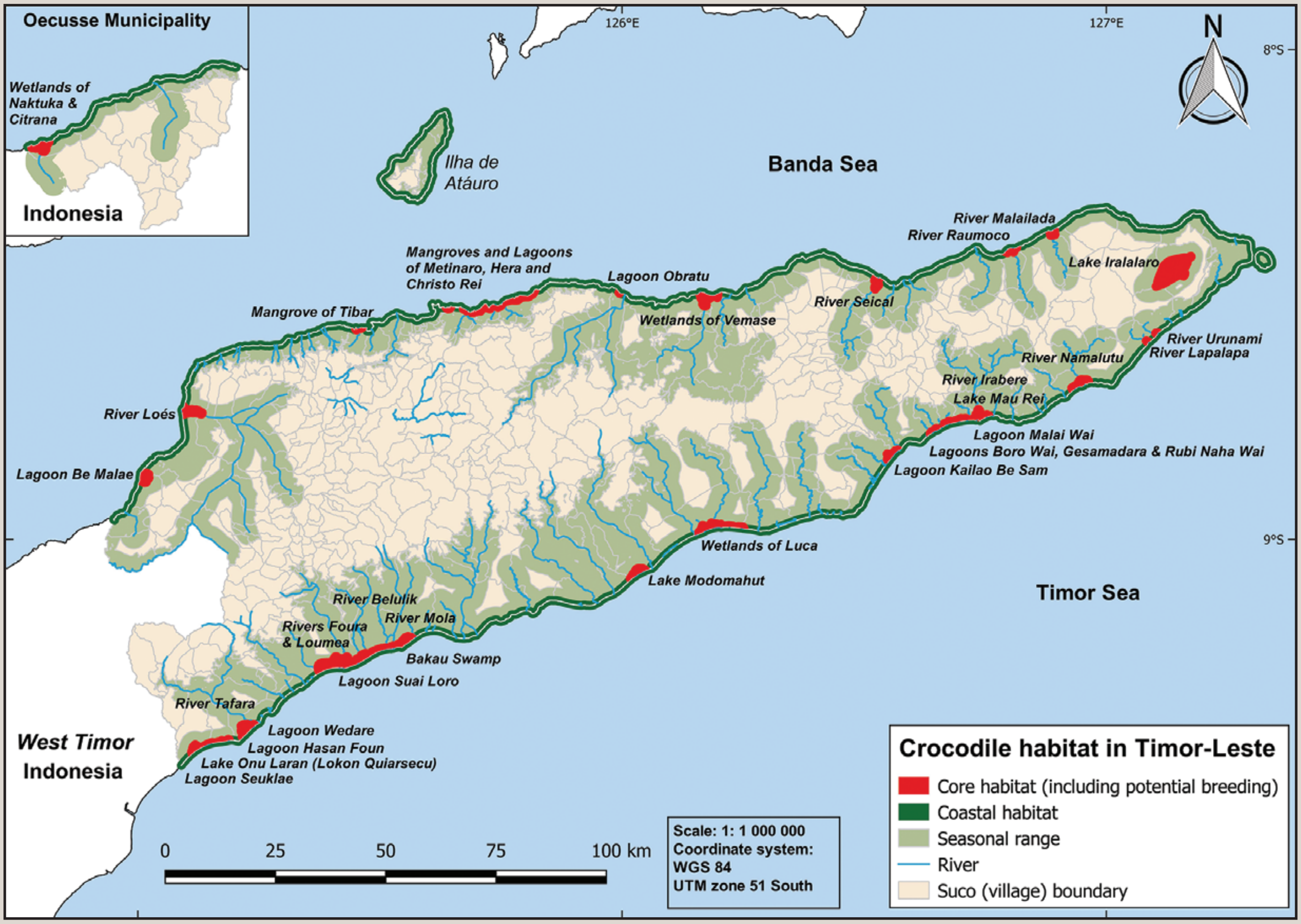

FIG. 1. Potential core, coastal, and seasonal habitats of Saltwater Crocodiles in Timor-Leste.

of the associated irrigation systems. As there were no reported sightings of *C. porosus* at altitudes above 500 m, we excluded all areas above this altitude from the identified core habitats and seasonal range using 3-arc second Shuttle Radar Topography Mission (SRTM) Digital Elevation Model (DEM). The highest observed record of a Saltwater Crocodile in Timor-Leste is at 422 m elev., in Lete Foho, Manufahí district (9.14331°S, 125.58883°E; S. Brackhane, pers. obs.).

All spatial analysis was performed using the free QGIS 2.18 software including the relevant extensions such as OpenLayers Plugin for spatial analysis of Google satellite imagery and QuickMapServices 18.4 Plugin for basemaps (QGIS Development Team 2017). SRTM DEM was downloaded from SRTM 90m Digital Elevation Database v4.1 (http://www.cgiar-csi.org/data/srtm-90m-digital-elevation-database-v4-1, accessed on 6 March 2017). Additional information such as administrative boundaries and waterbodies were downloaded at no cost from Geographic information Group (GIG) Timor-Leste (https://sites.google.com/site/gigtimorleste/data/administrative-boundaries, accessed on 6 March 2017) and Diva-GIS (www.diva-gis.orghttp://www.diva-gis.org/, accessed on 6 March 2017).

The map generated (Fig. 1) may be used by the CTF to identify priority areas for HCC management, i.e., to link management activities to areas where human and crocodile habitat frequently overlaps, especially by overlaying it with a current risk map showing hotspots of crocodile attacks or human density maps. Also, if made available through a website (e.g., www.common-environment.org or peskador.org), the map would help wildlife managers to raise public awareness and to inform local residents and tourists about areas of potential crocodile risk. The map will be improved as more crocodile-sighting and environmental data become available through field surveys or community-based monitoring approaches as described by Brackhane and Pechacek (2015). In particular, inclusion of crocodile sightings made by citizen scientists would be a cost-effective option to improve data availability (Brackhane et al. 2016). *Crocodylus porosus* habitat mapping will significantly improve when integrated with comprehensive vegetation data (Harvey and Hill 2003; Fukuda and Cuff 2013). In this context, the continuous assessment of available habitat suitable for nesting will be a crucial variable to estimate the capacity for a viable population of *C. porosus* in Timor-Leste (Magnusson 1980), and to inform wildlife managers on crocodile conservation needs.

Our case study here could hopefully serve as an example of the first step to habitat mapping for many developing countries, particularly those with limited resources, and facing HCC challenges like Timor-Leste. Whereas the methodology may be relatively easily applicable on islands with a rather simple habitat distribution such as Palau (Brazaitis et al. 2009), Vanuatu, Solomon Islands and several islands of volcanic origin in Papua New Guinea and Indonesia, its application to larger countries with extensive freshwater lakes and swamps, floodplains and large river systems may require more systematic assessment of habitat quality (e.g., Fukuda et al. 2007; Fukuda and Cuff 2013).

Particularly in these areas with high geographical complexity, the integration of local knowledge may constitute a valuable data source increasing the accuracy of habitat mapping. We caution that the presented methodology is designed for *C. porosus* and may be misleading for other crocodilian species.

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Record Life Span in a Population of Timber Rattlesnakes (*Crotalus horridus*)

Some living reptiles, including many turtles and crocodylians, and a few squamates, have evolved long life spans and typically display the following demographic traits: late age of maturity, low fertility, and high adult survival (Pough et al. 2016). These traits characterize many populations of *Crotalus horridus* throughout its North American range (Martin et al. 2008). Although *C. horridus* displays several life history correlates of long life span, longevity has seldom been measured in a wild population of any long-lived snake. At a study area in northeastern New York that has been continuously sampled over a 39-year period, we noted occurrences of recaptured snakes that evidently displayed exceptional longevities. This paper documents the longest-lived observed individuals—the record life span (Carey 2003)—in a wild population of timber rattlesnakes, and suggests evolutionary correlates in its life history and reproductive biology.

METHODS

Sampling and Marking Procedures.—The study locality (43–44°N latitude) comprises a series of dens (a metapopulation, herein called a population), spread out over a linear distance of ca. 13 km in mountainous terrain in northeastern New York. Most of the study site encompasses unbroken forests and woodlands over a large part of the total area occupied by the local populations (Brown 2008). Dens are interconnected by snake movements to summer mating areas and therefore show only modest levels of genetic differentiation, allowing the study area's population to be considered as a single panmictic deme (Clark et al. 2008). The study area has been sampled continuously by WSB every year from 1979 through 2017. Permanent marking of rattlesnakes was accomplished by ventral scale-clipping (Brown and Parker 1976); healed clip-scars were clearly identifiable, allowing all snakes to be certainly known at recapture. Once clipped, the scars persisted permanently over the natural life span of *C. horridus* at this locality.

A recent 15-year study period (2003–2017) was selected as an appropriate interval to evaluate records of snakes having capture histories of two decades or more; these snakes, if they could be aged at their initial capture (most as young individuals), permitted estimates of longevity by simply adding the intervening number of years in a snake's capture history to its initial age. We calculated rattlesnake life span estimates from eight different dens (range 5–22 estimates per den) in the population. We recorded all new (unmarked) snakes (N = 866) and recaptures (N = 820) that occurred during the 15-year recapture interval. Among the recaptures, we subtracted all repeat captures for the same individual in the same year (N = 63), arriving at a sample of adjusted recaptures (one recapture per individual per year). In this recapture group (N = 757), we recorded all known-age snakes that were at least 20 years of age; this arbitrary requirement yielded a subsample (N = 81) that contained snakes with remarkably long life spans.

Statistical tests of spatial (among dens), temporal (among years), and sex ratio effects were completed using Statistix 9 (Analytical Software, Tallahassee, Florida); probability level was set at 0.05. Mean values of samples are followed by ± one standard deviation (SD) with extremes in parentheses.

Aging Methods.—We adopted an aging procedure that allowed us to avoid biased over-estimates of the snakes' ages. This method produced a probable rattle size upon which the age estimate was based. At their initial capture, three characteristics of each snake's rattle were recorded as follows: 1) rattle entirety—string complete or broken; 2) degree of taper (full, slight, or none—generally found in young, middle-aged, and older snakes, respectively); and 3) estimated number of missing segments (button [b] only, 1+b, or 2+b). The sample consisted of 44 complete rattles (54%) that could be aged with the highest degree of accuracy, including 11 young-of-year and 10 1-yr-old or 2-yr-old snakes (b-only to complete 3+b rattles) whose ages were confidently ascertained by applying the finite (or actual) shedding rate of 1 shed/yr or 2 sheds/yr characterizing virtually all rattlesnakes in this population (Brown 1988). In addition, the sample consisted of 37 broken rattles (46%) among which 19 had a full taper with 2+b, 1+b, or b-only missing; thus, 63 of the 81 snakes (78%) with complete or broken but discernibly complete rattles could be aged most accurately. To deal with an additional 18 broken rattles with slight or no taper and lacking a reliable count of missing segments (i.e., missing >2+b), we first calculated the mean number of segments of all broken slight-taper rattles in the population—8.96 in 724 females, 8.59 in 564 males, or about 9 segments overall—then, using these data, we added 9 to the existing segment count of broken no-taper rattles (N = 6), thus providing an estimate of the number of segments that the rattle must have had, had it been complete. For those with broken slight-taper rattles (N = 12), 0 to 5 segments were added as appropriate for each individual's reproductive condition (nine were gravid females) and body mass. This technique is notably conservative, i.e., there may have been more than 9 segments in the remaining rattle, but only 9 or fewer additional segments were allowed in our additive procedure. Age estimates were calculated by dividing the actual or the estimated segment count by the mean annual shedding rate (i.e., the consequent addition of new segments) = 1.4 segments/yr (Brown 1988). It is prudent to assume a probable error of ±1 year in each snake's estimated age. Our correlations of rattle size and age are nearly identical with those of *C. horridus* in the Blue Ridge Mountains of Virginia (Martin 1993).

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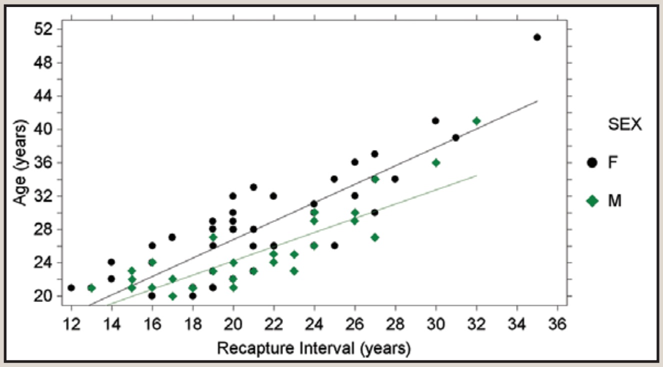

Fig. 1. Relationship between recapture interval and age in a sample of males (M; diamonds) and females (F; solid circles) that provided record life span estimates in a population of Timber Rattlesnakes (*Crotalus horridus*) in northeastern New York. Trend lines show best-fitting relationship in females (upper line) and males (lower line).

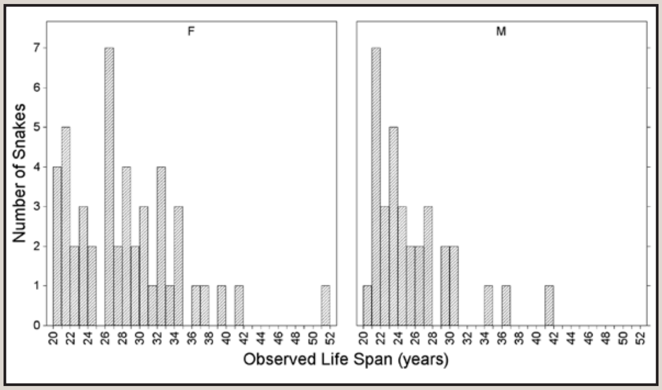

Fig. 2. Distributions of record life spans in a population of Timber Rattlesnakes (*Crotalus horridus*) in northeastern New York. Data are based on a sample of 81 recaptured snakes, 48 females (F, left side) and 33 males (M, right side) aged ≥ 20 years.

PHOTO BY MATT SIMON

Fig. 3. Male *Crotalus horridus* on 12 August 2012, estimated to be 41 years old.

PHOTO BY MATT SIMON

Fig. 4. Female *Crotalus horridus* on 12 May 2016, estimated to be 51 years old.

Results

Among a total of 757 recaptures (mean 51/yr, range 22–69/yr), 81 individuals (11%) were aged definitively, thus qualifying for inclusion in our 15-year subsample and providing data for estimation of the longevity in this species at the study locality. The record life spans were 41 years (in one male and one female) and 51 years (in one female).

Recapture intervals averaged 21.1 ± 4.8 (12–35) yr. At their initial capture, ages of snakes were 5.7 ± 3.4 (0–16) yr, and at their final recapture their ages were 26.8 ± 5.9 (20–51) yr. Sex ratio of snakes in the life-span subsample favored females (N = 48, or 0.59) over males (N = 33, or 0.41), as did the sex ratios among all recaptures (females N = 395, or 0.52; males N = 362, or 0.48) (two-by-two ratio test, Yates' corrected $\chi^2 = 1.20$, P = 0.27). Spatial and temporal tests showed that there was no significant difference in the final age estimates among dens (*F* = 1.09, P = 0.37, df = 7) or years (*F* = 0.97, P = 0.49, df = 14), indicating a population-level generality of the age estimates.

Females had a greater mean age (27.9 ± 6.3 yr, N = 48) than did males (25.2 ± 4.8 yr, N = 33) (*t* = -2.07, P = 0.042, pooled equal variances) (Fig. 1). In the determinative subsample, 22 individuals (27% of the sample) exceeded 30 years. Among males, there were five individuals ≥ 30 yr (15% of males); among females, there were 17 individuals ≥ 30 yr (35% of females) (Fig. 2). Among all individuals of both sexes, age distributions were: 20–29 yr (N = 59, or 73%), 30–39 yr (N = 19, or 23%), 40–49 yr (N = 2, or 2%), and 50– 59 yr (N = 1, or 1%).

The record life span among males belongs to one snake that was initially marked in May 1980 (8-yr-old: rattle broken–11, full taper, b-only missing) in his ninth year. He was recaptured five more times, finally as late as 2012 as a 41-yr-old (Fig. 3). A day prior to his final recapture, this male was observed accompanying an adult female, so he apparently was reproductively active.

The record life span among females belongs to one snake that was initially marked in May 1981 at an estimated minimum age of 16 yr (rattle broken–14, no taper, estimated total segment count = 23). She was recaptured four more times, finally as late as 2016 as a 51-yr-old (Fig. 4). The reproductive history of this female showed that she was gravid at her initial capture and at each of her three subsequent recaptures (one gravid record per decade in a 32-year interval between 1981 and 2012, but lacking captures in a number of probable intervening gravid years). Her final capture in 2016 showed that she was not gravid, with a robust body mass (904 g) typical of healthy adult females. Together with a small group of three other females that had moved ca. 200 m from their den in the early spring, she exhibited a normal escape attempt and a normal defensive posture when captured. Her advanced age was not evident.

Discussion

This analysis is not intended as a description of the age structure of this population of *C. horridus*, but rather draws attention to selected individuals that exhibited the longest recapture intervals and ages. Measuring maximum life spans and physiological correlates of aging in exceptionally long-lived species is an active field of gerontological research (Robert and Bronikowski 2010; Alper et al. 2015). Long-lived lepidosaurs generally are characterized by late age of first reproduction, infrequent reproduction, small clutches or litters, and high-latitude or cool-habitat geographic ranges (Scharf et al. 2015).

Our data demonstrate record life spans in the wild that have not previously been demonstrated or that could not be predicted in many, perhaps most, snake species as reported in three literature summaries: 1) among eight species of boids, colubrids, elapids, and viperids, maximum ages range from 21 to 29 years (Goin et al. 1978); 2) a survey of 115 species of snakes in eight families shows the maximum ages attained were about 30 years (Gibbons 1976); and 3) a recent global analysis of known longevity in squamates and tuatara includes summary data for 336 species of snakes whose maximum life span averages 15.8 yr (range 3.4–47.5 yr) (Scharf et al. 2015).

Among 36 species and subspecies of rattlesnakes (*Crotalus*), maximum life spans in captivity averaged 14.2 ± 6.3 yr (range 3.5–30.2 yr), with a median age of 13.6 yr (calculated from "Record Life Spans in Amphibians and Reptiles" in Carey and Judge 2000). A second more recent dataset for 48 taxa of *Crotalus* from captive records yielded a mean age of 15.6 ± 5.3 yr (range 5.7–30.2 yr), with a median age of 15.1 yr (calculated from data in Snider and Bowler 1992). There was no difference between median values in the two datasets for *Crotalus* spp. (median test $\chi^2 = 0.78$, $P = 0.378$). Interestingly, the maximum record in these data (30.2 yr) belongs to *C. horridus*. Other records available are for a single captive male that survived for 36.6 years (Cavanaugh 1994), and a marked wild-caught adult male recaptured after 24 years at an estimated age of 27 yr (Fitch and Pisani 2002).

A major problem in assessing the statistical context for record life spans in lizards and snakes has been a lack of reported sample sizes for maximum longevity, many of which are based on just a single record (Scharf et al. 2015). We have attempted to evaluate the longevity of *C. horridus* in the context of random annual samples of snakes of all sizes and ages over a period of almost four decades. Based on all recaptures, estimated ages of the oldest snakes so far known in this population of timber rattlesnakes may exceed 50 years in some exceptional individuals.

Among the individuals composing our 15-year determinative subsample, but lacking later recaptures after their final record, we cannot know whether mortalities in any snakes included in the sample might have occurred or might still occur. Therefore, our data represent the observed individuals that were alive at their final recapture but that may not represent a final age that could yet be attained. Some of these snakes, if they survive through the next several years, are expected to advance into their forties. Record life-span data reported here may be amended in the future if individual snakes manage to survive longer than the cut-off year (2017) currently adopted. Further, in the early years of the study when some snakes were already old and could not be aged initially, we suggest that the age distribution actually could be composed of a higher proportion of older snakes than is indicated by our "time slice" through the population.

Several researchers that study longevity and aging in vertebrates have pointed out a distinction between environmentally imposed mortality, or extrinsic mortality, caused by predation, disease, and weather; and mortality attributable to senescence, or intrinsic mortality, caused by the degradation of essential physiological or biochemical functions with age (Kardong 1996; Bronikowski 2008). Snakes, as ectotherms with indeterminate growth, have contributed to aging analyses and the so-called pace-of-life (or rate-of-living) hypothesis that focuses on several variables such as mitochondrial function and reactive oxygen species production, DNA damage and repair, and corticosterone-produced stress effects. These variables have been tested in contrasting populations of a colubrid (*Thamnophis elegans*) that are composed of fast-growing, short-lived ecotypes vs. slow-growing, long-lived ecotypes (Bronikowski and Vleck 2010; Robert and Bronikowski 2010). Results of this work demonstrate support for physiological mechanisms and their correlation with natural genetic variation, producing a number of divergent life-history traits in *T. elegans*—perhaps not unlike the geographic variation in morphology and life history documented in *C. horridus* (Martin et al. 2008).

Selection acting directly on reproduction and indirectly on adult survival rates through tradeoffs with reproduction will result in the evolution of a longer life span if extrinsic mortality rates decrease in older (and larger) animals, thus increasing the value of older individuals because of their increased contribution to reproductive success (Stearns and Hoekstra 2005). In the presently studied population of *C. horridus*, adult males with broken non-tapered rattles and body mass ≥ 1000 g averaged 1171 g (N = 117), but, other than quantifying maximum size in males, this datum is devoid of life history inferences which can be more plausibly applied to adult females.

In females, age at first reproduction is delayed (9.6 yr), reproductive cycles are long (4.2 yr), and lifetime reproductive rates are low (1.7 reproductive events per female) (Brown 2016). As adults, survival rates are high (≥ 90% per year, Brown et al. 2007) and apparent extrinsic mortality (predation, disease) is probably low, but there are physiological costs of reproduction in females (intrinsic mortality) that result in most (60%) being able to reproduce only once (Brown 2016). However, a considerable increase in reproductive potential in larger and older females is a prominent feature in these snakes. Grouped according to their expected sequence of lifetime reproduction ranging from one to four gravid events, body masses increased by 202 g (from 737 to 939 g; N = 196 records) in females from one focal den, and by 328 g (from 827 to 1155 g; N = 152 records) in females from a second focal den. These mass changes encompass the growth histories of snakes in their gravid years before reaching a size plateau by the fourth effort at an average age of approximately 22 years (Brown 2016). This increase in body mass would be sufficient to add two or three additional neonates to the litters (averaging 7.5 and 8.4 in the two dens, respectively) of older females weighing up to 1000 g or more, assuming an approximate ratio of 100 g of female mass required to produce one additional offspring (Brown 2017). Therefore, from available data on body size and growth in reproductive females, it may be inferred that the low lifetime reproductive rate is accompanied by selection for a long maximum life span. A slow-paced life history representing tradeoffs between reproduction and life span has evidently evolved in these rattlesnakes.

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Note Added in Proof.—In accord with our prediction, a 41-year-old female in 2016 was recaptured in July 2018 at an estimated age of 43 yr.

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Evaluating the Potential for Bias with Common Amphibian Protocols

Most ecological studies assume that standard protocols and sampling methods accurately sample populations in an unbiased way (Heyer et al. 1994). Violations of these assumptions can yield biased results or invalid conclusions, which could negatively influence management or conservation efficacy (Mazerolle et al. 2007; Kroll et al. 2008; Cecala et al. 2013). Extensive studies have evaluated methodological factors that change the detection probabilities of organisms including trapping methods, time or day of year, and habitat characteristics (Gamble 2006; Todd et al. 2007; Connette et al. 2015). However, few have investigated whether the captured individuals of a species are representative of the studied population as a whole (e.g., Willson et al. 2008; Michelangeli et al. 2016). Furthermore, little information is available for accuracy of measurements from standard protocols (e.g. Roitberg et al. 2011). Because amphibians are declining at unprecedented rates and represent an important taxon for understanding ecological and evolutionary phenomena (Stuart et al. 2004; Adams et al. 2013; Grant et al. 2016), determining if standard methods could introduce previously undocumented bias is important for future studies of amphibians (Grant 2014; Connette et al. 2015).

Variation in capture methodology has received recent attention as researchers become aware that passive capture methods can bias samples towards individuals with particular behavioral syndromes even with random sampling of available habitats (Biro and Dingemanse 2009; Carter et al. 2012; Biro 2013). Because behavioral traits are heritable (van Oers et al. 2004), differential capture rates associated with behavioral syndromes could introduce bias in studies of any number of physiological, behavioral, or life history traits (Biro and Stamps 2008; Wolf and Weissing 2012). Wilson and colleagues (1993) discovered that behavioral syndromes could contribute to extreme sampling bias where some individuals were trapped repeatedly whereas others were never captured. For individuals more likely to take risks and explore novel objects, they may be more likely to be captured in passive traps (Biro and Dingemanse 2009; Stuber et al. 2013). Furthermore, high activity levels common to bold individuals may also result in more encounters with passive sampling techniques (e.g., traps, gill nets, etc.; Stuber et al. 2013). In harvested populations of fish, these biases led to a population of individuals that were less active, less exploratory, and less likely to take risks (Biro and Post 2008). Similarly, individuals with different behavioral types could use habitats differently such that sampling could target only a particular behavioral type (Wilson et al. 2011). For example, sampling of open-water aquatic habitats could sample bold individuals relative to shallower areas that offer refugia for shy individuals (Wilson et al. 2011). Few studies have investigated potential biases associated with active capture techniques, but a study on Delicate Skinks (*Lampropholis delicate*) did not observe behavioral differences in individuals captured by hand relative to passive methods of capture (Michelangeli et al. 2016). Ultimately, population studies that do not account for bias associated with capture techniques could underestimate population sizes and lead to biased conclusions about the status of a population (Crespin et al. 2008; Pradel et al. 2010; Olivier et al. 2017).

Capture methods could also introduce biases associated with standard measurements particularly if species exhibit ontogenetic shifts or size-determined distribution patterns that could bias samples towards smaller or larger individuals of a population (Werner and Gilliam 1984; Hairston 1987; Todd and Winne 2006). Other implementations of standard protocols such as the use of anesthesia could result in higher accuracy of length measurements (Setser 2007), but impacts of anesthesia on measurements of mass are unknown. Furthermore, body length may change with environmental conditions making it critical that this variation can be attributed to environmental conditions rather than measurement error (Bendik and Gluesenkamp 2013). Mass could also be impacted by stomach contents that would result in larger mass measurements. Generally, feeding status or prey mass is unknown for wild-captured individuals, but holding individuals until digestion is complete could result in more accurate assessment of mass. For example, diet studies of Eastern Red-spotted Newts (*Notophthalmus viridescens*) found individuals consuming up to 55% of their body mass introducing a positive bias in morphological studies (Burton 1976; Dimmit and Ruibal 1980).

In this study, we evaluated if common practices in amphibian ecology could bias results. We determined if active versus passive capture techniques affected morphometric data or was biased towards a particular behavioral type. Once individuals were captured, we also investigated how time since capture and anesthetization impacted morphometric measurements. As a case study, we evaluated these methods on measurements of length (snout–vent length; SVL), mass, and exploratory behavior of *N. viridescens*.

METHODS

Adult *Notophthalmus viridescens* were captured from Lake Cheston in Franklin County, Tennessee, USA. We had two sampling periods from October to November 2016 and in March 2017. In the fall, we quantified the effects of anesthesia on morphometric data. In the fall and spring, we quantified the effect of capture method on morphometric data and behavioral data. We captured individuals by active dipnetting up to 1 m from the shore or by plastic minnow traps (Shaffer et al. 1994; Graeter et al. 2013). Minnow traps were set approximately 0.5 m from shore around the perimeter of the lake where emergent vegetation was absent. Traps were set at least 5 m from one another and checked every 24 h while deployed. Upon capture, newts were placed in

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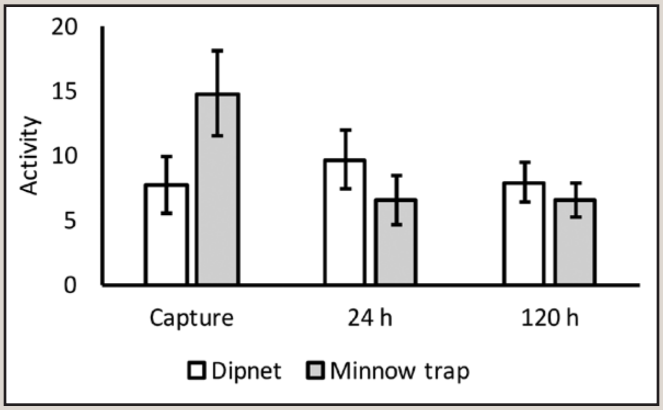

FIG. 1. Mean (± SE) activity of *Notophthalmus viridescens* at different time intervals after capture. Activity was measured as the number of 25-cm^2 squares entered by an individual in 5 minutes. Activity was highest immediately after capture for individuals captured in minnow traps.

a behavioral arena (described below), measured and weighed before placed in an 8 oz deli cup with lake water and a paper towel for the remainder of the experiment. Morphometric data was always collected after the behavioral assay to minimize the effects of handling stress on behavior. Individuals were kept without feeding at 11°C for five days before being released within 20 m of their capture location.

We evaluated morphometric and behavioral data from 52 individuals captured in the fall and 41 individuals captured in the spring. Effects of anesthesia were quantified from 40 additional individuals captured in the fall. We measured individual SVL to the nearest mm (to the posterior edge of the cloaca) and mass to the nearest 0.01 g (Fellers et al. 1994). Morphometric data were collected immediately upon capture, 24 h and 120 h after capture to evaluate if capture method or time since capture impacted measurements of length or mass. To determine if anesthesia impacted these same measurements, we measured length and mass before and during anesthetization. All evaluations of anesthesia were conducted within six hours of capture. Individuals were measured immediately before being placed in a 1 g Oragel L^{-1} solution with buffered dechlorinated water (Cecala et al. 2007). Once individuals were unresponsive to a toe pinch, we removed individuals from the anesthesia bath, rinsed and measured them. Individuals were allowed to recover on a wet paper towel until their righting reflex was restored before being released at their capture location.

To determine if behavioral traits were different between individuals captured in a passive (minnow trap) or active (dipnetting) method, we evaluated individual activity levels immediately upon capture in the field and again 24 h and 120 h after capture in the lab using the same behavioral arena. The behavioral assay was performed using a 5-gal tank with a 5-cm grid drawn on the bottom filled with 5 cm of lake water refreshed for each individual. Each individual was tested independently by being placed under a dark cup in a randomly selected corner for 30 s before the cup was removed. For five minutes, we recorded the number of boxes the individual entered with all four legs.

All data were analyzed using linear mixed models using individual as a random factor using package lme4 in R (Bates et al. 2015; R Development Corp 2016). Because time is a variable of interest, we included it as a fixed factor in each of our models of morphological and behavioral data alongside capture method. The effects of anesthesia only included time (before or during) as a fixed factor. Satterthwaite approximations of F-ratios were used to evaluate significance using package lmerTest (Kuznetsova et al. 2014), and posthoc tests of significant main effects were evaluated using Tukey p-value adjustments. Means are presented with standard errors (SE).

RESULTS

Notophthalmus viridescens mean SVL was 40.8 ± 0.28 mm, and mass was 2.32 ± 0.04 g. We captured 42 individuals using minnow traps in the fall and 21 in the spring. Active dipnetting surveys captured 20 individuals in the fall and 20 in the spring. Statistical models had neutral residuals. Capture method did not affect length ($F_{df = 1, 201} = 1.19$, P = 0.278) or mass of individuals ($F_{df = 1, 201} = 0.02$, P = 0.884), but activity was 79.2 ± 33.2% higher in individuals captured in minnow traps ($F_{df = 1, 201} = 5.20$, P = 0.033). Time significantly affected measurements of mass ($F_{df = 2, 201} = 4.41$, P = 0.039) and activity ($F_{df = 2, 201} = 4.72$, P = 0.033). Mass declined 0.079 ± 0.013 g between capture and 120 h (z = -3.18, p = 0.004) but was not different between capture and 24 h (Z = -1.41, P = 0.335). We observed a significant interaction between capture method and time on activity ($F_{df = 2, 201} = 5.20$, P = 0.025). Specifically, individuals captured using minnow traps exhibited high levels of activity at capture that returned to levels similar to individuals captured in dipnets and remained consistent between the two later time periods (Fig. 1). Anesthesia did not affect measurements of length ($F_{df = 1, 38} = 0.062$, P = 0.805) but did affect measurements of mass ($F_{df = 1, 38} = 6.12$, P = 0.019). Individuals weighed 0.032 ± 0.012 g less under anesthesia than they did before the process.

DISCUSSION

These studies documented that common amphibian capture methods of dipnetting and minnow traps capture individuals with similar characteristics. Significant effects of capture method on activity likely represented an immediate escape behavior that diminished through time (e.g., Morellet et al. 2009; Seress et al. 2017). We observed consistent negative effects of time and anesthesia on mass though these changes were less than 3% of adult mass in our study population. Finally, our study provides support for allowing comparisons of length among salamander studies that did or did not use anesthesia.

Novelty associated with introduction of passive sampling tools into a pond does not appear to introduce bias. Despite testing only a single behavior (Sih et al. 2004), we observed consistent exploratory behavior of individuals captured using passive or active techniques after the initial behavioral assay (Michelangeli et al. 2016). These results are counter to observations that passive techniques tend to capture bolder or more active individuals than shy, less active, or neophobic individuals (Biro and Dingemanse 2009; Stuber et al. 2013), but we recommend future evaluation of individuals from different habitat types (Wilson et al. 2011). Although individuals in our experiment might have demonstrated different behavioral traits in another assay, tendencies towards exploration or activity are frequently used traits to characterize behavioral syndromes (e.g., Sih et al. 2004; Dingemanse et al. 2007; Minderman et al. 2009). Furthermore, consistency between the two post-capture time intervals (24 and 120 h) provides additional support that these are indicative of individual tendencies (Sih et al. 2004). Results from this behavioral survey suggest that researchers carefully consider capture methods and timing until the first test of behavior. Individuals captured using active techniques exhibit immediate behavioral differences by exploring a novel enclosure more than individuals captured using passive traps or if individuals were contained for 24 h prior to testing. Capture can induce an acute stress response, and this initial exploratory behavior may be associated with capture stress and search for escape (Morton et al. 1995; Romero and Reed 2005). Therefore, we recommend that behavioral studies with individuals captured using passive methods refrain from initiating studies until 24 h after capture.

Small, but consistent, declines in mass measurements were observed through time and with anesthesia. Despite small changes, these could amplify if measurements are extrapolated for biomass estimates (e.g., Burton and Likens 1975; Semlitsch et al. 2014; Milanovich and Peterman 2016). We recommend that researchers maintain consistency in measurement protocols among samples taking into account time since capture and the use of anesthesia. Digestion and absence of feeding even over short temporal intervals were sufficient to change mass measurements. We are unaware of other studies documenting declines in mass associated with anesthesia, but suggest that it could be an osmotic response to the presence of a dissolved anesthetic or inhibition of physiological processes (Feder and Burggren 1992; Hillman et al. 2009). A future study should investigate if this effect is similar or more extreme in Plethodontid salamanders with highly permeable skin (Wells 2007; Hillman et al. 2009). Another study also found that body length and mass declined following preservation indicating that live measurements with or without anesthesia should not be compared to preserved specimens (Shu et al. 2017).

Methodological variation has been suggested as one alternative explanation for observations of declining salamander body size through time (Caruso et al. 2014; Grant 2014; Connette et al. 2015). No variation in length was linked to time or use of anesthesia in our study. Two additional variations of methodology should be tested. First, precision among personnel should be evaluated because confusion can exist between whether researchers measure to the anterior or posterior end of the cloaca, and experience with measuring amphibians could also impact precision (Roitberg et al. 2011). Secondly, anesthesia can relax the muscles of individuals potentially allowing for longer measurements such as in snakes (Setser 2007), but we did not observe this pattern for salamanders.

More research is needed to assess if common methodologies introduce bias into studies particularly if researchers wish to compare data among studies. This study also adds support for conclusions that passive sampling does not in every instance bias sampling towards more exploratory or active individuals (Biro and Dingemanse 2009; Michelangeli et al. 2016). Our research demonstrates that common practices in amphibian ecology do not introduce bias in measurements of length, but that time can influence measurements of behaviors and mass. Careful consideration of methodological practices continues to be the most important step in preventing the introduction of bias into scientific studies.

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Pelvic Constraint on Egg Size is Unlikely in Snapping Turtles (Chelydridae; *Chelydra*)

The width of the pelvic aperture in turtles is frequently invoked as a constraint on egg size in turtles (Congdon and Gibbons 1987; Table 1). It is often assumed that small-bodied turtles exhibit this pelvic constraint (e.g., Litzgus et al. 2008; Congdon and Gibbons 1987), and by implication (though usually not stated), that large turtles that produce large clutches of relatively small eggs (e.g., marine turtles, chelydrids, or trionychids) do not exhibit pelvic constraints. However, to our knowledge this possible constraint for such large species has not been explicitly tested. For practical reasons (body size relative to portable x-ray plate size, difficulty in counting and measuring large clutches from x-rays [see Congdon et al. 1987], etc.), large, gravid turtles are rarely x-rayed. For example, we know of only one study that reported x-ray egg widths for snapping turtles (*Chelydra serpentina*; Congdon et al. 1987) and to our knowledge no one has reported corresponding pelvic aperture widths for this species (or any other large turtle species).

While investigating reproductive output in snapping turtles at Gimlet Lake in Garden Co., western Nebraska, USA (Hedrick et al. 2017), we asked whether egg size is ever constrained by the pelvic aperture in that species. To answer this question we examined x-rays of gravid females from our field site, museum skeletal preparations, and dissections of road-killed and museum specimens of snapping turtles.

The ilia of nearly all turtles are parallel to one another in the transverse plane (e.g., Fig. 1; see also fig. 1 in Kern et al. 2015). However, the ilia of snapping turtles are not parallel in alignment, but as previously reported by Zug (1971), they instead diverge dorsally at angles of approximately 70 degrees (Figs. 2-3). Although not previously reported in the turtle reproductive literature, Zug (1971) found divergent ilia only in chelydrids among 38 species of 25 genera of six families of cryptodiran turtles. Morphological data available for pleurodires also suggest parallel ilia (fused to the carapace) in that clade (Wise and Stayton 2017). Whether the pelvic aperture constrains egg size in a pleurodire remains unexamined (Escalona et al. 2017).

The evidence for pelvic canal constraint on egg size in turtles is mixed (Table 1). Early work suggested that it was related to body size, with constraint in small-bodied taxa (e.g., Congdon and Gibbons 1987). However, nine of 15 species <150 mm carapace length (CL) listed in Table 1 apparently do not exhibit pelvic constraint on egg size, whereas all six species >200 mm CL lack evidence of pelvic constraint. In any case, there is certainly no such skeletal constraint on egg size in the snapping turtle (Figs. 3-4). No one has evaluated soft tissue pelvic canal constraints in any turtle (Rowe 1994), but such a constraint on egg size in *Chelydra* seems extremely unlikely given the divergent ilia and relatively small egg size. In future studies it should not be assumed that pelvic constraint on egg size is size-related in turtles (Table 1), nor that correlations between egg width and pelvic aperture width necessarily imply morphological constraint on egg size (Rollinson and Brooks 2008).

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FIG. 1. Typical pelvic structure of a turtle (*Kinosternon integrum*; Iverson, personal collection 85–105; Morelos, Mexico) illustrating how parallel ilia define the pelvic canal (maximum width = 28.7 mm) and can potentially constrict the passage of an egg (width = 17.4 mm).

FIG. 2. Pelvic structure of a snapping turtle (*Chelydra serpentina*; Earlham College Herpetology Collection uncatalogued; ca. 32 cm carapace length), illustrating the dorsally divergent ilia (dorsal divergence 77.5 mm) that likely contribute to the lack of pelvic constraint on egg size in the species.

Table 1. Evidence for or against pelvic constraint on egg size in various turtle species (listed in order of decreasing body size). SCL = mean straight-line carapace length of adult females. Asterisks indicate constraint only for small females. Existence of constraint in this table is based on the conclusions reported in the cited paper(s). [1]Curved plastron length.

Species	CL (mm)	Constraint	Source
Apalone spinifera	376	No	Iverson, unpubl.
Chelydra serpentina	321	No	Present study
Gopherus polyphemus	278	No	Rothermel and Castellón 2014
Pseudemys floridana	235	No	Wilkinson and Gibbons 2005
Trachemys scripta	ca. 210–220	No	Congdon and Gibbons 1987
Emydoidea blandingii	208	No?	Ruane et al. 2008
Graptemys sabinensis	ca. 196	Yes*	Fehrenbach et al. 2016
Chrysemys picta bellii	182	No	Iverson and Smith 1993
Mauremys leprosa	180	No	Lovich et al. 2010; Naimi et al. 2012
Kinosternon integrum	ca. 167	No	Macip-Ríos et al. 2013
Chrysemys picta bellii	156–174	No	Rowe 1994
Deirochelys reticularia	160	Yes	Congdon et al. 1983; Congdon and Gibbons 1987
Gopherus berlandieri	158	Yes	Long and Rose 1989
Malaclemys terrapin	153–160[1]	Yes*	Kern et al. 2015
Cuora flavomarginata	154	No	Chen and Lue 1999
Kinosternon integrum	ca. 144	Yes*	Macip-Ríos et al. 2012
Kinosternon integrum	ca. 141	Yes	Macip-Ríos et al. 2013
Kinosternon integrum	ca. 136	No	Macip-Ríos et al. 2013
Kinosternon sonoriense	130	No	van Loben Sels et al. 1997; Lovich et al. 2012
Kinosternon chimalhuaca	ca. 128	No	Macip-Ríos et al. 2013
Chrysemys picta marginata	125–135	Yes	Congdon and Gibbons 1987
Homopus femoralis	127–133	No	Loehr 2013
Terrapene ornata	119	Yes	Nieuwolt-Dacanay 1997
Sternotherus carinatus	117	No	Iverson 2002
Clemmys guttata	110	No	Rasmussen and Litzgus 2010
Kinosternon flavescens	102	No	Iverson 1991
Homopus signatus	93	Yes	Hofmeyr et al. 2005
Sternotherus odoratus	89–105	No	Clark et al. 2001
Kinosternon subrubrum	87	Yes	Wilkinson and Gibbons 2005
Sternotherus odoratus	78	No	Wilkinson and Gibbons 2005

Fig. 3. Ventral view of a radiograph of a young adult female snapping turtle (*Chelydra serpentina*; 11 winters old, believed to be primiparous; 285 mm maximum carapace length; clutch size 52 eggs) captured on her way to nest north of Gimlet Lake, Garden Co., Nebraska, on 5 June 2004. Note the diverging ilia (arrows), especially relative to egg width.

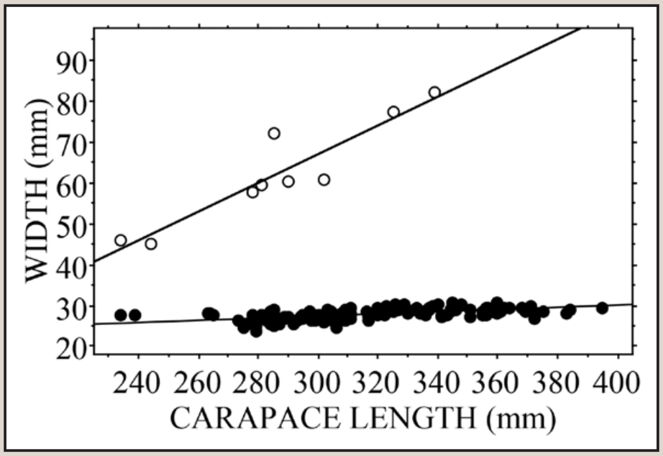

Fig. 4. Relationships of actual mean clutch maximum egg width (solid dots; N = 144 clutches) and maximum width of pelvic aperture (open circles; N = 9) with maximum carapace length in female snapping turtles in western Nebraska. Least squares linear regression for egg width is highly significant ($EW = 0.026CL + 19.683$; $r = 0.60$; $p < 0.0001$), as is that for aperture width ($PW = 0.352CL – 38.429$; $r = 0.93$; $p = 0.0003$).

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TECHNIQUES

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How to Measure a Turtle

Accurately and consistently measuring most vertebrates is a serious challenge (Ansell 1965; Jewell and Fullagar 1966; Rising and Somers 1989; Rivas et al. 2008; Önsoy et al. 2011; Ferner and Plummer 2016; Astley et al. 2017). However, the unique rigid shell of most turtles (Order Testudines) ought to make measuring body size simple, straightforward, and repeatable, even considering the general problem of measurement error (Yezerinac et al. 1992). Unfortunately, this has not been the case, as many different methods of measuring carapace (CL) and plastron length (PL) have been reported (Bolten 1999; see below), and many authors have not adequately described their measurement methods, complicating the situation.

Bolten (1999) attempted to standardize methods for measuring carapace and plastron length in marine turtles (see also Pritchard et al. 1983; Wyneken 2001); however, authors studying other hard-shelled turtles have not, as a group, developed nor standardized methods for measuring their subjects. As a result different authors have adopted different methods, and these methods have sometimes apparently changed over the authors' publishing careers.

The purpose of this note is to detail the most common methods of turtle shell measurement, point out the significant differences among them and their potential effects on meta-analyses, and call for more uniformity in measurements and more precision in describing those methods. We also urge authors to provide equations or ratios relating the measurements made using their method to measurements made by other authors.

METHODS

We began this study with an extensive but not exhaustive review of the freshwater and terrestrial turtle literature, with a focus on papers by the most prolific authors (Table 1). We also solicited method descriptions from a number of active colleagues. We tallied the methods used to measure only carapace and/or plastron length, ignoring other shell measurements for this review. It should be noted that all measurements by all cited authors were made parallel to the midline of the shell (i.e., the mid-sagittal plane). To illustrate differences among methods we measured specimens in the Joseph Moore Museum collections as well as Yellow Mud Turtles (*Kinosternon flavescens*) at our field site at Crescent Lake National Wildlife Refuge in Garden County, Nebraska.

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RESULTS

The following is a list of the eight best-described methods for measuring hard-shelled turtles (i.e., excluding softshell turtles). Table 2 provides an example of the measurements using these eight methods for three North American turtle species. Differences among measurements ranged up to 7% for straight-line measures. Table 3 illustrates the impact of using the various measures as input into meta-analyses such as sexual size dimorphism.

A. *Maximum carapace length*: measured parallel to the mid-plastral plane (the Cagle method, plate 1 in Cagle 1946; Fig. 1). Cagle (1946) used a modified shoe-sizing device to accomplish this measurement, although simply positioning the turtle plastron-down on a flat surface and using calipers with the anterior and posterior points both touching the flat surface (the plastral plane) records the same measurement. Apparently only Iverson (1977; method used consistently for CL since 1977), Vogt (1980 and pers. comm.), and Germano (1993; but see below) have used this method. This method purports to measure the biological length of the turtle as it exists in the environment, but is difficult to record reliably for some species (e.g., *Terrapene ornata*, for which both plastral lobes are movable and hence the plastral plane varies with the extent of plastral closure). A slight variation of this method was employed by Van Denburgh (1914) and followed by Gaymer (1968) and Cloudsley-Thompson (1970), by measuring maximum CL parallel to the plastral plane, but from the nuchal midline to the posterior-most margin of the carapace. We found no author who used this latter method since 1970. In addition, Vogt and Iverson appear to be the only contemporary turtle biologists who have published studies consistently using this method, and hence its future use is not recommended.

B. *Maximum carapace length*: measured as the straight-line distance along the frontal plane from the anterior-most to posterior-most margins of the carapace (Fig. 1; also fig. 1 in Ernst and Lovich 1986; fig. 7.1a in Legler 1990; fig. 1e in Bolten 1999; fig. 47 in Wyneken 2001; fig. 5.1 in Legler and Vogt 2013). Published users of this method (and the others) are compiled in Table 1. A number of authors have reported "maximum (straight-

Table 1. Author variation in measurement methods (see text) of carapace (CL) and plastron (PL) length in turtles. An asterisk indicates a method taken directly from the sources, but not precisely defined.

Measurement method from references	Method from this text	References
Maximum CL	A	Iverson (1977; method used consistently for CL since 1977), Vogt (1980 and pers. comm.), and Germano (1993).
Maximum CL	B	Carr (1952:49), Moll and Legler (1971), Bury and Smith (1986), Jackson (1988), Legler (1990), Ernst et al. (1998, pers. comm.), Lubcke and Wilson (2007), and Ashton et al. (2012).
Maximum straight-line CL*	B?	Graham (1979), Schleich (1981), Lovich et al. (1985, 1989), Fritz (1989), Yasukawa et al. (1992), Rowe (1994b), Jackson and Walker (1997), Yasukawa and Ota (1999), Riedle et al. (2008), and Germano and Bury (2009).
Maximum CL notch to tip	C	Grubb (1971), Hirth and Latif (1981), and Bjorndal and Bolten (1989).
Midline CL	D	Mitchell (1985a), Galbraith and Brooks (1987), Bjorndal and Bolten (1989), Yasukawa et al. (1992), Lambert (1995a, b), Dodd (1997), Litzgus and Brooks (1998), Yasukawa and Ota (1999), Tucker et al. (2001), Tuberville et al. (2005), Loehr et al. (2006), Greaves and Litzgus (2009), Ferner and Plummer (2016), Lovich et al. (2011, 2015, 2017), Ashton et al. (2012), Nafus et al. (2013, 2015), Germano (2016, pers. comm.), J. Congdon (pers. comm.), J.W. Gibbons (pers. comm.), F. Janzen (pers. comm.), and J. Litzgus (pers. comm.).
Maximum curved CL	E	Van Denburgh (1914), Gaymer (1968), Bourn and Coe (1978), and Zug et al. (1986).
Minimum midline curved CL	F	Bolten (1999); Bjorndal and Bolton (1989); and Shoop and Ruckdeschel (1986.)
Maximum plastron length	G	Carr (1952:49), Moll and Legler (1971), Iverson (1977, and all his subsequent papers measuring PL), Lovich et al. (1989), Legler (1990), Lubke and Wilson (2007), Rollinson and Brooks (2008b), and Ashton et al. (2012).
Maximum plastron length*	G?	Moll (1973), Auffenberg (1976), Graham (1979), Schleich (1981), Lovich et al. (1985), Fritz (1989), Yasukawa et al. (1992), Germano (1993), Jackson and Walker (1997), Yasukawa and Ota (1999), Riedle et al. (2008), and Rollinson and Brooks (2008a).
Midline (minimum) PL	H	Van Denburgh (1914), Gaymer (1968), Pritchard (1969), Grubb (1971), Lambert (1982, 1995a, b), Yasukawa et al. (1992), Germano (1993, 2016), Van Loben Sels et al. (1997), Yasukawa and Ota (1999), Litzgus and Brooks (1998), Tucker (1999, 2001), Tucker et al. (2001), Tuberville et al. (2005), Loehr et al. (2006), Ashton et al. (2012), Lindeman (2013:111), Ligon et al. (2014), Hofmyer and Branch (2018), J. Congdon (pers. comm.), J.W. Gibbons (pers. comm.), F. Janzen (pers. comm.), and J. Litzgus (pers. comm.).
Straight-line carapace length*	?	Legler (1960), Ernst et al. (1973), Ernst (1977, 1986), Lovich and Gibbons (1990), Congdon et al. (1993, 1994), Rowe (1994a), Nieuwolt-Dacanay (1997), Roosenberg and Dunham (1997), Rowe (1997), Van Loben Sels et al. (1997), Zug et al. (2002), Congdon et al. (2003), Zug et al. (2006), Spencer et al. (2006), Dodd and Dreslik (2008), Lovich et al. (2012, 2016), Richards-Dimitrie et al. (2013), Loehr (2016), and Hofmyer and Branch (2018).
Curved carapace length*	?	Broderick et al. (2003) and Spencer et al. (2006).
Curved plastron length*	?	Kern et al. (2016).
Straight-line plastron length*	?	Gibbons (1967), Ernst (1977, 1986), Lovich and Gibbons (1990), Congdon et al. (1993, 1994), Roosenberg and Dunham (1997), Rowe (1994a, 1997), Tucker and Moll (1997), Tucker (1999), Tucker et al. (1998a, 1998b, 1999), Van Loben Sels et al. (1997), Spencer et al. (2006), Richards-Dimitrie et al. (2013), and Ferner and Plummer (2016).
Carapace length/plastron length*	?	Tinkle (1961); Gibbons (1968a, 1968b), Ernst (1971, 1975), Ernst et al. (1973), Congdon and Tinkle (1982), Gibbons et al. (1982), Congdon and Gibbons (1983, 1987), Congdon et al. (1983, 1987, 2013), Gibbons (1983), Frazer and Ehrhart (1985), Mitchell (1985b, 1985c, 1988), Schwarzkopf and Brooks (1986), Germano (1988, 2010), Galbraith et al. (1989), Dunham and Gibbons (1990), Gibbons and Lovich (1990), Lamb and Lovich (1990), Lovich et al. (1990, 1998), Mitchell and Pague (1990, 1991), Moll and Moll (1990), Parker (1990), Vogt (1990), Brooks et al. (1992), Janzen (1993), Dreslik (1997), Tucker (1997), Harms et al. (2005), Wilkinson and Gibbons (2005), Litzgus et al. (2008), Buhlmann et al. (2009), Bury et al. (2010), Bowden et al. (2011), Joos et al. (2017), and Castellón et al. (2018).

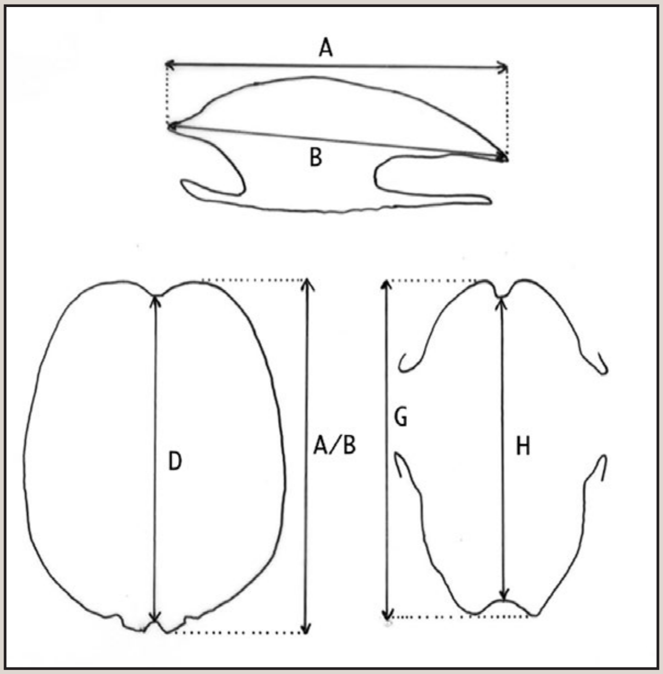

Fig. 1. Demonstration of the primary carapace and plastron length measurement methods outlined in the text. Subject is a hypothetical turtle exhibiting extensive variation in carapacial and plastral emargination.

line) carapace length" (Table 1) which is probably the same as this method (B). This method is straightforward but can be compromised by excessive shell flaring or damage to the rear margin of the shell.

C. *Maximum carapace length notch to tip*: measured as per method B except measured from the anterior midline of the nuchal scute to the posterior-most margin of the carapace (fig. 1a in Grubb 1971; fig. 1b in Bolten 1999; fig. 47 in Wyneken 2001). We found only three papers using this method (Table 1). Because it has been so rarely used in the published literature, we do not recommend its use for freshwater and terrestrial turtles.

D. *Midline carapace length (equivalent to minimum carapace length)*: measured as for method B, except measured along the midline (Fig. 1; also fig. 1a in Bolten 1999; fig. 2 in Seidel and Palmer 1991; fig. 47 in Wyneken 2001). This is the preferred straight measurement method advocated for sea turtles by Bolten (1999) and Bjorndal and Bolten (1989), but is subject to extensive individual variation in the depth of pygal notch in some species, and in the nuchal in others (e.g., fig. 2a in Moldowan et al. 2016). Ontogenetic changes in the serration of the rear of the shell also complicate this popular method.

E. *Maximum curved carapace length*: measured as the curved midline distance over the dome of the carapace using a flexible measuring tape extended along the midline from the anterior margin of the nuchal scute to the posterior-most margin of the carapace (Table 1; fig. 53 in Wyneken 2001). For large taxa in remote locations, this method is simpler than transporting large calipers. This method has the advantage of including a shell height component in its value, and hence crudely estimates shell volume. However, this method has not been used in over three decades, and hence we do not recommend its use.

F. *Minimum midline curved carapace length*: measured as the curved distance over the dome of the carapace using a flexible ruler extended along the midline (from the anterior margin of the nuchal scute to the medial caudal notch; fig. 51 in Wyneken 2001). This is the preferred curved length method for sea turtles (Bolten 1999; Bjorndal and Bolton 1989; Shoop and Ruckdeschel 1986). It also foregoes the need to transport bulky calipers to the field site to measure large turtles. Miller (1978) devised a method using modified calipers to take this measurement in smaller turtles. This method also has the advantage of including a shell height component in its value. However, lack of application to non-marine turtles argues against its use.

G. *Maximum plastron length*: measured from the anterior-most margin to the posterior-most margin of the plastron parallel to the plastral midline (Fig. 1; fig. 25 in Auffenberg 1976; fig. 1 in Ernst and Lovich 1986; fig. 7.1 in Legler 1990; fig. 60 in Wyneken 2001; fig. 5.1 in Legler and Vogt 2013). Most users of method G have clearly defined their measurements; however, many have simply reported "maximum plastron length" (Table 1). Although the latter are probably equivalent to Method G, the method was not precisely described as such by the original authors. This method is generally highly repeatable, but the lobes of turtles with plastral hinges must be oriented in the same plane for measurement. This measurement is also typically sexually dimorphic (see Gibbons and Lovich 1990), though not as much as mid-line measures, due to the deeper anal notch and/or the extended gulars in males of many taxa.

H. *Midline plastron length* (equivalent to minimum plastron length): measured from the anterior midline of the gular or intergular scutes to the midline of the anal scutes (i.e., into the anal notch) (Fig. 1; fig. 3 in Seidel and Palmer 1991). This method is also highly repeatable, is the most common measure used to quantify body size in freshwater turtles (Table 1), and is the preferred method for measuring the plastron in marine turtles (Bolten 1999). However, sexual dimorphism and ontogenetic changes in the depth of the anal notch can compromise size comparisons (Table 2, 3).

Unfortunately, many authors have not precisely described their measurement methods, leading to serious ambiguity in interpretation and limiting the ability for comparison with other studies. Examples include:

1) "Straight-line carapace length." Many authors have used this method without precise description (Table 1), and they could represent midline or maximum or notch to tip measures (e.g., methods A–D, above). In some cases where methods were imprecise, it may be possible to reconstruct the method used by a particular author by consulting their other papers.

2) "Curved carapace length." Broderick et al. (2003) and Spencer et al. (2006) reported this measure without description. Whether the measurement was maximum, midline, or notch to tip was unclear.

3) "Straight-line plastron length." This measure is frequently reported, but without description (Table 1), and hence, it is unclear whether the measurements were midline or maximum lengths (Method G or H).

4) "Curved plastron length." Kern et al. (2016) reported this measure without explanation.

5) "Carapace length" or "plastron length" (Table 1). This lack of precision in describing the precise method of measurement makes it very difficult to make comparisons with other studies, and precludes applying any methods to convert measurements to allow direct comparisons.

Several authors kindly provided further clarifications on their standard methods. J. Congdon, F. Janzen, J. Litzgus, and P. Meylan (all via pers. comm.) always recorded midline CL and

Table 2. Variation in methods (from the text) for measuring shell length in rigid-shelled turtles. Measurements are from an adult female *Trachemys scripta* (JMM T-1588), an adult male *Kinosternon flavescens* from our Nebraska field site, and an adult female *Gopherus polyphemus* (JMM HR-1672). Percentages in parentheses are the deviation from the values for method A for (carapace length) and G (for plastron length).

		Measurements (mm)		
Shell length component	Method	*T. scripta*	*K. flavescens*	*G. polyphemus*
Carapace	A	228	129	276
	B	230 (+1%)	132 (+2%)	288 (+4%)
	C	244 (+7%)	133 (+3%)	288 (+4%)
	D	224 (–2%)	132 (+2%)	288 (+4%)
	E	248 (+9%)	155 (+20%)	345 (+25%)
	F	243 (+7%)	153 (+19%)	345 (+25%)
Plastron	G	214	117	282
	H	209 (–2%)	109 (–7%)	262 (–7%)

Table 3. Variation in methods (from the text) for measuring shell length in adult Yellow Mud Turtles (*Kinosternon flavescens*) from our Nebraska field site. Sexual size dimorphism index (SSD) follows Lovich and Gibbons (1990; [larger sex/smaller sex] -1). Note the substantial difference in the indices based on carapace versus plastron length, a consequence of the relatively shorter plastron in males.

Method	Mean male (N)	Mean female (N)	SSD index
Carapace Length			
A	116.9 (24)	99.7 (20)	0.17
B	119.7 (24)	101.4 (20)	0.18
D	119.0 (24)	100.9 (20)	0.18
Plastron Length			
G	107.3 (24)	100.3 (20)	0.07
H	101.6 (24)	97.4 (20)	0.04

PL measurements (methods D and H). D. Germano (since about 2000) measured “straight-line carapace length over the midline” (Method H, pers. comm.). J. W. Gibbons recorded “maximum midline… with plastic ruler for both plastron and carapace” (Methods D and H; pers. comm.). C. Ernst, F. Janzen, and J. Mitchell measured maximum CL and PL (Methods B and G; pers. comm.). R. Brooks (pers. comm.) and R. B. Bury (pers. comm.) recorded both maximum and midline CL and PL (methods B, D, G, and H), even if they did not report all measuring in a given publication.

Discussion

Although most turtles have a rigid, easily measured shell, research demanding published body size data are compromised by the inconsistency among investigators in how their measures are taken and how precisely the methodology is reported. This diversity of reported methods and inconsistency in the reporting of methods creates great difficulty in making interpopulational (geographic) and/or interspecific comparisons for turtles. Depending on the species (i.e., whether it possesses distinct nuchal, caudal, intergular, or anal notches, or plastral kinesis), the differences among various shell length measurements can be substantial (see examples in Table 2). Furthermore, ontogenetic changes, sexual dimorphism, shell damage, and plastral kinesis can also complicate the reliability and comparability of measurements. Unfortunately, authors mining the published data for their own meta-analyses (e.g., Gosnell et al. 2009; Litzgus and Smith 2010; Jaffe et al. 2011; Ceballos et al. 2013; Itescu et al. 2014; Agha et al. 2018; among others) have uncritically accepted these diverse measures, potentially making their conclusions suspect.

Furthermore, even the most prolific authors rarely report measures for both CL and PL (no matter the method), making direct comparisons among studies that presented only one or the other difficult. Few studies actually report ratios or equations relating PL and CL, particularly by sex. However, Graham and Doyle (1979), Schleich (1981), Lambert (1982), Iverson (1988), Ernst et al. (1998), Mitchell and Pague (1990), Aresco (2004), Tucker et al. (2006), Hays and McBee (2010), Meylan et al. (2001), Litzgus and Smith (2010), Gradela et al. (2017), Jones (2017), Lewis et al. (*in press*), and Monzón-Argüello et al. (2018) are refreshing exceptions to that pattern.

Finally, because most turtles exhibit sexual dimorphism in plastron length relative to carapace length (males generally have a relatively shorter plastron and often a much deeper anal notch, or in many tortoises, a much longer gular projection), conclusions from meta-analyses of sexual size dimorphism using PL may differ significantly from those using CL (or body mass) (e.g., Gibbons and Lovich 1990; Litzgus and Smith 2010; Table 3).

We conclude with a number of recommendations. First, given that there is no clear preference among authors for maximum versus midline measurements, we urge authors to use the historically most popular methods B and D for carapace length and G and H for plastral measurements. Second, authors must precisely define their measurement methods, including retrospectively, in their future publications. Third, we urge authors to collect measurement data for both CL and PL and to provide summary statistics for both as well as equations relating those measurements. It would also be helpful if authors collected and published data relating maximum versus midline lengths and provided equations relating these measurements, perhaps including them as electronic supplements if publication in the paper itself is not possible or undesirable due to journal restrictions. As Dunham and Gibbons (1990) recommended nearly 30 years ago, “it may prove worthwhile, although time-consuming, to establish the mathematical relationship between the variables used for measurement.” Fourth, sexual dimorphism in measurements should always be considered when making measurements and when reporting them; data for the sexes should never be merged without good reason, either statistically or biologically, or without also reporting the measurements of the males and females separately somewhere in the paper. Finally, because repeatability is fundamental to the scientific method (e.g., see Yezerinac et al. 1991), we appeal to all authors, reviewers, and editors to demand the measurement method used be clearly stated in their papers and in the papers they review.

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An Evaluation of the Efficacy of Box Trap Arrays for Capturing Snakes and a Suggested New Trap Design

Traps combined with drift fence arrays are a common method for surveying herpetofauna (Enge 1997). Although capture rates can be influenced by an animal's body size, seasonal activity, individual behavior, and weather (Enge 1997; Willson et al. 2008), box traps with drift fences have been shown to be a cost-efficient and effective technique for capturing snakes (Rudolph et al. 1999; Burgdorf et al. 2005; Rudolph 2012). Other trap types associated with drift fences, such as mesh funnel traps, pitfall traps and cover objects, are also considered highly successful at capturing or detecting fossorial and small-bodied snakes but are less effective for larger-bodied snakes (Gibbons and Semlitsch 1982; Bury and Corn 1987; Greenberg et al. 1994; Crosswhite et al. 1999; Kjoss and Litvaitis 2001).

The standard box trap design for snakes typically consists of a box (of variable size) with mesh hardware cloth sides, with one to four funnels leading into the box (Imler 1945; Rudolph et al. 1999; Burgdorf et al. 2005; Rudolph 2012). Drift fences direct snakes to the entrance of the funnel and into the trap (Imler 1945; Dargan and Stickel 1949; Rudolph et al. 1999; Burgdorf et al. 2005; Rudolph 2012). While several publications suggest modifications to the designs used above (Steen et al. 2010), few studies have compared effectiveness of different designs. For example, Dargan and Stickel (1949) noted that small snakes can escape through the larger (≥ 0.64 cm or 1/4 in) mesh hardware cloth used for box traps and that they may also turn around inside the funnels before entering the trap. Moreover, when a snake encounters a drift fence, it may either turn toward or away from the trap at the center. Burgdorf et al. (2005) mentioned placing additional traps at the distal ends of drift fences; however, we found no published accounts of the relative effectiveness of this approach.

We employed box traps with drift fence arrays, following the design of Rudolph et al. (1999) and Burgdorf et al. (2005), at our study site for >14 years and anecdotally determined that some snakes were escaping from the box traps. We also captured very few small snakes and suspected our box traps, as designed, were biased against the capture of small-bodied snakes. Early in the study, in an attempt to increase captures of small snakes and to capture snakes at the distal ends of the arrays, we used single-ended funnel traps (four per trap) made from aluminum window screen (Campbell and Christman 1982). Very few snakes were captured by this method (0.007 snakes per trap-night per array, N = 29, unpubl. data), so we later replaced these with 19 L pitfall traps. Pitfall traps also yielded low capture rates (0.001 snakes per trap-night per array, N = 15, unpubl. data), and this capture method was also discontinued. To identify and remedy possible trap design flaws, we placed trail cameras at box traps to record snake activity including escape or avoidance of traps. To potentially increase captures of snakes, we designed and tested the efficacy of modified box traps placed at the distal ends of arrays. We used 0.12 cm (1/8 in) mesh siding for these traps to potentially increase captures of small snakes.

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MATERIALS AND METHODS

We conducted our study on Ichauway, the 11,800-ha research site of the Joseph W. Jones Ecological Research Center located in Baker County, Georgia. Our study site was dominated by a *Pinus palustris* savanna with an *Aristida stricta* understory. Overstory hardwood trees (primarily *Quercus* spp.) occurred at low densities throughout the site. Prescribed fire was applied on a two-year rotation and was the primary forest management tool used across the site. In 2003, we installed one 4-funnel box trap array (Burgdorf et al. 2005) in each of eight approximately 40-ha study plots. In 2005, we installed an additional eight trap arrays for a total of 16 arrays (two per study plot). We sampled trap arrays from March through November, the period when snakes were most active on our site (unpubl. data).

Our box traps consisted of a 1 m × 1 m 4-funnel box constructed with pressure-treated plywood and 0.64 cm (1/4 in) mesh hardware cloth. We attached four 30.5 m drift fences comprised of 0.64 cm hardware cloth to the four funnels (one per side) so that each array was comprised of a box trap in the center and drift fences arranged in a cross configuration. We placed 0.64 cm mesh hardware cloth vertically in the center of each funnel (ending within ca. 2.5 cm of the funnel opening) to act as a "lead" to deter snakes from turning around at the end of the drift fence (Burgdorf et al. 2005).

We selected 8 of the 16 snake traps with the greatest snake capture rates (unpubl. data) to use for this study. We deployed a total of 16 wildlife trail cameras (UWAY VH400HD, UWAY Outdoors Canada, Lethbridge, Alberta, Canada) with two cameras at each box trap. We attached each camera to a 2 m wooden stake and placed them 2.5 m diagonally from the trap corner (Fig. 1). We set the cameras to time-lapse mode, taking one standard-definition photo every 15 sec. We activated cameras for a ca. 48 h period per week or until the camera batteries died. We ran cameras at four of the eight snake traps for 31 weeks from 23 March 2016–11 November 2016. We ran cameras at the remaining four traps for only three weeks (23 March 2016 to 8 April 2016). Data were standardized as captures per trap-night to account for differences in sampling periods.

We examined all photos and when snakes were observed, we recorded behavior, specific escape method (where appropriate),

Fig. 1. Two UWAY VH400HD cameras (UWAY Outdoors Canada, Lethbridge, Alberta, Canada) were attached to a 2 m tall wooden stake and placed diagonally 2.5 m from opposite corners of a 4-funnel box trap to document interactions of snakes with the trap.

and identified snake species (where possible). We also recorded observations where snakes did not enter the trap but could be seen moving in the immediate area. We compared the number and species of snakes observed on camera to snakes captured during the following trap check to determine capture versus escape rate.

After observing specific ways in which snakes escaped, we designed new "Knapp-Murphy traps" (Fig. 2) placed at the distal ends of the drift fences. We used 1.8 cm (23/32 in) thick plywood cut into approximately 46 × 61 cm (18 × 24 in) sheets for the top and bottom of the trap. We attached plywood to two ca. 25 cm (10 in) tall frames made from 2.5 × 5 cm (1 × 2 in) boards (total trap height = ca. 30.5 cm [12 in]). We used 0.32 cm hardware cloth stapled to the plywood and boards for the walls of the traps. We connected a single hardware cloth funnel, with a circumference of 35 cm at the base and 6 cm at the tip, to one side of the box using galvanized steel wire. We installed a plywood door on the opposite side of the box and placed flush with the ca. 25.4 × 40.64 cm (10 × 16 in) opening (Steen et al. 2010). We positioned the door against an inset frame constructed from 2.5 × 5 cm (1 × 2 in) boards. We placed hardware cloth (0.32 cm mesh) leads, like those in the larger 4-funnel box trap (Burgdorf et al. 2005), vertically inside the funnel extending from the fence; however, these leads were extended approximately 2.5 cm past the end of the funnel into the trap. We used pressure-treated lumber for all wooden trap components.

We tested the efficacy of Knapp-Murphy traps by installing four traps each at two of the large 4-funnel box trap arrays (hereafter, Array 1 and Array 2) for a total of eight traps. We checked all traps at these arrays three times per week from 21 September 2016–11 November 2016 and from 13 March 2017–26 July 2017. We considered that the trapping effort was equal(same number of funnels per array) between one 4-funnel box trap and the four associated Knapp-Murphy traps. The following statistical analyses were done using Program R (version 3.4.1). We used a chi-square test to analyze differences in capture rate between the two trap types using the total number of snakes caught in each trap type in the same timeframe. Additionally, we used a Mann-Whitney U test to analyze differences in snake size (snout-vent length, SVL) between trap types.

Results

A total of 102 snakes were observed on camera during 31 ca. 48 h surveys at the snake box traps (ca. 5762 total trap-hours, ca. 240 trap-nights; Table 1). Only 32 (31%) of these snakes were captured in traps, while the remainder either escaped or avoided the trap completely. Of the 70 snakes that were not captured, 26 (37%) avoided capture by leaving the trap area before reaching the funnel, 20 (29%) entered the funnel but either subsequently backed out or turned around at the end of the lead, and 24 (34%) entered through the funnel into the trap but escaped before the trap was checked. The most common means of escape once a snake entered the box trap was through the hardware cloth mesh of the trap walls and funnels (N = 11, 46% of all escapes after capture). However, snakes also escaped through small gaps and damaged sections of the hardware cloth (N = 7, 29%). We also observed four snakes (17%) escape back through the funnel opening. Two additional snakes (8%) escaped by unknown means before the traps were checked.

Trap escape rates varied among species (Table 1). *Coluber flagellum* had the lowest escape rate (33%, N = 9) while all *Heterodon simus* (N = 3) and *Cemophora coccinea* / *Lampropeltis elapsoides* (N = 5) escaped (Table 1). *Coluber constrictor* (N =

Table 1. Fate of all identifiable snakes captured on camera at snake trap arrays at the Joseph W. Jones Ecological Research Center, Newton, Georgia, USA, from 23 March 2016 to 11 November 2016. A trap captured snake was one that entered the trap and did not escape before the trap was checked. Some snakes observed at night or only from long distances were unidentifiable due to lack of color in infrared photos (at night) or insufficient photo definition. * Snakes were grouped together because, in some cases (i.e. at night), a positive identification could not be made for snakes with similar patterns and morphology.

Species	Left trap area	Turned around in funnel	Escaped through wire mesh	Escaped through funnel	Unknown escape method	Total escapes	Trap captures
Agkistrodon contortrix	0	0	0	0	1	1	0
Cemophora coccinea / *Lampropeltis elapsoides**	1	1	3	0	0	5	0
Coluber constrictor	0	3	5	1	0	9	7
Coluber flagellum	0	2	1	0	0	3	6
Crotalus horridus	0	0	0	0	0	0	1
Crotalus adamanteus	0	0	0	0	0	0	1
Heterodon platirhinos	0	0	0	0	0	0	1
Heterodon simus	0	3	0	0	0	3	0
Lampropeltis getula	0	1	0	1	0	2	2
Nerodia sp.	0	0	1	0	0	1	0
Pantherophis guttatus / *Pantherophis spiloides**	2	2	3	1	0	8	6
Pituophis melanoleucus	0	1	0	0	0	1	2
Thamnophis sirtalis	4	2	3	0	0	9	6
Unidentifiable snakes	19	5	2	1	1	28	0
TOTAL	26	20	18	4	2	70	32

Table 2. Distribution of snout–vent lengths (SVL) of snakes captured in Knapp-Murphy traps and the 4-funnel box traps over 186 trap-nights at the Joseph W. Jones Ecological Research Center, Newton, Georgia, USA, from 21 September 2016 to 11 November 2016 and from 13 March 2017 to 6 July 2017. Two snakes captured in the Knapp-Murphy trap during this time were not measured and are not reported in the counts in this table.

Trap type	< 500 mm	500–1000 mm	> 1000 mm
Knapp-Murphy trap	56	28	10
4-funnel box trap	9	17	12

16), *Thamnophis sirtalis* (N = 15), and *Pantherophis* spp. (N = 14) were observed most often in our study and each had similar escape rates of ca. 60% (Table 1). Snakes avoided capture in a variety of ways (Table 1) but *T. sirtalis* had the highest propensity to avoid the trap entirely after traveling down the drift fence (ca. 44% of *T. sirtalis* escapes/avoidances, N = 4).

We caught significantly more snakes in the eight Knapp-Murphy single-funnel traps (N = 96) compared to the two 4-funnel box traps (N = 38) over the same period ($\chi^2 = 25.104$, $p < 0.001$; 186 trap-nights). The Knapp-Murphy traps caught snakes from a wide range of sizes (SVL range: 204–1645 mm), including more small (< 500 mm SVL) and medium-sized (500–1000 mm SVL) snakes than the 4-funnel box traps (SVL range: 142–1690 mm) ($U = 2440.5$, $p < 0.001$) (Table 2). At Array 1, we captured 20 snakes at the 4-funnel box trap (0.110 snakes per trap-night) and 51 snakes at the four single-funnel Knapp-Murphy traps (0.274 snakes per trap-night). At Array 2, we captured 18 snakes at the 4-funnel box trap (0.097 snakes per trap-night) and 45 snakes at the four single-funnel Knapp-Murphy traps (0.242 snakes per trap-night). Snake captures per trap-night in the Knapp-Murphy traps (0.258 snakes per trap-night) were much higher than both previously used aluminum window screen funnel traps (0.007 snakes per trap-night) and pitfall traps (0.001 snakes per trap-night). We successfully captured three species (*H. simus*, [N = 3]; *C. coccinea* [N = 6]; *L. elapsoides* [N = 6]) in the Knapp-Murphy traps that escaped capture 100% of the time from the 4-funnel box traps during our camera study. One individual *C. coccinea* was a juvenile weighing only 9 g with a SVL of 320 mm that could have easily moved through the 0.64 cm (1/4 in.) mesh in the larger 4-funnel box trap. We also captured a new species (*Virginia valeriae* [N = 2]) that had not been previously recorded at these box trap arrays (unpubl. data). We captured more *C. constrictor* in the Knapp-Murphy traps ($\chi^2 = 16.82$, $p < 0.001$) and the average SVL of these snakes was smaller ($U = 158$, $p = 0.004$) (avg SVL = 460 mm, SD = 191.8, N = 31) than in the associated 4-funnel box traps (avg SVL = 707 mm, SD = 183.6, N = 6). The number ($\chi^2 = 0.36$, $p = 0.549$) and average SVL ($U = 82$, $p = 0.404$) of *C. flagellum* captured were

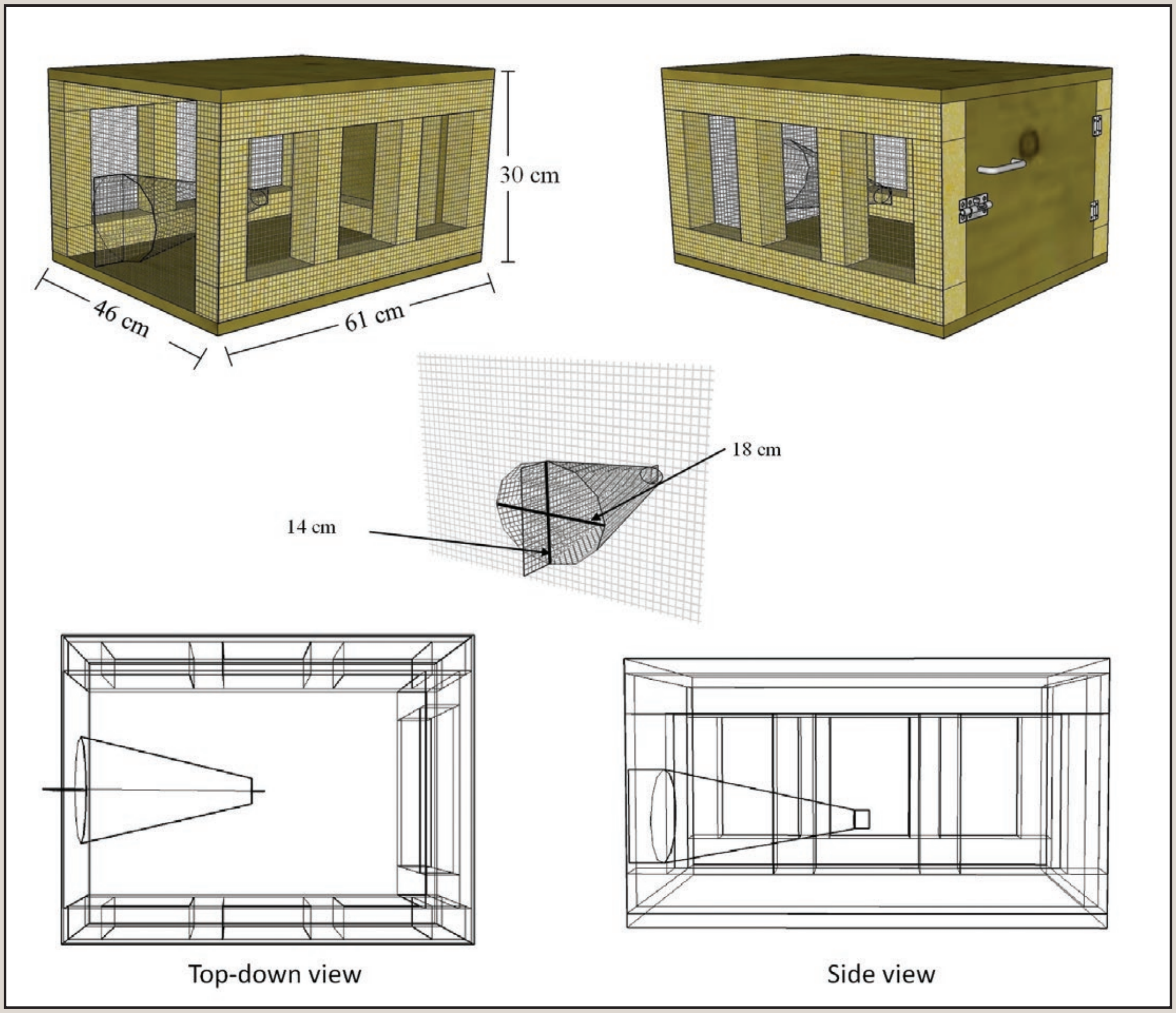

Fig. 2. Snake trap design schematics for the Knapp-Murphy Trap. This trap was designed to increase the capture of smaller snake species. This trap should either be used at the distal ends of a cross-configuration drift fence array in association with a 4-funnel box trap or simply using a single drift fence and two Knapp-Murphy traps, one placed on either end.

similar between the Knapp-Murphy traps (avg SVL = 1067 mm, SD = 454.3, N = 14) and the central box traps (avg SVL = 1140 mm, SD = 446.8, N = 11).

Discussion

We found that 4-funnel box traps with 0.64 cm mesh were biased towards capture of large snakes and that a large proportion of snakes (70 of 102 snakes, 69%) escaped or otherwise avoided capture. Our camera observations allowed us to determine that the majority of snakes that escaped did so through the hardware cloth mesh, and gaps or damaged areas in the funnels; some snakes also escaped by turning around or escaping back through the funnels. Based on these observations we designed a new trap (Knapp-Murphy Trap) that incorporated modifications aimed at increasing capture rates, especially for small snakes (Fig. 2). Our new trap design included extending the hardware cloth leads suggested by Burgdorf et al. (2005) for the funnels. By extending the lead past the end of the funnel it becomes an effective extension of the drift fence and may deter slower moving species such as *H. simus* from turning around within the funnel. Additionally, we used a smaller-sized mesh hardware cloth (0.32 cm) for the walls, funnel, and lead in the Knapp-Murphy trap to address small and juvenile snakes escaping through the mesh. Ford et al. (1991) also used 0.32 cm hardware cloth and reported captures of small species, such as *Storeria* spp. and *V. valeriae*. We also took extreme care during trap construction not to leave gaps in the hardware cloth. We recommend careful monitoring of wear and tear of traps through time to prevent even minor damage to hardware cloth which can allow snakes to escape.

Our new trap design did not address a snakes' ability to escape back through the funnel opening after capture. Although this was the least common method for escape, some trap adjustments can be made to address this. For example, Dargan and Stickel (1949) attempted to mediate this by constructing a one-way hardware cloth door over the funnel. This was effective in reducing escapes, however, they noticed that one species, *H. nasicus*, was hesitant to enter a funnel when the door was in place. Farallo et al. (2010) used double-funnel traps to prevent escape back through the funnel. Halstead et al. (2013) constructed a one-way valve using cable ties in the funnels of aquatic minnow traps which increased capture rates of *T. gigas*.

Any of these modifications could be used for the Knapp-Murphy trap; however, this method of escape was rare in our study of terrestrial snakes and may not need a specific trap modification.

If traps are targeting certain species, trap modifications may not be necessary. For example, Burgdorf et al. (2005) was targeting *Pituophis ruthveni*, a large-bodied snake, and therefore a trap with a finer mesh size would not be needed. However, if capture of juveniles of these large bodied species is desired, modifications may still be necessary. Additionally, snakes that have low escape rates such as *C. flagellum* may require less trap augmentation. *C. flagellum* is a fast-moving, diurnal snake (Tuberville and Gibbons 2008) and its speed may make it more susceptible to entering the funnel and being captured. Although the two trap types caught a similar number and average SVL of *C. flagellum*, the Knapp-Murphy trap still performed slightly better at capturing *C. flagellum* than the original box traps. This is likely due to the extended lead and smaller mesh size which prevented the two observed ways ("turned around in funnel" and "escaped through mesh") that *C. flagellum* was shown to escape. Another study suggests that box trap size may influence captures of large snakes. Hyslop et al. (2009) were targeting the large species *Drymarchon couperi* and they used a reduced trap height (0.3 m tall) versus the 0.45 m tall traps described by Burgdorf et al. (2005). To prevent snakes from avoiding the funnel by crawling over these shorter box traps, they installed horizontal wooden panels above the funnel. Lastly, we encourage monitoring of snake traps via trail camera to evaluate their effectiveness and for tailoring a trap design to target certain species.

The addition of Knapp-Murphy traps at the end of each drift fence not only increased capture rates of small and medium-sized snakes but also allowed for the capture of large snakes, and snakes that chose to move along the drift fence in the opposite direction of the center 4-funnel box trap. Additionally, Knapp-Murphy traps can be used in a single drift fence array configuration with two traps, one placed on either end of a drift fence. Overall, this trap design would work well for studies where the objective is to attempt to document all snake species and size classes.

Acknowledgments.—This work used humane and ethical treatment of animals and complies with Georgia Department of Natural Resources Scientific Collecting Permit #029. We thank Owen T. Jenkins and Jillian Josimovich for their assistance in the field and with data entry. Micheal Simmons assisted with data management.

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Promising Surgical Implantation Method for Radio-tracking Eastern Tiger Salamanders (*Ambystoma tigrinum tigrinum*)

A critical objective of radio-telemetry is detecting transmitter signals to repeatedly relocate individual animals as they move across a landscape. For mammals sufficiently large to wear GPS-collars powered by large batteries, this is not a problem. Small animals (e.g., smaller snakes, lizards, anurans, and salamanders), require small batteries and weaker transmitters that produce shorter detection ranges, requiring frequent trips to the field to prevent losing tagged animals. Many anurans can be fitted with one of several belt designs (e.g., Rathbun and Murphey 1996; Bartelt and Peterson 2000; Muths 2003; Burrows et al. 2012), that attach transmitters externally around the waist, leaving the whip antenna to trail behind, maximizing the detection range (e.g., 400–600 m, depending on topography, the anuran's behavior, such as sitting in a burrow; pers. obs.).

The smooth, linear body form of snakes precludes externally attaching transmitters. Instead, snake ecologists surgically implant the radio and battery of a transmitter into the body cavity, then run the long whip antennae under the skin to maximize the detection range of these transmitter (Reinert and Cundall 1982).

The body shape, delicate skin, and burrowing habits of salamanders also generally preclude any external attachment of transmitters; instead, they also are surgically implanted (e.g., Stebbins and Barwick 1968; Kroll et al. 1973; Reinert and Cundall 1982; Blouin-Demers and Weatherhead 2001; Carfagno and Weatherhead 2008). These transmitters typically are equipped with an antenna tightly wound into a helical coil ("helical antenna") to prevent it from puncturing the body wall or entangling internal organs. A disadvantage of a helical antenna is their apparent and substantially shorter detection ranges (e.g., 25–60 m, Eggert 2002; 4–60 m, Goldberg et al. 2002), often less than an animal's average daily movement.

We study Eastern Tiger Salamanders (*Ambystoma t. tigrinum*) in northern Iowa, where they typically breed in semi-permanent, fishless wetlands (Lannoo 1996). After breeding, these highly cryptic salamanders enter terrestrial habitats and spend much time underground in burrows they dig (Semlitsch 1983) or that are excavated by small mammals (Collins et al. 1993; Kolbe et al. 2002). They can disperse long distances from the breeding pond (up to 10 miles; Lannoo 1996), though many studies report shorter distances traveled throughout the active season, from breeding to hibernation (e.g., 229 m, Gehlbach 1967, *cited in* Lannoo and Phillips 2005; 162 m, Semlitsch 1983; 500 m, Madison and Farrand III 1998). We wanted an alternate antenna configuration on implanted transmitters that would extend the detection range and help us track Tiger Salamanders as they dispersed among multiple, recently restored wetlands. We also wanted an alternate configuration that would not injure the animal nor interfere with its movements or burrowing habits.

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We purchased 23 BD-2 radio transmitters (1.8 g, 150–152 Mhz, pulse rates between 0.50–0.65 s; Holohil Systems, Ltd., Carp, Ontario, Canada), each equipped with a 10-cm flexible, whip antenna with a typical range of 400–600 m and a ca. 4-mo battery life (based on work completed 2009–2011; Bartelt and Klaver 2017). After modifying each antenna, we implanted these transmitters into adult salamanders that we trapped with unbaited minnow traps placed among 10 restored wetlands in Winnebago County, Iowa, in 2015 (13 salamanders) and 2016 (23 salamanders; 10 of these salamanders received new transmitters and 13 refurbished units). We tracked the salamanders for the season and compared the performance of these transmitters to identical transmitters (equipped with helical antennae) and tracked with identical or similar equipment in other studies.

Modifying the Antenna.—To minimize the reduction in detection range, we gently bent the whip antenna into a single loop, reducing its length by about half (the exact amount of reduction was based on the size of each animal). To stabilize the antenna and prevent it from damaging internal organs, we encased the antenna loop in a thin layer of biologically inert silicone caulk (flattened by compressing between two layers of wax paper; Fig. 1). During the silicone curing process (24–48 h), toxic methanol and acetic acid is released (General Electric, 2015; DAP Products Inc. 2015). We allowed the silicone to completely cure ≥ 72 h before surgery to minimize risks of toxicity. We aimed to minimize the additional weight of the silicone so that each transmitter would weigh < 10% of the animal's body weight (Heyer et al. 1994, cited in Johnson 2006; Rowley and Alford 2007; Dodd 2010). During our first season (Spring 2015), the mean (± SD) added weight was 0.76 ± 0.144 g; during Spring 2016, we reduced these added weights to 0.21 ± 0.08 g (Table 1). Hence, for three salamanders in Spring 2015, the total weight exceeded the recommended weight limit (10.7, 11.2, and 11.3%); for the other nine, weights were < 10% (within the range used by Eggert 2002). Learning from this experience, we were able to reduce mean (± SD) total weights in Spring 2016 to 5.44 ± 0.01% of salamander weights. To maximize battery life, we waited until just before surgical implantation to remove the magnet, activating the radio and signal.

Surgical Methods.—To minimize infection during surgery, we used sterile utensils and techniques, and treated salamanders with 2.27% Enroflox (enrofloxacin, an antibiotic) up to 5 d before and 5 d after surgery. Using a ½-cc insulin syringe, we mixed 0.03 mL of 100-mg Enroflox with 0.07 mL of distilled water per 40 g (B. Wayne D.V.M., pers. comm.*)* and dripped the mixture onto the animal's skin. We also used a standard amphibian Ringer's solution (sterilized) to prep animals and maintain their moisture. We anesthetized each salamander by immersing it in 0.03% MS-

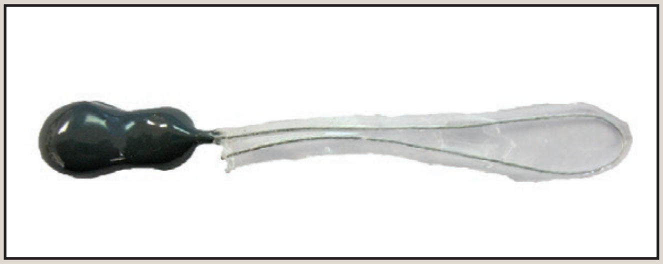

Fig. 1. Holohil BD-2 radio transmitter with a 10-cm whip antenna looped and encased in a thin layer of clear silicone. Transmitter length varies due to varying loop lengths to fit the coelomic cavity of individual animals.

Table 1. Maximum detection ranges for BD-2 radio transmitters with modified whip-antennae.

No.	Range (m)	Year
1	75	2015
2	75	2015
3	75	2015
4	100	2015
5	117	2016
6	137	2016
7	140	2016
8	140	2016
9	152	2016
10	154	2016
11	154	2016
12	155	2015
13	156	2016
14	156	2015
15	183	2015
16	183	2015
17	187	2016
18	189	2015
19	192	2016
20	195	2016
21	204	2015
22	205	2015

222 (tricaine methanesulfonate) solution, mixed in a ratio of 5 g/200 mL Ringer's solution for 10–15 min or until the animals were unresponsive.

We made a 2–3 cm incision slightly offset and parallel to the midline of the ventral surface (avoiding any ventral abdominal veins). The transmitter apparatus was soaked in ethanol for 1 min, retrieved with sterile forceps, air dried, and immediately implanted. We placed the bulk of the unit (radio and battery) into the pelvic area of the coelomic cavity, with the thin, flexible antenna loop laid into the thoracic region. Putting the bulk of the unit posteriorly avoided placing pressure on vital thoracic organs (e.g., heart and major arteries). After implantation, the incision was closed with sterile sutures (6/0 VISORB© absorbable sutures; C.P. Medical, Portland, Oregon, USA). We placed each salamander into a sterilized 1-L beaker with a sterilized paper towel soaked with amphibian Ringer's solution for post-surgical observations; we monitored the incision site and daily treated each salamander with antibiotics. Earthworms were offered to salamanders after 24-48 h. We released animals 5 d post-surgery to minimize the effects of prolonged activity that may delay or shut down normal field behavior (Madison and Farrand III,1998). Previous studies involving radio implantation in amphibians have allowed similar recovery times (24 h, Werner 1991, and Johnson 2006; 1–9 d, Madison and Farrand III 1998; 2 d, Eggert 2002).

Post-implantation.—Surgical implantation for all salamanders was successful, though one died 2 d post-surgery (likely because we retained it for an extended time pre-surgery and it had poor body condition). During the 5-d recovery periods, the remaining individuals were alert, active, and without signs of infection. Incision sites healed quickly with substantial healing at the time of release. After being released into its pond, each salamander vigorously swam into the water. As a control (to help us evaluate the health effects of this new technique), we implanted a transmitter into another salamander that was a "resident" in our laboratory, fed it weekly, and retained for observations.

We relocated each salamander every 2–3 d with a TR-2 or TR-4 receiver and a 2-element RA-2H directional antenna (Telonics, Inc., Mesa, Arizona, USA). Animals were handled and assessed only if they were visible above ground or if there was concern for their well-being (usually 1–2 times per season).

Detection Range and Tracking.—We measured the detection range for each transmitter as we released each salamander (the presumed maximum range). This silicone-loop technique achieved a mean (± SD) detection range of 151 ± 41 m (range = 75–205 m; Table 1). Differences in this range relate largely to the size of the animal: smaller animals requiring a shorter loop to fit the coelomic cavity had the shorter ranges of detection, and larger animals had the longer ranges.

In 2015, we tracked the animals a mean of 125 ± 46 d, with the longest length of time being 177 d from implantation. We made a total 93 observations of 12 salamanders over 89 d (17 April–15 July; one salamander died before release). They traveled a mean (± SD) total distance of 286 ± 168 m (135–659 m), and a mean daily distance of 12.4 ± 8.2 m (max = 147 m). The duration of telemetry was substantially shortened for all but three salamanders. Predators caught five, possibly seven, salamanders; one likely died when sutures came loose; another was killed by heavy agricultural machinery after it moved into a soybean field; and we lost track of three when the batteries expired. The control salamander, a laboratory resident, remained active, demonstrated usual movements and behaviors, and accepted food offerings consistently. The radio was retrieved after 6 mo.

In 2016, we made a total 300 observations of 23 salamanders over 109 d (3 April–21 July). They traveled a mean daily distance of 5.8 ± 4.3 m (max = 308 m) and a mean (± SD) total distance of 251 ± 179 m (50–708 m). We recovered 12 radios in the field with signs of digestion and/or visible predator bite marks; four more signals were lost, either by animals moving out of detectable range or carried away by predators. Two bodies were recovered: one animal died within a couple days of the prairie habitat being burned (it was matted with ash); the other was killed and the radio removed by a predator (both the body and radio were found). Two radio signals never left the pond (release site); we were unable to retrieve the radios and suspect the salamanders were eaten by an aquatic predator. We retrieved three animals alive at the end of the tracking season; all were active and had increased body weight with slight to no scarring at the implant site. We successfully recovered these transmitters through surgery.

Mortality.—Predation was a major challenge to salamander survival during our two seasons of telemetry. Of our 35 animals implanted and released, 16 radios were recovered without the salamander, with similar signs of digestion as a radio recovered from a garter snake (*Thamnophis* sp.; digestive juices stained the silicon a yellow-green color) or, in many cases, frayed antenna or signs of chewing of a larger animal. Assessing the natural rate of predation of adult salamanders is difficult, but they do fit within the diet of a plethora of predators within wetlands (e.g., Common Snapping Turtle, *Chelydra serpentina*) and prairie sites (e.g., Raccoon, *Procyon lotor*; Coyote, *Canis latrans*; Red Fox, *Vulpes vulpes*; Striped Skunk, *Mephitis mephitis*). Are these high predation rates typical? We were unable to develop a method to compare the predation of our implanted salamanders with other free-ranging salamanders in these same habitats, but the mortality rates experiences by our salamanders reflected those observed in other studies for *A. tigrinum* (e.g., Madison and Farrand III 1998). In addition, D. Madison (pers. comm.) has documented very high predation rates in both implanted and non-implanted adults, and is confident that documented high mortality had very little to do with any increased susceptibility of implanted salamanders to predators.

Summary.—This antenna modification did not alter salamander behavior, compared to results from other studies. It also increased the detection range compared to transmitters with helical antennae. Holohil Systems, Ltd., lists typical ranges of the BD-2H ("H" = helical antenna) transmitter as 50–100 m and the BD-2HX ("X" = helical around a short post) as 100–200 m. This assumes, however, the use of a 3-element Yagi antenna, not a 2-element antenna (e.g., RA-2H), for which the range would be less (J. Edwards, pers. comm.).

Various factors (e.g., battery size and power, type of receiving antenna, animal behavior) can affect detection range. To better estimate the benefit of our method, we compared our results with two independent studies that used nearly identical equipment. Bartelt (2000) used four BD-2H transmitters and a Telonics TR-2 receiver and a 2-element RA-2H directional antenna to monitor Western Toads (*Anaxyrus boreas*) in southeastern Idaho; the detection range for these four varied between 30 to a max. of ca. 50 m. Swanson (2017) used 25 BD-2H transmitters in 2015 and 2016 to track Northern Leopard Frogs (*Lithobates pipiens*) in northern Iowa. She detected the signals with a model 2400 receiver (Advanced Telemetry Systems, Isanti, Minnesota, USA) and a Telonics 2-element RA-14 directional antenna (same performance as a RA-2H antenna; Telonics, Inc., pers. comm.); the detection range in this study ranged from 20 to a max. of ca. 70 m. She used ca. 10 BD-2HX units in 2016; maximum ranges for these were 20–30 m greater (J. Swanson, pers. comm.). Transmitters with the shortest range in this study exceeded the best results of Bartelt (2000) by 25 m and those of Swanson (2017) by 5 m; ranges of the other transmitters were at least twice greater than BD-2H transmitters in the two other studies.

As with any other transmitter, the animal's behavior (e.g., underground or under water) caused some shortening of the range (e.g., est. usually 10–20%; 25% in the most extreme situations). Regardless, this longer range allowed us to follow individuals more effectively. For example, on 11 different occasions in 2015, 7 salamanders moved > 90 m (93–205 m) in one day; on 15 occasions in 2016, 12 salamanders moved ≥ 80 m (80–318 m) in one day. Given the more limited range of helical antennas, the above data suggests we may have lost over half of the salamanders (because they moved well beyond the detection range of a helical antenna).

Limitations.—Limitations with this method include the need for a longer incision and more sutures, when compared to helical antenna radios. If not applied carefully, the addition of silicone can substantially increase the percent body weight ratio, but with practice we were able to keep the total weight ratio to ca. 5%. Finally, because longer antenna loops provide greater ranges than shorter loops, one will achieve best results when using medium to larger animals (e.g., ca. 35 g or more). Except for the largest species, we suspect this method to have limited use with anurans, due to their shorter, truncate bodies.

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AMPHIBIAN AND REPTILE DISEASES

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First Confirmed Occurrence of *Ophidiomyces ophiodiicola* in Indiana, USA

Snake fungal disease (SFD) is an emerging fungal pathogen in North America (Allender et al. 2011). The causative agent of SFD has been identified as *Ophidiomyces ophiodiicola* (Allender et al. 2015a; Lorch et al. 2015), which can persist in soil and grows at a wide range of temperatures (Allender et al. 2015b). Snake fungal disease causes skin lesions ranging from minor-scale abnormalities to severe swelling, disfiguration, loss of tissue and bone, and death (Allender et al. 2011). The known mortality rate for some species can be greater than 50% (e.g., Timber Rattlesnakes, Clark et al. 2011; and Eastern Massasaugas, Allender et al. 2016). In the United States, SFD has been documented in 21 states and in more than 15 genera and 30 species of captive and free-ranging snakes (Lorch et al. 2016). No surveillance for the pathogen has been conducted in Indiana, USA despite confirmed occurrence of SFD in bordering states (Illinois, Allender et al. 2011; Kentucky, Price et al. 2015; Michigan, Tetzlaff et al. 2015; Ohio, Lorch et al. 2016). Many of the genera reported to be susceptible to SFD occur in Indiana (e.g., *Crotalus, Sistrurus, Nerodia*), so the lack of surveillance in this state is a gap in the understanding of the distribution of this pathogen. In 2017, we initiated sampling for the presence of SFD in Indiana.

Live snakes were located via visual encounter or road cruising surveys in central and southern Indiana (Fig. 1) from August–October 2017, captured, inspected, marked, and released. Captures were given a wellness exam consisting of a behavioral assessment (alertness, activity level, and response to capture) and visual assessment of the skin for lesions or abnormalities. If present, lesion number and location were recorded. Live snakes were sexed via cloacal probing and marked with a scale clip to minimize repeat sampling of the same individual. Snout–vent length (SVL) was measured to the nearest 0.1 cm, mass was measured to the nearest gram (Ohaus™ digital scale), and capture location was recorded. Venomous species were restrained in a clear plastic tube during sampling. Each snake was swabbed by rubbing a sterile cotton-tipped applicator from snout to tail 2–3 times on both the dorsal and ventral surfaces. Lesions consistent with SFD infection (e.g., displaced scales, raised pustules, necrotic ulcers) were swabbed separately. Skin swabs were collected from all snakes whether or not lesions were present. Swabs were placed in 2-mL Eppendorf tubes labeled with species, collection site, and date, and stored in a -20°C freezer until processing. All equipment was sterilized with 10% bleach solution (Rzadkowska et al. 2016) between individuals, and snakes were released at their point of capture. Disposable nitrile gloves were worn when possible. If speed required for capture did not allow time to put on gloves, hands were immediately cleaned with alcohol-based hand sanitizer. Snakes found dead on roads were collected opportunistically when encountered by Indiana Department of Natural Resources property staff during their regular duties and stored in a -20°C freezer until swabbing could be completed (1–24 months post-collection).

Swabs were tested for *Ophidiomyces ophiodiicola* DNA using quantitative PCR (qPCR). DNA was extracted using a Qiagen DNEasy kit and DNA concentration and purity were determined using a spectrophotometer (Nanodrop, Thermo Scientific, Waltham, MA, USA). A TaqMan PCR assay was performed using primers targeting a 68-bp segment of internal transcribed spacer, between 18S and 5.8S rRNA genes of *Ophidiomyces* (Allender et al. 2015c). The qPCR assays were conducted using a real-time PCR thermocycler and data were analyzed using the associated software (7500 ABI real-time PCR System, Sequence Detection Software v2.05, Applied Biosystems, Carlsbad, California, USA). Differences in infection rates among species were determined using a chi-squared test performed in Microsoft Excel.

We sampled 53 individual snakes of 14 species from 10 Indiana counties (Table 1). Thirty-one snakes were captured live or found freshly killed (within 24 hours) on roads and 22 snake carcasses were collected by property staff and stored in the freezer for later sampling. Timber Rattlesnakes (*Crotalus horridus*; n = 16) were the most commonly sampled species, followed by Northern Watersnake (*Nerodia sipedon*; N = 10), Queensnake

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Table 1. Results of snakes tested for *Ophidiomyces ophiodiicola* from Indiana, USA in 2017. Quantitative PCR (qPCR) results indicated as positive or negative and number of fungal copies detected in the sample was standardized per nanogram of DNA amplified (fungal copy/ng DNA).

Species	Disposition	Date (month/day/year)	County	qPCR result	Fungal copy/ ng DNA
Agkistrodon contortix	Live	8/10/2017	Brown	Negative	0
Agkistrodon contortix	Live	8/10/2017	Brown	Negative	0
Coluber constrictor	Live	8/10/2017	Brown	Positive	734.77
Coluber constrictor	Live	8/10/2017	Brown	Positive	3264.37
Coluber constrictor	Dead in Freezer	10/23/2017	Gibson or Pike	Negative	0
Crotalus horridus	Dead in Freezer	10/23/2017	Brown	Negative	0
Crotalus horridus	Dead in Freezer	10/23/2017	Morgan	Negative	0
Crotalus horridus	Dead in Freezer	10/23/2017	Morgan/Monroe	Negative	0
Crotalus horridus	Dead in Freezer	10/23/2017	Brown	Negative	0
Crotalus horridus	Dead in Freezer	10/23/2017	Unknown	Negative	0
Crotalus horridus	Dead in Freezer	10/23/2017	Morgan/Monroe	Negative	0
Crotalus horridus	Dead in Freezer	10/23/2017	Brown	Negative	0
Crotalus horridus	Dead in Freezer	10/23/2017	Brown	Negative	0
Crotalus horridus	Dead in Freezer	10/23/2017	Monroe	Negative	0
Crotalus horridus	Dead in Freezer	10/23/2017	Brown	Negative	0
Crotalus horridus	Dead in Freezer	10/23/2017	Morgan/Monroe	Negative	0
Crotalus horridus	Dead in Freezer	10/23/2017	Unknown	Negative	0
Crotalus horridus	Dead in Freezer	10/23/2017	Morgan/Monroe	Negative	0
Crotalus horridus	Dead in Freezer	10/23/2017	Brown	Negative	0
Crotalus horridus	Dead in Freezer	10/23/2017	Marion*	Negative	0
Crotalus horridus	Dead in Freezer	10/23/2017	Lawerence	Negative	0
Diadophis punctatus	Live	8/11/2017	Brown	Negative	0
Diadophis punctatus	Live	9/5/2017	Harrison	Negative	0
Lampropeltis calligaster	Dead on Road	8/23/2017	Gibson	Negative	0
Lampropeltis triangulum	Live	9/7/2017	Harrison	Negative	0
Lampropeltis triangulum	Live	9/7/2017	Harrison	Positive	74.89
Lampropeltis triangulum	Dead in Freezer	10/23/2017	Monroe	Positive	1.24
Lampropeltis triangulum	Dead in Freezer	10/23/2017	Jay	Negative	0
Nerodia sipedon	Live	8/10/2017	Brown	Negative	0
Nerodia sipedon	Live	8/11/2017	Brown	Positive	21.73
Nerodia sipedon	Lesion	8/11/2017	Brown	Positive	0.99
Nerodia sipedon	Live	9/6/2017	Harrison	Positive	1.56
Nerodia sipedon	Live	9/6/2017	Harrison	Positive	0.28
Nerodia sipedon	Live	9/6/2017	Harrison	Positive	778.51
Nerodia sipedon	Live	9/6/2017	Harrison	Positive	3.45
Nerodia sipedon	Live	9/6/2017	Harrison	Positive	873.03
Nerodia sipedon	Live	9/6/2017	Harrison	Positive	1.18
Nerodia sipedon	Live	9/6/2017	Harrison	Negative	0
Nerodia sipedon	Live	9/6/2017	Harrison	Positive	1.03
Opheodrys aestivus	Dead on Road	9/6/2017	Harrison	Negative	0
Opheodrys aestivus	Live	9/7/2017	Harrison	Negative	0
Pantherophis spiloides	Dead on Road	8/10/2017	Brown	Negative	0
Pantherophis spiloides	Live	9/8/2017	Spencer	Negative	0
Pantherophis spiloides	Dead in Freezer	10/23/2017	Brown	Negative	0
Pantherophis spiloides	Dead in Freezer	10/23/2017	Scott	Negative	0
Regina septemvittata	Live	9/6/2017	Harrison	Positive	1.23
Regina septemvittata	Live	9/6/2017	Harrison	Negative	0
Regina septemvittata	Live	9/6/2017	Harrison	Negative	0
Regina septemvittata	Live	9/6/2017	Harrison	Negative	0
Regina septemvittata	Live	9/6/2017	Harrison	Negative	0
Storeria dekayi	Live	9/7/2017	Harrison	Negative	0
Thamnophis sauritus	Dead in Freezer	10/23/2017	Pike	Negative	0
Thamnophis sirtalis	Live	8/10/2017	Brown	Negative	0
Virginia valeriae	Live	9/7/2017	Harrison	Negative	0

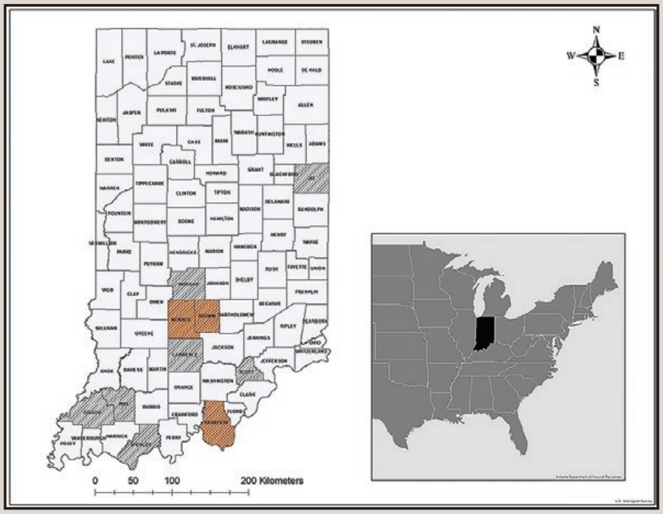

Fig. 1. Counties sampled for *Ophidiomyces ophiodiicola* in Indiana, USA, 2017. Counties with positive samples (N = 3) are shaded orange, counties with only negative samples are shaded grey (N = 7), unshaded counties were not sampled.

(*Regina septemvittata*; N = 5), Eastern Milksnake (*Lampropeltis triangulum*; N = 4), Gray Ratsnake (*Pantherophis spiloides;* N = 4), Racer (*Coluber constrictor* ; N = 3), Copperhead (*Agkistrodon contortrix*; N = 2), Ring-necked Snake (*Diadophis punctatus*; N = 2), Rough Greensnake (*Opheodrys aestivus*; N = 2), Dekay's Brownsnake (*Storeria dekayi*; N = 1), Garter Snake (*Thamnophis* sp.; N = 1), Prairie Kingsnake (*Lampropeltis calligaster*; N = 1), Eastern Ribbonsnake (*Thamnophis saurita*; N = 1), and Smooth Earthsnake (*Virginia valeriae*; N = 1).

We detected SFD in 14 samples from 13 individuals of four species (all colubrids), and SFD was not detected in 41 snakes. The overall prevalence of individuals testing positive for *Ophidiomyces* was 24.5% (95% CI: 13.8–38.3%). Infected individuals included two of three Racers, eight of ten Northern Watersnakes, two of four Eastern Milksnakes, and one of five Queensnakes. Clinical signs were only observed in three individuals: an area of abnormal scales was found on a Racer (chin) which tested SFD-positive by qPCR, one lesion (pustule) was SFD-positive on a Northern Watersnake (vent), and one lesion (scab/crust) was SFD-negative on a Black Ratsnake (dorsum). Three counties of ten sampled accounted for all SFD-positive individuals (Fig. 1). Snakes sampled in Brown County had a SFD prevalence rate of 19% (95% CI: 4–46%) and there was a 45% prevalence in Harrison County (95% CI: 23–69%). Infection rates differed significantly among the 14 species examined (p = 0.0016).

This study is the first to detect the presence of SFD in Indiana. Future sampling efforts are planned to span the entire active season and a larger geographic area to obtain better estimates of statewide prevalence and seasonal occurrence. Additionally, as more individuals are sampled, we will be able to determine if prevalence patterns are associated with geographic region, species, sex, or body condition.

Our results did not detect *O. ophiodiicola* from either of the venomous species sampled, which is encouraging because pitvipers may be especially sensitive to this pathogen (Clark et al. 2011; Allender et al. 2016). However, sample size was small and more individuals need to be tested to confirm this result. No Eastern Massasaugas (*Sistrurus catenatus*) were sampled in 2017, which will be a target species in 2018 given the prevalence of SFD in this species in neighboring Illinois, USA (Allender et al. 2016). Most positive samples (64%) came from Northern Watersnakes, a potentially intriguing result given that *O. ophiodiicola* is thought to be a soil-dwelling pathogen (Allender et al. 2015b). The Northern Watersnakes and Queensnakes from Harrison County came from the same small stream, with some individuals found under the same rock, yet SFD prevalence differed between these species. Further investigations are needed to determine if this is a real difference in species susceptibility or an artifact of small sample size.

These results confirm that *O. ophiodiicola* is present in Indiana. It is prudent for land managers and researchers to take precautionary measures and to consider biosecurity procedures when handling snakes or traveling between sites, particularly where threatened and endangered species occur. This includes either wearing disposable nitrile gloves when handling snakes and changing gloves between individuals, or using alcohol-based hand sanitizer. In addition, disinfection of equipment and boots between snake captures and sites using a 10% bleach solution can preclude accidental human-mediated transmission of the pathogen (Rzadkowska et al. 2016). Although positive detections have come from only three counties in Indiana, there is no reason to believe *O. ophiodiicola* does not occur elsewhere as statewide sampling has not yet been completed. Further, because many sites sampled were state parks, there is a potential risk of human-facilitated transport if *O. ophiodiicola* is present in soils on or near trails or other high use areas.

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The Identification of *Janthinobacterium lividum* on Wisconsin, USA Amphibians

One threat to worldwide amphibian populations is the parasitic chytrid fungus *Batrachochytrium dendrobatidis* (*Bd*: Berger et al. 1998; Daszak et al. 1999; Daszak et al. 2003; Pounds et al. 2006; Schloegel et al. 2006). This heterotrophic fungus causes the epidermal infection leading to chytridiomycosis, including dysfunction of osmoregulation, eventually leading to cardiac arrest and host death (Rollins-Smith et al. 2011; Voyles et al. 2007). *Bd* has caused large-scale mortalities, resulting in amphibian population declines throughout Australia (Laurance et al. 1996; Retallick et al. 2004; Schloegel et al. 2006), Central America (Berger et al. 1998; Lips et al. 2006), and the western United States (Padgett-Flohr and Hopkins 2009; Vredenburg et al. 2010). *Bd* is now panzootic with positive detections in 71 of 105 (68%) countries sampled and 48% of the species sampled (Olson et al. 2013; Olson and Ronnenberg 2014). The spread of *Bd* and the emergence of a similar species, *B. salamandrivorans* (Martel et al. 2013), has spurred increased interest regarding the various mechanisms that might forestall disease emergence in infected hosts [e.g., nutrition (Venesky et al. 2012; Hess et al. 2015), and anthropogenic factors (Greenspan et al. 2017). Several studies support the role that numerous symbiotic host epidermal bacteria may play in the amphibian immune defense repertoire (Bresciano et al. 2015; Harris et al. 2006; Lauer et al. 2007; Lauer et al. 2008; Rollins-Smith et al. 2006; Woodhams et al. 2018). One species, *Janthinobacterium lividum* (*Jliv*), appears to hold particular promise as it produces a potent antifungal metabolite, violacein (Becker et al. 2009; Brucker et al. 2008), which may contribute to the innate immune system of amphibians (Becker and Harris 2010; Harris et al. 2009a,b). Probiotic bacterial communities appears to vary between different locations or species (Bletz et al. 2017; McKenzie et al. 2012; Walke et al. 2014) and can influence an individual's survival following *Bd* exposure (Lam et al. 2010; Woodhams et al. 2006; Woodhams et al. 2007). However, antifungal species do not appear to be universally beneficial in protecting different amphibian species such as the Panamanian Golden Frog (*Atelopus zeteki*) (Becker et al. 2011).

A recent study that examined the prevalence of *Bd* in Wisconsin, USA showed that *Bd* was present at varying levels (mean = 6435, SD = 50920 copies/ng DNA) throughout numerous amphibian species and habitats (Standish et al. 2018). However, these detections were accompanied by a lack of chytridiomycosis disease signs (Standish et al 2018). It is unclear whether suboptimal environmental conditions (Forrest and Schlaepfer 2011), the presence of a less virulent endemic *Bd* lineage (rather than the Global Panzootic Lineage (GPL, Rosenblum et al. 2013)), a vigorous adaptive immune response (Ramsey et al. 2010), or perhaps the presence of symbiotic bacteria such as *Jliv* may play a role in the innate defense against this pathogen. Herein, we examine a range of amphibian samples collected in 2017 for the presence of *Jliv* using conventional polymerase chain reaction (Harris et al. 2009a). This study represents the first such survey for the symbiotic bacteria in the upper Mississippi River Basin, USA and serves as an initial assessment of the presence and role of *Jliv* in Wisconsin amphibians.

A portion of the field-collected samples (amphibian skin swabs; water samples for environmental DNA assessment) reported by Standish et al. (2018) was used for this analysis, and the following is a brief summary of these methods. Samples were opportunistically collected without apparent bias from numerous species, life stages, and habitats. Aquatic habitat types were visually determined based on size, water flow, vegetation, etc. (Bain and Stevenson 1999). We examined 335 amphibian samples from 12 species and 12 water samples from March 2017 to August 2017 (Fig. 1; Table 1; see Standish et al. 2018). Sampling was non-lethal in accordance with the amphibian skin swabbing protocol described by Hyatt et al. (2007). Sterile rayon fine-tipped swabs (Puritan Medical Products LLC, Guilford, Maine, USA) were used to first swab keratinized epidermal areas such as the tooth rows in larval anurans (Fellers et al. 2001; Kadekaru et al. 2016) followed by swabbing of the cloacal area (Gray et al. 2012). One-mL water samples were collected in sterile 1.5-mL tubes (USA Scientific, Ocala, Florida, USA) from undisturbed

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TABLE 1. *Janthinobacterium lividum* sampling site, habitat type, County in Wisconsin, USA where sampling occurred, and the number of samples per amphibian species and life stage, or number of water samples taken.

Site	Habitat type	County	Species or H_2O	N
Captive	NA	NA	Mudpuppy adults (*Necturus maculosus*)	62
A	Spring fed pond	Vernon	American Toad eggs (*Anaxyrus americanus*)	1
			American Toad larvae	4
			Green Frog adults (*Rana clamitans*)	5
			Green Frog larvae	25
			Tree frog larvae (*Hyla* spp.)	16
			H_2O	1
B	Spring fed pond	Vernon	American Toad eggs	1
			Boreal Chorus Frog adults (*Pseudacris maculata*)	3
			Boreal Chorus larvae	1
			Green Frog adults	7
			Green Frog juveniles	10
			Green Frog larvae	11
			Spring Peeper adults (*Pseudacris crucifer*)	3
			Spring Peeper larvae	1
			Wood Frog adults (*Rana sylvatica*)	6
			Wood Frog juveniles	5
			H_2O	3
C	Mature forest	Bayfield	Eastern Red-Backed Salamander adult (*Plethodon cinereus*)	1
D	Lake	Vernon	American Toad adults	4
			American Toad larvae	2
			Green Frog adult	1
			Green Frog juveniles	3
			Green Frog larvae	6
			H_2O	4
E	Urban	Monroe	American Toad juveniles	17
F	Marsh/Swamp	La Crosse	Green Frog larvae	4
G	Spring fed pond	Monroe	American Toad eggs	6
			American Toad larvae	1
			Bullfrog adults (*Rana catesbeiana*)	4
			Green Frog adults	7
			Green Frog juveniles	3
			Green Frog larvae	32
			Tree frog larvae	1
			H_2O	3
H	Spring fed pond	Vernon	American Toad eggs	1
			Bullfrog larvae	4
			Central Newt adults (*Notophtalmus viridescens*)	3
			Green Frog adult	1
			Green Frog juveniles	2
			Green Frog larvae	9
			Tree frog larvae	5
			H_2O	1
I	River/Marsh/Swamp	La Crosse	American Toad adult	1
			Green Frog adult	1
			Green Frog juveniles	11
			Green Frog larvae	8
			Leopard Frog adults (*Rana pipiens*)	17
J	Spring fed pond	Monroe	American Toad adult	1
			Green Frog adult	1
			Green Frog juveniles	5
			Green Frog larvae	3
K	Mature forest	Fond du Lac	Blue-spotted Salamander adult (*Ambystoma laterale*)	9
Total (N)				347

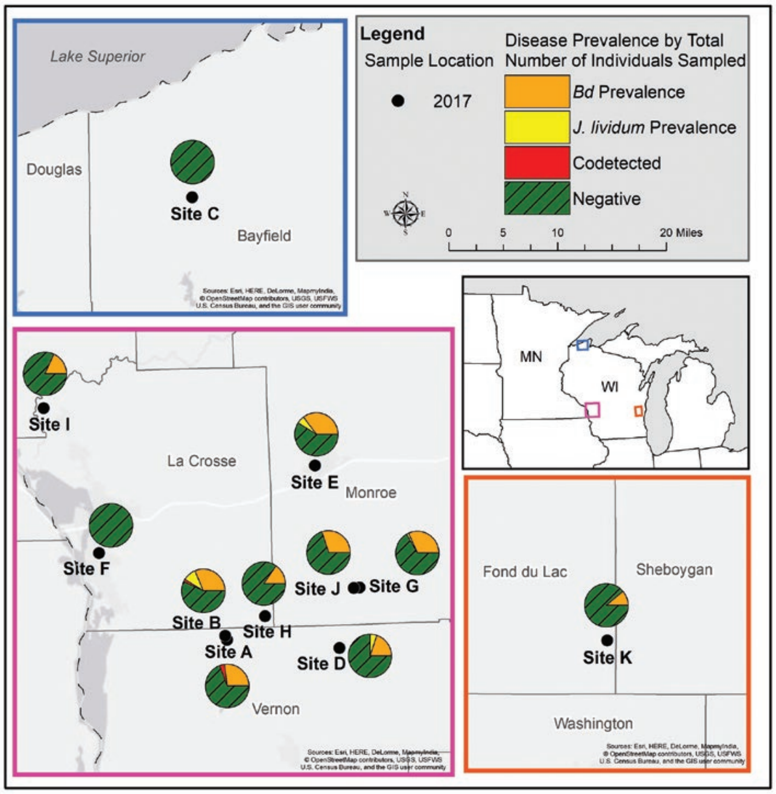
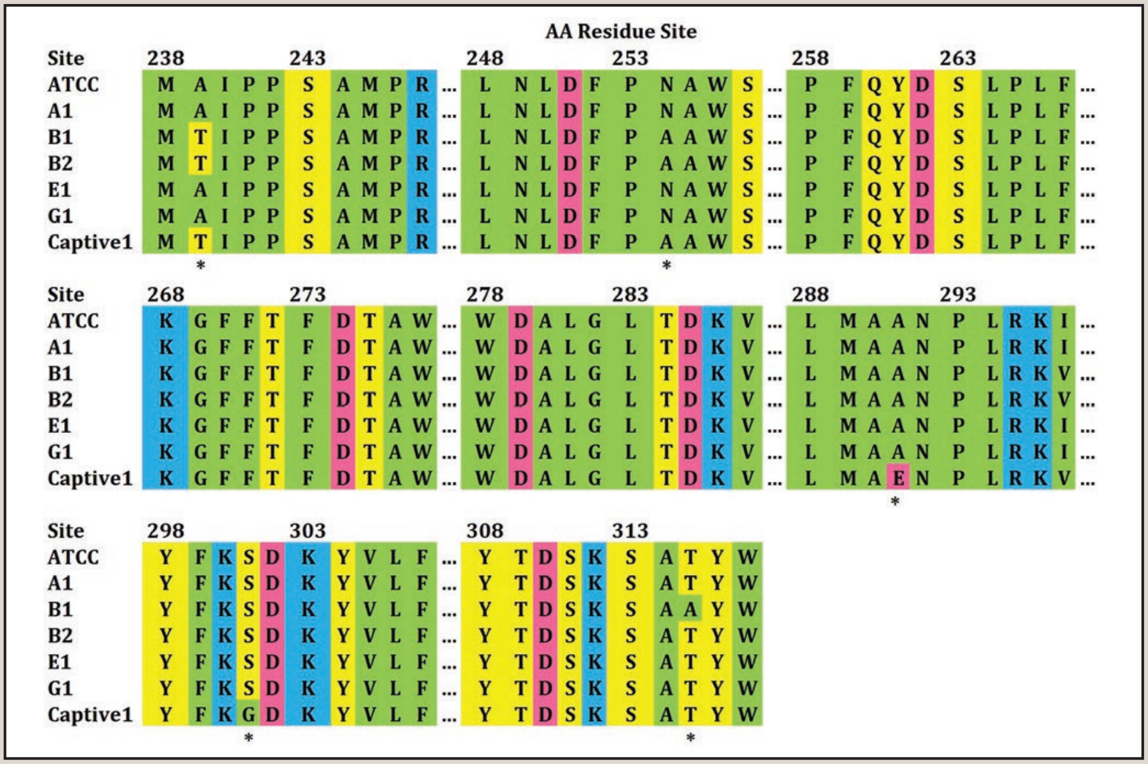


FIG. 1. Results of Wisconsin (WI), USA sampling for *Batrachochytrium dendrobatidis* (*Bd*) prevalence, *Janthinobacterium lividum* prevalence, codetected individuals, and negative individuals - shown as a portion of the total number of individuals sampled from each site. Counties are labeled on site maps. Maps were created using ArcGIS® software (V. 10.5, ESRI, Redlands, California, USA).



AA Residue Site

Site	238					243						248					253						258					263					
ATCC	M	A	I	P	P	S	A	M	P	R	...	L	N	L	D	F	P	N	A	W	S	...	P	F	Q	Y	D	S	L	P	L	F	...
A1	M	A	I	P	P	S	A	M	P	R	...	L	N	L	D	F	P	N	A	W	S	...	P	F	Q	Y	D	S	L	P	L	F	...
B1	M	T	I	P	P	S	A	M	P	R	...	L	N	L	D	F	P	A	A	W	S	...	P	F	Q	Y	D	S	L	P	L	F	...
B2	M	T	I	P	P	S	A	M	P	R	...	L	N	L	D	F	P	A	A	W	S	...	P	F	Q	Y	D	S	L	P	L	F	...
E1	M	A	I	P	P	S	A	M	P	R	...	L	N	L	D	F	P	N	A	W	S	...	P	F	Q	Y	D	S	L	P	L	F	...
G1	M	A	I	P	P	S	A	M	P	R	...	L	N	L	D	F	P	N	A	W	S	...	P	F	Q	Y	D	S	L	P	L	F	...
Captive1	M	T	I	P	P	S	A	M	P	R	...	L	N	L	D	F	P	A	A	W	S	...	P	F	Q	Y	D	S	L	P	L	F	...
		*																*															

Site	268					273						278					283						288					293					
ATCC	K	G	F	F	T	F	D	T	A	W	...	W	D	A	L	G	L	T	D	K	V	...	L	M	A	A	N	P	L	R	K	I	...
A1	K	G	F	F	T	F	D	T	A	W	...	W	D	A	L	G	L	T	D	K	V	...	L	M	A	A	N	P	L	R	K	I	...
B1	K	G	F	F	T	F	D	T	A	W	...	W	D	A	L	G	L	T	D	K	V	...	L	M	A	A	N	P	L	R	K	V	...
B2	K	G	F	F	T	F	D	T	A	W	...	W	D	A	L	G	L	T	D	K	V	...	L	M	A	A	N	P	L	R	K	V	...
E1	K	G	F	F	T	F	D	T	A	W	...	W	D	A	L	G	L	T	D	K	V	...	L	M	A	A	N	P	L	R	K	I	...
G1	K	G	F	F	T	F	D	T	A	W	...	W	D	A	L	G	L	T	D	K	V	...	L	M	A	A	N	P	L	R	K	I	...
Captive1	K	G	F	F	T	F	D	T	A	W	...	W	D	A	L	G	L	T	D	K	V	...	L	M	A	E	N	P	L	R	K	V	...
																										*							

Site	298					303						308					313				
ATCC	Y	F	K	S	D	K	Y	V	L	F	...	Y	T	D	S	K	S	A	T	Y	W
A1	Y	F	K	S	D	K	Y	V	L	F	...	Y	T	D	S	K	S	A	T	Y	W
B1	Y	F	K	S	D	K	Y	V	L	F	...	Y	T	D	S	K	S	A	A	Y	W
B2	Y	F	K	S	D	K	Y	V	L	F	...	Y	T	D	S	K	S	A	T	Y	W
E1	Y	F	K	S	D	K	Y	V	L	F	...	Y	T	D	S	K	S	A	T	Y	W
G1	Y	F	K	S	D	K	Y	V	L	F	...	Y	T	D	S	K	S	A	T	Y	W
Captive1	Y	F	K	G	D	K	Y	V	L	F	...	Y	T	D	S	K	S	A	T	Y	W
				*															*		

FIG. 2. Representative sequence data from cPCR-positive detections of *Janthinobacterium lividum* from Wisconsin amphibians. Tryptophan oxidase gene amplicons were Sanger sequenced, contigs were aligned and translated using MEGA 7.0. Green amino acids are hydrophobic non-polar, yellow amino acids are hydrophilic polar, pink are hydrophilic negatively charged and blue are hydrophilic positively charged. An asterisk (*) denotes variable sites.

sites prior to amphibian sampling. Stringent biosecurity and disinfection measures were followed between individuals and sampling areas as detailed by Gray et al. (2017). All equipment, including buckets, coolers, waders, boots, and nets was disinfected between sites using a 2% solution of Virkon® Aquatic (DuPont, Wilmington, Delaware, USA) (Gray et al. 2017).

Samples were stored at 4°C for immediate extraction or -20°C if extraction could not take place within 24 h of collection. Water samples were vortexed and combined with an equivalent volume of PrepMan™ Ultra Sample Preparation Reagent (Thermo Fisher Scientific, Waltham, Massachusetts, USA); for swabs, 150 µL of the PrepMan™ Ultra Sample Preparation Reagent was added. Extractions then proceeded following the manufacturer's instructions and quantified using a Qubit™ 3.0 fluorometer (Thermo Fisher Scientific). Extracted samples were stored at 4°C until analysis. Samples were analyzed by Standish et al. (2018) for the presence of amphibian pathogens using a multiplex qPCR and stored at -20°C. Copy number/ng of extracted DNA was determined using a synthetic gBlock® standard (Standish et al. 2018). A two-tailed t-test was used to test for a significance difference in *Bd* copy number (copies/ng DNA extract) in samples positive and negative for *Jliv*. Captive mudpuppies and water samples were not included in this analysis.

Janthinobacterium lividum cPCR.—The cPCR was conducted using the *Jliv* specific primer set described by Harris et al. (2009a) for the detection of the bacteria's tryptophan oxidase gene, a precursor in violacein synthesis: JlivF (5-TACCACGAATTGCTGTGCCAGTTG-3) and JlivR (5-ACACGCTCCAGGTATACGTCTTCA-3). PCR reactions (52 µL) contained 46 µL Platinum® PCR Supermix (ThermoFisher Scientific), 2 µM of both the JlivF and JlivR primers, and 4 µL of sample DNA. The negative control was master mix without any sample extract, and the positive control contained extracted DNA from a *Jliv* reference strain 12473 (ATTC®, Manassas, Virgina, USA). Amplification was conducted using a Biometra T3000 Thermocycler (Biometra GmbH, Germany) with the following conditions: 4 min at 94°C followed by 35 cycles of 30 s at 94°C, 60 s at 58°C, and 2 min at 72°C. Reactions concluded with a final extension for 10 min at 72°C. Reactions were visualized using the E-Gel Electrophoresis System (Thermo Fisher Scientific) and comparing bands to the TrackIt™ 100 bp ladder (ThermoFisher Scientific).

Sequence Analysis.—Reactions producing a band approximately 500 bps in length were considered presumptively positive for the *Jliv* violacein gene and sent to the Whitney Genetics Laboratory (U.S. Fish and Wildlife Service; Onalaska, WI, USA) for direct Sanger sequencing. Contigs were assembled using Codon Code Aligner (CodonCode Corporation, Centerville, Massachusetts, USA). Contigs were searched for similarity using BLASTn (NCBI, National Center for Biotechnology Information, National Institutes of Health, Bethesda, Maryland, USA) and compared to the ATCC positive control (GenBank Accession LC000629.1). MEGA7 (Kumar et al. 2016) was used in the alignment and translation of sequences to allow for amino acid comparison. DNA sequences were deposited in GenBank (Accession numbers MH270664–MH270675).

Jliv was detected at 5 of 11 field sites sampled, with site prevalence ranging from 1.8 to 11.8% (Fig. 1; Table 2). When coupled with the findings reported previously by Standish et al. (2018), *Jliv* was detected more often at sites where *Bd* was most prevalent and was not detected at any site where *Bd* was not detected (Table 2). Though, on wild amphibians, *Bd* copy number was not significantly different between *Jliv* positive (Mean = 2263, SD = 2264) and *Jliv* negative (Mean = 9307, SD = 63915) samples (t (83) = -0.189, p = 0.849).

Table 2. The total number of animal and water samples tested from each site, *Batrachochytrium dendrobatidis* (*Bd*) detection results by qPCR (assay, results and sample set reported in Standish et al. 2018) and *Janthinobacterium lividum* (*Jliv*) detection results from cPCR.

Site	N	% *Bd* detected	% *Jliv* detected	% Co-detected
Captive	62	71.0	1.6	1.6
A	52	30.8	3.8	3.8
B	51	31.4	11.8	1.9
C	1	0.0	0.0	0.0
D	20	20.0	5.0	0.0
E	17	35.3	5.9	0.0
F	4	0.0	0.0	0.0
G	57	31.6	1.8	0.0
H	26	15.4	0.0	0.0
I	38	18.4	0.0	0.0
J	10	30.0	0.0	0.0
K	9	11.1	0.0	0.0

Habitat type did not appear to influence the detection of *Jliv*, as samples from spring-fed ponds (sites A, B, G; Appendix 1), a lake (D), and an urban site (E) tested positive for the probiotic bacteria. Detections of *Jliv* similarly varied across species (Table 3) with detections in American Toad (*Anaxyrus americanus*) juveniles and eggs, Boreal Chorus Frog (*Pseudacris maculata*) and Green Frog (*Rana clamitans*) adults and tadpoles, H_2O samples, and captive Mudpuppies (*Necturus maculosus*). The highest *Jliv* prevalence was observed in the Boreal Chorus Frogs (33.3%), followed by H_2O samples (25.0%) and eggs from American Toads (11.1%).

The contig length of positive cPCR reactions ranged from 308 to 448 bps. (Table 4). All contigs were 93–100% similar to the ATCC *J. lividum* reference sequence (GenBank Accession LC000629.1). Differences in this violacein precursor gene prompted the examination of amino acid variability (Fig. 2). For example, amino acid residues at sites 239, 254, 291, 301 and 315 varied between hydrophilic and hydrophobic amino acids residues.

Our preliminary sampling effort of Wisconsin amphibians confirmed the presence of the probiotic bacteria *Jliv* co-occurring with *Bd*. Using non-lethal field collection methods and cPCR, we were able to detect the bacteria in several locations and species. However, there were relatively few detections of *Jliv* at our study sites (i.e., the bacteria was detected at 5 of 11 sites). Our findings appear to corroborate the low prevalence of *Jliv* noted in investigations of wild *Rana sierrae* and *Rana mucosa* populations in California, USA (Woodhams et al. 2007; Jani and Briggs 2014). However, these findings are contrary to a study noting the consistent presence of *Jliv* on Red-backed Salamanders (*Plethodon cinereus*) in Virginia, USA (Loudon et al. 2014) as well as with amphibians in higher elevations of Ecuador (Bresciano et al. 2015). A recent study using next-generation sequencing did find *Jliv* on all 36 amphibian species examined, with prevalence ranging from 11–100% (Woodhams et al. 2018). These studies suggest the possibility of false negatives in our sampling methods, leading to lower prevalence

Table 3. The total number of each amphibian species sampled and prevalence that tested positive for *Janthinobacterium lividum* (*Jliv*) using cPCR.

Sample/Species/Lifestage	N	% *Jliv*-positive
American Toad adults (*Anaxyrus americanus*)	6	0.0
American Toad eggs	9	11.1
American Toad juveniles	17	5.9
American Toad larvae	7	0.0
Blue-spotted Salamander adult (*Ambystoma laterale*)	9	0.0
Boreal Chorus Frog adults (*Pseudacris maculata*)	3	33.3
Boreal Chorus Frog larvae	1	0.0
Bullfrog adults (*Rana catesbeiana*)	4	0.0
Bullfrog larvae	4	0.0
Central Newt adults (*Notophtalmus viridescens*)	3	0.0
Eastern Red-backed Salamander adult (*Plethodon cinereus*)	1	0.0
Green Frog adults (*Rana clamitans*)	23	8.7
Green Frog juveniles	34	0.0
Green Frog larvae	98	3.1
H_2O	12	25.0
Leopard Frog adult (*Rana pipiens*)	17	0.0
Mudpuppy adults(*Necturus maculosus*)	62	1.6
Spring Peeper adults(*Pseudacris crucifer*)	3	0.0
Spring Peeper larvae	1	0.0
Tree Frog larvae (*Hyla* spp.)	22	0.0
Wood Frog adults (*Rana sylvatica*)	6	0.0
Wood Frog juveniles	5	0.0
Total (N)	347	

estimates. Moreover, with so few detections, it is difficult to draw conclusions regarding the extent that *Jliv* may be involved in the *Bd*-immune defense of Wisconsin amphibians. Though, we did isolate *Jliv* more frequently from aquatic amphibians, it is unclear if *Jliv* is more closely associated with the aquatic than the terrestrial environments. For instance, we analyzed only a few water samples, but 25% were positive for *Jliv*, as were samples from captive Mudpuppies and American Toad eggs, an aquatic species and life stage, respectively. This supports the hypothesis that bacterial communities differ between aquatic and terrestrial life cycle phases, with a higher proportion of antifungal taxa in aquatic larvae (Sabino-Pinto et al. 2017). However, more detections in aquatic samples may simply be an artifact of our sampling methods, such as the capillary action of swabs allowing for greater sampling efficiency for aquatic samples/environments.

One of the more interesting findings of this study is the variability we found in the L-tryptophan oxidase gene (VioA). There appears to be nucleotide variability at several codons, which results in translational changes. Whether these alterations result in either functional or conformational changes in the enzyme or violacein production downstream remains unclear, but it does suggest the occurrence of amino acid variation in the VioA gene. The VioA enzyme is responsible for the initial oxidative conversion of L-tryptophan into indole-3-pyruvic acid (IPA) imine, which is then coupled into a dimer by VioB (Füller et al. 2016). A mutagenesis and enzymatic kinetic study on VioA from *Chromobacterium violaceum* indicates that VioA activity can vary between different tryptophan analogue substrates (Füller et al. 2016). The study identified several key catalytic residues (Arg^{64}, Lys^{269} and Tyr^{309}) (Füller et al. 2016), two of which appear conserved in all of our *Jliv* sequences (residues Lys^{268} and Tyr^{308}). It is unclear if these translational alterations affect violacein production. Differences in the activity or quantity of this antifungal compound could explain why *Jliv* displays varying levels of *Bd* inhibition (Woodhams et al. 2006, 2007; Lam et al. 2010; Becker et al. 2011) and supports the notion that this may be due to differences in phenotypic expression (Jani and Briggs 2014).

Future studies focusing on collecting and genotyping various isolates of *Jliv* while using them in inhibition assays with *Bd* could help elucidate if such differences exist. Whereas there has been much emphasis placed on the potential for *Jliv* to provide protection to amphibians from *Bd*, in this study, we did not observe a significant correlation with *Bd* burden. Examining larger sample sizes and using more sensitive detection methods (i.e., next-generation sequencing) may help further elucidate the extent of the protection provided to wild amphibians by *Jliv*. Additionally, future investigations can survey for additional symbiotic bacteria as *Jliv* represents only one such species.

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HERPETOLOGICAL HISTORY

Herpetological Review, 2018, 49(3), 480–484.

C. J. P. Ionides: A Commemoration

Fifty years ago, on the night of 22 September 1968, C. J. P. Ionides (Fig. 1) died in Nairobi Hospital, as a result of complications following the amputation of both legs. His death was marked by a diary note in the British literary magazine *The Spectator*, (Anonymous[a] 1968) and obituaries in both *The Times* and *The Observer* (Anonymous [b]; Richardson 1968), two of the United Kingdon's most venerable newspapers. It was a surprising indication of the respect and affection that the public held for this slight, elfin-like reclusive naturalist and herpetologist, a man who had spent the previous 25 years living in the remote woodlands of southern Tanzania, collecting reptiles, in particular highly venomous snakes. A man of steely determination, he was single-minded in pursuit of his hobbies, initially hunting rare mammals, and then collecting reptiles. He never married. As a young man he was in love, but faced with the dilemma of either marrying the girl or devoting himself to hunting, cold bloodedly chose hunting.

Known to his family as "Bobby" and to his friends as "Iodine," Constantine John Philip Ionides was born in Hove, United Kingdom, on 17 January 1901. The son of a prominent surgeon in Brighton, Ionides was British but of Greek descent. He was educated at the famous Rugby School, where he got into trouble after a search of his study (for stolen money, of which he was innocent) revealed, among other things, a sawn-off shotgun, two pistols, six rabbit nets, a cosh, and several animal skins; the rebellious young man was a prolific poacher and enthusiastic trainee taxidermist. But the tradition of Rugby School was that no cognizance could be taken of things in a pupil's study, other than the specific items being searched for.

His initial career was checkered. He enlisted in the British Army, trained at Sandhurst, and was posted to central India in 1922. Never a model officer, as he wryly recounts in his 1965 autobiography, "the day I was not called a bloody young bastard I was concerned for the health of my superior officer." But he did a lot of hunting; he shot two rogue elephants, and in India he also handled and kept snakes for the first time. On his leave, before returning to Britain, he went to Kenya on a hunting safari. It was a pivotal journey, opening his eyes to the African continent. Back in UK, he applied for secondment to the Kings African Rifles, a colonial regiment drawn from East African troops, and in 1925 was posted to Dar Es Salaam, in Tanganyika Territory (now Tanzania).

He only lasted two further years in the military. Ionides was no soldier, and also upset the deputy commanding officer's wife. Ionides objected to her joining the troops' Swahili classes, due to the "earthy" terms they would need to learn; enraged, she pressured her husband into writing a highly critical confidential report. This had the desired effect. Ionides resigned his commission and became an ivory poacher and a professional hunter. Over the next six years, between taking clients out to shoot big game, he poached elephants, playing the border between Tanzania and the Congo. In 1932 he was lucky to survive an attack

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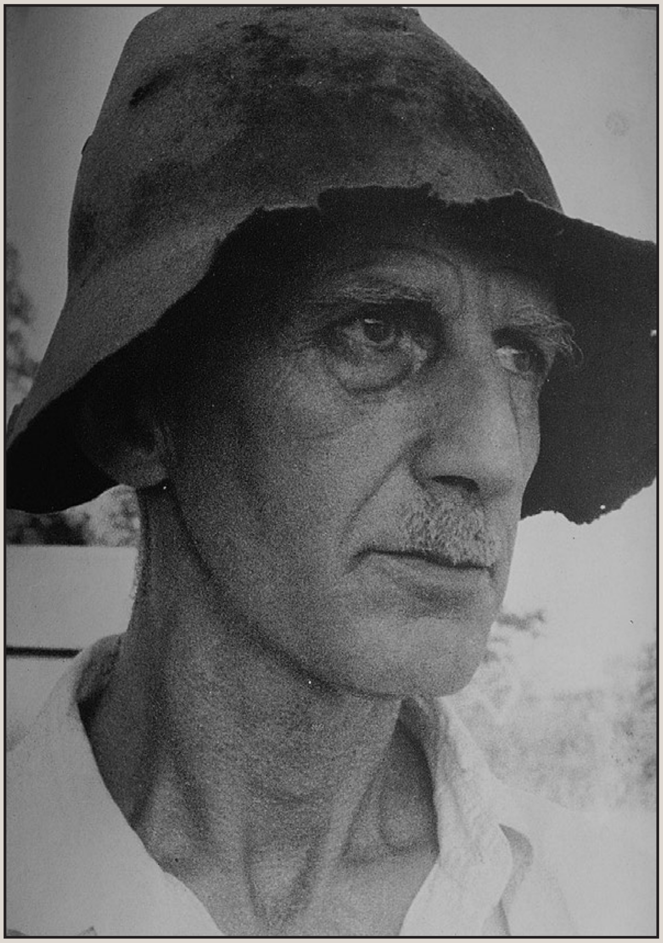


PHOTO BY BRIAN NICHOLSON

Fig. 1. C. J. P. Ionides and his "magic hat," an ancient 'Terai,' and a fetish believed by many to be the source of his legendary snake-handling ability (1962).

by a cow elephant that had been wounded by a local hunter. She attempted to stab him with her tusks and kicked him about; miraculously, Ionides' only damage was to the hearing of his left ear. He was left partially deaf, although, as he dryly commented in his autobiography, this sometimes proved useful.

In 1933, Ionides joined the Tanganyika Game Department. For an admitted poacher, this must sound, as the man himself said (Ionides 1965), like a gangster saying that all he ever wanted to do was become a policeman. But he rationalized it by stating that his knowledge of poaching would be invaluable to the department. And it was. A game ranger's business was preventing poaching and dealing with problem animals. And problematic some were. Apart from minor nuisances like fruit-stealing baboons and potato-thieving warthogs, Ionides found himself dealing with crop-raiding and rogue elephants, and man-eating lions and leopards. Killer leopards in Tanzania usually took children. One that Ionides hunted killed 18 children before he finally trapped it. A rogue elephant, wounded by an incompetent Englishman with an army .303 rifle, killed 28 people before Ionides shot it. With his protégé Brian Nicholson, Ionides was charged with keeping elephants out of the massive, British-government funded sugar cane farming scheme on the Ruaha River; they had to shoot 600 of the huge beasts. Such slaughter nowadays would be greeted with horror. But in the 1930s in Tanzania, elephants and other big game were seen merely as a dangerous nuisance, preventing development. A similar agricultural scheme in Kenya in the 1940s saw the professional hunter John Hunter and his men shoot over a thousand black rhinoceros (Hunter 1952). No one had any idea that in the future, visitors would pay good money for the privilege of seeing such animals. But Ionides and Nicholson were later instrumental in the creation of the Selous Game Reserve, a massive conservation area in central Tanzania.

At the time, Ionides' interest in reptiles had not blossomed; his ambition was to make a mounted collection of Africa's rare mammals, and during periods of leave he hunted these. He spent all his free time and money in pursuit of trophies, from Malawi to the Sudan, often undergoing many days of desperate hardship to obtain his quarry. Ionides was a hunter of the old school, scorning such practices as following his quarry by vehicle or shooting over a bait; nor would he ever take the shot unless he could see enough of his quarry to be certain of the outcome. He went into the field on foot and kept at it; a Yellow-backed Duiker (*Cephalophus silvicultor*) that he shot in the Mau Forest of Kenya took him 49 days in the high-altitude cold and wet. His specimens, expertly mounted by his friend Norman Mitton, for years formed the subjects of dioramas at the National Museum in Nairobi. Despite changes in layout, some are still on display; the Okapi he shot in the Ituri Forest remains prominent in the central hall and has been seen by many thousands of Kenyan schoolchildren.

Ionides re-enlisted in the Second World War, in the Kings African Rifles, his old regiment. His expertise was recognized, whatever misgivings the authorities may have had about his previous service. He was given an independent command and sent to sort out insurrection in Somalia and Ethiopia. The Italians had withdrawn from much of Somalia, but before departing had thoughtfully given the Somalis rifles, and instructions to make trouble for the British. Ionides, trying to deal with armed young men in northern Somalia stealing their neighbors' stock and shooting up all and sundry, solved the problem in a novel way. He ordered his company to round up and incarcerate all the young women from the nearby villages, he then sent a message to the local youths: "return the rifles or I'll let my sex-starved troops loose on your women." Most of the guns came back, although in order to get all of them, Ionides also had to threaten to hang the local Sheikh.

Discharged from the military in 1943, Ionides was asked by the paleoanthropologist Louis Leakey if he could collect snakes for the display at the Museum in Nairobi, where Leakey was the curator. The request re-kindled Ionides' interest in reptiles. As well as the spectacular snakes that Leakey wanted for his display, Ionides began systematically collecting smaller reptiles, started a correspondence with Charles Pitman (author of the 1938 pioneering *A Guide to the Snakes of Uganda*) and donated specimens to various museums. He struck up a friendship with Arthur Loveridge, Africa's most eminent herpetologist, and regularly sent material to the Museum of Comparative Zoology at Harvard. Loveridge repaid the compliment by naming several new species after Ionides, and wrote regular bulletins describing his collections (Loveridge 1951, *et seq.*).

In 1956 a series of thromboses in his right leg led to surgery and the surgeon's verdict that his long-distance walking days were over; there would be no more rarity hunting. Ionides was bitterly disappointed, but philosophical. "Luck is not a word I like to use," he said. "Let us just say that nature played a little practical joke" (Ionides 1965). But his misfortune was herpetology's gain, for he then threw himself with gusto into fieldwork. He settled in southern Tanzania, initially at Liwale and then at Newala near the Mozambique border, caught local reptiles and made collecting forays all over eastern Africa. He captured huge numbers of snakes around Newala. In a short paper (Ionides and Pitman 1965a), he describes collecting 973 Green Mambas (*Dendroaspis angusticeps*) and 689 Gaboon Vipers (*Bitis gabonica*) over a seven-year period. His personal notebooks list the totals of dangerous snakes he collected; in his lifetime he caught, among others, 6633 green mambas, 2159 Gaboon Vipers, 210 black mambas, 312 North-east African Carpet Vipers, and 386 Black-necked Spitting Cobras. The majority of these were sold to zoos, snake parks, and venom research institutes worldwide; the smaller non-commercial specimens were donated to museums.

During these years, Ionides lived an idyllic herpetological life; relaxing, smoking, and reading in his bungalow until a snake report came in, whereupon he and his assistants travelled quickly to the locality, caught the snake, paid the fee and returned to base. If the reptile was some distance off the road Iodine was wheeled in a single-wheeled chair to save his legs. In the wet season, his assistants routinely turned ground cover, collecting the smaller species. From time to time he loaded his truck with his collecting team and their retinue and travelled to other parts of East Africa in search of venomous species. He visited the Aberdare Mountains in Kenya in pursuit of the Kenya Montane Viper (*Montatheris hindii*), Lake Tanganyika in pursuit of the Banded Water Cobra (*Naja annulata*), and northern Kenya looking for North-east African Carpet Vipers (*Echis pyramidum*). He also made expeditions to Al Abr, in the Hadramaut in Yemen in 1965; there he collected 10 Arabian Horned Vipers, *Cerastes gasperettii* , and to Thailand in 1966, with Jonathan Leakey, where he collected 16 King Cobras (*Ophiophagus hannah*).

Ionides was a purist where snake collecting was concerned, insisting that snakes reported to him were not restrained in any way. He was known to refuse to catch snakes that had been trapped under a basket, for example. He taught himself snake catching; at Nairobi Snake Park we used a modified version of the snake sticks that he designed. Fifty years later I have yet to

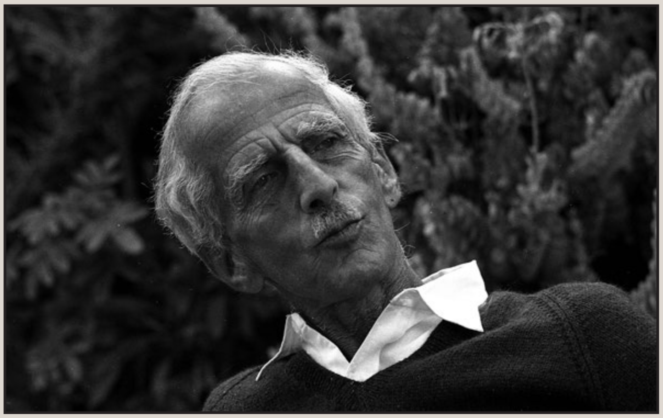


PHOTO BY ALEC FORBES-WATSON

Fig. 2. Ionides in Nairobi, Kenya in 1966, after his return from Thailand. He wore the same brown sweater for 25 years.

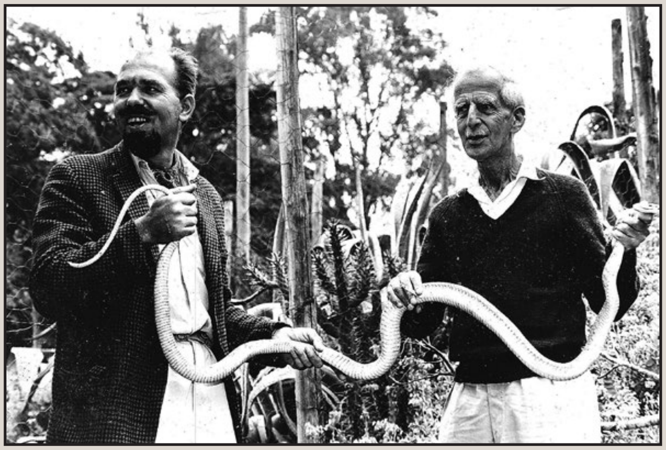


PHOTO COURTESY OF BIO-KEN ARCHIVE

Fig. 3. Ionides and James Ashe with a big Blanding's Tree Snake (*Toxicodryas blandingii*) at Nairobi Snake Park, 1965. A cast of this huge specimen is still on display in the National Museum in Nairobi.

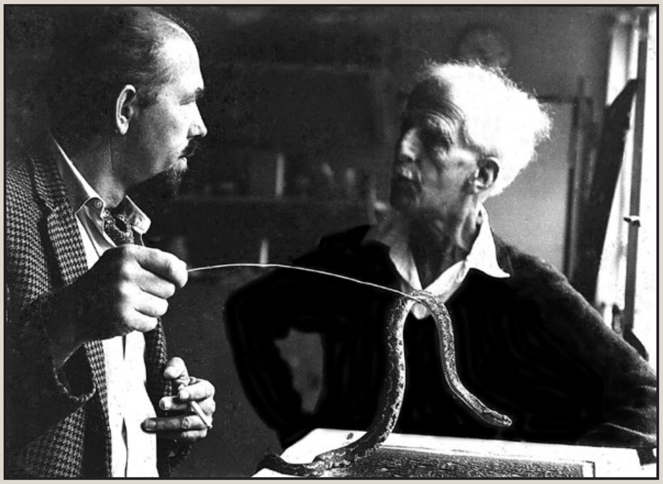


PHOTO COURTESY OF BIO-KEN ARCHIVE

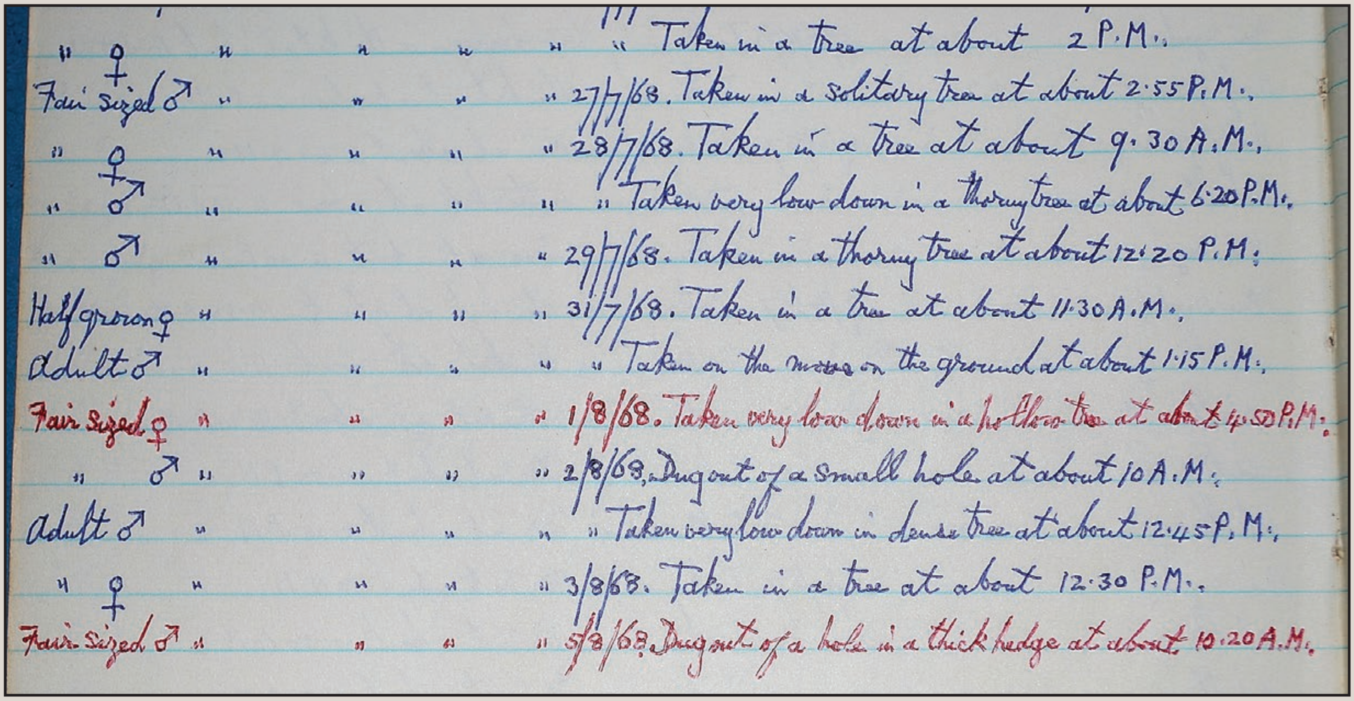
Fig. 4. Ionides and James Ashe discuss the new Mount Kenya Bush Viper (*Atheris desaixi*), in the Quarantine/Research backroom at Nairobi Snake Park, 1968.

see a better stick. Ionides developed the "reverse-bag technique" for bagging a big deadly elapid snake, whereby the handler puts their hand into the inverted bag, grasps the head through the bag and then reverses it, the snake's body is then pushed into the bag, and when it has been tied the handler releases the head.

The 1960s were a remarkable time in East African herpetology. Jonathan Leakey, Louis Leakey's eldest son, had set up the Nairobi Snake Park and then started his own snake farm at Lake Baringo in Kenya. Subsequent curators of the Nairobi snake park included the husbandrist Cecil Webb, and then the dynamic and charismatic herpetologist James Ashe. Iodine always stopped at Nairobi Snake Park when he passed through the capital, and spent time with James. It was there that I first met him, in 1966, I was an impressionable 13-year old and he certainly made an impression on me. He had time for anyone if they were interested in natural history, and his conversation was precise, scintillating, and inspiring (Fig. 2). Away from home, he invariably sat in an old-fashioned folding deck chair that he took everywhere with him. All were welcome at the snake park; to spend an afternoon drinking tea and in herpetological conversation with Iodine, James Ashe (Fig. 3), and often an eminent visitor like Joy Adamson, Jonathan Leakey or the Tsavo wardens David Sheldrick or Bill Woodley, was like an afternoon in Paradise. You went home inspired to do herpetology. I once turned up with a Cape Wolf Snake (*Lycophidion capense*) that I was unable to identify, and Iodine talked to me for half an hour, pointing out the salient features and explaining how to distinguish between it and a dangerous burrowing asp (*Atractaspis*). He himself had twice been bitten by these innocuous-looking, but dangerous little snakes. Ionides suffered 13 bites in total from dangerous species, almost all, as he self-deprecatingly noted, the results of carelessness and inexperience. One bite, however, was from a night adder that he forced to bite him, to test his immunity after undergoing a traditional local immunization involving plant concoctions, rubbed into razor cuts on his limbs. No symptoms resulted.

In the mid-1960s, Iodine's leg problems became worse. Nevertheless, he continued travelling and collecting. In 1967 an American Peace Corps volunteer, Frank DeSaix, had found a new species of bush viper at Chuka on the southeastern slopes of Mount Kenya, later named *Atheris desaixi*, and in early 1968 Iodine travelled there and collected one (Fig. 4). In July of 1968 he mounted an expedition to Chesegon, north of Lake Baringo in the Kerio Valley in Kenya, with Peter Nares, one of Jonathan Leakey's field team leaders. It turned out to be his final safari. He collected 27 Black Mambas in the valley, taking his lifetime haul to over 200. But on 5 August Nares heard a scream from Ionides' tent, and found him lying on the floor in agony. Medically evacuated to Nairobi, both his legs were amputated above the knee. Ionides received many visitors, and joked about his snake-proof legs. But his surgical wounds did not heal, complications set in, and he died on Sunday, 22 September 1968. His remains were cremated after a service at the Greek Orthodox Church in Nairobi. His ashes were buried in a grave near that of the pioneer Frederick Courtenay Selous, in the magnificent Selous Game Reserve in Tanzania, the haven that he had helped found.

There are three books about Ionides. The first, *Snake Man* (1960) by the writer Alan Wykes, is very readable and well written. It was Wykes' best-selling book and brought the snake hunter to the public's attention. But Ionides detested it—he described it to Ann Mitton Simon, daughter of his old friend Norman Mitton, as "filthy tripe"—as it sensationalized both the man and his snakes, which was anathema to this most accurate of naturalists. Any herpetologist reading *Snake Man* might guess at this, for Wykes opens with an incident where a Green Mamba enters a hut at night and kills eight people; Ionides then catches it. In another story Wykes describes Ionides sitting in the dark on a long-drop toilet at night when a Black Mamba crawls across



" ♀	"	"	"	"	" Taken in a tree at about 2 P.M.,
Fair sized ♂	"	"	"	" 27/7/68.	Taken in a solitary tree at about 2·55 P.M.,
" ♀	"	"	"	" 28/7/68.	Taken in a tree at about 9·30 A.M.,
" ♂	"	"	"	"	" Taken very low down in a thorny tree at about 6·20 P.M.,
" ♂	"	"	"	" 29/7/68.	Taken in a thorny tree at about 12·20 P.M.;
Half grown ♀	"	"	"	" 31/7/68.	Taken in a tree at about 11·30 A.M.,
Adult ♂	"	"	"	"	" Taken on the moss on the ground at about 1·15 P.M.,
Fair sized ♀	"	"	"	" 1/8/68.	Taken very low down in a hollow tree at about 4·50 P.M.
" ♂	"	"	"	" 2/8/68.	Dug out of a small hole at about 10 A.M.;
Adult ♂	"	"	"	"	" Taken very low down in dense tree at about 12·45 P.M.,
" ♀	"	"	"	" 3/8/68.	Taken in a tree at about 12·30 P.M.,
Fair sized ♂	"	"	"	" 5/8/68.	Dug out of a hole in a thick hedge at about 10·20 A.M.,

PHOTO BY STEPHEN SPAWLS 2018

Fig. 5. Final Black Mamba entry in Ionides' notebook, written with a ballpoint pen. All of Ionides' records save these final expedition notes were written in an elegant longhand with an old-fashioned dip pen with a steel nib that he dipped in blue ink.

his legs. Both stories are fiction; mambas are diurnal snakes. The second book, *Life with Ionides*, by the acclaimed British author Margaret Lane, was published in 1963, and describes her three-month visit to Ionides' home in southern Tanzania. Like many others including the American writer John Gunther and the photographer Sally Anne Thompson, Lane made the long pilgrimage down to Newala, drawn by the magnetism of her subject. Lane was a beauty, and her affection for Ionides, manifest in the book, has lead to rumors that there was something between them, although Iodine once commented gruffly to James Ashe that he felt her book made him seem somewhat unhygienic. The third book, published in hardback as *A Hunter's Story* (1965), and in softback (1968) as *Mambas and Man-eaters*, was his autobiography, although the writer Dennis Holman did most of the editorial work and kindly insisted that it was published under Iodine's name alone. This is a super book, and conveys something of the true nature of the man, his motives, his wry humor, rigorous accuracy, and the sheer excitement of the chase. He tells a gripping story. But it must be read with the zeitgeist in mind; Ionides' attitudes to women and black Africans were often anachronistic.

Ionides is mentioned in a number of other books. Not everyone approved of his collecting activities or manners. In 1980, the famous Kenyan writer Elspeth Huxley published *Nellie; Letters from Africa*, an edited version of her mother, Nellie Grant's, Kenya letters. Iodine had camped at the Grants' farm at Njoro in 1959, on the hunt for Kenya Horned Vipers (*Bitis worthingtoni*). Nellie wrote "I have the great snake catcher staying here… He is a bit round the bend. He asked for chameleons…[twelve] were brought to him. I said…what were the chameleons having for tea and they were all dead! It made me feel sick….He is very deaf and boring." She mentioned that Ionides disliked fridges, newspapers, and the radio, and other writers have commented on his contempt for what he called modern gadgets. He wrote with an old-fashioned ink pen, and hated ballpoints, although he used them in the end, his final mamba entries in his notebooks are in ballpoint pen (Fig. 5).

Sadly, the man himself published little, save a few nature notes (Ionides 1953; Ionides and Pitman 1965a, b, etc.) At his death he was working on a guide to the reptiles of East Africa; three of his notebooks form a draft manuscript. But it was never completed. He died before he could research the species he was not familiar with. The incomplete manuscript, kindly made available to me by Jonathan Leakey, contains much remarkable material; he noted in 1953 that shovel-snout snakes (*Prosymna*) ate reptile eggs, but this was not reported in the literature until the 1980s. He found Hinkel's Red-flanked Skinks (*Lepidothyris hinkeli*) in the Kakamega Forest in 1963, although the first specimens were not formally recorded there until the 1980s. This reluctance to publish (Arne Schiøtz [2004] called it the "Nairobi Museum Cramp") has bedevilled East Africa herpetology. I'm not complaining; the fact that Ionides, James Ashe, Alex MacKay, and Jonathan Leakey hardly ever put pen to paper opened the path for others to do so. But Iodine's legacy rests with the many thousands of crucial specimens that he donated to museums. His fieldwork clarified our knowledge of the distribution of the East African herpetofauna; for that and the stimulating memories of a genuinely larger-than-life naturalist, he will be endearingly remembered.

Acknowledgments.—I thank my old friend Jonathan Leakey, who kindly made available Ionides' field notebooks, and Julia Leakey and Dena Crain, who organized this. Joy MacKay, Bob Drewes, Mark O'Shea, and Sandra Harwood supplied press cuttings and photographs. My thanks are also due to Royjan Taylor and Nic Cahill of Bio-Ken Snake Farm, for kindly allowing use of material from the Bio-Ken archive. This commemoration is dedicated to the memory of the late James and Sanda Ashe, of Bio-Ken, Watamu, Kenya, for many years of stimulating support to herpetology in East Africa.

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HERPETOCULTURE

HERPETOCULTURE NOTES

CAUDATA — SALAMANDERS

***EURYCEA RATHBUNI* (Texas Blind Salamander). OOPHAGY AND CANNIBALISM OF LARVAE.** *Eurycea rathbuni* is a troglobitic spelerpine salamander endemic to the San Marcos Springs of the Edwards Aquifer in Hays County, Texas, USA (Hammerson and Chippindale 2004. IUCN Red List of Threatened Species 2004: e.T39262A10173274). Because of its occurrence in water-filled subterranean caverns, field observations on the behavior and habits of *E. rathbuni* have been limited (e.g., Uhlenhuth 1921. Biol. Bull. 40:73–104), and much of what is known about the species' biology has come from observations of individuals maintained in captivity (e.g., Norman 1900. Amer. Nat. 34:179–183; Uhlenhuth, *op. cit.*; Maruska 1982. Proceeding of the 5th Annual Reptile Symposium on Captive Propagation and Husbandry 5:151–161; Bechler 1986. Proceedings of the 9th International Congress of Speleology 5:120–122; Bechler 1988. Southwest. Nat. 33:124–126).

Audubon Zoo has maintained a captive colony of *E. rathbuni* since 2001 and has been successfully reproducing this species since 2004. An adult breeding group comprised of 10 individuals of unknown sexes is maintained on public display in a 190-L aquarium at the zoo's Reptile Encounter building. Round river stones of varying sizes cover the floor of the exhibit as a substrate and provide potential sites for egg deposition. To facilitate public viewing, fluorescent lighting illuminates the exhibit on a 10-h photoperiod. Water parameters, photoperiod, and feeding frequency remain constant throughout the year.

Eggs are periodically produced throughout the year, although it is unclear which individual or individuals produce them. Eggs from clutches ranging from around 20–60 eggs are usually adhered singly or in small clusters to the surfaces of submerged rocks during each laying event. Eggs have typically been removed from the exhibit as soon as they are discovered and set up in a separate aquarium for hatching and development, but on several occasions, adult *E. rathbuni* were observed predating the eggs before keeper staff could retrieve them. Additionally, on rare occasions when undetected eggs hatched on exhibit, some adults were observed predating the larvae. Due to a lack of discernable physical features to facilitate visual identification, it is also unclear which individual or individuals have been observed consuming eggs or larvae.

The most recent observed case of oophagy by an adult *E. rathbuni* was closely monitored by one of us (RWM) and recorded with video. A clutch of around 20 eggs was laid on 10 February 2018 but could not be retrieved by keepers until two days later. On the morning of 12 February, one of the adults was observed directly above an egg that was adhered to the side of a rock (Fig. 1a). Using suction to detach the egg from the rock, the animal drew the egg into its mouth and then proceeded to reposition the egg inside its mouth for several seconds using chewing-like jaw movements before exuding a large cloud of opaque liquid (presumably the albumin escaping from the ruptured egg; Fig. 1b). The remaining eggs were quickly retrieved and moved to a separate rearing tank to prevent further predation.

Like other troglobitic species of *Eurycea*, *E. rathbuni* is believed to feed on aquatic subterranean invertebrates including crustaceans and snails (Goriki et al. 2012. *In* White and Culver [eds.], Encyclopedia of Caves, pp. 665–676. Elsevier, Waltham). Captive specimens have been reported to accept *Ambystoma* larvae (Uhlenhuth, *op. cit.*), strips of muscle from the tail of crayfish (Norman, *op. cit.*), and *Artemia* shrimp (Maruska, *op. cit.*; RWM et

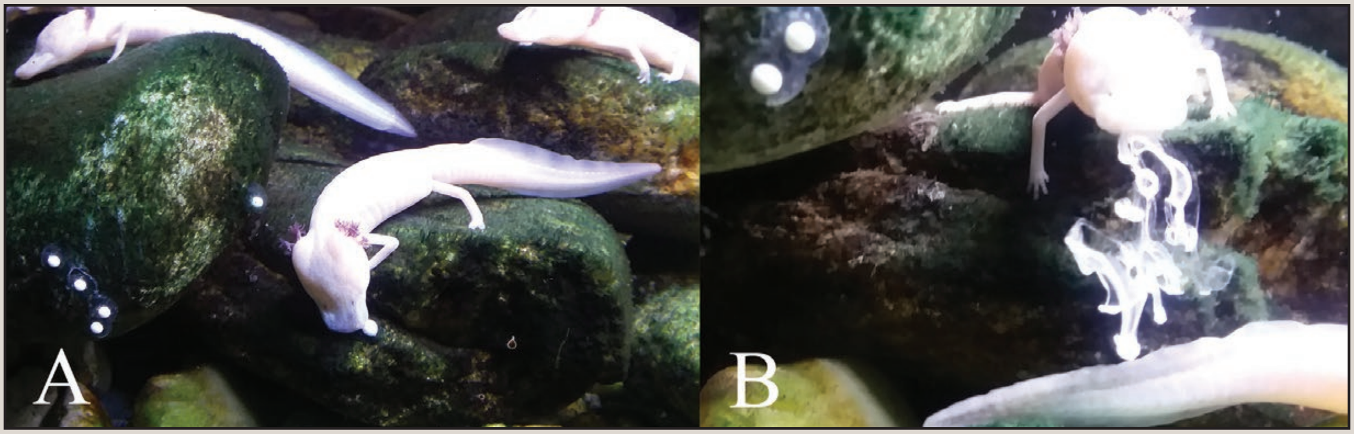

Fig. 1. Oophagy in *Eurycea rathbuni* in a captive setting.

al., unpubl). Oophagy and cannibalism have been documented in several species of plethodontid salamanders (Petranka 1998. Salamanders of the United States and Canada. Smithsonian Institution Press, Washington, D.C. 587 pp.; Chavez. 2017. Herpetol. Rev. 48:791–793) including oophagy in the spring-dwelling *E. sosorum* in captivity (Chavez, *op. cit.*). Given the paucity of available information on the ecology of *E. rathbuni*, it is unclear whether the feeding observations described here for *E. rathbuni* might represent natural elements of its diet and behavioral repertoire or an artifact of captivity. It might not be unreasonable, however, to suspect that *E. rathbuni* might occasionally take such opportunistic food items in nature where prey diversity and availability in aquatic subterranean environments might be limited.

These observations also have important implications for the management and breeding of *E. rathbuni* in captivity. As a vulnerable species with an extremely restricted range that has seen dramatic population declines over the past several decades (Hammerson and Chippendale, *op. cit.*), future conservation measures for *E. rathbuni* may rely on captive-breeding and repatriation as a way to bolster or restore diminished wild populations. If oophagy and cannibalism of larvae by adults pose a significant threat to captive breeding efforts, such behavior may dictate how individuals are grouped and housed to maximize egg and larval survivorship and reproductive success.

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TESTUDINES — TURTLES

***CUORA BOURRETI* (Bourret's Box Turtle). BRUMATION, OVIPOSITION AND INCUBATION.** *Cuora bourreti* is listed as critically endangered (McCormack and Stuart 2016. The IUCN Red List of Threatened Species 2016: e.T163447A115303472; accessed 14 Jul 2018) and populations continue to experience declines in the wild. Recommendations to develop globally integrated captive breeding colonies have been prioritized as a conservation measure for *C. bourreti* (Horne et al. [compilers] 2012. Conservation of Asian Tortoises and Freshwater Turtles: Setting Priorities for the Next Ten Years. Wildlife Conservation Society Singapore Ltd, Singapore. 28 pp.). In North America, the captive zoo population of *C. bourreti* is managed by a Species Survival Plan (SSP) of the Association of Zoos and Aquariums (AZA), with a current population of 83 individuals held in both zoological parks and private collections.

The Smithsonian's National Zoological Park (NZP) maintains a breeding group of *C. bourreti* comprised of two males and one female. All three animals are wild-caught in origin and have been in captivity for over ten years. NZP acquired the female in late 2012, one male in 2013, and the second male in 2014. The *C. bourreti* at NZP are cycled annually by providing seasonal changes in diet, photoperiod, humidity and temperature. During the months of June, July and August, the animals are provided with fourteen hours of daylight, fed three times a week, and misted daily. These parameters are then gradually reduced to eight hours of daylight, mistings once a week, and no feedings during the winter months of December, January, and February. Captives were initially cooled indoors from an ambient temperature of 28° to 18°C in the winters of 2013 and 2014. In the winters of 2015 and 2016, they were brumated in an outdoor greenhouse and cooled below the goal temperature of 10°C, reaching minimum temperatures of 4°C and 7°C, respectively. During brumation, animals were housed individually in 113.5-liter aquaria with large, shallow water bowls (ca. 50 cm in diameter and 5 cm deep), and a PVC hide buried in deep soil and leaf litter substrate for refugia. From March through October, the animals were housed individually in large enclosures (males: 105.4 × 28.5 × 63.5 cm; female: 165.1 × 88.9 × 30.5 cm) with peat moss and soil substrate. Each enclosure had a basking spot providing an 8–10°C gradient within the enclosure, multiple water features, leaf litter, and several refugia. The female was introduced into both male enclosures for breeding two to four times a week during this period.

Nesting occurred as early as February and the female laid two to three clutches of one to two eggs annually (Table 1). The female excavates a shallow ca. 2-cm deep divot in the substrate for oviposition and covers the eggs with leaf litter using her rear limbs. Although this female laid eggs in 2013, hatchlings were not produced from this breeding group until 2017. In 2013, 2014, and 2015, eggs never showed banding or other signs of development. They were incubated in the following methods: inside a small plastic container (ca. 21 × 15 × 8 cm) where the eggs were partially buried in a mixture of moist vermiculite at a 6:5 ratio to water by weight; and in a small (ca. 20 × 17 × 12 cm) suspended incubation container (S.I.M. containers; Squamata Concepts®, Staten Island, New York, USA) with the eggs suspended over saturated vermiculite. In 2016, three eggs banded, but two embryos died within the first month and the third died after 82 days of incubation. The third embryo appeared fully developed and a cause of death was not determined. The successful development of eggs in 2016 and 2017 might be the result of lower brumation temperatures experienced during those winters than in previous years.

In 2017, eggs were incubated using three different methods. One egg from the first clutch, laid in March, was incubated in a large (ca. 33 × 23 × 15 cm) suspended incubation container (S.I.M. Containers; Squamata Concepts, *opt. cit.)* suspended over saturated vermiculite. The second egg from this clutch was incubated in a small plastic container (ca. 21 × 15 × 8 cm) and partially buried in a mixture of moist vermiculite at a 6:5 ratio to water by weight. A second clutch of two eggs laid in April was incubated in the substrate taken from the enclosure where they were laid (peat and soil mixture) inside a small plastic container (ca. 21 × 15 × 8 cm).

Relatively little has been documented on the reproduction of turtles in the genus *Cuora.* The first reported hatchings of *C. bourreti* occurred in 1998 and 1999 and documented eggs (N = 5) from three different females averaging 5.54 × 3.0 cm and incubating for 85–117 days at 24–30°C (Fiebig and Lehr 2000. Salamandra 36:147–156). Here, we document a single female *C. bourreti* producing 19 eggs between 2013 and 2017, including five eggs produced in three different clutches in 2015 (Table 1). Although two other *Cuora* species have been reported to triple-clutch: *C. flavomarginata* (Connor and Wheeler 1998. Tortuga Gazette 34:1–7) and *C. amboinensis* (Ernst et al. 2000. World Biodiversity Database, CD-ROM Series. Springer-Verlag, Berlin), to our knowledge this is the first record of *C. bourreti* triple clutching in a single breeding season. Published accounts on egg production in *C. bourreti* note one or two clutches comprised of one to four eggs annually (McCormack et al.

TABLE 1. Egg and hatchling data from a single female *Cuora bourreti* housed at the Smithsonian's National Zoo over a four–year period.

Year	Date	Weight (g)	Length (cm)	Width (cm)	Egg banding?	Incubation temperature (°C)	Results	Hatchling weight (g)
2017	21 March	37.6	6.59	3.17	Y	26–27	Hatched: 83 days incubation	24.4
	21 March	34.5	6.44	3.09	Y	26–27	Hatched: 83 days incubation	22.7
	29 April	31.51	5.73	3.09	Y	27	Hatched: 88 days incubation	23.7
	29 April	32.13	5.68	3.08	Y	27	Hatched: 89 days incubation	19.5
2016	20 April	33.4	6.08	3.06	Y	28–29	Died early in development	–
	20 April	31.21	5.65	3.02	Y	28–29	Died late in development– 82 days incubation	–
	11 July	30.62	6.03	3.06	Y	28–29	Died early in development	–
2015	17 April	40.45	6.51	3.18	N	28–29	Never developed	–
	19 May	35.78	3.16	5.88	N	28–29	No development	–
	19 May	35.47	3.19	5.86	N	28–29	No development	–
	26 June	32.12	5.85	3.11	N	28–29	No development	–
	26 June	30.86	5.6	3.04	N	28–29	No development	–
2014	26 April	33.7	3.17	6.28	N	28–29	No development	–
	16 June	32.9	5.88	3.11	N	28–29	No development	–
	16 June	33.5	6.07	3.05	N	28–29	No development	–
2013	20 May	30	5.75	3.07	N	28–29	No development	–
	19 June	29.2	5.74	3.08	N	28–29	No development	–
	19 June	28.8	5.51	3.05	N	28–29	No development	–
	23 August	27.6	5.77	2.81	N	28–29	No development	–

2010. Asian Turtle Program [ATP], Hanoi, Vietnam 35; Fiebig and Lehr 2000, *op. cit.*; Heuberger and Heuberger 2008. Reptilia 61:20–26).

Despite efforts to protect wild *C. bourreti*, populations are continuing to decline. An increased understanding of the reproductive biology of species in the genus *Cuora* is needed to develop and refine conservation efforts through captive propagation. The hatching of the four *C. bourreti* at NZP represents an important step towards the development of a sustainable captive breeding population in North America.

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***CUORA FLAVOMARGINATA* (Yellow-margined Box Turtle). LONGEVITY AND MACROCEPHALY.** *Cuora flavomarginata* is a small (to 190 mm carapace length) geoemydid turtle native to portions of southeastern mainland China, Taiwan, and the southern Ryuku Islands of Japan (Ota et al. 2009. *In* Rhodin et al. [eds.], Conservation Biology of Freshwater Turtles and Tortoises: A Compilation Project of the IUCN/SSC Tortoise and Freshwater Turtle Specialist Group. Chelonian Research Monographs 5:035.1–035.10). This turtle is considered Endangered, and is probably Critically Endangered in the mainland portion of its range (Asian Turtle Trade Working Group 2000. [errata version published in 2016]. The IUCN Red List of Threatened Species 2000: http://dx.doi.org/10.2305/IUCN.UK.2000.RLTS.T5960A11965283.en; accessed 5 June 2018). The potential longevity of *C. flavomarginata* is poorly known; a commonly published maximum known lifespan is 19 years, 1 month for a captive female (Snider and Bowler 1992. Longevity of Reptiles and Amphibians in North American Collections, 2nd Ed. SSAR. 40 pp.). This note documents a considerably longer lifespan for two captive individuals.

FIG. 1. A) Male *Cuora flavomarginata*, obtained in 1974, displaying macrocephaly. B) Close-up image of head.

Three presumably wild-caught *Cuora flavomarginata,* two males and one female, were obtained in 1974 from a commercial source. These specimens appeared to be fully mature at this time, though no initial measurements are available. This group produced numerous fertile eggs and healthy hatchlings each year, until the female died accidentally several years ago. The two males are still alive and healthy at this writing (June 2018). The age of sexual maturation in this species has been estimated as six or seven years for wild individuals (Ota et al., *op. cit.*); thus a conservative estimate of age for the two males discussed here would be 50 years. Given that these turtles appeared to be fully mature when obtained, they could be several years older than this estimate.

One of the two living male *C. flavomarginata* reported here has developed very noticeable macrocephaly (Fig. 1), with the postocular dorsal musculature greatly hypertrophied. Although this species is considered relatively "big-headed" within its genus, and males are noted to have broader heads than females (Ota et al., *op. cit.*), true macrocephaly has apparently not been described in this species. Based on observations of numerous captive specimens and on published photographs of others (e.g., Vetter and van Dijk 2006. Terralog: Turtles of the World Vol. 4: East and South Asia. Edition Chimaira, Frankfurt am Main. 160 pp.), the male specimen described here appears to display exceptional head development.

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RHINOCLEMMYS PUNCTULARIA **(Spot-legged Turtle). DOUBLE-CLUTCHING.** The reproductive biology of *Rhinoclemmys punctularia* is not well known. It has been reported that females deposit one or two eggs per clutch in March and April (Vogt 2008. Amazon Turtles. Gráfica Biblos, Lima. 104 pp.), and nesting was also recently reported in a captive specimen in July (Soares et al. 2017. Herpetol. Rev. 48:85–86). Other species of this genus have varied reproductive strategies. Because of the lack of specific published data on how many times per year species of *Rhinoclemmys* nest, it is important to document cases of individual turtles nesting in a single season so that more accurate estimates of annual reproductive potential for these species can be calculated (Páez 2012. *In* Páez et al. [eds.], Biologia y Conservacion de las Tortugas Continentales de Colombia, pp. 189–203. Serie Editorial Recursos Hidrobiologicos y Pesqueros Continentales de Colombia,. Instituto de Investigacion de Recursos Biologicos Alexander von Humboldt, Bogotá).

In Colombia, *Rhinoclemmys annulata, R. diademata, R. melanosterna,* and *R. nasuta* nest year-round (Páez, *op. cit.*). However, with the exception of *R. diademata,* which nests every two months and has an estimated reproductive potential of 6–18 eggs per year, it is unclear how many times per year individual females of these species will nest. *Rhinoclemmys nasuta* has been reported to nest twice a year, while *R. funeria* and *R. pulcherrima* nest up to four times a year, and *R. puncutlaria* nests year-round (Bonin et al. 2006. Turtles of the World. Johns Hopkins University Press, Baltimore, Maryland. 416 pp.); however, no citations were provided for these data. In Mexico, Legler and Vogt (2013. Turtles of Mexico: Land and Freshwater Land Forms. University of California Press, Berkeley, California. 416 pp.) noted that *R. areolata* lays up to five clutches annually and *R. pulcherrima* up to four clutches per year, whereas the number of clutches laid by *R. rubida* is unknown.

A captive female *R. punctularia* maintained at the Centro de Estudos de Quelônios da Amazônia – CEQUA, of the Instituto Nacional de Pesquisas da Amazônia, Manaus, Amazonas, Brazil, was observed nesting on 8 July 2016 at 1430 h and then again at 0930 h on 22 August 2016, representing an inter-nesting interval of 45 days. The first clutch consisted of one egg (38 g; 71 × 31 mm) (Soares et al. *op. cit.*); the second clutch was comprised of two eggs measuring 66 × 28 mm and 42 g, and 60 × 26 mm and 36 g, respectively. The female, which measured 235 mm in straight line carapace length and 2085 g after oviposition, is part of a colony of six males and eight females originally collected from the Manaus area of Brazil that have been maintained in captivity for 22 months. The indoor terrarium housing the colony measures 10 × 10 m, features six ponds each measuring 1 m in diameter and 20 cm deep, and is maintained on natural light and temperature cycles, with two 2 × 4 m windows allowing natural light to enter the structure. One nest was constructed adjacent to a palm tree trunk and the second nest at the base of a small palm tree, both in silica sand. The nesting female was positively identified during both nesting events by its PIT tag number. This is the first record of double-clutching from direct nesting observations of captive *R. punctularia.*

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SQUAMATA — LIZARDS

ANOLIS GARMANI **(Jamaican Giant Anole). HERMAPHRODITISM.** Hermaphroditism, a condition in which both male and female reproductive organs are expressed in an individual, is rare among higher vertebrates. Some cases have been reported in the Reptilia (e.g., Risley 1941. J. Morphol. 68:101–121), and in lizards, documented cases appear to be limited to lacertids: *Lacerta viridis* (Tayler 1918. Proc. Zool. Soc. Lond. 88:223–230), *Darevskia saxicola* (Lantz 1923. Bull. Soc. Zool. France 48:289–290; Darevsky 1966. J. Ohio Herpetol. Soc. 1966:115–152); anguids: *Hyalosaurus koellikeri* (Bons and Bons 1969. Compt. Rend. l'Acad. Sci 268:695–696); varanids: *Varanus exanthematicus* (Frye et al. 1999. Proc. Assoc. Reptil. Amphib. Vet. 1999:59–62), *V. acanthurus* (Brown 2008. Biawak 2:87–88); and phrynosomatids: *Sceloporus occidentalis* (Goldberg 1989. Copeia 1989:486–488).

In November 2015, a captive-bred adult *Anolis garmani* maintained at Audubon Zoo was presented to veterinary staff due to lethargy, inappetence, and overall decline in health, and subsequently euthanized due to concerns over quality of life. Upon gross necropsy and histopathological analysis, this animal, a suspected female, was determined to be hermaphroditic with both male and female reproductive organs that showed signs of active folliculogenesis and spermatogenesis.

Upon its arrival in mid-2010, this animal was originally housed together with an adult male *A. garmani.* Together, this pair produced more than 30 eggs (most of which were non-viable) over the course of one year, resulting in at least two live offspring and several mid-term and full-term embryos that died during incubation. From mid-2011 onward, this animal was periodically transferred between enclosures housing additional *A. garmani* of both sexes. Although eggs continued to be laid in these enclosures over the next several years, it was unclear which animals produced them. Eggs were also received when this hermaphroditic individual was housed with up to seven female *A. garmani*; however, since no attempts were made to incubate these eggs, it is unclear whether this individual was also capable of successfully reproducing in a male capacity. Post-mortem histopathological analysis of the animal noted some follicular degeneration with yolk resorption, suggesting full female reproductive capabilities at the time of death.

As far as it can be determined, this represents the first documented case of hermaphroditism in a dactyloid iguanian. This case highlights the importance of necropsy examinations and post-mortem histopathological analyses for detecting internal anatomical and developmental aberrations that would otherwise go unnoticed in living specimens.

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***LIOLAEMUS CHALTIN*. PREFERRED AND CRITICAL THERMAL TEMPERATURES.** *Liolaemus chaltin* is distributed in northwestern Argentina and southern Bolivia between 3400–3750 m elev. in the Puna ecoregion (Lobo and Espinoza 2004. Copeia 2004:850–867; Quinteros 2012. Herpetologica 68:100–120). Lobo and Espinoza (*op. cit.*) report that these lizards seek refuge in grasses (*Festuca* sp.) and thorny shrubs (*Adesmia* sp.), feed on arthropods, and lay 4–5 eggs from September to November. The average field body temperature (T_b) recorded by these authors in January 1995 and 1996, between 1000 and 1800 h was 31.3 ± 2.7 °C (range 25.3–36.9; N = 40). This note aims to provide additional information on the thermal biology of *L. chaltin.*

Five *L. chaltin* (4 adult males and 1 juvenile female) were collected 30 km N of Villazón, Potosí, Bolivia (21.83°S, 65.62°W, WGS 84; 3551 m elev.) on 5 October 2015. Lizards were transported in cloth bags to the laboratory of the Colección Boliviana de Fauna (CBF) in La Paz, Bolivia. They were kept in a terrarium measuring 60 × 30 × 35 cm (length × width × height) and acclimatized two-weeks prior to laboratory thermal data collection. The terrarium was set in direct sun as a heat source and lizards had access to places to hide and bask. They were fed crickets and mealworms three times a week, and had free access to water. Individuals were held in captivity for two months before being deposited as voucher specimens (CBF 4301–4305). No specimens died and no injuries were observed during or after laboratory tests. Individual snout-vent lengths (SVL) were measured with a vernier caliper (0.05 mm), and weights (W) were taken with a CM 60-2N Kern Electronic Pocket Balance (± 0.01 g) before starting data collection.

Preferred temperature (T_{pref}) is an estimate of the optimal temperature a lizard would attain without biotic and abiotic constraints, generally measured in a thermal gradient (Hertz et al. 1993. Am. Nat. 142:796–818). A thermal gradient was created in a glass terrarium (120 × 40 × 40 cm) that was divided internally into four 10-cm wide lanes separated by 10-cm high opaque walls. A 100-watt incandescent bulb was placed on the same end of each lane, registering substrate temperatures from 15 to 80°C. A 42-watt fluorescent bulb was suspended in the middle of the terrarium to standardize ambient lighting. Light bulbs were turned on at 0730 h, and specimens were placed in the middle of the thermal gradient at 0800 h. Preferred temperatures of individuals were measured every hour from 0900 to 1700 h for two days, obtaining 18 records per individual. To determine body temperatures, the tip of the thermocouple connected to a digital thermometer (Fluke 52-II, Everett, Washington, USA) was inserted up to 0.5 cm into the cloaca of each individual. For calculating set point preferred temperatures, central 50% interquartile observations were considered (sensu Hertz et al., *op. cit.*).

The thermal limits at which physiological functions of lizards do not operate correctly are given by critical thermal temperatures (Cowles and Bogert 1944. Bull. Am. Mus. Nat. Hist. 83:265–296; Angilletta et al. 2002. J. Therm. Biol. 27:249–268). Measurements for critical thermal data were performed three weeks after T_{pref} data collection. Critical thermal minimum (CT_{Min}) was taken exposing individuals to a 10 × 10 × 5 cm empty plastic container within a 40 × 30 × 20 cm plastic container filled with ice. Experimental voluntary maximum temperature (T_{vol}), panting temperature (T_{pant}) and critical thermal maximum (CT_{Max}) were taken simultaneously under a 100-watts bulb placed 10 cm above the individuals. After critical thermal records, lizards were placed immediately in another 10 × 10 × 5 cm plastic container at room

TABLE 1. Voucher number code of the Colección Boliviana de Fauna (CBF), sex, snout–vent length (SVL), weight (W), individual preferred temperature (T_{pref} mean ± standard deviation, and range in parenthesis), lower and upper set-point preferred temperature values, critical thermal minimum (CT_{Min}), experimental voluntary maximum temperature (T_{vol}), panting temperature (T_{pant}), critical thermal maximum (CT_{Max}) and thermal tolerance range (TTR) for *Liolaemus chaltin* from Potosí, Bolivia.

CBF	Sex	SVL (mm)	W (g)	T_{pref} mean range (°C)	Lower set point (°C)	Upper set point (°C)	CT_{Min} (°C)	T_{vol} (°C)	T_{pant} (°C)	CT_{Max} (°C)	TTR
4301	♀	36.5	1.15	34.98 ± 0.73 (32.20–37.70)	33.73	35.78	6.30	38.90	39.20	43.50	37.20
4302	♂	52.9	4.22	36.13 ± 1.00 (33.50–37.30)	35.95	36.80	4.80	37.70	41.50	43.60	38.80
4303	♂	53.2	3.91	35.16 ± 1.38 (33.00–36.60)	34.70	35.88	5.90	37.60	40.60	43.50	37.60
4304	♂	51.3	3.65	36.23 ± 1.61 (35.20–37.60)	35.65	36.65	5.40	38.60	40.30	42.50	37.10
4305	♂	52.8	3.03	34.66 ± 1.11 (32.10–38.20)	33.40	35.93	6.30	38.50	40.90	43.10	36.80

temperature (20°C), to warm them in the case of CT_{Min} test, or to cool them in the case of CT_{Max} test. Thermal tolerance range (TTR) was estimated as CT_{Max} - CT_{Min}, according to Moreno-Azócar et al. (2013. Oecologia 171:773–788).

Results are shown in Table 1. Mean SVL was 49.30 mm (± 0.72, range: 36.50–53.20 mm), and mean weight (W) was 3.10 g (± 1.25, range: 1.15–4.22 g). Mean T_{pref} was 35.38°C (± 1.38, range: 32.10–38.20°C). The 25% and 75% interquartiles of T_{pref} were 34.69°C and 36.21°C, respectively. Mean value for CT_{Min} was 5.70°C (± 0.64), for T_{vol} was 38.30°C (± 0.58), for T_{pant} was 40.50°C (± 0.85) and for CT_{Max} was 42.77°C (± 1.88). Mean thermal tolerance range (TTR) was 37.50°C (± 0.78).

Our T_{pref} was 4°C lower than the T_b of *L. chaltin*, obtained by Lobo and Espinoza (*op. cit.*); this might suggest that the accuracy of thermoregulation (d_b) could be constrained in the wild. Field body temperature in the *L. alticolor-bibronii* group (Abdala 2015. South Amer. J. Herpetol. 10:104–115) is generally less than reported T_{pref} (see Labra et al. 2009. Am. Nat. 174:204–220; Medina et al. 2012. J. Therm. Biol. 37:579–586; Valdecantos et al. 2013. J. Therm. Biol. 38:126–134), settling the suggestion that higher altitude thermal environments present in Puna habitats keep *Liolaemus* species from reaching their T_{pref} (e.g., Marquet et al. Oecologia 81:16–20); although cases with high accuracy of T_b are reported, as in *L. yanalcu*, a related viviparous species (d_b= 1.16±1.46; Valdecantos et al. 2013, *op. cit.*). On the other hand, reported T_{pref}, CT_{min}, T_{vol}, T_{pant}, CT_{max} and TTR values for *L. chaltin* fall within known records for several species of *Liolaemus* (see Carothers et al. 1997. Rev. Chil. Hist. Nat. 70:297–309; Labra et al. 2009, *op. cit.*; Bonino et al. 2011. J. Exp. Zool. 315:495–503; Moreno-Azócar et al. 2013, *op. cit.*; Cruz et al. 2014. Ecosistemas 23:37–45; Bonino et al. 2015. Zoology 118:281–290).

Obtained physiological temperatures can give us information to forecast to what extent thermo-environmental deviations caused by habitat modification and climate change could affect species (Clusella-Trullas and Chown 2014. J. Therm. Biol. 184:5–21), but future research must also broaden the information available on species thermoregulatory efficiency in the field.

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SCELOPORUS TORQUATUS **(Torquate Lizard). SELECTED BODY TEMPERATURE.** Selected body temperatures under laboratory conditions have become one of the most important traits studied in the thermal biology of ectotherms (Sinervo et al. 2010. Science 328:894–899). Selected temperatures represent the range of core temperatures within which ectotherms are comfortable while performing their behaviors (IUPS Thermal Commission 2003. J. Therm. Biol. 28:75–106). The lizard *Sceloporus torquatus* is endemic to central Mexico, where it lives in rocky areas. Several reports on this species' biology have been published, describing its hematological traits (González-Morales et al. 2015. Can. J. Zool. 93:377–388), muscle contractions (Quintana et al. 2014. Acta Zool. 95:264–271), and reproduction (Guillete and Méndez-de la Cruz. J. Herpetol. 27:168–174). However, the selected body temperature of *S. torquatus* has not yet been documented. Here, we describe the thermal preferences of *S. torquatus* under laboratory conditions.

During September 2016, we captured 17 adult *S. torquatus* by noose or hand (> 87 mm SVL: 7 males and 10 females) in Texcalyacac, State of Mexico (19.12694°N, 99.49472°W; 2500 m elev.). The capture site was a rocky area dominated by pine (*Pinus* spp.).

In the laboratory, the lizards were separated by sex, maintained at 25°C in plastic containers with soil and stones, and hydrated by periodically spraying the cages with water. Laboratory experiments were conducted two days after the lizards were captured. We used a thermal gradient made from a polycarbonate box (150 × 150 × 70 cm [length x width x height]) divided into ten tracks each measuring 15 cm in length, to prevent interactions between the lizards. The box was located in a room with a controlled temperature of 20°C. Six 100-W lamps distributed along the box generated a thermal gradient ranging from 20–50°C. Body temperature data was collected manually using a digital thermometer with a probe inserted into the lizard's cloaca every two hours between 0900 and 1930 h. We used a Mann-Whitney U test to compare selected body temperatures between the sexes.

Males and females did not differ significantly in selected temperature (U = 1175.00, p = 0.66) or measurement of temperature (H_5 = 7.63, p = 0.178). Mean preferred body temperature among all lizards measured was 33.21°C (SD = 2.82, range = 22.40–33.70°C). The 25% and 75% quartiles were 31.95°C and 35.20°C, respectively. These results suggest that *S. torquatus* can be considered a eurythermic species, due to its wide range of preferred temperatures. These data can be used as a reference for future studies that evaluate traits in this species that may depend on temperature, such as color measurements and performance.

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SQUAMATA — SNAKES

LYCODON JARA **(Twin Spotted Wolf Snake). FEEDING.** *Lycodon jara* is a small snake found in varied habitats including forests, open areas with bushes and scattered trees, and agricultural areas (Das 2002. A Photographic Guide to Snakes and Other Reptiles of India. New Holland Publishers UK Ltd, Garfield. 38 pp.). It is distributed in northeastern India, Bangladesh, Nepal, Bhutan, and Myanmar (Chaudhuri et al. 2015. Hamadryad 37:95–103). Little is known about the natural history and behavior of this snake, although it has been reported to feed on geckos (Whitaker and Captain 2004. Snakes of India. Macmillan India Limited, New Delhi. 196 pp.).

On 23 June 2012 at 2130 h, we came across a *L. jara*, (snout–vent length = 31.75 cm; tail length = 7.62 cm) in a cowshed in Nadia District, West Bengal, India (22.96503°N, 88.52388°E, WGS 84;

Fig. 1. *Lycodon jara* feeding on *Lygosoma albopunctata.*

16 m elev.). It was collected and housed indoors for observations in a small plastic container measuring 24 × 18 × 10 cm (L × W × H) with ambient fluorescent room lighting. Thirty minutes after the snake's capture, it was offered a House Gecko (*Hemidactylus flaviviridis*) measuring approximately 7 cm in total length (TL). The gecko was kept with the snake in the container for the next two hours, but the snake showed no interest in feeding. At 0030 h a skink (*Lygosoma albopunctata*) measuring ca. 9 cm TL was introduced to the container, replacing the gecko. The snake immediately seized the skink and held on to its head while throwing a loose coil around its prey to get a better grip. It swallowed the skink within 5 min. Four days later, we introduced another *H. flaviviridis* measuring ca 6 cm TL and replaced it after one hour with another gecko species (*H. frenatus*) that measured ca. 5 cm TL. In both cases the snake showed no interest in feeding on the geckos. Thirty minutes after introducing the second gecko, it was replaced by another similarly sized *L. albopunctata*, as before. As soon as the skink was introduced, the *L. jara* showed the same predatory behavior and swallowed the skink within approximately 5 min. The snake was released back in the same habitat where it was found the following evening.

From these observations of a recently captured individual, it appears that *L. jara* may prefer skinks over geckos and may have a skink-specific dietary interest. Further investigations into the dietary habits of this species are needed to understand its feeding habits.

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CONSERVATION

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The Lessons of History and the Future of American Crocodile Conservation in Belize

The American Crocodile *(Crocodylus acutus*) is the most widely distributed crocodilian in the New World, inhabiting coastal and lowland wetlands from southern Florida, USA to the limits of mangrove forest in Peru (Ernst et al. 1999; Platt and Thorbjarnarson 2000a; Fig. 1). In 1973, the American Crocodile was listed as endangered by the International Union for the Conservation of Nature (IUCN), and listed under Appendix I by the Convention of International Trade in Endangered Species (CITES) because of overexploitation, deliberate killing, illegal hunting, habitat loss, and drowning in fishing nets (Thorbjarnarson 1989; Thorbjarnarson et al. 2006). These threats led to a 30% decline in many wild populations of *C. acutus* in the 75 years prior to the establishment of the IUCN, CITES, and national protection laws (Thorbjarnarson et al. 1992). With the implementation of new national laws, international laws, and management strategies over the last 45 years, a few populations have recovered, thus the down-listing of *C. acutus* as Vulnerable by the IUCN (IUCN 2012). Despite population recovery in some areas, American Crocodile abundance and density throughout its historical range remain low and some populations remain at risk of extirpation, requiring continued conservation and evaluation of management strategies (Thorbjarnarson et al. 2006). This holds specific relevance for populations in Belize (Fig. 2), a past stronghold of the American Crocodile (Ross 1998; Platt and Thorbjarnarson 2000b).

Belize is a small country (22,791 km^2) nestled south of Mexico on the Caribbean side of Central America, rich in marine and terrestrial biodiversity; a likely reflection of the diverse marine and terrestrial biomes that include the Belize Barrier Reef and Mesoamerican Biological Corridor. Belize is also home to a melting pot of various cultures (i.e., Maya, Creole, Garifuna, East Indian, Mestizo, East Asian, Mennonite and Central American), each with its own unique connection to the native flora and fauna. A preliminary ethnozoological study investigating the relationship between wildlife (particularly crocodiles) and the residents of Belize (which include recent immigrants from North America) illustrates diverse perceptions and relationships among the different ethnic groups towards the archaic predator (Tellez, pers. obs.). Understanding the diverse views, myths, and misguided information people have of crocodiles is essential in moving forward with the development of an effective management and educational program as conservation is not necessarily just about wildlife, it is about people. The efforts and actions under the umbrella of conservation will rarely be successful if there is a lack of support by the local people who interact with the focal species daily. Currently, there is no management program for the American Crocodile in Belize, and any actions towards its conservation and future management are still in its infancy. As key stakeholders move forward in the decision-making process to protect the American Crocodile, it is pertinent to consider current human-crocodile relations while simultaneously proactively educating the facts to extinguish any false beliefs to further the support of crocodile conservation, or at least establish a tolerance for co-existence.

To further understand human-crocodile relations, and the threats and possible future population trends of the American Crocodile of Belize, an exploration of historical data as well as identifying cultural perspectives and relationships could be advantageous in the creation of a successful conservation program. Here, we provide a brief, yet detailed, review of the historical population trends of the American Crocodile in Belize, and

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FIG. 1. *Crocodylus acutus* (American Crocodile) in Belize.

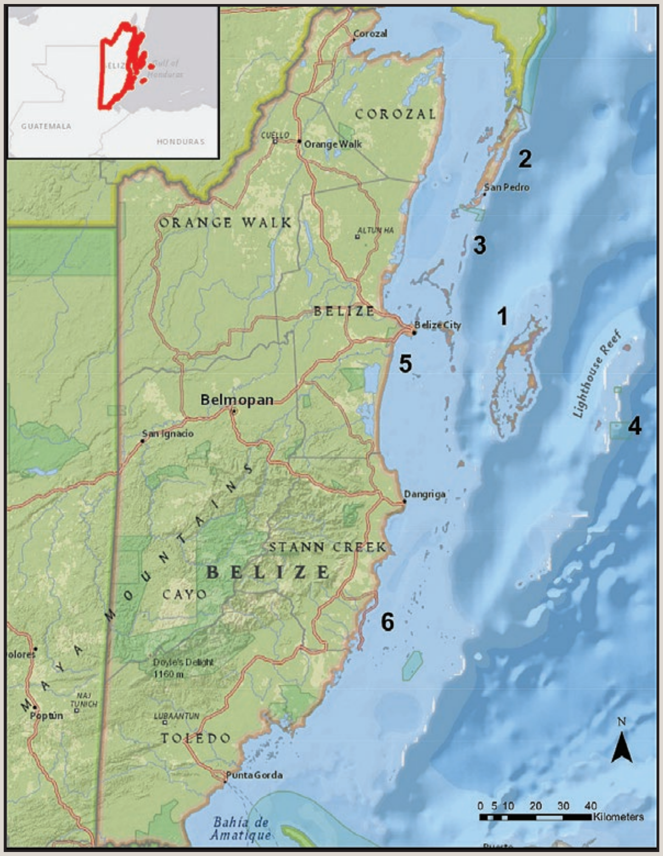

FIG 2. Map of Belize with numbers demarking approximate locations for which survey data was found. Turneffe Atoll (1), Ambergris Caye and Bacalar Chico (2), Caye Caulker (3), Lighthouse Reef (4), mainland Belize (5), other offshore cayes and atolls (6).

conclude with a discussion of current and future conservation efforts by key stakeholders, which include the creation of a crocodile conservation and management program, and community outreach programs that endorse active community participation in crocodile conservation and their habitat.

METHODS

Literature review.— Prior to addressing collected data, several considerations merit review. Recorded surveys are sparse for Belize and contiguous data is unavailable for most sites with the exception of Ambergris Caye and Turneffe Atoll. Additionally, credible data of antiquity are nearly non-existent and consist primarily of anecdotal observations and reports.

We collected material for review by conducting rigorous database searches, including Science Direct, Wiley Online Library, and BioOne, using the terms "population; status; survey of American Crocodiles; *C. acutus*; *Crocodylus acutus*; Belize/British Honduras" as search criteria. We retrieved further documentation from publicly sourced newsletters and proceedings of the IUCN/Species Survival Commission-Crocodile Specialist Group (CSG), and by direct request from the journal *Oryx* and from the InterLibrary Loan Internet Accessible Database (ILLiad) of West Virginia University. Following collection of relevant materials we extracted quantitative data and collated nest and population survey results into a collective spreadsheet. We recorded metadata for each entry to include; survey year, location, site, kilometers surveyed, nest counts, total crocodiles encountered, and crocodile encounter rate. Our data is not structured appropriately for statistical analyses as available data was not sufficient, and the purpose of our report is to provide a review of past work. Rather, our results are presented in discussion and as figures or tables for collected data.

RESULTS

History of Crocodylus acutus in Belize.—"Alligators of large size infest most of the inland lagoons, and provide good sport at the out-stations" (Morris 1883). The earliest accounts of crocodiles in Belize suggest a relatively abundant population of both species, with few confirmed records of *C. acutus* on the mainland (Schmidt 1924). American Crocodile populations still appeared healthy and abundant in the late 1930s despite the rise of crocodile hunting in the 1920s, a response to the fashion industry demand for crocodile products (Sanderson 1941). By the late 1950s, indiscriminate hunting of the last 30 years led to near extirpation of the species (Wright et al. 1959; King et al. 1980; Ross 1998; Platt and Thorbjarnarson 2000a). The rarity of *C. acutus* sightings along the coast mirrored the decline of the once thriving populations of crocodiles amongst the cayes, now devoid of the aquatic predator (Charnock-Wilson 1970; Powell 1971; King et al. 1980; Abercrombie et al. 1982). This scenario provides a glimpse of the possible pivotal role of caye populations as the source population of American Crocodiles in the region.

Forty years of intense commercial harvesting and trophy hunting of *C. acutus* (particularly among the cayes) decimated the American Crocodile population in Belize to near extirpation by the late 1970s (Powell 1971; King et al. 1980). Interestingly, description of crocodiles observed during a 1978 study in saltwater mangrove lagoons, swampy creeks or sandy beaches describe animals that did not adhere implicitly to the standard morphometric characteristics of *C. moreletii* or *C. acutus* (Abercrombie et al. 1982). Given our current observations of crocodiles along the coast of southern Belize, perhaps the lack of *C. acutus* sightings is a glimpse of evidence of hybridization between the two species (Tellez, pers. obs.). Identifying *C. moreletii* x *C. acutus* hybrids in the wild can be troublesome, and misidentification of an individual is probable (Sanchez-Hererra et al. 2012; Pacheco-Sierra et al. 2016; Tellez pers. obs.). Although hybridization may be a common phenomenon between the two species (Hekkala et al. 2015; Pacheco-Sierra et al. 2016), perhaps the 1978 population survey reflects a relatively recent rise of hybridization as parental species (particularly *C. acutus*) became scarce due to past overexploitation.

In 1981, Belize gained its independence from the United Kingdom, and created the Wildlife Protection Act (WPA), which included strict legislation against hunting and trade of wildlife products. Although crocodiles finally became legally protected under this national mandate, officially banning commercial hunting of crocodiles in Belize, opportunistic killing continued due to a lack of tolerance and fear (Aguilar, pers. obs.). Some members of the older generation in Belize contribute the lack of involvement in wildlife management and decisions pre-Independence as a principal factor causing the lack of tolerance or co-existence with wildlife (Manglar, pers. obs.), particularly with crocodiles. Despite the fear and lack of tolerance, the protection of American Crocodiles under the WPA Chapter 220 likely initiated the slow recovery of the species on the atolls (Turneffe Atoll, N = 200–300), with few confirmed sightings of the species along the mainland (Perkins and Carr 1985; Meerman

Table 1. Population survey data of American Crocodiles (*Crocodylus acutus*) in Turneffe Atoll, Belize, from 1994–2010. Data from Platt and Thorbjarnarson (1996, 2000a), Platt et al. (2004), Rainwater (2008, 2010), and Rainwater and Platt (2009).

Year	Location	Distance surveyed (km)	Crocodiles spotted	Encounter rate (crocodiles/km)
1994	Turneffe Atoll	94.60	50	0.53
1995	Turneffe Atoll	37.60	17	0.45
1996	Turneffe Atoll	20.80	19	0.91
1997	Turneffe Atoll	41.60	49	1.18
1997	Turneffe Atoll (countrywide)	156.80	152	0.96
2002	Turneffe Atoll	40.10	49	1.22
2008	Turneffe Atoll	91.90	31	0.34
2009	Turneffe Atoll	56.8	33	0.58
2010	Turneffe Atoll	52.2	19	0.34

Table 2. Nest survey data of American Crocodiles (*Crocodylus acutus*) on Turneffe Atoll from 1994–2008. Note that 1995 survey data are incomplete, NA = not available. Data from Platt and Thorbjarnarson (1996), Platt et al. (2004), Rainwater (2008, 2010), and Rainwater and Platt (2009).

Location	1994	1995	1996	1997	2002	2004	2008	2009	2010
Calabash Cay	0	___	0	0	1	2	0	1	5
Blackbird Caye (south)	0	___	5	3	1	3	0	1	2
Blackbird Caye (west)	2	1	1	2	0	0	0	0	0
Northern Caye	8	___	7	10	6	11	2	4	4
Total	10	1	13	15	8	16	2	6	11

1994). However, any achievements made between 1980 and the early 1990s by the implementation of this law are unclear. With the perceived small population of the American Crocodile inhabiting Belize, as well as low numbers throughout its range, the IUCN declared the American Crocodile Endangered in 1986.

By the late 1980s–early 1990s, concern for the conservation status of the American Crocodile throughout its range initiated discussion amongst CSG members for increased population assessments and investigation into the ecology of American Crocodiles for the development of a conservation management plan of the species (Thorbjarnarson et al. 1992). The commencement of American Crocodile population surveys in Belize sparked a crocodile renaissance in-country, creating an increased collection of relevant quantitative survey data from various locations, generating a multitude of data sets. As a result, a few locations began encompassing contiguous years of vital data, such as Turneffe Atoll.

Turneffe Atoll and its surrounding cayes have had the most comprehensive American Crocodile survey data to-date (Table 1). Published surveys commenced in 1994 (0.53 crocodiles per km shoreline surveyed) with encounter rates increasing linearly from 1996, 1997, and 2002, (Platt and Thorbjarnarson 1996; Platt and Thorbjarnarson 2000a; Platt et al. 2004). In 2008, the linear population increase came to a halt with a severe drop in the crocodile encounter rate (0.34 crocodiles per km), mirrored by a decrease nesting activity (Rainwater and Platt 2009). However, crocodile nesting and encounter rates both began to increase again over the next five years (T. Rainwater and S. Platt, pers. comm.). The rise in development on Turneffe Atoll may be a primary factor contributing to the population and nesting declined observed. Even with adequate aquatic habitat, limited nest site availability has the potential to limit the growth of crocodile populations and decrease recruitment (Platt et al. 1998, Platt and Thorbjarnarson 2000b). However further monitoring is required to determine if the decline is legitimately the result of anthropogenic disturbance, natural variation in nesting effort in relation to the small number of adult females (Rainwater and Platt 2009), both.

Ambergris Caye is another area in Belize with somewhat contiguous population survey data since the 1990s. The first official crocodile population survey on Ambergris Caye recorded a total encounter rate of 0.34 crocodiles/km across 96.9 km (Platt and Thorbjarnarson 1997). Almost 15 years later encounter rates increased to 0.71–0.96 crocodiles per km (Chenot-Rose et al. 2011; Chenot-Rose 2013), likely reflecting a decrease in illegal hunting and opportunistic killings in conjunction to a population recovering from past exploitation. Observations from an ongoing project examining immunology and parasitology of American Crocodiles on Ambergris Caye further support the hypothesis of recovery given the identification of 276 individuals across ~177.6 km (1.55 crocodiles/km; Partyka, unpubl. data). However, this recovery might also reflect dispersal of American Crocodiles from Mexico moving south as habitat reaches carrying capacity in Mexico. Thus, even with the ongoing rapid loss of habitat, in addition to a resurgence of opportunistic killings (particularly to sell crocodile products to tourists and illegally sell the meat), the American Crocodile population may continue to grow and remain stable via immigrants from the north. A collaboration between researchers from Mexico and Belize is currently underway to investigate dispersal patterns of American Crocodiles in this area to validate the aforementioned hypothesis. Interestingly, American Crocodiles appear to be seeking refuge from loss of habitat around the caye's sewage

Table 3. Population survey data of American Crocodiles (*Crocodylus acutus*) from the coastal zone of Belize, including the mainland. Data from Platt (1995) and Platt and Thorbjarnarson (1997).

Year	Location	Distance surveyed (km)	Crocodiles spotted	Encounter rate (crocs/km)
1994	Maps Cay	12.9	11	0.85
1997	Maps Caye	21.0	3	0.14
1995	Bacalar Chico	42.7	8	0.19
1997	Frenchman's Caye	12.0	1	0.08
1997	Hick's Caye	13.7	5	0.36
1997	Lighthouse Atoll	5	1	0.20
1997	Long Caye	8.4	4	0.47
1997	Middle Long Caye	27.6	2	0.07
1997	Caye Caulker	14.1	48	3.4
2016	Caye Caulker	22.15	55	2.4
2004	Twin Cayes	___	3	___
1997	North Mainland	206.1	3	0.01
1997	South Mainland	369.5	11	0.02

ponds, as data illustrate an increase in density within this area (Tellez and Boucher, unpubl. data).

Since the nationwide survey of *C. acutus* in 1997, the population of Caye Caulker has been surprisingly ignored (given the size of the population relative to the size of the caye). During the initial survey, Platt and Thorbjarnarson (2000a) reported a relatively high encounter rate compared to other locations in Belize (N = 52, 3.40 crocodiles per km), yet, a population still in a state of recovery from past over-exploitation. Almost 20 years later, a recent survey describes the Caye Caulker population as relatively stable (N = 55, 2.4 crocodiles per km) (Tellez et al. 2016). However, a rapid rise of habitat destruction in 2017 in response to increase residential and hotel development in conjunction to an increase illegal trade of crocodile products have exponentially increased human–crocodile conflict, resulting in a rise of crocodile deaths, thus the stability of this population is in question (Tellez, pers. obs.).

Population data of other offshore islands are available, yet sparse in comparison to the aforementioned islands (Table 3). It is likely these islands have been largely ignored given the initial small populations of *C. acutus*, i.e., Maps Caye in 1995 (N = 11) and 1997 (N = 3); Lighthouse Atoll from 1996–1997 (N = 2 adults, 1 hatchling, 1 nest) and Twin Cayes in 2004 (N = 3) (Platt and Thorbjarnarson 1996, 1997; Platt et al. 1999; McKeon and Feller 2004). Other confirmed nest sites found among surveyed islands include Maps Caye during the 1994 and 1997 survey, and Long Caye in 1997 (Platt and Thorbjarnarson 1996, 1997).

Similar to offshore cayes, mainland survey data are limited to those from the countrywide survey completed in 1997 (Platt and Thorbjarnarson 2000a). A total of 14 *C. acutus* in only 5 locations were observed over 574.6 km, the lowest encounter rate of any data found (Platt and Thorbjarnarson 2000a). However, this may no longer be representative of mainland populations as in recent years there has been increased concern by northern communities of Belize about the increase of American Crocodiles encountered around human developments (Sandoval, pers. obs.). Additionally, the recent discovery of two large populations of *C. acutus* on the mainland not previously identified in southern Belize reforms the past conjecture that *C. acutus* is not common on the mainland (Tellez and Boucher, pers. obs.).

Learning from History to Write a New Conservation Chapter

So what has history taught us about the American Crocodile in Belize? Exploitation of crocodiles for their skin and meat almost led to the extirpation of *C. acutus* in Belize by the 1950s. The population remained desolate for the next 30–40 years as a result of the continuous lack of wildlife enforcement and laws to protect these animals, and recovery did not transpire until the establishment of legal protection and enforcement in the early 1980s (Rainwater and Platt 2009; Tellez et al. 2016; Rainwater, pers. obs.; Tellez and Boucher, pers. obs.). The recovery of the American Crocodile in Belize correlated to the increase of crocodile abundance among the cayes, suggestive of the cayes' importance as a source population within the region. Perhaps mainland populations of *C. acutus* represent dispersed individuals or progeny from the cayes as habitat reached its carrying capacity of the species. However, given the rise of *C. acutus* in some locations along the coast, conservationists and researchers should be open to the idea that we currently may be witnessing a shift in location of source and sink populations. Resident crocodiles of Turneffe Atoll, Ambergris Caye, and Caye Caulker are already facing the pressure of dispersing and finding new suitable (nesting) habitat as the cayes are currently under rapid development relative to the coast of Belize, and it's possible that more individuals are dispersing towards the coast seeking refuge. The development among the cayes is likely not to stop in the near future given the increased popularity of Belize as a retirement and tourist destination, thus, it is significant to locate key habitat and nesting locations to ensure the stability and reproductive vitality of *C. acutus*. Perhaps the creation of an American Crocodile reserve or sanctuary among some closely-linked cayes could provide the necessary habitat (Hekkala et al. 2015) and refuge for *C. acutus*.

Undoubtedly the American Crocodile population succumbed to the hands of hunters in the past 100 years in Belize, however illegal hunting is no longer the principal danger. Pollution, and development seem to be the primary dangers threatening the American Crocodile (Platt and Thorbjarnarson 1996, 2000a; Rainwater and Platt 2009; Rainwater 2010; Chenot-Rose 2013;

Fig. 3. Populations of *Crocodylus acutus* on Ambergris Caye are currently under threat by heavy metal pollution: A) a relatively young 2.4-m *C. acutus* captured in 2008 with no teeth and sloughed skin (other crocodiles in this location were illustrating similar symptoms), and B) the consumption of trash has become common among a population of crocodiles given the use of trash as landfill for development.

Tellez and Chenot-Rose 2015). Shoreline development decreases available nesting and nursery habitat, and large-scale mangrove clearance impacts overall habitat use by American Crocodiles. As a result, American Crocodiles may increasingly seek more unconventional habitats such as residential communities, inland wetlands, and man-made structures in which they would not normally be found. The thriving population of American Crocodiles in the wastewater treatment ponds of an offshore caye is a prime example. The area is surrounded by extensive development, mangrove clearance, and dredging. However, the ponds themselves are isolated and see little human traffic. Additionally, the raised berms of the ponds are, not by design but function, extremely suitable for nesting and are used annually by the resident adult population. The raw sewage does not appear to hamper the growth or health of individuals observed and captured in the ponds, however, pollution has had considerable impact on American Crocodiles in other locations in Belize. Current research addressing increases of pollution within crocodilian habitat is indicating that crocodiles and aquatic biota are beginning to show the effects of their toxic environment, such as on Ambergris Caye (Tellez, unpubl. data). Crocodiles of all sizes have been caught emaciated, with no teeth, skin sloughing off, physical deformities (particularly in hatchlings), or illustrating symptoms of neurological disorders such as star-gazing (Fig. 3). These abnormalities may be a result of living in an environment in which trash is being used as landfill and chemical waste is seeping into the aquatic habitat. Moreover, stomach content data and behavior observations infer crocodiles are exposed to non-organic chemicals through the consumption of trash (Boucher and Tellez, pers. obs.). Collaborative efforts among various environmental and conservation organizations are gathering increasing evidence of the adverse effects of environmental toxicity on crocodiles and adjacent wildlife in Belize (Tellez, unpubl. data). However, the negative impacts of pollution seem to outpace the scientific research.

Although not as formidable a threat to survival such as habitat destruction and pollution, hybridization between *C. acutus* and *C. moreletii* has recently raised concerns among key stakeholders about the future genetic integrity of the parental species (Hekkala et al. 2015). Hybridization or introgression is a common phenomenon between the sympatric crocodylian species, possibly existing for several generations as a result of natural processes (Ray et al. 2004; Hekkala et al. 2015; Pacheco-Sierra et al. 2016). The present level or occurrence of hybridization between *C. acutus* and *C. moreletii* is unknown, however, of concern is the possible increase rate of admixing as *C. acutus* loses more habitat along the coast, dispersing further inland into historically *C. moreletii* habitat. Individual specimens recently caught (May 2016 to August 2016) from a field site inhabited by *C. acutus* and *C. moreletii* (N = 83) illustrate morphological features of both species as described in Herrera et al (2012) and Hekkala et al (2015); it is almost becoming commonplace (particularly south of the Belize River) to find individuals not adhering to the standard morphological characteristics of the parental species (Tellez and Boucher, pers. obs.). It would seem that if conservationists are concerned about the loss of genetic integrity of American and Morelet's crocodiles, the preservation of key (nesting) habitat is essential.

Although the collection of scientific data is necessary in advancing any conservation efforts of the American Crocodile, acceptance and interest in the conservation of the species among communities is fundamental for the success of any current and future management program. Many Belizeans are unaware of the protection status of crocodiles, do not understand the possible important ecological role crocodiles serve in their environment in conjunction with the possible economic opportunity via eco-tourism, or unaware of the knowledge to live in co-existence with species that are relatively shy and timid. The preliminary data from an on-going community perception survey about crocodiles in Belize suggests that the lack of educational outreach, sensationalized TV documentaries or shows, and the previous ideology of relocating non-problematic crocs (which ultimately subconsciously teaches fear and the lack of desire to co-exist with crocodiles) has resulted in wide-spread misguided information and false facts about the predators (Tellez, unpubl. data). The lack of tolerance has led to the continual harassment and illegal hunting of crocodiles as they are seen as vicious predators or pests. Key stakeholders (i.e. the Crocodile Research Coalition (CRC), The Belize Zoo, and the Belize Forest Department) are currently combating opportunistic killings or false beliefs through intensive crocodile educational outreach, which include festival and school presentations, radio and TV interviews, interactive social media posts, local internship and volunteer opportunities, and community involvement programs.

For example, Next Gen Croc initiated by the CRC in collaboration with the Forest and Marine Reserve of Caye Caulker (FAMRACC), is a crocodile monitoring program that mentors Ocean Academy high school students on Caye Caulker to conduct official monthly nocturnal eyeshine surveys in addition to monthly community outreach to educate locals and tourists about the American Crocodile, the environment, and adjacent wildlife. Students of Next Gen Croc are already proving themselves as local advocates for crocodile conservation, pioneering the establishment of a long-term American Crocodile monitoring program on the caye. We anticipate that outreach programs such as Next Gen Croc will motivate young conservationists to establish local initiatives, as well as build a strong foundation for the future stewards of conservation management in Belize. Given the logistical support received by the local communities and government for programs similar to Next Gen Croc, it would appear integrative management and mentorship programs will emerge as a new approach of conservation management in contrast to previous programs, which tentatively ignored community participation in local conservation efforts.

Similar to other wildlife species in threat of survival, the long-term management of American Crocodiles of Belize would likely benefit from ongoing conservation efforts, not simply a one-time positive intervention. Education and the re-establishment of cultural linkage between crocodiles and Belizeans could reignite pride in the species, further promoting a countrywide sentiment of species protection. For example, discussing the importance of crocodiles within the Maya culture (such as being the only animal recognized to connect the celestial, terrestrial and underworld, as well as being the first symbol of the Maya Calendar) has warranted further positive interest to learn more about crocodiles amongst communities and tourists, as well as stimulating tour guides and tour operators to take positive action towards crocodile conservation. Encouraging local participation can help identify a champion, one who can lead the conservation efforts for the American Crocodile in the perspective region. This is extremely important to gain further community support for wildlife conservation efforts. Lastly, an updated countrywide population survey is warranted to update the status of *C. acutus* in Belize, and reevaluate data deficient areas such as the mainland. It is unlikely that the population of American Crocodiles will ever reach its once historical abundance given the current threats of habitat destruction and pollution, however, stakeholders can work together to ensure the population remains stable while simultaneously mitigating local extirpation. Given the recent interest in crocodile management, rise in crocodile research, and collaborative efforts by various stakeholders and young conservationists, we believe Belize is entering a Second Renaissance of crocodile conservation that will be favorable for both *C. acutus* and local communities for generations to come.

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GEOGRAPHIC DISTRIBUTION

CAUDATA — SALAMANDERS

***AMBYSTOMA JEFFERSONIANUM* (Jefferson Salamander).** USA: PENNYLVANIA: Montour Co.: 6.5 km SW Washingtonville and 5.2 km NW Mooresburg (41.01702°N, 76.74674°W; WGS 84). 18 April 2018. S. Hartzell. Verified by Coleman M. Sheehy, III. Florida Museum of Natural History (UF 181114; photo voucher). New county record (Hulse et al. 2001. Amphibians and Reptiles of Pennsylvania and the Northeast. Cornell University Press. Ithaca, New York. 419 pp.). The nearest records are ca. 40 km to the northeast in adjacent Columbia County (UF 181103, 181104).

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***DESMOGNATHUS FUSCUS* (Northern Dusky Salamander).** CANADA: NEW BRUNSWICK: York Co.: unnamed stream, Miramichi watershed, 1.9 km SSW of Boiestown (46.4405°N, 66.4297°W; WGS 84). 24 July 2011. Gregory F. M. Jongsma. Verified by D. F. McAlpine. New Brunswick Museum (NBM 9241–9243). New watershed record. New Brunswick (NB), Canada represents the most northeasterly extent of *Desmognathus fuscus* distribution. Within NB, it has been thought that the species was limited to the lower Saint John River and Fundy coast (COSEWIC 2012. Assessment and Status Report on the Northern Dusky Salamander, *Desmognathus fuscus*, in Canada. Committee on the Status of Endangered Wildlife in Canada, 1–69). This new watershed record is significant because *D. fuscus* mainly disperses via waterways (Miller et al. 2015. Ecosphere 6:1–9) and this extends the potential distribution of the species by ca. 13,547 km² to the northeast. An additional specimen (NBM 9587) within the Miramichi watershed was collected 20 June 2012, 10.2 km NW of Juniper, Carleton County, (46.61552°N, 67.29642°W), 67 km NNW from the original record. Specimens collected under a collecting permit from the Department of Natural Resources (SP11-013).

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***HEMIDACTYLIUM SCUTATUM* (Four-toed Salamander).** USA: NORTH CAROLINA: Transylvania Co.: DuPont State Recreational Forest, 6.0 airline km NNE of the town of Cedar Mountain (specific locality withheld due to conservation concern and on file with the North Carolina Wildlife Resources Commission). 16 April 2018. Lori A. Williams and Charles R. Lawson. Verified by Jeffrey C. Beane. North Carolina Museum of Natural Sciences (NCSM 13567; photo voucher). Adult male (69 mm TL, 43 mm SVL) temporarily collected in wetland habitat under woody debris (147 cm × 40 cm), 4 m from standing water. New county record (NCSM files; Beane et al. 2010. Amphibians & Reptiles of the Carolinas and Virginia. Second Edition. University of North Carolina Press, Chapel Hill, North Carolina. 274 pp.; North Carolina Natural Heritage Program. 2018. Biotics Database. Department of Environment and Natural Resources, Raleigh, North Carolina). Pending lab analysis, this record may represent one of only three known populations in the state of "Clade E," the undescribed Evolutionarily Significant Unit in the upper French Broad River drainage in southwestern North Carolina (Herman and Bouzat 2015. J. Biogeog. 43:666–678). The closest previous historical record in the state is from Henderson County, 14.1 airline km to the north.

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***NYCTANOLIS PERNIX* (Nimble Long-limbed Salamander).** GUATEMALA: ALTA VERAPAZ: MUNICIPALITY OF SAN CRISTÓBAL VERAPAZ: Finca PAMAC II, 11 km W of San Cristóbal Verapaz (15.39553°N, 90.57367°W; WGS 84), 2165 m elev. 8 April 2018. Diegopáblo Pineda-Schwarz. Verified by Jonathan A. Campbell. Colecciones Biológicas de la Universidad del Valle de Guatemala, Ciudad de Guatemala, Guatemala (UVG A-1761). First record for the municipality, filling a gap in the species' Guatemalan distribution between the two closest known localities, ca. 34 km SE of Palo Viejo Hydroelectric Dam on road to La Gloria, Municipality of San Miguel Uspantán, El Quiché and 42 km northwest of Biotopo del Quetzal, Municipality of Purulhá, Baja Verapaz (www.amphibiaweb.org; accessed 17 July 2018). The new locality is at the highest elevation recorded for *N. pernix* from throughout its known range; by 555 m in Guatemala over an elevation of 1610 m previously reported by Wilson and Johnson (2010. *In* Wilson et al. [eds.], Conservation of Mesoamerican Amphibians and Reptiles, pp. 32–235. Eagle Mountain Publishing, LC, Eagle Mountain, Utah), and 20 m higher than the 2145 m elevation reported for a site in the Municipality of Las Margaritas in adjacent Chiapas, México (Barrio-Amorós et al. 2016. Mesoamer. Herpetol. 3:534–536). The salamander was found at 1520 h trapped in the top fabric of our tent, presumably after falling out of a tree during a previous nighttime wind storm. The vegetation community at our campsite was cloud forest.

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ANURA — FROGS

***HOPLOBATRACHUS LITORALIS* (Coastal Bullfrog)**. INDIA: WEST BENGAL: Khordanahala District: 24 Parganas South (22.24772°N, 88.14408°E; WGS 84), 7 m elev. 20 June 2018. K. Mondal. Lee Kong Chian Natural History Museum, National University of Singapore (ZRC [IMG] 1.149; photo voucher). Individual encountered on bank of village pond at ca. 2123 h. Previously reported from Ukhia, Teknaf Upazila and Cox's Bazar, Bangladesh (Hasan et al. 2012. Zootaxa 3312:45–48) and northern Tripura State, India (Purkayastha and Basak 2018. Hamadryad 38:25–26). First record from the state of West Bengal, India.

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***HYLA CINEREA* (Green Treefrog)**. USA: GEORGIA: Gordon Co.: private residence 180 m NW of the junction of Fields Ferry Drive NE and Woodedge Drive NE (34.54968°N, 84.84117°W; WGS 84), 205 m elev. 26 June 2014. Maisie Grace MacKnight. Verified by Coleman M. Sheehy III. Florida Museum of Natural History (UF 183919; photo voucher). An adult male found calling from a suburban yard within 300 m of several golf course ponds. Private residence 230 m N of the junction of Dews Pond Road NE and Towne Lake Drive NE (34.50525°N, 84.92999°W; WGS 84), 215 m elev. 25 February 2018. Maisie Grace MacKnight. Verified by Coleman M. Sheehy III. UF 183918 (specimen and tissue sample). An adult (48 mm SVL, 6.4 g) collected from a suburban yard 50 m from a 0.8-ha pond. Private residence 320 m SE of the junction of Nelson Lake Road SE and Spencer Drive SW (34.44715°N, 84.95991°W; WGS 84), 270 m elev. 2 May 2018. Erin Kennedy Box. Verified by Coleman M. Sheehy III. UF 184113 (specimen, tissue sample, photo voucher). An adult male (43 mm SVL, 4.7 g) collected from a rural yard within 900 m of a 1.0-ha pond. First records from the county (Jensen et al. 2008. Amphibians and Reptiles of Georgia. University of Georgia Press, Athens, Georgia. 575 pp.). The 2014 specimen from Woodedge Drive may not represent an established population, but the Towne Lake Drive and Spencer Drive records do appear to represent separate, well-established populations, as breeding choruses have been heard at both sites within the past three years. These records add to a growing body of evidence that *H. cinerea* is expanding its range in northwest Georgia (Rothermel 2008. *In* Jensen et al. [eds.], Amphibians and Reptiles of Georgia, pp. 59–61. University of Georgia Press, Athens, Georgia; Jensen et al. 2011. Herpetol. Rev. 42:250–255; Stevenson et al. 2015. Herpetol. Rev. 46:597–601), and they fill a gap between adjacent Polk and Murray counties with the nearest vouchered records occurring in Polk (Georgia Museum of Natural History [GMNH] 50794) and Murray (GMNH 50094) counties. Work approved under authority of University of Georgia IACUC AUP #A2016 02-001-Y2-A0, and GA DNR Scientific Collecting Permit #029.

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***INCILIUS NEBULIFER* (Gulf Coast Toad)**. USA: MISSISSIPPI: Pearl River Co.: MS Hwy 607, 1.6 km SE of Nicholson (30.46500°N, 89.68470°W; WGS 84). 23 June 2018. Erin Y. Kreiser and Brian R. Kreiser. Verified by Jennifer Y. Lamb. Florida Museum of Natural History (UF 184944; photo voucher). New county record based off a review of issues of *Herpetological Review* from 1967–2018 and museum records on VertNet (www.vertnet.org) found no documentation of this species from Pearl River County. A record from the Mississippi Museum of Natural Science (MMNS 8560) is listed as Stone County. However, the site description (19 miles west of Wiggins) would place this specimen in Pearl River County. Given the ambiguity in the collection locale, our record provides definitive evidence of the presence of this species in Pearl River County (R. L. Jones, pers. comm.). This specimen was located ca. 50 km to the southwest of the ambiguous MMNS record. The closest records to this site are about 20 km to the southeast in Hancock County (Louisiana Museum of Natural History [LSUMZ] 64073, 64106, 64128, 64149, 64167, 64225). Records for the species exist in all of the Louisiana Parishes along the southern border with Mississippi. The confirmed presence of the species in Pearl River County expands the list of Mississippi counties located across the border or along the coast that presumably represent the natural part of the range. Given this, we predict that the species should also be found in the remaining southern Mississippi counties (Amite, Marion, and Walthall) adjacent to Louisiana.

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***LITHOBATES SYLVATICUS* (Wood Frog)**. USA: ILLINOIS: Randolph Co.: Road bordered by forest and agricultural field (37.8820°N, 89.7540°W; WGS 84). 20 February 2018. Jill Schumacher and Jeremy Schumacher. Verified by Chris Phillips and Scott Ballard. Illinois Natural History Survey Collection (INHS Unvouch 44285; photo voucher) and HerpMapper (HM 211264; http://www.herpmapper.org/record/211264). Frog was observed immediately after a heavy flash thunderstorm moving from a forested area towards a flooded agricultural ditch. Phillips et al. (1999. Field Guide to Amphibians and Reptiles of Illinois. Illinois Natural History Survey Manual 8, Champaign, Illinois. 300 pp.) does not list any previous records for this species in Randolph County. To the best of our knowledge this observation represents a new county record for *L. sylvaticus* in Illinois. We thank Christopher Smith for his help in writing this observation account and Chris Phillips for verifying our observation.

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***PSEUDACRIS CRUCIFER* (Spring Peeper)**. USA: TENNESSEE: Roane Co.: wetland adjacent to Black Creek, on the S side of Kirby Ln in Rockwood (35.85554°N, 84.70192°W; WGS 84). 11 March 2018. Andy Edington. Verified by A. Floyd Scott. David H. Snyder Museum of Zoology, Austin Peay State University (APSU 19867; audio voucher). Although one prior record exists for this county, it lacks specific locality data (University of Michigan Museum of Zoology [UMMZ] 56367). Our audio recording represents the first county record with specific locality information (Redmond and Scott 1996. Atlas of Amphibians in Tennessee. Austin Peay State University, Clarksville, Tennessee. Misc. Publ. No. 12. The Center for Field Biology, Austin Peay State University, Clarksville, Tennessee. 94 pp.; http://www.apsubiology.org/tnamphibiansatlas/; 16 April 2018). Records exist in four neighboring counties, and this observation is ca. 17 km east of the nearest previously documented locality (Indiana State University [ISU] 1751). We thank A. Floyd Scott for help in locating museum records.

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PSEUDACRIS FOUQUETTEI **(Cajun Chorus Frog)**. USA: TEXAS: Waller Co.: ca. 10.5 km W of Magnolia (30.2152°N, 95.8614°W; WGS 84), 92 m elev. 30 December 2017. Andrew MacLaren. Verified by Emily Moriarty Lemmon. Cornell University, Macaulay Library (ML 225183; audio voucher). New county record (Dixon 2013. Amphibians and Reptiles of Texas: with Keys, Taxonomic Synopses, Bibliography, and Distribution Maps. Texas A&M University Press, College Station, Texas. 447 pp.). This record fills a gap in the known range for this species that exists among Grimes, Montgomery, Harris, Fort Bend, and Austin Counties, while the species remains undescribed from neighboring Washington County (Dixon 2013, *op. cit.*). The nearest specimen in collection comes from Austin County, ca. 35 km to the southwest (Biodiversity Research and Teaching Collections, Texas A&M University [TCWC] 97796, as *P. triseriata*). The breeding call of this species was recorded using an automated recording unit (Wildlife Acoustics SM2+) on multiple occasions throughout the winter and early spring of 2018.

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TESTUDINES — TURTLES

APALONE MUTICA **(Smooth Softshell)**. USA: MISSISSIPPI: Clarke Co.: Chickasawhay River, Hwy 512 crossing west of Quitman (32.04203°N, 88.74449°W; WGS 84). 7 May 2018. Peter V. Lindeman. Verified by Grover J. Brown. Florida Museum of Natural History (UF 184621; photo voucher). Basking adult male photographed *in situ* with an 83× Nikon CoolPix camera. New county record based on records on VertNet (www.vertnet.org) and in the Mississippi Museum of Natural History. The nearest record for the species in the Chickasawhay River, the major eastern tributary of the Pascagoula River drainage, is from northern Greene County (UF 184622), two counties and 133 river km downstream of the locality reported herein, and no records are known from farther upstream in the Chickasawhay or from any of its tributaries.

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APALONE SPINIFERA **(Spiny Softshell)**. MEXICO: COAHUILA: Municipality of Zaragoza: Zaragoza, Rio Escondido at the bridge along Mexico Hwy 29 (28.48516°N, 100.91707°W; WGS 84), 355 m elev. 10 March 2018. Sean P. Graham, Mark Black, Mark Herr, and Tomas Hernandez. Verified by Toby Hibbitts. James Scudday Vertebrate Collections, Sul Ross State University (SRSU-D 132; photo voucher). First municipality record located ca. 80 km northeast from the closest previous record at Múzquiz, Municipality of Múzquiz (Lemos-Espinal and Smith 2007. Anfibios y Reptiles del Estado de Coahuila, México/Amphibians and Reptiles of the State of Coahuila, Mexico. UNAM, CONABIO, México, D.F. 613 pp.). This research was conducted on behalf of the Maderas del Carmen Flora and Fauna Protection Area under SEMARNAT collecting permit SGPA/DGVS/00047/18 issued to DL, with SPG listed as a sub-permittee. We thank B. Pat, B. McKinney, J. Villalobos, H. Sotelo Gallardo, and other members of El Carmen conservation project for logistical support.

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GRAPTEMYS GEOGRAPHICA **(Northern Map Turtle)**. USA: MICHIGAN: Wexford Co.: ca. 8.4 km SE of Buckley (44.46896°N, 85.50562°W; WGS 84), 286 m elev. 23 June 2015. John G. Phillips. Verified by Kirsten E. Nicholson. Central Michigan University Museum of Cultural and Natural History (MCNH 2018.005; photo voucher). Multiple individuals (both adult and juvenile) were seen in the general vicinity basking on logs in the Big Manistee River, a waterway in which this species is commonly seen. New county record (Holman 2012. The Amphibians and Reptiles of Michigan: A Quaternary and Recent Faunal Adventure. Wayne State University Press, Detroit, Michigan. 528 pp.; Phillips 2016. J. North Am. Herpetol. 1:45–69). This specimen fills a gap in the distribution, as *G. geographica* has been recorded from all surrounding counties except Missaukee County to the east (Phillips 2016, *op. cit.*), through which the Big Manistee also flows through. Field support was provided by M. Phillips, G. McPheeters, and M. Phillips.

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GRAPTEMYS OUACHITENSIS **(Ouachita Map Turtle)**. USA: TEXAS: Wilbarger Co.: Beaver Creek, 17.2 km SW Electra on state FM 1811 (33.95893°N, 99.06623°W; WGS 84). 16 September 2015. John Karges. Verified by Carl J. Franklin. Amphibian and Reptile Diversity Research Center, University of Texas at Arlington (UTADC 8666–8669; photo voucher). New county record extending known range. In the southwesternmost portion of its range, the Ouachita Map Turtle (*Graptemys ouachitensis*) is confined to the Red River Basin of Louisiana, Arkansas, and Texas. Although it has been introduced elsewhere in Texas, as released or escaped captives (C. Franklin, pers. comm.), the native range in Texas is primarily along the Red River and extends no farther westward along the mainstem of the Red River than Clay County, east of Wichita Falls (Lindeman 2013, The Map Turtle and Sawback Atlas: Ecology, Evolution, Distribution and Conservation. University of Oklahoma Press, Norman, Oklahoma. 460 pp.). In Clay County, the Little Wichita River confluences with the Red River coursing from Baylor and Archer counties to the southwest where it originates as an intermittent waterway in Texas' Rolling Plains. Dixon (2013. Amphibians and Reptiles of Texas: with Keys, Taxonomic Synopses, Bibliography, and Distribution Maps, 3rd ed. Texas A&M University Press, College Station, Texas. 447 pp.) maps both Baylor and Archer counties within the range verified by records or specimens, although Lindeman (2013) suspected the Baylor County record was in error. No such scrutiny was given the Archer County record. Presumably, records for either county are from the Little Wichita River drainage within those counties although I have no details of confirmed locations in either county. On 16 September 2015, I found *Graptemys ouachitensis* at Beaver Creek. I returned on 17 September for photographs and estimated 15 adults at the pool beneath a low dam upstream of the highway bridge, either swimming or basking on bridge fixtures, emerged logs, or portions of the remnant paved low-water crossing there. Both Beaver Creek and the Little Wichita River have extremely sinuous main channels braided through their respective floodplains and thus problematic to calculate true distances of stream reaches. However, straight-line measures are 24.5 km downstream from this location to Beaver Creek's confluence with the Little Wichita River and another 65 km downstream to the confluence of the Little Wichita River with the Red River in Clay County. It is unknown whether both reaches are perennially flowing on the surface for their entirety or whether during low flow periods, surface water is confined solely to deeper pools but it does confirm hydro-ecological contiguity or connectivity from the Red River upstream to at least this newly detected occurrence via this tributary drainage basin. This new record for Wilbarger County and more specifically within the Beaver Creek drainage supports the likelihood of *G. ouachitensis* also being present and persisting in the Little Wichita River basin of Baylor and Archer counties and, also for likely occurring in adjacent Wichita County, between Wilbarger and Clay counties along Beaver Creek and the Little Wichita River.

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***PSEUDEMYS CONCINNA* (River Cooter)**. USA: MISSISSIPPI: Smith Co.: Caney Creek, Hwy 481 crossing E of Polkville (32.20218°N, 89.59998°W; WGS 84). 29 May 2018. Peter V. Lindeman. Verified by Grover J. Brown. Florida Museum of Natural History (UF 184628; photo voucher). Basking adult female photographed *in situ* with an 83× Nikon CoolPix camera. Strong River, Smith County Rd 563 crossing SE of Polkville (32.17979°N, 89.63357°W; WGS 84). 30 June 2015. Peter V. Lindeman. Verified by Grover J. Brown. UF 184629 (photo voucher). Basking adult female photographed *in situ* with an 83´ Nikon CoolPix camera. New county record and first vouchered records of *P. concinna* for the Strong River drainage based on records on VertNet (www.vertnet.org) and in the Mississippi Museum of Natural History. Presence of the species at two localities in the lower Strong River in Simpson County was previously reported from basking surveys (Lindeman 1998. Chelon. Conserv. Biol. 3:137–141). The nearest vouchered records of the species in the Pearl River drainage are from the Pearl River in the vicinity of Georgetown in Copiah and Simpson counties, a few river km above the confluence of the Pearl and the Strong, but additional photo vouchers of the species from the Strong River farther downstream have also recently been deposited (Rankin County: UF 184630; Simpson County: UF 184631), thus a range of at least 114 river km in the Strong River plus 4.4 river km in its tributary, Caney Creek, is indicated by these new records.

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***PSEUDEMYS GORZUGI* (Rio Grande Cooter)**. MEXICO: COAHUILA: Municipality of Zaragoza: Zaragoza, Rio Escondido at the bridge along Mexico Hwy 29 (28.48516°N, 100.91707°W; WGS 84), 355 m elev. 10 March 2018. Sean P. Graham, Mark Black, Mark Herr, and Tomas Hernandez. Verified by Toby Hibbitts. James Scudday Vertebrate Collections, Sul Ross State University (SRSU-D 130; photo voucher). First municipality record situated ca. 20 km north from the closest reported locality in Coahuila at Allende, Municipality of Allende (Lemos-Espinal and Smith 2007. Anfibios y Reptiles del Estado de Coahuila, México/Amphibians and Reptiles of the State of Coahuila, Mexico. UNAM, CONABIO, México, D.F. 613 pp.). This research was conducted on behalf of the Maderas del Carmen Flora and Fauna Protection Area under SEMARNAT collecting permit SGPA/DGVS/00047/18 issued to DL, with SPG listed as a sub-permittee. We thank B. Pat, B. McKinney, J. Villalobos, H. Sotelo Gallardo, and other members of El Carmen conservation project for logistical support.

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***PSEUDEMYS NELSONI* (Florida Red-bellied Cooter)**. USA: GEORGIA: Glynn Co.: Jekyll Island (31.03318°N, 81.41348°W; WGS 84). 8 September 2015. Katie Mascovich. Verified by John Jensen. Georgia Museum of Natural History (GMNH 50519; photo voucher). This male specimen was located on the beach on Jekyll Island, Georgia, and then marked and released (212 mm straight carapace length, 1357 g). The same individual was later observed on two separate occasions, once again on the beach 3 km southwest from the original location and a third time on a game camera in secondary dune/ephemeral wetland habitat 0.5 km north of the second location. In addition to being a new county record (Jensen et al. 2008. Amphibians and Reptiles of Georgia. University of Georgia Press, Athens, Georgia. 575 pp.), this observation is a northeastern range extension; the species was previously documented on Cumberland Island, an adjacent barrier island to the south in Camden County (Shoop and Ruckdeschel 1986. Herpetol. Rev. 17:51).

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***TERRAPENE NELSONI* (Spotted Box Turtle)**. MÉXICO: NAYARIT: Municipality of Huajicori: Huajicori (22.66021°N, 105.31178°W; WGS 84), 85 m elev. 15 February 2017. Jesús A. Loc-Barragán. Verified by Paulino Ponce-Campos. Amphibian and Reptile Diversity Research Center, University of Texas at Arlington (UTADC 9177; photo voucher). First record for the municipality, third locality known from Nayarit, and a range extension of 27.7 airline km southeast of the closest site at Ejido de la Ciénega, municipality of Escuinapa, Sinaloa (CONABIO 2018. Naturalista. Comisión Nacional para el Conocimiento y Uso de la Biodiversidad, México, D.F., Mexico; http://www.naturalista.mx/observations/1832317; accessed 4 February 2018), and ca. 30 airline km northwest of the closest reported site in Nayarit near Mesa de Pedro y Pablo, Municipality of Acaponeta (National Museum of Natural History, Smithsonian Institution [USNM] 46252; Legler and Vogt 2013. The Turtles of Mexico: Land and Freshwater Forms. University of California Press, Berkeley, California. 416 pp.). The turtle was found around 0500 h while it was active on wet forest litter in tropical deciduous forest after heavy rainfall the previous night. We thank the people of Huajicori for their help in the field during the program *Community Monitors to the Natural Patrimony of Sierra Madre Occidental de Nayarit*, which was funded by the project "Payments for Environmental Services" from Consultoría Forestal y Servicios Ambientales Nayarit through the Comisión Nacional Forestal (CONAFOR).

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***TRACHEMYS SCRIPTA* (Pond Slider)**. MEXICO: COAHUILA: Municipality of Zaragoza: Zaragoza, Rio Escondido at the bridge along Mexico Hwy 29 (28.48516°N, 100.91707°W; WGS 84), 355 m elev. 10 March 2018. Sean P. Graham, Mark Black, Mark Herr, and Tomas Hernandez. Verified by Toby Hibbitts. James Scudday Vertebrate Collections, Sul Ross State University (SRSU-D 131; photo voucher). First municipality record located ca. 80 km northeast of the closest reported locality in Coahuila at Múzquiz, Municipality of Múzquiz. This specimen was consistent with the taxon *Trachemys scripta elegans*. Although it is possible this population could be the result of a translocation, wild populations are known from downstream in the Rio Grande drainage. (Lemos-Espinal and Smith 2007. Anfibios y Reptiles del Estado de Coahuila, México/Amphibians and Reptiles of the State of Coahuila, Mexico. UNAM, CONABIO, México, D.F. 613 pp.). This research was conducted on behalf of the Maderas del Carmen Flora and Fauna Protection Area under SEMARNAT collecting permit SGPA/DGVS/00047/18 issued to DL, with SPG listed as a sub-permittee. We thank B. Pat, B. McKinney, J. Villalobos, H. Sotelo Gallardo, and other members of El Carmen conservation project for logistical support.

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***TRACHEMYS SCRIPTA ELEGANS* (Red-eared Slider)**. USA: NEW MEXICO: Curry Co.: Clovis, Greene Acres Park Lake (34.41850°N, 103.20644°W; WGS 84), 1300 m elev. 17 April 2013. J. N. Stuart. University of Kansas Digital Archives (KUDA 12156; photo voucher). Clovis, Hillcrest City Park, fishing pond (34.41016°N, 103.18544°W; WGS 84), 1456 m elev. 21 April 2015. J. N. Stuart. KUDA 12157 (photo voucher). Ned Houk Memorial Park, ca. 13 km NNE of Clovis (center), fishing pond (34.51606°N, 103.17080°W; WGS 84), 1298 m elev. 21 April 2015. J. N. Stuart. KUDA 12158 (photo voucher). All verified by Leland J. S. Pierce. Multiple individuals seen at each of these three sites. Reported as an introduced species in Curry County by Painter et al. (2017. West. Wildl. 4:29–60) but without locality data. The species has been introduced to the few perennial water bodies (all isolated ponds) in the county and is potentially established there. Nearest other confirmed record is ca. 97 km to the north-northwest in the Canadian River near Logan, Quay County, where it is native (Degenhardt and Christiansen. 1974. Southwest. Nat. 19:21–46; Degenhardt et al. 1996. Amphibians and Reptiles of New Mexico. University of New Mexico Press, Albuquerque, New Mexico. 431 pp.).

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SQUAMATA — LIZARDS

***BRACHYMELES BONITAE* (Stub-limbed Burrowing Skink)**. PHILIPPINES: LUZON ISLAND: CAMARINES NORTE PROVINCE: Municipality of Labo: Barangay of Tulay na Lupa, Mt. Labo (14.0802°N, 122.77815°E; WGS 84), 264 m elev. 21–23 May 2017. J. B. Fernandez, E. Schaper, and E. Freitas. Verified by Luke J. Welton. Sam Noble Oklahoma Museum of Natural History (OMNH 45575, 45577, 45578). Collected between 0800 and 1430 h from rotting tree stumps among young herbaceous plants and roots in coconut plantation or beneath piles of decaying coconut husks, leaves, and roots in secondary growth forest. New municipality and province record, and first confirmed record of a member of *B. bonitae* complex from Bicol Peninsula, southeastern Luzon Island (Davis et al. 2014. J. Herpetol. 48:480–494; Davis et al. 2016. Zootaxa 4132:30–43), extending distribution 113.6 km SE in neighboring province. True *B. bonitae* restricted previously to Central Luzon and Polillo Island (Davis et al. 2014, *op. cit.*). Fieldwork supported by NSF IOS 1353683 to CDS and NSF IOS 1353703 to PJB. Fieldwork was conducted under the Memorandum of Agreement with the BMB of the Philippines (2015–2020) and Gratuitous Permits to Collect No. 247.

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***HEMIDACTYLUS TURCICUS* (Mediterranean Gecko)**. USA: NEW MEXICO: Eddy Co.: Whites City campground, Carlsbad (34.17527°N, 104.38039°W; WGS 84), ca. 1113 m elev. 20 June 2018. Korry J. Waldon and Thanchira Suriyamongkol. Verified by Carl J. Franklin. Amphibian and Reptile Diversity Research

Center, University of Texas at Arlington (UTADC 9204–9208; photo voucher). First confirmed record for Eddy County, and ca. 166 km E from the nearest known record in Otero County (Murry and Painter 2003. Herpetol. Rev. 34:166). The adult male (47 mm SVL, 58.8 mm TL) was captured by hand at 2100 h along an exterior bathroom wall of Whites City tent campground.

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MARISORA BRACHYPODA **(Middle American Short-limbed Skink)**. MEXICO: SINALOA: Municipality of San Ignacio: near Highway 6, ca. 1.2 km SW prior to road to Lo de Ponce (23.80987°N, 106.55012°W; WGS 84), 177 m elev. 16 September 2011. Scott J. Trageser and Robert A. Schell. Verified by T. J. Papenfuss. Natural History Museum of Los Angeles County (LACM PC 2361; photo voucher). An individual of unknown sex was observed and photographed at 1230 h near the bank of a large dry arroyo. First record for Sinaloa, northernmost locality for the species in Mexico, and a range extension of ca. 185 km NE of the closest documented locality in Nayarit near the Sinaloan border (Andrade-Soto et al. 2012. Mesoamer. Herpetol. 4:224–230). Andrade-Soto et al. (2012, *op. cit.*), using Maxent to generate a species distribution model, predicted that *M. brachypoda* should be expected to occur even further north than our record, probably at least 40 km northward to the El Salto area (Municipality of Sinaloa) along contiguous riparian corridors.

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PLESTIODON INEXPECTATUS **(Southeastern Five-lined Skink)**. USA: TENNESSEE: Davidson Co.: Couchville Cedar Glade State Natural Area (36.10221°N, 86.53406°W; WGS 84), 165 m elev. 2 April 2018. Steven Hromada. Verified by A. Floyd Scott. David H. Snyder Museum of Zoology, Austin Peay State University (APSU 19871; photo voucher). Juvenile found under rock at edge of cedar glade. New county record filling a gap in the central Nashville basin of Middle Tennessee (Scott and Redmond 2008. Atlas of Reptiles in Tennessee. http://www.apsubiology.org/tnamphibiansatlas/; accessed 3 April 2018). This record is ca. 12 km northwest from closest vouchered record in Rutherford County (Niemiller et al. 2007. Herpetol. Conserv. Biol. 6:135–149).

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SCELOPORUS COUCHII **(Couch's Spiny Lizard)**. MEXICO: COAHUILA: Municipality of Múzquiz: rock cliffs near La Cuesta de Malena along Coahuila Hwy 53 (28.72409°N, 102.50215°W; WGS 84), 1521 m elev. 11 March 2018. Sean P. Graham, Mark Black, Mark Herr, and Tomas Hernandez. Verified by Jackson Shedd. James Scudday Vertebrate Collections, Sul Ross State University (SRSU 7085–7086).

Municipality of Ocampo: limestone cliffs within Cañón San Isidro, Área de Protección de Flora y Fauna Maderas Del Carmen (28.85643°N, 102.58134°W; WGS 84), 1343 m elev. 14 March 2018. Sean P. Graham, Mark Black, Mark Herr, and Tomas Hernandez. Verified by Adrián Nieto-Montes de Oca. James Scudday Vertebrate Collections, Sul Ross State University (SRSU 7091).

First records for the two municipalities, extending the range of this species in Coahuila ca. 160 and 180 km, respectively, north along the Sierra Madre Oriental to the Sierra del Carmen, from the nearest known locality in the Municipality of Cuatro Ciénegas (Lemos-Espinal and Smith 2007. Anfibios y Reptiles del Estado de Coahuila, México/Amphibians and Reptiles of the State of Coahuila, Mexico. UNAM, CONABIO, México, D.F. 550 pp.). This research was conducted on behalf of the Maderas del Carmen Flora and Fauna Protection Area under SEMARNAT collecting permit SGPA/DGVS/00047/18, issued to DL, with SPG listed as a sub-permittee. Both specimens are on indefinite loan to CEMEX El Carmen project office at Rancho Los Pilares, Coahuila, México. We thank B. Pat, B. McKinney, J. Villalobos, H. Sotelo Gallardo, and other members of El Carmen conservation project for logistical support.

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SCELOPORUS COWLESI **(Southwestern Fence lizard)**. USA: NEW MEXICO: De Baca Co.: Fort Sumner Lake eastern campgrounds, Lake Sumner 88119 (34.61416°N, 104.3689°W; WGS 84), ca. 1300 m elev. 28 April 2018. Derek Jamerson and Thanchira Suriyamongkol. Verified by Carl J. Franklin. Amphibian and Reptile Diversity Research Center, University of Texas at Arlington (UTADC 9196–9200; photo voucher). The adult male specimen (58.47 mm SVL, 58.17 mm tail length) was caught at Fort Sumner Lake around arid vegetation after running under a large rock for cover. *Sceloporus cowlesi* has been spotted in multiple locations in the surrounding Torrance, Sandoval, and Bernalillo counties (Painter et al. 2017. West. Wildl. 4:29–60). The first report of this species in New Mexico was in Mimbres Canyon (Bernalillo County) in 1935 (San Diego Natural History Museum [SDNHM] 224293). This specimen is the first verified record of *S. cowlesi* found in De Baca County. The closest verified record is in Torrance County, ca. 246 km to west of the current location (Museum of Southwestern Biology [MSB] 98705).

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SCINCELLA LATERALIS **(Little Brown Skink)**. USA: TENNESSEE: Cocke Co.: North Cherokee National Forest (35.93573°N, 82.97504°W; WGS 84). 4 April 2018. Katie Harris and Wade GeFellers. Verified by A. Floyd Scott. David H. Snyder Museum of Zoology, Austin Peay State University (APSU 19872; photo voucher). One adult found on a south-facing slope on a fallen Post Oak (*Quercus stellata*). This Cocke County record is a new county record extending the known range eastward (Scott and Redman 2008. Atlas of Reptiles in Tennessee. http://www.apsubiology.

org/tnreptileatlas/; accessed 25 May 2018). The nearest known locality is 57.8 km SW in Sevier County (Great Smoky Mountains National Park herp collection, catalogue nos. 3857–3858).

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SQUAMATA — SNAKES

***BOIRUNA MACULATA* (Mussurana, Víbora Luta)**. ARGENTINA: SAN JUAN PROVINCE: Departamento Valle Fértil: La Majadita (30.70035°S, 67.49781°W; WGS 84), 972 m elev. 19 December 2013. N. Pelegrin, A. Laspiur, and C. Piantoni. Verified by J. C. Stazzonelli. Museo Argentino de Ciencias Naturales Bernardino Rivadavia, Buenos Aires, Argentina (MACN 51231). Subadult female (980 mm total length and 170 mm tail length) collected at 2300 h. This species has a wide distribution in southern South America (Bolivia, Paraguay, Uruguay, Brazil, and Argentina). *Boiruna maculata* is known in Argentina from 18–39°S and 55–69°W in the provinces of Jujuy, Salta, Formosa, Chaco, Santiago del Estero, Tucumán, Catamarca, La Rioja, Córdoba, Santa Fe, Misiones, Corrientes, Entre Ríos, Mendoza, San Luis, and La Pampa (Scott et al. 2006. Pap. Avul. Zool. 46:77–105); it was reported from San Juan without voucher (Acosta et al. 2017. Los Reptiles de San Juan. Editorial Universidad de San Juan, Argentina. 132 pp.). First vouchered province record, filling the gap between Los Molinos, La Rioja Province (28.80709°S; 66.94130°W; 215 km to the north), Lafinur, San Luis Province (32.06671°S, 65.33335°W, 250 km to the southeast), and Cerro Bola, Mendoza Province (34.64775°S, 68.58387°W, 450 km to the south) the nearest records of the species (Scott et al. 2006, *op. cit.*). It also represents the first record from the natural protected area Parque Provincial Valle Fértil. Specimen collected under permission of Secretaria de Estado de Ambiente y Desarrollo Sustentable, San Juan (Exp. N° 1300-2643). Financial support was provided by CONICET fellowship to AL.

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***CARPHOPHIS AMOENUS* (Common Wormsnake)**. USA: TENNESSEE: Greene Co.: forested hillside S of Paint Creek in the Cherokee National Forest (35.95097°N, 82.89078°W; WGS 84). 22 April 2018. Morgan Fleming and Todd W. Pierson. Verified by A. Floyd Scott. David H. Snyder Museum of Zoology, Austin Peay State University (APSU 19868; photo voucher). First county record (Scott and Redmond 2008. Atlas of Reptiles in Tennessee. http://www.apsubiology.org/tnreptileatlas/; accessed 28 April 2018), although records exist in three neighboring counties. This observation is ca. 15 km NE of the nearest previously documented locality in Tennessee (Florida Museum of Natural History [UF] 113786) and 9.5 km NW of the nearest documented locality in North Carolina (North Carolina Museum of Natural Sciences [NCSM] 15144; Palmer and Braswell 1995. Reptiles of North Carolina. University of North Carolina Press, Chapel Hill, North Carolina. 412 pp.). This observation was made while operating under UTK-IACUC # 2541-0617.

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***GLOYDIUS HIMALAYANUS* (Himalayan Pitviper)**. INDIA: WEST BENGAL: Kalimpong District: Kolakham (27.1132°N, 88.6786°E; WGS 84), 1320 m elev. 26 June 2012. S. Mukherjee and A. Chaudhuri. Verified by Gernot Vogel. Lee Kong Chian Natural History Museum, National University of Singapore (ZRC [IMG] 2.385a–b; photo voucher). Individual encountered basking on rock near Changey waterfall at ca. 1320 h. Species reported from Kashmir, Himachal Pradesh, Northern Punjab, Uttakhand, northern Haryana and Uttar Pradesh States in India, besides Bhutan, Nepal, and Pakistan (Whitaker and Captain 2008. Snakes of India, The Field Guide. Draco Books, Chennai. 385 pp.; Wallach et al. 2014. Snakes of the World: A Catalogue of Living and Extinct Species. Taylor and Francis, CRP Press, Boca Raton, Florida. 310 pp.; Koirala et al. 2016. J. Threat. Taxa 8:9461–9466). First record from West Bengal State.

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***LAMPROPELTIS GENTILIS* (Western Milksnake)**. MEXICO: COAHUILA: Municipality of Múzquiz: Coahuila Hwy 53, SE of La Cuesta de Malena (28.72506°N, 102.46445°W; WGS 84), 1272 m elev. 23 May 2018. Fabiola Baeza-Tarin, Sean P. Graham, Laine Giovanetto, and Tomas Hernandez. Verified by Toby Hibbitts. Herpetology Collection, Universidad Autónoma de Nuevo Léon, San Nicolas de los Garza (UANL 8273). First verified record for this species in Mexico (Lemos-Espinal and Smith 2007. Anfibios y Reptiles del Estado de Chihuahua, México/Amphibians and Reptiles of the State of Chihuahua, Mexico. UNAM, CONABIO, México, D.F. 613 pp.; Lemos-Espinal and Smith 2007. Anfibios y Reptiles del Estado de Coahuila, México/Amphibians and Reptiles of the State of Coahuila, Mexico. UNAM, CONABIO, México, D.F. 550 pp.; Ruane et al. 2013. Syst. Biol. 63:231–250). The closest reported locality for *L. gentilis* (as *L. triangulum celaenops*) is ca. 80 km to the northwest in Brewster County, Texas, USA (Werler and Dixon 2000. Texas Snakes: Identification, Distribution, and Natural History. University of Texas Press, Austin, Texas. 437 pp.). This research was conducted on behalf of the Maderas del Carmen Flora and Fauna Protection Area under SEMARNAT collecting permit SGPA/DGVS/00047/18 issued to DL, with SPG listed as a sub-permittee. We thank B. Pat, B. McKinney, J. Villalobos, H. Sotelo, and other members of El Carmen conservation project for logistical support.

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***LYCODON JARA* (Twin-spotted Wolf Snake).** INDIA: JHARKHAND: Hazaribagh District: Hazaribagh (24.01349°N, 85.39629°E; WGS 84), 617 m elev. 28 December 2017. Arshad Khan and Vivek Sharma. Verified by Anirban Chaudhuri. Lee Kong Chian Natural History Museum, National University of Singapore (ZRC [IMG] 2.387 a–b; photo voucher). Adult specimen found basking at Canary Hills at 1000 h. New record for Jharkhand State and Chota Nagpur Plateau (Chaudhuri et al. 2015. Hamadryad 37:95–103). New locality is ca. 264 km NW from nearest published locality of Kharagpur, West Bengal. India.

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***MASTICOPHIS SCHOTTI* (Schott's Whipsnake).** MEXICO: COAHUILA: Municipality of Zaragoza: Mexico Hwy 29 just north of Ejido Zaragoza (28.56282°N, 100.90971°W; WGS 84), 347 m elev. 10 March 2018. Sean P. Graham, Mark Black, Mark Herr, and Tomas Hernandez. Verified by Toby Hibbitts. James Scudday Vertebrate Collections, Sul Ross State University (SRSU 7083). Specimen collected DOR. First record for the municipality, located ca. 50 km northwest from the nearest recorded localities within Coahuila in the Municipality of Guerrero and adjacent Municipality of Villa Union (Lemos-Espinal and Smith 2007. Anfibios y Reptiles del Estado de Coahuila, México/Amphibians and Reptiles of the State of Coahuila, Mexico. UNAM, CONABIO, México, D.F. 550 pp.), that fills a gap in its distribution between those records and records to the north in Texas (see Powell et al. 2016. Peterson Field Guide to Reptiles and Amphibians of Eastern and Central North America. Houghton Mifflin Harcourt Publishing Co., New York, New York. 494 pp.). This research was conducted on behalf of the Maderas del Carmen Flora and Fauna Protection Area under SEMARNAT collecting permit SGPA/DGVS/00047/18, issued to DL, with SPG listed as a sub-permittee. The specimen is on indefinite loan to the CEMEX El Carmen project office at Los Pilares, Coahuila, México. We thank B. Pat, B. McKinney, J. Villalobos, H. Sotelo Gallardo, and other members of El Carmen conservation project for logistical support.

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***MASTICOPHIS TAENIATUS* (Striped Whipsnake).** MEXICO: COAHUILA: Municipality of Músquiz: Coahuila Hwy 53, 26 km NW of Músquiz (28.25167°N, 101.60980°W; WGS 84), 571 m elev. 10 March 2018. Sean P. Graham, Mark Black, Mark Herr, and Tomas Hernandez. Verified by Toby Hibbitts. James Scudday Vertebrate Collections, Sul Ross State University (SRSU 7084). Specimen collected DOR. First municipality record, located ca. 100 km SE from the closest reported locality in Coahuila within the Municipality of Ocampo (Lemos-Espinal and Smith 2007. Anfibios y Reptiles del Estado de Coahuila, México/Amphibians and Reptiles of the State of Coahuila, Mexico. UNAM, CONABIO, México, D.F. 550 pp.). This research was conducted on behalf of the Maderas del Carmen Flora and Fauna Protection Area under SEMARNAT collecting permit SGPA/DGVS/00047/18, issued to DL, with SPG listed as sub-permittee. The specimen is on indefinite loan to the CEMEX El Carmen project office at Rancho Los Pilares, Coahuila, México. We thank B. Pat, B. McKinney, J. Villalobos, H. Sotelo Gallardo, and other members of El Carmen conservation project for logistical support.

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***NERODIA ERYTHROGASTER* (Plain-bellied Watersnake).** USA: TENNESSEE: Franklin Co.: unnamed pond in western end of Arnold Air Force Base (35.35181°N, 86.14055°W; WGS 84). 19 July 2017. Brian T. Miller. Verified by A. Floyd Scott. David H. Snyder Museum of Zoology, Austin Peay State University (APSU 19788; photo voucher). New county record (Scott and Redmond 2016. Atlas of Reptiles in Tennessee. Austin Peay State University, Clarksville, Tennessee. Misc. Pub. No. 18. The Center for Field Biology, Austin Peay State University, Clarksville, Tennessee. 188 pp.; http://www.apsubiology.org/tnreptileatlas/; accessed 19 January 2018). This species is widespread in western Tennessee, but has a limited occurrence in middle Tennessee with scattered populations within the Cumberland River basin and one population within the Duck River watershed (Scott and Redmond 2016, *op. cit.*). This new county record, and first record for the Elk River watershed, extends the known distribution approximately 9.25 km southeast from the Duck River watershed record in Coffee County (Scott and Redmond 2016, *op. cit.*).

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***NERODIA FASCIATA* (Southern Watersnake).** USA: ARIZONA: Yuma Co.: Laguna Dam Rd below Imperial Dam, ca. 22.5 km NE of Yuma (32.87155°N, 114.455917°W; NAD 83), ca. 49 m elev. 23 October 2015. J. Clayton Sharp. Verified by Thomas R. Jones. Arizona State University Herpetological Collection (ASU 36351). The snake was found alive on the road. Laguna Dam Rd crosses the complex wetlands that empty into Mittry Lake along the Colorado River. A second individual was seen two days later but was not captured. This is the first record of *Nerodia fasciata* in Arizona (Brennan and Holycross. 2006. A Field Guide to Amphibians and Reptiles in Arizona. Arizona Game and Fish Department, Phoenix, Arizona. 150 pp.). *Nerodia fasciata* is new to the fauna of Arizona, and since the first animal was collected there have been numerous observations and collections made at various locations below Imperial Dam (JCS, pers. obs., C. Meachum, pers. comm., D. Ortiz, pers. comm., R. Reed, pers. comm.). *Nerodia fasciata* is native to the southeastern United States (Powell et al. 2016. Peterson Field Guide to Reptiles and Amphibians of Eastern and Central North America. Houghton Mifflin Harcourt, Boston, Massachusetts. 494 pp.). It has also been introduced into California where it has become established in at least two sites, neither of which is near the Colorado River (Rose and Todd 2014. PLoS ONE 9:e100277; Reed et al. 2016. Herpetologica 72:130–136). Subsequent investigations by U.S. Geological Survey staff strongly suggest that a breeding population of *N. fasciata* is established in these Colorado River wetlands (R. Reed, pers. comm.), and further spread of *N. fasciata* upstream along the Colorado or Gila rivers could impact native aquatic wildlife. Specimen collected under an Arizona Game and Fish Hunting/Fishing License (#4624).

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***RHADINAEA FLAVILATA* (Pine Woods Snake)**. USA: FLORIDA: MIAMI-DADE CO.: Amelia Earhart Park (25.89559°N, 80.28683°W; WGS84). 3 February 2017. Emily Powell, Hunter Howell, and Stephanie Clements. Verified by Leroy Nunez. Florida Museum of Natural History (UF 182309; photo voucher). Adult individual (122 mm SVL, 1.69 g) was found under woody debris from the invasive Melaleuca (*Melaleuca quinquenervia*). First county record in Miami-Dade County and southern range extension of this species in South Florida. The nearest record is from ca. 10 km to the north in Broward County (UF 151119). The surrounding habitat is dominated by non-native vegetation including large stands of Australian pines (*Casuarina* spp.) and Melaleuca. The persistence of this species within a largely modified habitat composed predominantly of non-native vegetation and surrounded by an urban matrix suggests that it may be able to persist within heavily modified landscapes in developed areas.

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***RHADINAEA HESPERIA* (Western Graceful Brown Snake)**. MÉXICO: NAYARIT: MUNICIPALITY OF SAN BLAS: Ecoaldea Ojo de Cielo (21.57812°N, 105.05755°W; WGS 84), 458 m elev. 2 July 2017. Jesús Alberto Loc Barragán. Verified by Guillermo Alfonso Woolrich-Piña. Amphibian and Reptile Diversity Research Center, University of Texas at Arlington (UTADC 9178; photo voucher). The snake was found in the afternoon crawling on leaf litter in oak forest. First municipality record, which fills a distributional gap in Nayarit between localities situated ca. 12 km to the northwest at Rancho la Noria, Sierra San Juan (Municipality of Xalisco; Luja and Grünwald 2015. Herpetol. Rev. 46:223–225), and 81 km to the southwest at a place on the southeastern slope of a high ridge north of the village of Mesa del Nayar (Municipality of El Nayar), on road to Santa Teresa (California Academy of Sciences [CAS] 169688).

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***SALVADORA DESERTICOLA* (Big Bend Patch-nosed Snake)**. MEXICO: COAHUILA: MUNICIPALITY OF OCAMPO: Rancho Pilares at El Mazón, Maderas del Carmen, 180 airline km NW from Nueva Rosita (28.88345°N, 102.70766°W; WGS 84), 1097 m elev. 25 August 2002. Equipo El Maderas del Carmen. Universidad Autónoma de Nuevo León (UANL 8142, adult male). Rancho Pilares at Campo Pilares, Maderas del Carmen, 173.5 air km NW from Nueva Rosita (28.84979°N, 102.65144°W; WGS 84), 1186 m elev. 20 September 2010. Equipo El Maderas del Carmen. UANL 8143 (adult female). First records for Coahuila, both verified by Larry David Wilson. The species is known from along the border in Brewster County, Texas, USA (Werler and Dixon 2000. Texas Snakes: Identification, Distribution, and Natural History. University of Texas Press, Austin, Texas. 437 pp.), and in Mexico the closest records are from the Municipality of Camargo, Chihuahua (Lemos-Espinal and Smith 2007. Anfibios y Reptiles del Estado de Chihuahua, México/Amphibians and Reptiles of the State of Chihuahua, Mexico. UNAM, CONABIO, México D.F. 613 pp.), but is expected to occur in Coahuila (Lemos-Espinal and Smith 2007. Anfibios y Reptiles del Estado de Coahuila, México/Amphibians and Reptiles of the State of Coahuila, Mexico. UNAM. CONABIO, México, D.F. 550 pp.). The snakes were found in a mixed Chihuahuan Desert microphyllous scrub and grassland community containing numerically dominant Creosote Bush (*Larrea tridentata*), Lechuguilla (*Agave lechuguilla*), Purple Prickly-pear (*Opuntia macrocentra*), Sotol (*Dasylirion berlandieri*), varius yuccas (*Yucca* spp.), and several native and invasive grass species. We thank the authorities of Rancho Maderas del Carmen and Cementos Mexicanos (CEMEX) for permitting us access to their collections. Fieldwork was partly funded by a grant (No. 445411) to Manuel Nevárez de los Reyes from CONACYT and Universidad Autónoma de Nuevo León (Programa de Apoyo a la Investigación Científica y Tecnológica [PAICYT CN315-15]). Collecting permit was issued to DL by SEMARNAT (OFICIO NUM.SGPA/DGVS/08374/16).

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***TANTILLA CUCULLATA* (Trans-Pecos Black-headed Snake)**. MEXICO: COAHUILA: MUNICIPALITY OF MÚZQUIZ: Coahuila Hwy 533 at La Cuesta de Malena (28.72621°N, 102.50342°W; WGS 84), 1519 m elev. 24 May 2018. Fabiola Baeza-Tarin, Sean P. Graham, Laine Giovanetto, and Tomas Hernandez. Verified by Toby Hibbitts. Herpetology Collection, Universidad Autónoma de Nuevo León, San Nicolas de los Garza (UANL 8275). First record for Coahuila (Lemos-Espinal and Smith 2007. Anfibios y Reptiles del Estado de Coahuila, México/Amphibians and Reptiles of the State of Coahuila, Mexico. UNAM, CONABIO, México, D.F. 613 pp.), a range extension of ca. 120 km E of the first Mexican record of this species in Chihuahua (Herr et al. 2017. Herpetol. Rev. 48:816), and a range expansion of ca. 80 km SE from the nearest known records in Brewster County, Texas, USA (Werler and Dixon 2000. Texas Snakes: Identification, Distribution, and Natural History. University of Texas Press, Austin, Texas. 437 pp.). This research was conducted on behalf of the Maderas del Carmen Flora and Fauna Protection Area under SEMARNAT collecting permit SGPA/DGVS/00047/18 issued to DL, with SPG listed as a subpermittee. We thank B. Pat, B. McKinney, J. Villalobos, H. Sotelo, and other members of the El Carmen conservation project for logistical support.

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THAMNOPHIS SIRTALIS **(Common Gartersnake)**. USA: TENNESSEE: Sullivan Co.: South Holston Weir Dam Wetland, across from Osceola Island Parking Area along Holston View Dam Road (36.5237°N, 82.1108°W; WGS 84). 7 May 2018. M. Kevin Hamed. Verified by A. Floyd Scott. David H. Snyder Museum of Zoology, Austin Peay State University (APSU 19881; photo voucher). New county record (Scott and Redmond 2008 [latest update: 9 June 2018]. Atlas of Reptiles in Tennessee. The Center of Excellence for Field Biology, Austin Peay State University, Clarksville, Tennessee; http://www.apsubiology.org/tnreptileatlas/; accessed 6 July 2018). This adult was found at the edge of a vernal pool and its eyes appeared cloudy suggesting it was about to shed. The nearest known locality is ca. 27 km NE in adjacent Johnson County near Backbone Rock Campground (National Museum of Natural History [USNM] 464681).

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TRIMORPHODON VILKINSONII **(Texas Lyresnake)**. MEXICO: COAHUILA: Municipality of Múzquiz: Coahuila Hwy 53 W of La Cuesta de Malena (28.72639°N, 102.54500°W; WGS 84), 1419 m elev. 21 May 2018. Fabiola Baeza-Tarin, Sean P. Graham, Laine Giovanetto, and Tomas Hernandez. Verified by Toby Hibbitts. Herpetology Collection, Universidad Autónoma de Nuevo León, San Nicolas de los Garza (UANL 8269). First record for Coahuila (Lemos-Espinal and Smith 2007. Anfibios y Reptiles del Estado de Coahuila, México/Amphibians and Reptiles of the State of Coahuila, Mexico. UNAM, CONABIO, México, D.F. 550 pp.), with the closest record located ca. 80 km to the northwest in Brewster County, Texas, USA (Werler and Dixon 2000. Texas Snakes: Identification, Distribution, and Natural History. University of Texas Press, Austin, Texas. 437 pp.; LaDuc and Johnson 2003. Herpetologia 59:364–374; Devitt et al. 2008. Copeia 2008:370–387). This research was conducted on behalf of the Maderas del Carmen Flora and Fauna Protection Area under SEMARNAT collecting permit SGPA/DGVS/00047/18, issued to DL, with SPG listed as a sub-permittee. We thank B. Pat, B. McKinney, J. Villalobos, H. Sotelo, and other members of El Carmen conservation project for logistical support.

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Two New Additions to the Terrestrial Reptiles Established on Bermuda, with Notes on Other Species

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Bermuda is an island archipelago totaling 54 km^2 in area, divided administratively into nine parishes, and situated in the Atlantic Ocean north of the Caribbean Sea and east of the USA (Fig. 1). The climate and natural history of Bermuda is heavily influenced by the Gulf Stream, which transports warm sea water and disperses biota from the Caribbean and southeastern coastal areas of the USA (Glasspool 1994; Meylan and Sterrer 2000; Grady et al. 2001; Parham et al. 2008). Despite its isolation, the overall endemism rate on Bermuda is low (ca. 3%) because of habitat loss and species extinction events associated with multiple Pleistocene sea-level fluctuations (Sterrer 1998). Colonization beginning during 1609 resulted in further dramatic changes to Bermuda's biodiversity, particularly with regards to exotic species and significant habitat modification as a result of human development (Sterrer et al. 2004). Currently, over 70% of Bermuda's land area is considered developed, the majority of which is used for residential housing (Anderson et al. 2001; Thomas 2004).

Bermuda has one endemic terrestrial reptile; the skink, *Plestiodon longirostris* (Brandley et al. 2010), which is believed to have been living on this archipelago from one to two million years

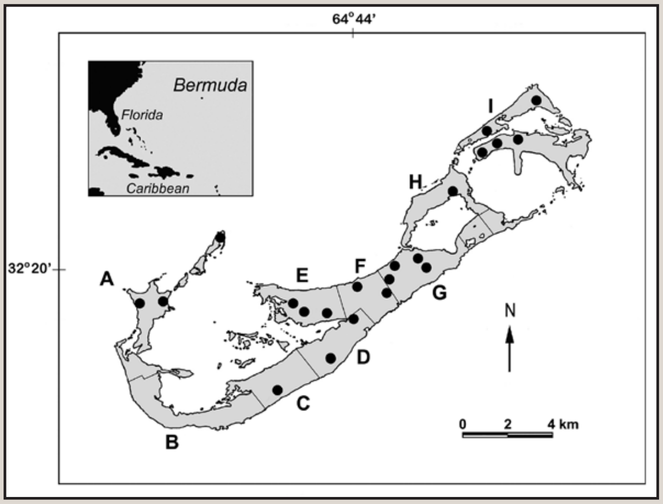

Fig. 1. Distribution map for *Hemidactylus mabouia* and *Hemidactylus turcicus* across Bermuda. A = Sandys Parish, B = Southampton Parish, C = Warwick Parish, D = Paget Parish, E = Pembroke Parish, F = Devonshire Parish, G = Smiths Parish, H = Hamilton Parish, and I = St. George's Parish.

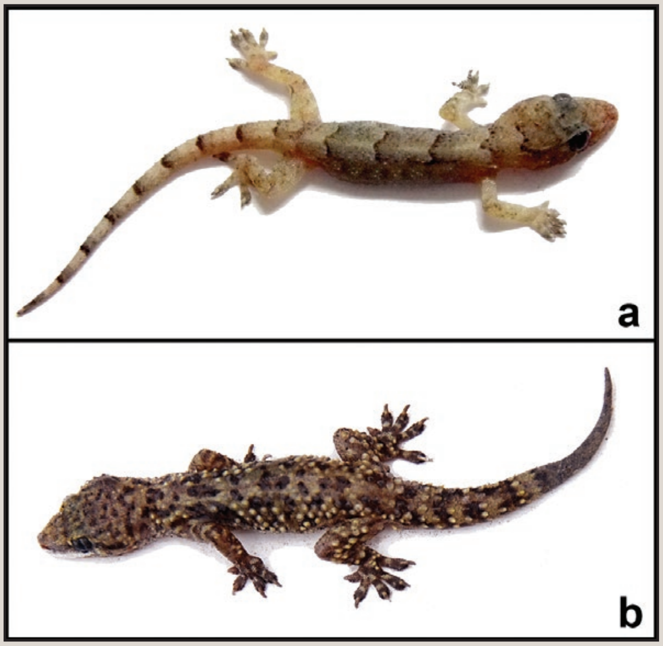

Fig. 2. Non-native geckos, *Hemidactylus mabouia* (a) and *Hemidactylus turcicus* (b) collected during haphazard nocturnal surveys on Bermuda in 2014 at the international airport in St. George's Parish (NHM 2014 289 005) and a plant nursery in Paget Parish (NHM 2014 289 009), respectively. Images not to scale.

(Olson et al. 2006), and four non-native species of dactyloid lizards that became established since the mid-twentieth century; *Anolis grahami, A. leachii, A. extremus* (Bacon et al. 2006), and *A. sagrei* (Stroud et al. 2017). A single record for *A. carolinensis* exists (Stroud et al. 2016), but it is not presently thought to represent an established population. Only *A. grahami* was intentionally brought to Bermuda; the remaining species of anoles were inadvertently introduced to the islands (see Wingate 1965; Stroud et al. 2017). *Plestiodon longirostris* is declining in abundance and geographical distribution within Bermuda, which has been attributed to habitat loss and predation by introduced species (Davenport et al. 2001; Bacon et al. 2006). Conversely, *A. grahami* and *A. leachii* have rapidly expanded across mainland Bermuda and many of its outlying islands (Losos 1996; Macedonia et al. 2016; Stroud, pers. comm.). *Anolis extremus* and *A. sagrei* both have very limited distributions for the time being. This report updates our knowledge on the distribution of lizards on Bermuda and increases the number of extant established terrestrial reptile species to seven.

On 26 November 2007, a gecko was captured in the arrivals area of the L. F. Wade International Airport, St. George Parish (32.36046°N, 64.70025°W; WGS 84). The specimen was accessioned into the Natural History Museum collection at the Department of Environment and Natural Resources (NHM 2008 261 018) and later verified by S. R. Smith, Curator for the Bermuda Natural History Museum, as a Tropical House Gecko, *Hemidactylus mabouia*, using Krysko and Daniels (2005). *Hemidactylus turcicus* (Mediterranean Gecko) was confirmed on Bermuda 6 March 2011 when a specimen was collected from a warehouse in the city of Hamilton, Pembroke Parish (32.29635°N, 64.78251°W; WGS 84) and given to the Natural History Museum collection (NHM 2011 202 007) and later verified by Smith. Successful propagation by *H. mabouia* was confirmed on 10 August 2011 when a clutch of 10 eggs was found under a metal paint can at the edge of the runway at the international airport. The eggs were transferred to the Department of Environment and Natural Resources where five subsequently hatched; the hatchlings were euthanized and accessioned into the Natural History Museum collection (NHM 2010 271 046).

Hemidactylus mabouia and *H. turcicus* are nocturnal lizards native to continental Africa and the Mediterranean basin, respectively; however, they have been introduced into many other parts of the globe, including several New World countries (Lever 2003). They are associated with human development and are highly adaptable, thereby becoming successful invaders throughout their non-native range. From November 2007 through December 2016, a total of 57 *Hemidactylus* geckos were reported from eight of the nine Bermuda parishes (Fig. 1) to the Department of Environment and Natural Resources by Bermuda's general public. Those reports included 49 physical specimens, four photo vouchers, and four from e-mail communications. Nineteen of the physical specimens were originally identified by us and verified by Kenneth L. Krysko as *H. turcicus* and 21 as *H. mabouia*. The remaining specimens were unavailable for examination. Of the records that have confirmed species identifications, 47% came from commercial and/or industrial properties, 36% came from the L. F. Wade International airport, 11% from private residences in St. George's, Hamilton, Smiths, Warwick, and Paget parishes; 6% had no locality information.

Haphazard surveys were subsequently undertaken from 9 September through 4 October 2014 at three locations where geckos had been reported: the international airport at the east end of Bermuda, the Royal Naval Dockyard at the west end in Sandys Parish, and a private plant nursery in the center of the main island in Paget Parish. Specimens were spotlighted and captured by hand during evening hours. Ten specimens were captured over a four-night period and identified; nine were *H. mabouia* (Fig. 2a) and one was a *H. turcicus* (Fig. 2b). Five *H. mabouia* were collected from the fortified wall surrounding the National Museum in the Dockyard and four on construction materials at the plant nursery. The one *H. turcicus* was found on an exterior wall of the Customs freight shed at the airport.

Yearly increases of frequencies of geckos reported by the general public averaged 0.7 geckos per year from 2007 through 2009, 5.7 geckos per year from 2010 through 2012, and 8.5 geckos per year from 2013 through 2016. Morphometric data from available specimens showed that there were all age classes present on Bermuda for both of these non-native species; SVL for *H. turcicus* ranged from 20–53 mm (mean 36.1 mm, N = 17) and SVL for *H. mabouia* ranged from 22–68 mm (mean 44.9 mm, N = 27).

Human-mediated dispersal is thought to be a major cause for the dramatic range expansion of some *Hemidactylus* species across their potential global ranges (Carranza and Arnold 2006; Locey and Stone 2006), so that is likely responsible for the introduction of *H. turcicus* and *H. mabouia* onto Bermuda. Presently these species are the only two members of the genus *Hemidactylus* known to occur here.

Acknowledgments.—We thank those members of the public who captured and deposited *Hemidactylus* geckos with the Department of Environment and Natural Resources during the past decade. We are also extremely grateful to Robbie Smith for assisting us with some of the nocturnal surveys, to Kenneth L. Krysko for confirming the identity of the *H. turcicus* and *H. mabouia* geckos from Bermuda, and to James Stroud for commenting on an earlier draft of this note. This is contribution #267 of the Bermuda Biodiversity Project (BBP), Bermuda Aquarium, Natural History Museum and Zoo, Department of Environment and Natural Resources.

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NATURAL HISTORY NOTES

CAUDATA — SALAMANDERS

***ANEIDES AENEUS* (Green Salamander). POST EGG DEPOSITION.** Egg deposition and hatch date have been documented in many states throughout the range of *Aneides aeneus*; however, a better understanding of geographic variation within this species requires more data from other areas. At 1000 h on 29 August 2017, we found three *A. aeneus* egg masses, approximately 30 cm apart from one another in a rock crevice within Mountain Bridge Wilderness Area, Greenville County, South Carolina, USA (precise locality withheld due to conservation concerns). Each egg mass was guarded by a brooding female *A. aeneus* (Fig. 1). Little is known about breeding habits of *A. aeneus* in South Carolina, and communal nesting behavior in *A. aeneus* has only been documented once (Gordon 1952. Am. Midl. Nat. 47:666–701). In the same crevice at 1100 h on 10 October 2017 (presumably the hatch date), we found five *A. aeneus* neonates (several of which were upside down). At 1200 h on 20 December 2017, we found one *A. aeneus* neonate climbing ~2 m up a dead beech tree sapling located ~2 m from the same rock outcrop. Neonates have been seen on trees in association with tree-nesting (Waldron and Humphries 2005. J. Herpetol. 39:486–292); however, we believe this observation may represent dispersal away from the rock outcrop, as described by Gordon (1952, *op. cit.*).

In West Virginia and North Carolina, *A. aeneus* deposit eggs in June and eggs hatch late August through September (Snyder 1971. Ph.D. dissertation, University of Notre Dame, South Bend, Indiana. 140 pp.; Canterbury and Pauley 1994. J. Herpetol. 28:431–434). In Kentucky and Mississippi, *A. aeneus* deposit eggs in mid to late July and eggs hatch 70–80 days later (Woods 1969. Ph.D. dissertation, University of Southern Mississippi, Hattiesburg, Mississippi.182 pp.; Cupp 1991. J. Tennessee Acad. Sci. 66:171–174). In South Carolina, *A. aeneus* appear to exhibit reproductive timing similar to *A. aeneus* in Kentucky and Mississippi (as eggs were first observed in late August and hatched in October). To our knowledge, these are the first nesting and developmental data for this rare species in South Carolina.


PHOTO BY DEREK F. THORNTON

FIG. 1. One of the three *Aneides aeneus* egg clutches guarded by a female salamander in the brooding crevice.

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***NOTOPHTHALMUS VIRIDESCENS* (Eastern Newt). DIET.** The diet of *Notophthalmus viridescens* consists primarily of terrestrial and aquatic invertebrates (MacNamara 1977. Herpetologica 33:127–132; Petranka 1998. Salamanders of the United States and Canada. Smithsonian Press, Washington, D.C. 587 pp.; Bliss et al. 2015. Herpetol. Rev. 46:609). Additionally, *N. viridescens* has been reported to consume vertebrate prey such as small fish, the eggs and larvae of amphibians (Petranka 1998, *op. cit.*), and carrion (Carlson 2014. Herpetol. Rev. 45:475). At 1600 h on 30 March 2018, I observed an adult *N. viridescens* consume a portion of an *Ambystoma maculatum* (Spotted Salamander) spermatophore within a vernal pool in Columbia County, Pennsylvania, USA (41.24040°N, 76.37049°W; WGS 84). Upon initial observation, the *N. viridescens* was within a shallow section of the vernal pool in which approximately 50 *A. maculatum* spermatophores had been deposited. The *N. viridescens* held the spermatophore in its mouth, and with a series of sharp, lateral jerking movements broke off and consumed a small portion of the spermatophore. This process was observed two more times during approximately five minutes of observation. The eggs and larvae of vernal pool-breeding amphibians may form an important, seasonal component of the diet of *N. viridescens* (Pitt et al. 2011. Herpetol. Rev. 42:263). This observation suggests the spermatophores of pond-breeding ambystomid salamanders might also be a seasonal component of the diet of *N. viridescens.*

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***OEDIPINA ELONGATA* (White-crowned Worm Salamander). ARBOREAL BEHAVIOR.** *Oedipina elongata* is a least concern species of worm salamander that occurs in tropical and subtropical wet forest of northern Chiapas, México, central and southern Belize, central and eastern Guatemala, and northwestern Honduras (Parra-Olea et al. 2008. http://www.iucnredlist.org/details/59312/0; 15 Jul 2018). At 2000 h on 20 October 2017, a subadult *O. elongata* was found on the leaf of a

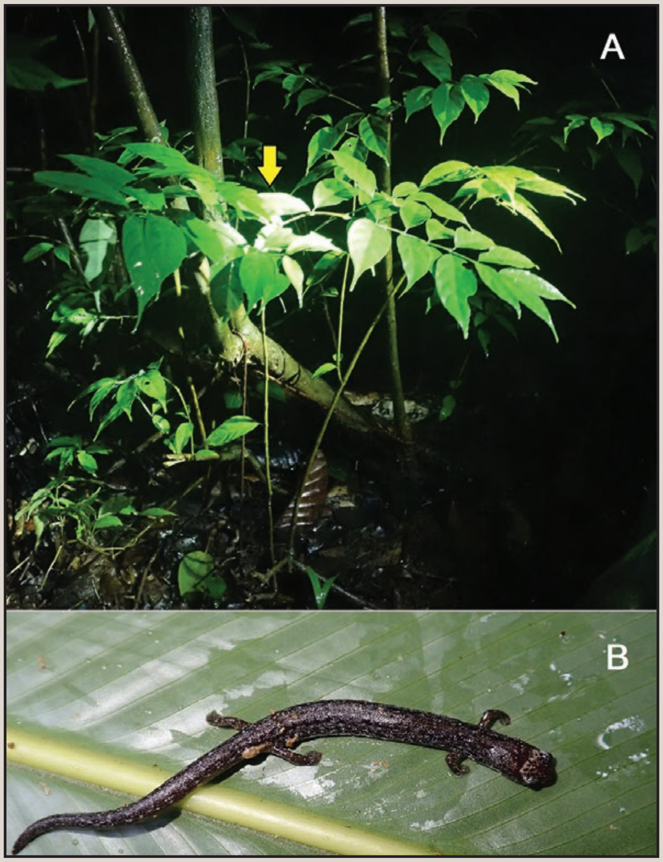

Fig. 1. *Oedipina elongata* arboreal behavior: A) location where the subadult *O. elongata* was found resting marked by the yellow arrow; B) *O. elongata* the day following the observation, posed for a photo on the plant.

plant (Piperaceae) approximately 1 m above the ground (Fig. 1) along a stream within the Cerro San Gil, Izabal, Guatemala (15.69056°N, 88.65269°W, WGS 84; 305 m elev.). This species is generally associated with moist microhabitats where there are abundant logs on the ground, and it is typically found in the channels of fallen logs, old termite nests on the ground, in leaf litter, in holes, and in tree stumps (Campbell 1998. Amphibians and Reptiles of Northern Guatemala, the Yucatán, and Belize. University of Oklahoma Press, Norman, Oklahoma. 400 pp.; Parra-Olea et al. 2008, *op. cit.*). Arboreal behavior in this species was reported by Townsend et al. (2006. Salamandra 42:61–62) in Honduras, where an adult *O. elongata* was found crawling on a broken branch within a log pile approximately 1 m above the ground; however, the authors suggested that the salamander could have been dislodged from one of the logs that they collected to make a wood fire, or to avoid the smoke and heat that their campfire radiated.

Arboreal behavior in other *Oedipina* is reported for *O. poelzi*, *O. pseudouniformis*, and *O. uniformis* (Brame 1963. Nat. Hist. Mus. Los Angeles Co. 65:3–12; Brame 1968. J. Herpetol. 2:2–64) but this is the first report of arboreal behavior without direct human intervention in *O. elongata*. This species may use the surface tension to climb to branches rather than specialized limbs and feet like arboreal salamanders (Wake 1987. Ann. Missouri Bot. Gard. 74:242–264). Facultative climbing *O. elongata* may be in response to variation in the availability of leaf litter versus arboreal prey (increased foraging potential), to increase detection of olfactory cues (increased chemosensory information), or to shelter in moss mats (McEntire 2016. Copeia 104:124–131).

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PLETHODON CINEREUS **(Eastern Red-backed Salamander). FLUORESCENCE.** Natural fluorescence is rare in terrestrial systems, and it has only been recently documented in amphibians (Taboada et al. 2017. Proc. Natl. Acad. Sci. 114:3672–3677). Between September and November of 2014, using a black light, I documented 45 individual salamanders exhibiting natural fluorescence during mark-recapture surveys. Five individuals were from one population in Centre County, Pennsylvania, USA (40.85670°N, 78.08580°W; WGS84), and the other 40 individuals were from Union County, Pennsylvania, USA (40.85570°N, 77.25500°W; WGS84), approximately 165 km away. Fluorescence occurred predominantly on the ventral side of the tail and around the cloaca, and they appeared as several dozen yellow-green dots (Fig. 1). Of the 45 fluorescent individuals caught, 33 were male and 12 were female. Of the total salamanders captured from that season, 18% of Union County and 6% of Centre County animals had fluorescent tails.

Animals were being marked using visual implant elastomer, an artificial fluorescent compound (see Muñoz et al. 2016. J. Herpetol. 50:570–581). Initially the tail markings were thought to

Fig. 1. A ventral view and the use of a common black light reveal several dozen fluorescent yellow-green markings on *Plethodon cinereus.*

be defective elastomer marks. Several pieces of evidence rule this out. Elastomer marks never break uniformly or into more than one or two small pieces, and no elastomer marks were given near the tail. Twenty-eight salamanders were previously marked with elastomer, but 24 of them did not have marks containing yellow elastomer—the only color that could have been confused with the natural fluorescence. Lastly, 17 of the individuals had never been previously marked and showed no evidence of marking (poorly injected marks still leave evidence at site of injection).

In treefrogs under twilight and nighttime light levels, fluorescence increased emergent light levels by 18–29%, and the extra light is hypothesized to aid treefrog vision (Taboada et al. 2017, *op. cit.*). Because salamander eyes are sensitive to green light (Chen et al. 2008. J. Photochem. Photobiol. 64:855–862), the compounds may be a similar adaptation to improve night vision (or subterranean vision), enabling enhanced vision of the surroundings or increased visibility of other salamanders. However, it is not known whether these compounds are generated by the salamander or are a byproduct of diet or infection. It is also unknown under what natural light conditions they fluoresce. Future lines of research should discern the source of the compounds and how common fluorescence is within the species. Even if this fluorescence is not important ecologically or evolutionarily, other scientists who use fluorescent marking techniques on salamanders should be aware that naturally fluorescent compounds can possibly create confusion.

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***PLETHODON YONAHLOSSEE* (Yonahlossee Salamander) and *PLETHODON GLUTINOSUS* (Slimy Salamander). INTERSPECIFIC COURTSHIP.** Courtship behaviors in intraspecific pairings of plethodontid salamanders have been well documented, though most of these observations have been made under laboratory settings (Pierson et al. 2017. Herpetol. Conserv. Biol. 12:1–15). In most plethodontids, courtship typically consists of various stages, beginning with the initiation of a behavior referred to as the tail-straddling walk, where a female follows a male while straddling his tail as it undulates and arches, until eventually he deposits a spermatophore which she collects into her oviduct (Arnold 1976. Ethology 42:247–300). Courtship can be disrupted and discontinued at any stage throughout this process.

Fig. 1. Courtship displays observed between *Plethodon yonahlossee* and *P. glutinosus*.

At approximately 2200 h on 22 September 2017, in Mitchell County, North Carolina, USA (36.09992°N, 82.28192°W, WGS 84; 930 m elev.), we observed courtship behavior involving the tail-straddling walk between a female *Plethodon yonahlossee* and a male *P. glutinosus* (Fig. 1; Video available at: https://youtu.be/3S8Ksosdoq4). Further, the male *P. glutinosus* appears to be in the "stationary with tail flexed" stage (described in Pierson et al. 2017, *op. cit.*), which is beyond the point at which most interspecific pairings often break down in a laboratory setting (Dawley 1986. Herpetologica 42:156–164; Kozak 2003. Southeast. Nat. 2:281–292). In Smyth County, Virginia, these species have been observed sympatrically, along with "probable" hybrid offspring, however this observation was not verified with genetic testing (Highton and Peabody 2000. *In* Bruce et al. [eds.], The Biology of Plethodontid Salamanders, pp. 31–93. Kluwer Academic/Plenum Publishers, New York).

To my knowledge, this is the first recorded occurrence of this interspecific pairing engaging in courtship behaviors, and could be indicative of potential hybridization between these species.

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ANURA — FROGS

***ALLOBATES FEMORALIS* (Pan-Amazonian Frog). ENDOPARASITES.** *Allobates femoralis* is a widely distributed frog in South America (Bárrio-Amoros et al. 2010. Check List 6:208–209). This species has diurnal and terrestrial habits and is commonly found in foliage (Simões et al. 2010. Zootaxa 2406:1–28). In the Neotropical region, the only species of helminths reported parasitizing frogs of the genus *Allobates* are *Cosmocerca podicipinus* in *A. femoralis* and *Cylindrotaenia americana* and *Physaloptera* sp. in *A. marchesianus* from Peru (Campião et al. 2014. Zootaxa 3893:1–93). In the present note, we provide a new host record for nematodes of the genus *Rhabdias*.

On 21 April 2017, a single specimen of *A. femoralis* was collected in the Cancão Municipal Natural Park, about 400 m E of the Amapari River and 2.5 km NW of the village of Pedra Preta, Municipality of Serra do Navio, Amapá, Brazil (0.90083°N, 52.01347°W; WGS84). As part of an unrelated study, the frog was dissected and examined for the presence of parasites. We found three nematodes infecting the lungs of the host and the helminths were fixed in 70% hot ethanol and cleared in Aman's lactophenol for light microscopic observation. The helminths collected in the lungs of *A. femoralis* were assigned to the genus *Rhabdias*, based on their morphology, site of infection, and known parasitism in anurans. Neither the frog nor the nematodes were deposited in a museum collection.

A study conducted by Kuzmin et al. (2016. Folia Parasitol. 63:015) reports *Rhabdias galactonoti* parasitizing the lungs of *Adelphobates galactonotus*, a dendrobatid frog, however, this study presents the first report of nematodes of the genus *Rhabdias* infecting anurans of the family Aromobatidae and for *Allobates femoralis* in the eastern Amazon.

Instituto Chico Mendes de Conservação da Biodiversidade granted permission to collect under license SISBIO #48102-2.

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***ANAXYRUS BOREAS* (Western Toad). HABITAT.** The vast majority of natural history data on *Anaxyrus boreas* is from inland and high elevation populations with little reference to coastal populations (Dodd 2013. Frogs of the United States and Canada. The John Hopkins University Press, Baltimore, Maryland. 460 pp.). Terrestrial habitat use by juvenile *A. boreas* is also poorly documented but this life stage is suspected to use wetland habitats during summer months in contrast to damp terrestrial or subterranean habitats used by adults (Muths and Nanjappa 2005. *In* Lannoo [ed.], Amphibian Declines: the Conservation Status of United States Species, pp. 392–396. University of California Press, Berkeley, California). Some of the terrestrial microhabitat cover reported in the Pacific Northwest includes logs, stumps, vegetation, loose soil, and rodent burrows (Olson 2005. *In* Jones et al. [eds.], Amphibians of the Pacific Northwest, pp. 162–165. Seattle Audubon Society, Seattle, Washington.). Here we report an observation of *A. boreas* in a Coastal Redwood (*Sequoia sempervirens*) forest utilizing an arboreal habitat.

At 1400 h on 8 August 2017, while searching cracks and crevices of old-growth redwood log cut-ends along a trail in the Lady Bird Johnson Grove, Redwood National Park, Humboldt County, California, USA (41.30342°N, 124.01813°W; WGS 84), we observed a juvenile (SVL ca. 30 mm) *A. boreas*. The log was located along a ridge, ca. 0.89 km to the nearest perennial stream. The toad was located at the opening of a crevice (opening: height = 20 mm, width = 45 mm, and depth ca. 280 mm). The crevice was 1.71 m above the ground and was the highest horizontal cavity on a 1.27-m diameter *S. sempervirens* log. The opening of the cavity was dry compared to the damp interior. Access to this microhabitat would have required the toad to climb. The toad presumably used the opening as a foraging site and demonstrated it provided suitable refuge habitat as it retreated from the opening and wedged itself into a narrow corner of the crevice while we inspected the dimensions of the cavity. This observation provides an example of an alternative above-ground summer foraging and refuge habitat for juvenile *A. boreas* in coastal old growth *S. sempervirens* forests compared to the subterranean habitats and cover types more commonly reported for the species elsewhere in the range.

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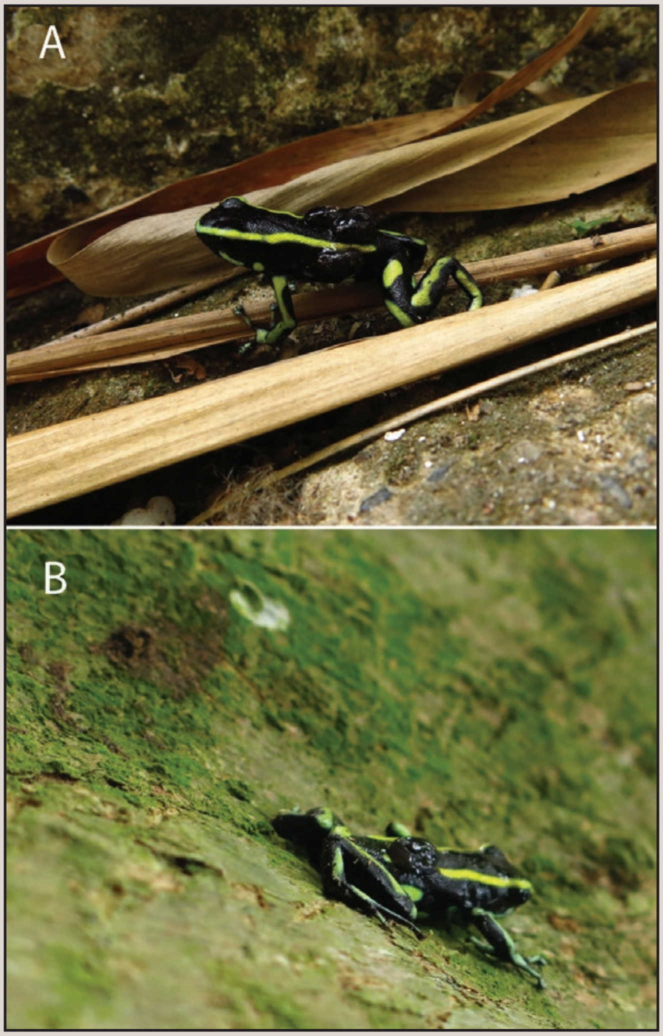
***DENDROBATES TRUNCATUS* (Yellow-striped Poison Frog). MALE PARENTAL CARE.** *Dendrobates truncatus* is a diurnal endemic Colombian species that occurs throughout the Caribbean lowlands of Colombia, towards the Magdalena River drainage, inhabiting both wet and seasonally dry forests from 70–120 m elev (Gualdrón-Duarte et al. 2016. *In* Kahn et al. [eds.], Aposematic Poison Frogs [Dendrobatidae] of the Andean Countries: Bolivia, Colombia, Ecuador, Perú and Venezuela, pp. 323–328. Conservation International, Arlington, Texas). Gualdrón-Duarte et al. 2016 (*op. cit.*) describe this species as reproducing year round but exhibiting a peak during the dry season. Females lay 1–8 eggs in the leaf litter on the ground or inside phytotelmata, and in captivity the species breeds inside artificial crevices provided by photographic film containers (Londoño and Tovar 2008. Int. Zoo. Yb. 42:71–77, Guayara-Barragan and Bernal 2012. Caldasia 34:483–496). In species of *Dendrobates*, tadpoles are transported by the male and deposited in forest pools or phytotelmata, where they fully develop without further parental care (Summers et al. 1999. Herpetologica 55:254–270). Observations of a closely related species (*D. auratus*) show that males carry tadpoles to water individually (Wells 1978. Herpetologica 34:148–155). The use of vertical microhabitats for breeding by *D. truncatus*, and the number of tadpoles a male can carry have not been previously reported.



PHOTOS BY MAURICIO BERNAL

FIG. 1. Male *Dendrobates truncatus* carrying tadpoles at Jardín Botánico de Cartagena, Turbaco Bolívar, Colombia during May 2015; A) male carrying three tadpoles on its back; B) male carrying a single tadpole on its back.

During May 2015, biologist Mauricio Bernal photographed two different males carrying one and three tadpoles on their backs; these males were observed during morning hours while being active on the forest floor (Fig. 1). An additional adult male *D. truncatus* was also photographed on 5 May 2015 at Jardín

PHOTOS BY MAURICIO BERNAL

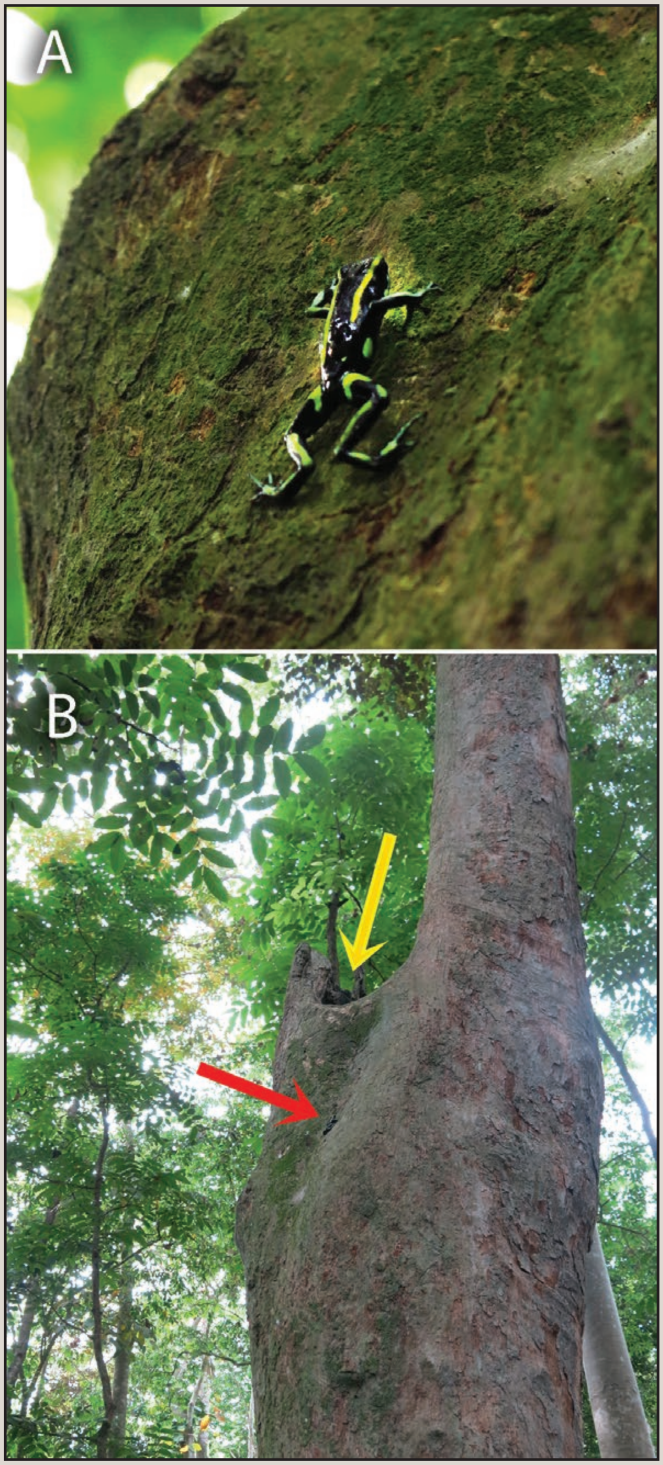


FIG. 2. A) Male *Dendrobates truncatus* climbing up a tree (*Brosimum alicastrum*) carrying a single tadpole; B) red arrow shows the male ascending the tree; yellow arrow shows the location of the pool inside the tree hole. Jardín Botánico de Cartagena, Turbaco Bolívar, Colombia, May 2015.

Botánico de Cartagena, Turbaco, Bolívar, Colombia (10.35447°N, 75.42803°W, WGS 84; 133 m elev). The frog was observed climbing the vertical surface of a Guaimaro (Breadnut) Tree (*Brosimum alicastrum*), carrying a single tadpole on its back (Fig. 2A). The tadpole was deposited in a small pool located inside a tree hole, 4 m above ground (Fig. 2B). Tadpoles from this species were also observed at this locality living in pools that formed inside holes on the buttress of *Ficus* trees, a few centimeters above ground. These observations suggest that the species may use a wide spectrum of vertical microhabitats to breed in, including small pools formed inside tree holes that are located several meters above ground. These microhabitats may play an important role in seasonally dry tropical forests, as they maintain a stable environment for the developing larvae.

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DUTTAPHRYNUS MELANOSTICTUS **(Asian Common Toad) and *RHACOPHORUS DENNYSI*** **(Chinese Flying Frog). HETEROSPECIFIC AMPLEXUS.** Heterospecific amplexus in anurans is well known (Marco and Lizana 2002. Ethol. Ecol. Evol. 14:1–8). The behavior has been suggested to arise due to a lack of available females for male anurans, such that they will engage in amplexus with almost anything, from dead conspecifics (Ayers 2010. Herpetol. Rev. 41:192–193), to the fingers of human observers, to inanimate objects (Streicher 2008. Herpetol. Rev. 39:75). There has been documentation of *Duttaphrynus melanostictus* being amplexed by another species (Reilly et al. 2016. Herpetol. Rev. 47:114), but this is the first record of *D. melanostictus* amplexing another species. Here we report on the case of heterospecific amplexus by *D. melanostictus* on *Rhacophorus dennysi.*

At 1438 h on 6 April 2016, a male *D. melanostictus* was observed engaging in axillary amplexus with a *Rhacophorus dennysi* (Fig. 1) of undetermined sex in Duijiang village, northwestern Guangxi province, China (25.32818°N, 110.27071°E, WGS84; 164 m elev.). Though both species are primarily nocturnal, during the breeding season *D. melanostictus* will continue calling and breeding throughout the daytime (pers. obs). However, *R. dennysi* is usually hidden in the daytime. One possible explanation for finding this pair in mid-day is that the *D. melanostictus* was not releasing its grip on the *R. dennysi* and was impairing its movement to a daytime refuge. Interspecific amplexus can lead to impairment and even death in some cases (Cheong et al. 2008. Anim. Cells Syst. 12:93–96). The pair was found at the base of a small karst mountain on the outskirts of an agricultural farm.

A photographic voucher of the behavior was deposited with HerpMapper.org (HM 151097 and 151098, http://www.herpmapper.org/record/151097).

FIG. 1. Male *Duttaphrynus melanostictus* amplexing *Rhacophorus dennysi* in northwestern Guangxi, China.

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***ELACHISTOCLEIS CESARII* (Oval Frog). DEFENSIVE BEHAVIOR.** Frogs present several strategies to avoid predation (Duellman and Trueb 1994. Biology of Amphibians. The John Hopkins University Press, Baltimore, Maryland. 670 pp.). One of these defensive strategies, known as "body-tilting" consists of inflating and tilting the body toward the predator displaying its glands, cutaneous secretions, or aposematic coloration (Toledo et al. 2011. Ethol. Ecol. Evol. 23:1–25). For the genus *Elachistocleis*, this behavior was previously observed in *E. erythrogaster* (Kwet and Solé 2002. Herpetol. Rev. 33:45) and *E. ovalis* (Kokubum and Menin 2002. Herpetol. Rev. 33:198). We report here for the first time body tilting defensive behavior performed by *E. cesarii*, a species native to Brazil (Caramaschi 2010. Bol. Mus. Nac. Rio de Janeiro. 527:1–30). Around 2200 h in August 2014, in the municipality of Guapó, Goiás, Brazil (16.87596°S, 49.45314°W, WGS 84; 1021m elev.), an individual of *E. cesarii* was found vocalizing in a swampy environment associated with a Cerrado phytophysiognomy known as "Vereda." After manipulation the frog inflated and elevated its body exposing its inguinal and femoral characteristic coloration (Fig. 1). The individual remained inflated and elevated for approximately two minutes. We also observed that the individual shifted its position as we moved, and thus oriented the display in our direction.

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Fig. 1. Defensive behavior of *Elachistocleis cesarii* recorded in the municipality of Guapó, Goiás State, Brazil.

***FEIHYLA HANSENAE* (Hansen's Bush Frog). MULTIMALE AMPLEXUS.** *Feihyla hansenae* (Rhacophoridae) is a nocturnal treefrog that breeds in temporary ponds in Thailand and parts of Cambodia during the rainy season (Taylor 1962. Univ. Kansas Sci. Bull. 43:526–529; Aowphol et al. 2013. Zootaxa 3702:101–123). Males attract females by calling from emergent vegetation or vegetation at the edge of the pond. After forming an amplectic pair, females will move around the vicinity and select the final location for oviposition (S. Poo, pers. obs.). Eggs are deposited in hemispherical gelatinous masses attached to vegetation overhanging the pond. Once the last egg is laid, males leave, while females remain, finish constructing the egg mass, and provide parental care by maintaining egg hydration (Poo and Bickford 2013. Ethology 119:671–679) and deterring egg predators (Poo et al. 2016. Biol. J. Linn. Soc. 118:901–910) until the eggs hatch and fall into the pond below. Here we report the first record of multiple males forming an amplectic group with a single female in this species.

On 18 September 2015 between 2100 and 2200 h, we observed four *F. hansenae* males attempting to mate with one *F. hansenae* female (Fig. 1) at a seasonal pond at the Sakaerat Environmental Research Station in northeastern Thailand (14.5090°N, 101.9537°E; WGS 84). When first observed, the female was in the process of laying eggs and constructing the gelatinous egg mass, with eggs visible both in the female's abdomen and on the grass blade beneath the female's vent. Of the four males, one male (Male 1) was in the normal, axillary amplectic position with the female, holding on to the female's dorsum (Fig. 1). The second male (Male 2) was positioned to the right dorsolateral side of Male 1 (Fig. 1). The third male (Male 3) was positioned dorsal inferiorly to Male 1 (Fig. 1). Finally, the fourth male (Male 4) was positioned laterally to the left of Male 1 and had all four limbs extended to wrap around the female, Male 1, and Male 3 (Fig. 1). Vents of all males were positioned in close proximity to the vent of the female, and the female continued to lay eggs and construct the egg mass by kicking up gel and foam with her hindlimbs. Males made slight movements with their limbs and body without changing their relative position to each other within the amplectic group. Seven minutes after observation started, Male 4 left the group by moving to the opposite side of the grass blade (relative to the female) for one minute, then jumping away (all observations rounded to the nearest minute). Similarly, one minute later, Male 3 moved to the opposite side of the grass blade, paused for one minute, and then jumped away. After Male 3 left, Male 2 repositioned itself to the dorsal side of Male 1, forming amplexus with Male 1. Male 1 and Male 2 maintained their positions until the last egg was laid. Eight minutes later, both Male 1 and Male 2 exited amplexus and moved to the opposite side of the grass. Male 1 paused for two minutes before jumping away, while Male 2 stayed for another minute before jumping away. The female remained at the oviposition site and continued to construct the egg mass with its hindlimbs.

We observed another multimale amplectic group nearby (~3 m away) at the same time, with two males attempting to mate with one female. One male was in the normal, axillary amplectic position with the female and was using its hindlimbs to kick or

FIG. 1. Female *Feihyla hansenae* laying eggs while in amplexus with four males.

push the second male away. The second male was positioned laterally to the amplectic pair with one arm on the female and another on the side of the grass blade. The vents of both males were positioned in close proximity to the female vent, and the female was in the process of laying its eggs. No further observations were made for this amplectic group.

Our observations of multimale amplexus occurred in a year where the onset of rainfall and accumulation of pond water was delayed in comparison to past years. At the time our observations were made (mid-September), water depth in the pond was less than 0.5 m compared to the average of 2.5 m in 2010–2013 (range = 1–5 m). The delay in heavy rain may have increased pressures on males to secure mates before the end of the breeding season. In comparison, multimale amplexus was not observed in over 200 night surveys performed in July–Oct from 2010 to 2013. To our knowledge, this is the first report of multimale amplexus in an arboreal-breeding species with a gelatinous egg clutch. Polyandrous behavior has been observed in the congeneric foam-nesting species *C. xerampelina* and *C. rufescens* (Coe 1974. J. Zool. 172:13–34; Jennions et al. 1992. Anim. Behav. 44:1091–1100), and in two other foam-nesting genera, *Polypedates*, and *Rhacophorus*, within the Rhacophoridae (Jennions and Passmore 1993. Biol. J. Linn. Soc. 50:211–220). In comparison, multimale amplexus appears to be more common in African rhacophorids compared to their Asian counterparts.

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LITHOBATES CATESBEIANUS (American Bullfrog). DIET. *Lithobates catesbeianus* is highly aquatic and occurs across North America in many types of mesic habitats (Bury and Whelan 1984. USFWS Resource Publication 155:1–26; Dodd 2013. Frogs of the United States and Canada, Volume 2. The Johns Hopkins University Press, Baltimore, Maryland. 982 pp.). Numerous studies have examined the diet of *L. catesbeianus*, which includes many invertebrate and vertebrate species. Herpetofauna in the diet includes salamanders, lizards, frogs, and a few snakes. At least seven species of snakes are known in the diet of *L. catesbeianus*: *Crotalus atrox*, *Lampropeltis getula*, *Rena dulcis*, *Micrurus fulvius*, *Nerodia* sp., *Thamnophis eques*, and *T. sauritus*; Dodd 2013, *op. cit.*). Prey size often is proportional to body size (Bruneau and Magnin 1980. Can. J. Zool. 58:175–183), so adult bullfrogs consume a larger variety of prey items of greater sizes.

Here, we report the first record of *L. catesbeianus* consuming a *Pituophis catenifer sayi* (Bullsnake). An adult *L. catesbeianus* (ca. 16.5 cm SVL) was captured in late August or early September 2015 on Hackberry Lake, Valentine National Wildlife Refuge, Cherry County, Nebraska (42.56153°N, 100.67891°W; WGS 84). A dietary study of *L. catesbeianus* from the refuge did not document snakes in the diet (Lingenfelter et al. 2014. J. N. Am. Herpetol. 2014:81–86). *Pituophis catenifer sayi* is distributed largely west of the Mississippi River from southern Alberta and Saskatchewan into Mexico and west of the Rocky Mountains (Ernst and Ernst 2003. Snakes of the United States and Canada. Smithsonian Books, Washington, D.C. 668 pp.). The total length of the predated *P. c. sayi* was ca. 40 cm, a size corresponding to a hatchling. Known predators of this snake species include mid-sized mammals and predatory birds, but previously, no species of frog has been documented to predate *P. catenifer* (Ernst and Ernst 2003, *op. cit.*).

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LITHOBATES SYLVATICUS (Wood Frog). PREDATION. *Lithobates sylvaticus* has the most extensive native range of any North American anuran and is common throughout this range (Martof and Humphries 1959. Am. Midl. Nat. 61:350–389). Typical avian predators of adult *L. sylvaticus* include wading birds, raptors, and ducks (Dodd Jr. 2013. Frogs of the United States and Canada Vol. 2. John Hopkins University Press, Baltimore, Maryland. 982 pp.). In June 2014 we witnessed an adult *Sterna paradisaea* (Arctic Tern) circling overhead and diving into a shallow, ephemeral wetland in the tundra and emerging with a single *L. sylvaticus* individual in its beak before flying away. This event occurred in Churchill, Manitoba, Canada, just a few kilometers inland from Hudson Bay (58.72919°N, 93.76882°W; WGS 84). This is the first record of *S. paradisaea*, a seabird, reported as predator of *L. sylvaticus*.

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***OSTEOCEPHALUS TAURINUS* (Slender-legged Treefrog). ENDOPARASITES.** *Osteocephalus taurinus* is widely distributed in Brazil, Suriname, Guiana, and French Guiana (Lima et al. 2005. Guide to the Frogs of Reserva Adolpho Ducke, Central Amazonia. Atema Design Editorial, Manaus, Brazil. 168 pp.). In the Neotropical region, helminths reported parasitizing *Osteocephalus taurinus* are: *Ochoterenella vellardi*, *Physalopteroides venancioi*, *Polystoma naponensis*, *Kentropyxia hylae*, and *Parapharyngodon politoedi* (Campião et al. 2014. Zootaxa 3893:1–93; Feitosa et al. 2015. Syst. Parasitol. 92:251–259; Santos et al. 2018. J. Helminthol. :1–6; doi:10.1017/S0022149X18000093). In the present study, we provide a new host record for nematodes of the genus *Rhabdias*.

Three specimens of *O. taurinus* were collected in the Cancão Municipal Natural Park, on the right bank of the Amapari River, Serra do Navio municipality, Amapá, Brazil (0.90083°N, 52.01347°W; WGS 84), during a survey of amphibians and reptiles and their associated parasites conducted in March 2018 (collecting permit SISBIO/ICMBio #48102-2). We found one specimen of nematode infecting the lungs of one *O. taurinus*. The nematode was rinsed in saline and fixed in 70% hot ethanol. For morphological analysis, the nematode was cleared with Aman's lactophenol for light microscopic observation. The helminth collected in the lungs of *O. taurinus* is assigned to the genus *Rhabdias*, based on its morphology (presence of a body covered by a cuticular inflation, esophagus claviform, with a buccal capsule, intestines filled with a dark content), site of infection, and known parasitism in anurans. Neither the frog nor the nematode were deposited in a museum. This study presents the first report of these nematodes infecting frogs of the genus *Osteocephalus*.

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***PELOPHYLAX SHQIPERICUS* (Albanian Pool Frog). ENDOPARASITE.** *Pelophylax shqipericus* is a ranid frog distributed from Skadar Lake in Montenegro to coastal regions of central Albania (Speybroeck et al. 2016. Field Guide to the Amphibians and Reptiles of Britain and Europe. Bloomsbury Natural, London, UK. 432 pp.). Little is known about its natural history and ecology (Uzzell and Crnobrnja-Isailovi 2009. http://www.iucnredlist.org/details/58715/0; 10 May 2018). In this note we report for the first time the occurrence of a helminth parasite in *P. shqipericus*. A parasitized frog was found among the 50 *P. shqipericus* individuals (males, females, and juveniles) sampled at a single locality: Nishaj (41.69°N, 19.59°E; WGS84), Lezhë district, northwestern Albania, on 26 April 2017. The frogs were assigned to the species on the basis of their morphological traits (Günther 1990. Die Wasserfrösche Europas. Die Neue Brehm-Bücherei, A. Ziemsen Verlag, Wittenberg Lutherstadt, Germany. 288 pp.; Plötner 2005. Die westpaläarktischen Wasserfrösche. Laurenti-Verlag, Bielefeld, Germany. 160 pp.).

A helminth was expelled from the cloaca of an adult *P. shqipericus* male during handling and was subsequently stored in 70% ethanol and shipped to CRB for identification. On the basis of its morphology (female; length 35 mm; body almost cylindrical, with slight widening toward anterior end; proboscis with 16 longitudinal rows of 5 hooks; eggs thin, fusiform, 0.13 mm in length, middle membrane of egg forming long, narrow protrusions at poles), the helminth was identified as *Acanthocephalus ranae* (Schrank, 1788) Lühe, 1911, Acanthocephala, Echinorhynchidae. The specimen is deposited in the Harold W. Manter Parasitology Laboratory, University of Nebraska, Lincoln, Nebraska, USA, as HWML 110367. *Acanthocephalus ranae* is a widely distributed species parasitizing the small and large intestines in European amphibians (Yildirimhan et al. 2006. Comp. Parasitol. 73:237–248), including water frogs (Günther 1990, *op. cit.*). *Pelopyhalax shqipericus* represents a new host for this parasite.

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***PHILAUTUS PETERSI* (Peters' Bush Frog). ENDOPARASITES.** *Philautus petersi* is an upland forest inhabiting species that occurs in central Peninsular Malaysia and is found throughout Borneo (Grismer 2011. Amphibians and Reptiles of the Seribuat Archipelago [Peninsular Malaysia]—A Field Guide. Edition Chimaira, Frankfurt am Main. 239 pp.). We know of no published reports of helminths in *P. petersi*. In our note we report the presence of one cestode and three species of Nematoda, thereby establishing the helminth list for this rhacophorid frog.

Eight *P. petersi* (mean SVL = 24.0 mm ± 6.8 SD, range = 17–36 mm) were collected by hand during 2004–2011 from Peninsular Malaysia and deposited in the herpetological collection (LSUHC) of La Sierra University, Riverside, California, USA and examined for helminths. By state, they were: Kedah (LSUHC 10475), Pahang (LSUHC 6124, 8363, 9104, 10250, 10663, 10698), Perak (LSUHC 9720). The frogs were euthanized by soaking in Tricaine Methanesulfonate, fixed in neutral-buffered 10% formalin, and stored in 70% ethanol. The body cavity was opened by a longitudinal incision and the digestive tract was removed and opened. The esophagus, stomach, and small and large intestine were examined for helminths under a dissecting microscope. Helminths were placed on a glass slide in a drop of lactophenol, a cover slip was added, and identification was made from these temporary wet mounts. Identifications of nematodes were made utilizing Anderson et al. (2009. Keys to the Nematode Parasites of Vertebrates, Archival Volume. CAB International, Wallingford, Oxfordshire. 463 pp.), Gibbons (2010. Keys to the Nematode Parasites of Vertebrates, Supplementary Volume. CAB International, Wallingford, Oxfordshire, UK. 416 pp.), and by comparisons to original descriptions. The cysticercoid was identified utilizing Roberts et al. (2013. Gerald D. Schmidt & Larry S. Roberts' Foundations of Parasitology, Ninth Edition. McGraw Hill, New York, New York. 670 pp.). Parasitology terms are according to Bush et al. (1997. J. Parasitol. 83:575–583).

Found were one cestode cysticercoid in the small intestine, prevalence = 13% and three species of Nematoda, *Cosmocerca ornata* (in large intestine), N = 1, prevalence = 13%, *Falcaustra purchoni* (in small and large intestines), N = 6, prevalence = 25%, mean intensity = 3.0 ± 2.8 SD, range = 1–5 and *Foleyellides malayensis* (in body cavity) N = 1, prevalence = 13%. Voucher helminths were deposited in the Harold W. Manter Parasitology Laboratory (HWML), The University of Nebraska, Lincoln, Nebraska, USA: cestode cysticercoid (HWML 99969), *Cosmocerca ornata* (HWML 99966), *Falcaustra purchoni* (HWML 99967), *Foleyellides malayensis* (HWML 99968).

Cysticercoids are larval forms of cyclophyllidean cestodes which occur in intermediate hosts; development to the adult occurs when the cyst is ingested (Roberts et al. 2013, *op. cit.*). *Cosmocerca ornata* is widespread and occurs in Europe, Africa, Malaysia, China, India, and South America (Baker 1987. Mem. Univ. Newfoundland, Occas. Pap. Biol. 11:1–325). However, Moravec and Kaiser (1994. Parasitol. Res. 80:29–32) reassigned the South American specimens to *Cosmocerca paraguayensis.* The report of *C. ornata* in the microhylid frog, *Chiasmocleis capixaba* from Brazil by Van Sluys et al. (2006. Brazil J. Biol. 66:167–173) should perhaps also be reassigned. Recent lists of *C. ornata* hosts are in Yildirimhan et al. (2009. Comp. Parasitol. 76:247–257) and Halajian et al. (2013. Comp. Parasitol. 80:80-95). Additional hosts for *Cosmocerca ornata* include *Cnemaspis mcguirei* (Bursey et al. 2014. Acta Parasitol. 59:643–652), three bufonids, *Duttaphrynus melanostictus, Ingerophrynus parvus*, and *Phrynoidis asper* (Goldberg et al. 2017. Pac. Sci. 71:367–375), and four ranids, *Chalcorana labialis, Hylarana erythraea, Pulchrana picturata*, and *Sylvirana mortensi* (Goldberg et al. 2017. Pac. Sci. 71:229–235). *Falcaustra purchoni* was described from the bufonid *Phrynoidis asper* (as *Bufo asper*) by Yuen (1963. J. Helminthol. 37:241–250) from Peninsular Malaysia. To our knowledge, *P. petersi* is the second host to harbor *F. purchoni. Foleyellides malayensis* was originally described as *Waltonella malayensis* by Petit and Yen (1979. Bull. Mus. Nat. d'Histor. Nat., Paris, Sect. A. Zool. 1:213–218), but was moved to *Foleyellides* by Esslinger (1986. Proc. Helminthol. Soc. Washington 53:218–223). It was previously found in frogs from Malaysia: *Pulchrana glandulosa* (as *Rana glandulosa*) by Petit and Yen (1979, *op. cit.*), *Limnonectes macrodon* (as *Rana macrodon*), *Amolops larutensis* by Mak and Yong (1981. Asian J. Trop. Med. Publ. Health 12:617–618), *Limnonectes blythii* (Goldberg et al. 2017. Pac. Sci. 71:535–540), and *Philautus vermiculatus* (Goldberg et al. 2017. Herpetol. Rev. 48:113).

Cestode cysticercoid, *Cosmocerca ornata, Falcaustra purchoni, Foleyellides malayensis* in *P. petersi* are new host records.

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***RANA UENOI* (Ueno's Brown Frog). LEECH PARASITISM.** Leech infestation on tadpoles and adult amphibians may lead to weakness, sickness, or death (Berven et al. 2001. Copeia 2001:907–915). There are only a few studies on the ectoparasites of amphibians in the Republic of Korea. Only the leeches *Parabdella quadrioculata* and *Torix tagoi* are known to parasitize Korean species of *Rana* (Sim et al. 2012. Korean J. Herpetol. 4:1–7), but mortality has not been recorded. It is however possible for leeches to kill adult frogs (Merilä et al. 2002. Ann. Zool. Fennici 39:343–346).

Fig. 1. *Rana uenoi* parasitized by leeches (*Torix tagoi*).

At 1525 h on 7 May 2017, we observed a *Rana uenoi* parasitized by leeches (*Torix tagoi*) at Yumyeong Mountain, Republic of Korea (37.58909°N, 127.49077°E, WGS84; Fig. 1). The individual was found on the edge of a slow-flowing stream. It is not unusual for this species to be found in this habitat during the day. At the beginning of the observation, the individual was alive (Fig. 1), but it died within minutes, and we handled it only to confirm death. We counted 30 *T. tagoi* on the skin of the dead *R. uenoi* (Fig. 1). They were mostly located on its lateral sides. Leeches were blood-feeding, leaving wounds on the frog. We did not preserve the leeches or the frog.

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***RANA UENOI* (Prevernal Frog). PREDATION.** *Rana uenoi*, previously considered part of *Rana dybowskii* (Matsui 2014. Zool. Sci.31:613–620), is widespread in the Korean peninsula (Borzée et al. 2016. Herpetol. Rev. 47:421), where it lives in multiple habitats and often breeds in rice paddies. There is anecdotal evidence of birds preying on *R. uenoi*; however, published data on specific predators are lacking. One general frog predator is *Mustela sibirica* (Siberian Weasel; e.g., Tatara and Doi 1994. Ecol. Res. 9:99–107; McDonald et al. 2000. J. Zool., Lond. 252:363–371), although the specific frog species preyed upon are undocumented. We found a road-killed *M. sibirica* on a country road between rice paddies in South Gyeongsang Province, Republic of Korea (34.54899°N, 126.72664°E, WGS84; 12 m elev.). Upon dissection of the weasel's stomach, we found frog remains. We identified the frog species through DNA extraction (DNeasy Tissue Kit; Qiagen, Valencia, USA) and PCR with the 16S primer pair for DNA barcoding from Jeong et al. 2013 (Mol. Ecol. Res. 13:1019–1032). The sequencing results identified the consumed frog as *R. uenoi*; a BLAST search comparison with data from Genbank showed 99.7% similarity. This is the first record of *M. sibirica* preying upon *R. uenoi.*

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***RHINELLA MIRANDARIBEIROI*. PREDATION.** *Rhinalla mirandaribeiroi* is a medium-sized bufonid belonging to the *R. granulosa* group found in the Cerrado Biome and in Cerrado enclaves in the Amazon (Narvaes and Rodrigues 2009. Arq. Zool. 40:1–73). Here, we report predation of *R. mirandaribeiroi* by *Leptodeira annulata* (Banded Cat-eyed Snake; Fig. 1). At 2050 h on 15 August 2014, in a remnant of cerrado vegetation within a rock outcrop in the municipality of Iporá, state of Goiás, Brazil (16.45175°S, 51.38838°W, WGS 84; 460 m elev.), we observed an individual of *R. mirandoribeiroi* being consumed by *L. annulata* (Fig. 1). Intake of the prey started from the head and the ingestion process lasted approximately 15 min. This is the first record of *R. mirandaribeiroi* being preyed by *L. annulata*. This record contributes to the increase of knowledge about the potential predators of this species, as well for the diet of *L. annulata*.

Fig. 1. *Rhinella mirandaribeiroi* being preyed upon by *Leptodeira annulata*.

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***RHINELLA MARINA* (Cane Toad). PREDATION BY A CROCODILE.** The invasive *Rhinella marina* has been introduced to many countries and islands worldwide (Lever 2001. The Cane Toad. The History and Ecology of a Successful Colonist. Westbury Academic and Scientific Publishing, Otley, UK. 230 pp.), including the Philippines where it was introduced as a biological pest control agent in the 1930s (Merino 1936. Philipp. J. Agric. 7:283–286). *Rhinella marina* has large parotid glands that produce bufotoxins, which can cause cardiac distress when consumed; therefore, it is assumed that few predators can safely consume adult *R. marina* (Toledo and Jared 1995. Comp. Biochem. Physiol. A 111:1–29), especially in newly *R. marina*-invaded habitat.

In Australia, the interaction between the *R. marina* invasion and crocodilian populations is extensively studied. In some locations, *Crocodylus johnstoni* (Freshwater Crocodile) populations displayed mass mortalities after *R. marina* invaded their habitat (e.g., Letnic et al. 2008. Biol. Conserv. 141:1773–1782; Britton et al. 2013.Wildl. Res. 40:312–317). In contrast, negligible impacts were observed in other populations (Doody et al. 2009. Anim. Conserv. 12:46–53; Somaweera and Shine 2012. Anim. Conserv. 15:152–163). In line with the latter, *C. porosus* (Estuarine Crocodile) has shown tolerance towards *R. marina* ingestion (Smith and Phillips 2006. Pac. Conserv. Biol. 12:40–49). Previous studies indicate that predator size may play a substantial role in *R. marina* tolerance (Smith and Phillips 2006, *op. cit.*), with intermediate-sized (0.6–1.5 m) crocodiles most at risk (Letnic et al. 2008, *op. cit.*; Britton et al. 2013, *op. cit.*). Here, we suggest that some individuals of the relatively small, critically endangered *C. mindorensis* (Philippine Crocodile) may prey on introduced *R. marina* without ill effects.

PHOTO BY S. P. TELAN

Fig. 1. A juvenile *Crocodylus mindorensis* with an individual *Rhinella marina* in its mouth, grasping it by the head and parotid glands.

The Mabuwaya Foundation regularly monitors breeding sites of *C. mindorensis* in the Sierra Madre mountain range on Luzon, the Philippines, since 2001 (van Weerd and van der Ploeg 2012. The Philippine Crocodile: Ecology, Culture and Conservation. Mabuwaya Foundation, Cabagan, Philippines. 152 pp.). All these sites are located in human-dominated landscapes and have been colonized by *R. marina*, which occurs in high densities (pers. obs.) and is the only member of the Bufonidae on Luzon (Diesmos et al. 2015. Proc. California Acad. Sci. 62:457–539; Brown et al. 2013. ZooKeys 266:1–120). One of the sites is Dinang Creek, a small tributary to the Ilaguen River with a narrow riparian forest zone (ca. 2–5 m) and otherwise surrounded by agricultural lands and grassland. In 2010, a juvenile *C. mindorensis* (intermediate-sized, ca. 1 m total length) was observed mouthing an adult *R. marina* (Fig. 1) in Dinang Creek in the municipality of San Mariano (16.79329°N, 122.04489°E; WGS84). It is unknown whether the crocodile consumed the toad. However, it is suggested that some *C. johnstoni* die from just mouthing *R. marina* (Somaweera et al. 2013. Anim. Conserv. 16:86–96). Neither a deceased toad nor crocodile were found in the subsequent two days, which suggests that the *C. mindorensis* killed and consumed the *R. marina* without ill effects. In all surveys and at all survey sites, no *C. mindorensis* mortality without human interference was recorded even though *R. marina* is common in these sites.

Our observation of an intermediate-sized *C. mindorensis* mouthing a *R. marina* with no observed ill effects suggests some individuals are tolerant to bufotoxin but we do not know how variable this tolerance is between individuals and how that might translate to tolerance at the population level. Coevolution with other bufonids may have resulted in bufotoxin tolerance.

Crocodylus mindorensis overlaps in distribution with native toads in Mindanao and Calauit Island (Diesmos et al. 2015, *op. cit.*), although the latter population is likely introduced from the Visayas (Tabora et al. 2012. Zootaxa 3560:1–31). Luzon Island, however, does not have any native toad species (Diesmos et al. 2015, *op. cit.*). If bufotoxin tolerance has evolved in *C. mindorensis* in Mindanao, this trait could have spread throughout the Philippines when the *C. mindorensis* population was still large and contiguous (van Weerd and van der Ploeg 2012, *op. cit.*).

The potential resistance to bufotoxin of *C. mindorensis* deserves further study, due to the possibility of heterogeneity of bufotoxin tolerance between and within populations, as is shown in *C. johnstoni* (Somaweera et al. 2013, *op. cit.*). However, no crocodilian mass mortality was recorded in relation to the range expansion of *R. marina* in the Philippines. Nevertheless, *C. mindorensis* remains severely threatened by anthropogenic impacts such as hunting and habitat loss (van Weerd and van der Ploeg 2012, *op. cit.*).

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TRACHYCEPHALUS MESOPHAEUS **(Golden-eyed Treefrog). MICROHABITAT.** *Trachycephalus mesophaeus* is an endemic species of the Atlantic Forest of Brazil and is generally associated with vegetation around temporary and permanent bodies of water (Haddad et al. 2013. Guia Dos Anfíbios da Mata Atlântica - Diversidade e Biologia. Anolis Books, São Paulo. 544 pp.), including inside bromeliads. Their eggs are deposited on the surface of temporary and permanent bodies of water (Prado et al. 2003. Bol. Mus. Nac., N.S., Zool. 510:1–11). Here we describe an observation of *T. mesophaeus* using the pitcher of the plant *Nepenthes ventricosa*, in a plant nursery greenhouse in the Atlantic Forest, Juquitiba, São Paulo, Brazil (23.54490°S, 46.59230°W; WGS 84). At 1200 h on 12 December 2017, a *T. mesophaeus* (SVL = 5 cm) was observed for the first time in a *Nepenthes* pitcher (opening = 4 cm, length = 14.4 cm; Fig. 1). Every time we approached, it retreated into the pitcher, with half of the body submerged. The liquid of the pitcher was full of dead invertebrates, and when the *T. mesophaeus* moved, there was a smell of decaying animals. The *T. mesophaeus* was seen in the pitcher for five days, and was last seen at 1340 h on 19 December 2017.

Carnivorous plants of the genus *Nepenthes* have leaves modified into pitchers. In the operculum (pitcher hood) there is a liquid used to attract vertebrates and invertebrates. The pitcher also contains liquid, which is responsible for digesting prey, however, these liquids are not able to kill all organisms and some animals use the pitcher as temporary or permanent habitat and for breeding (Adlassnig et al. 2010. Annal. Bot. 107:181–194). *Nepenthes ventricosa* is indigenous to the Philippines and lives in tropical forests, and can be found for sale in nurseries in Brazil. Some animals (e.g., amphibians) that use pitchers of carnivorous plants are opportunistic and can take advantage of the amount of prey attracted by the plant as a food source (Adlassnig et al. 2010, *op. cit.*). We did not observe the *T. mesophaeus* feeding, and instead we assume it was using the pitcher as a diurnal retreat.

Fig. 1. *Trachycephalus mesophaeus* inside the pitcher of *Nepenthes ventricosa.*

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TESTUDINES — TURTLES

CHELODINA BURRUNGANDJII **(Sandstone Snake-Necked Turtle). MAXIMUM SIZE.** *Chelodina burrungandjii* is a medium-sized, long-necked chelid turtle native to tropical northern Australia, where it inhabits lotic waters and associated pools in

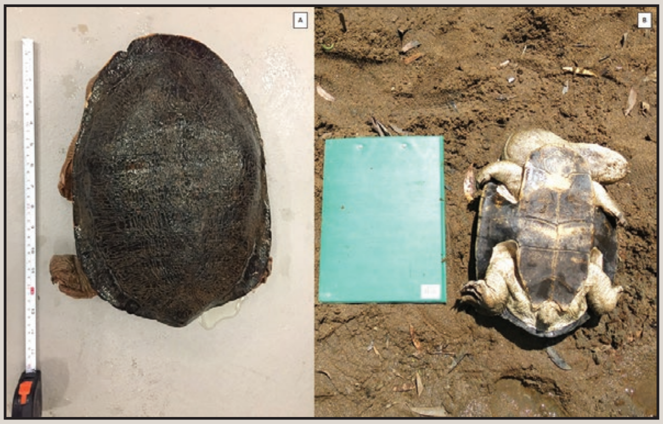

Fig. 1. Largest specimens of *Chelodina burrungandjii* on record.

Table 1. Size comparisons of different specimens of *Chelodina burrungandjii* from field measurements or museum collections. (CL - Carapace Length; CDL - Carapace Dome Length; CW - Carapace Width; CDW - Carapace Dome Width; TL - Total Length).

Origin	Current location	CL (cm)	CDL (cm)	CW (cm)	CDW (cm)	TL (cm)	Weight (kg)
-17.403875°, 124.910345° Captured during a survey by Department of Biodiversity, Conservation and Attractions on 10 May 2017 (Fig. 1B)	Voucher not collected	36	42	25.5	33.3	67	5.5
-17.41667°, 124.95° Collected by A. M. Douglas on 7 May 1966 (Fig. 1A)	WAM R26800	35.8	38.2	23.7	31	62	–

sandstone plateaus, and escarpments. The carapace is oblong with a recorded maximum size of 31.6 cm for a female from the Kimberley region of Western Australia (Thomson 2011. Chelon. Res. Monogr. 5:056.1–056.7). Here we report two individuals collected from Lennard River in the Kimberley that exceed the maximum size record (Table 1, Fig. 1).

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CHELONIA MYDAS (Green Sea Turtle). UNUSUAL TERRESTRIAL ACTIVITY AND OCCURRENCE. On 9 April 2018, a subadult female *Chelonia mydas* was encountered at 0625 h on Habhakhana, an Olive Ridley nesting beach, Kujanga Forest Range, Odisha, India (20.118666°N, 86.489888°E; WGS 84). The turtle was crawling towards the high tide line. I estimated the weight of the turtle at 20–22 kg; Curved Carapace Length and Curved Carapace Width were 48.7 cm and 40.3 cm, respectively, corresponding with the presumption that this was a subadult turtle. Its carapace was almost completely covered by what appeared to be filamentous algae. Barnacles also were evident on many parts of the turtle's body, particularly the head and front limbs (Fig. 1). Taken together, these observations suggest that this turtle may have been in poor health, as most Green Sea Turtles use behavioral means to limit algal and barnacle growth (Losey et al. 1994. Copeia 1994:684–690; Mettee 2014. Parasites. Marine Turtle Trauma Response Procedures: A Veterinary Guide. WIDECAST Technical Report No. 16. Accessed online 2 Aug 2018). The turtle was followed up to 27 m above the high tide line and observed near the Olive Ridley nesting beach.

Nesting of Green Sea Turtles has not been recorded along the Odisha coast. To the best of our knowledge this is the first time a female subadult Green Sea Turtle has been recorded from this Olive Ridley nesting rookery. Four species of sea turtles are reported to occur in the coastal waters of Odisha: the Leatherback (*Dermochelys coriacea*), Hawksbill (*Eretmochelys imbricata*), Green (*C. mydas*), and Olive Ridley (*Lepidochelys olivacea*) sea turtles, of which the Olive Ridley is by far the most common (Kar and Bhaskar 1982. *In* Bjorndal [ed.], The Biology and Conservation of Sea Turtles, pp. 365–372. Smithsonian Institution Press, Washington D.C.). However, only nesting by the Olive Ridley has been confirmed along the Odisha coast, a distance of about 480 km. The remaining three species are extremely rare in these waters. Juvenile Hawksbill Sea Turtles were recovered stranded on the sea coast near the Devi River mouth on three occasions and a juvenile Green Sea Turtle was recovered from a monofilament gill net at the Rushikulya rookery during a study conducted by Wildlife Institute of India (WII, 1999). A Leatherback Sea Turtle was found dead and washed ashore the Gahirmatha coast. Anecdotal information was received from fishermen on observations of Green Sea Turtles in the coastal waters off Odisha. However, information on the occurrence of Green Sea Turtles in this area is inadequate and systematic surveys on the potential occurrence and nesting of this species in the coastal waters and on the beaches of Odisha are needed.


PHOTO BY SRI MANAS KUMAR DASH

Fig. 1. Green Sea Turtle (*Chelonia mydas*) on Olive Ridley nesting beach, Odisha coast, India.

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CHELYDRA SERPENTINA (Snapping Turtle). DIET. The diet of *Chelydra serpentina* is opportunistic, omnivorous, and extremely diverse, and includes insects, spiders, isopods, amphipods, shrimp, crayfish, crabs, water mites, clams, snails, earthworms, leeches, tubificid worms, planarians, freshwater sponges, fish (eggs to adults), amphibians, small turtles, snakes, birds, and mammals, as well as plants (including algae, duckweed, cattails,

Table 1. Frequency of occurrence of primary food types in the intestines of 49 *Chelydra serpentina* from Island Lake, Garden County, Nebraska, USA, in June 2008. Size indicates range of maximum carapace length in mm measured by the Cagle Method (1946. Am. Midl. Nat. 36:685–729).

Size (CL)	Sex	N	Bird	Salamander	Fish	Crayfish	Snails	Algae
270–275	M	3	1	0	0	0	1	2
	F	3	0	0	0	0	3	2
275–300	M	2	1	1	2	1	0	1
	F	5	0	0	0	0	2	3
300–325	M	4	2	0	1	1	1	4
	F	5	0	0	1	1	0	4
325–350	M	1	1	0	0	0	0	1
	F	8	6	0	2	1	2	7
350–375	M	5	4	0	3	2	1	5
	F	4	2	0	0	1	0	3
375–400	M	4	2	0	1	0	0	4
	F	1	1	0	0	0	1	1
400–425	M	4	2	0	0	3	1	3
	F	0	–	–	–	–	–	–
ALL	M	23	13	1	7	7	4	20
	F	26	9	0	3	3	8	20
Total		49	22	1	10	10	12	40

Table 2. Comparison of frequency of occurrence (in percent) of major food items in gastrointestinal tracts of *Chelydra serpentina*. NR = not reported.

		Vegetation						Birds		
State	N	w/ algae	algae	Crayfish	Mollusks	Fish	Amphibians	game	nongame	Source
PA	19	26.3	10.5	63.1	36.8	10.5	5.2	[total = 5.2]		Surface 1908 (State Dept. Agric. 6:105–196)
NY/MA	18	61.1	11.1	27.8	22.2	27.8	5.6	[total = 5.6]		Pell 1940 (Copeia 1940:131)
CT	470	60	17.2	27	0.6	40.4	2.5	0.4	0.2	Alexander 1943 (*op. cit.*)
MI	281	>93.5	NR	NR	NR	>51.7	NR	[total = <7.2]		Lagler 1943 (Am. Midl. Nat. 29:257–312)
MI*	21	>88.2	>88.2	>30.8	>47.1	>52.9	NR	>15.4	NR [total > 29.4]	Lagler 1943 (*op. cit.*)
ME*	157	80.2	NR	NR	37.6	65.6	15.9	<15.3 [total = 29.4]	<15.9	Coulter 1957 (J. Wildl. Mgmt 21:17–21), 1958 (Maine Field Nat. 14:53–62)
NE	22	>68.2	>36.4	0	95.4	>27.3	0	8.1 [total = 22.7]	4.5	Hammer 1969 (J. Wildl. Mgmt. 33:995–1005)
FL	59	100	NR	83	100	0	94.9	0	0	Punzo 1975 (J. Herpetol. 9:207–210)
NE	49	81.6	81.6	32.7	24.5	20.4	2	0	45	This study

*Only high density waterfowl areas sampled

pondweeds, bulrush, and water lilies) and carrion (Ernst and Lovich 2009. Turtles of the United States and Canada. The Johns Hopkins University Press, Baltimore, Maryland. 840 pp.). Substantial evidence indicates that Snapping Turtles include a diversity of game waterfowl in their diet (reviewed in Davis and Buckland 2017. Herpetol. Rev. 48:174–175); however, it is unclear whether this predation is frequent enough to compromise the work of waterfowl management agencies. Under the assumption that predation rates must be high, some managers cull Snapping Turtles as a preventative strategy (Alexander 1943. J. Wildl. Mgmt. 7:278–282; among others). Clearly, more data are needed that quantify the actual rates of predation of these turtles on game waterfowl populations.

As part of a predator-control program, the Crescent Lake National Wildlife Refuge (Garden County, Nebraska, USA) removed 49 *C. serpentina* from Island Lake during June of 2008. With permission from refuge managers, we were allowed to dissect the carcasses of these turtles to examine their gut contents before disposal. Digestive tracts were removed and frozen for later study, and within two days the tracts were thawed, dissected, and the food types present were recorded (Table 1). Stomach contents were disregarded due to the possible presence of baitfish during trapping. Quantitative volumetric analysis was not possible due to time constraints.

Algae and bird material were the most frequent items found in the gastrointestinal tract (Table 1), although fish, snails, and crayfish were also common. A single neotenic *Ambystoma mavortium* was the only amphibian found. Algae was present in 40 of the 49 individuals (82%), and bird remains (primarily feathers) were found in 22 turtles (45%). Generally, the feathers found in the gut were from small, unidentifiable birds; all large identifiable feathers and/or legs or bones were from coots (*Fulica*). Birds increased in frequency in the diet when body size reached ca. 325 mm carapace length (Table 1). Only 4 of the 22 (18%) smaller turtles included bird remains whereas 18 of the 27 (67%) of larger turtles did ($\chi^2 = 26.5$; $P < 0.0001$).

A comparison of our data with previous studies (Table 2) confirms that Snapping Turtles are omnivorous and likely opportunistic in their diet, and hence diet varies considerably across their range. Except in habitats with dense waterfowl populations (Table 2), the frequency of game birds in the diet is quite low, supporting the statement by Breckenridge (1944. Reptiles and Amphibians of Minnesota. University of Minnesota Press, Minneapolis. 202 pp.) who noted: “It is probable that the seriousness of the snapper’s preying upon our waterfowl has been greatly exaggerated.”

We thank the staff at Crescent Lake National Wildlife Refuge for allowing us to salvage the gastrointestinal tracts from the culled turtles.

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CHRYSEMYS PICTA (Painted Turtle). MOUTH ANOMALY.

Chrysemys picta is a small omnivorous freshwater turtle common throughout much of the eastern and central United States (Carr 1952. Handbook of Turtles: The Turtles of the United States, Canada, and Baja California. Comstock Publishing Associates, Ithaca, New York. 542 pp.). There are few reports of anomalies in *C. picta* outside of occasional developmental malformations in juveniles, which may result in lower survival and low prevalence in adults. Facial deformities such as unfused premaxilla and maxilla bones, shortened jaws, or missing eyes in aquatic turtle embryos may have detrimental effects and are seldom observed in adults (Bell et al. 2006. Environ. Pollut. 142:457–465). However, proportions of injury from predation and deformity in other aquatic turtles such as the Northern Map turtle, *Graptemys geographica*, can be as high as 14% in some populations (Bennett and Litzgus 2014. J. Herpetol. 48:262–266) and recessed jaws have been observed in 3.8% of Snapping Turtles, *Chelydra serpentina* (Bishop et al. 1998. Environ. Pollut. 101:143–156). However, in *Chrysemys*, facial or jaw deformities have been observed to be relatively rare in studied populations, ~1% (Davy and Murphy 2009. Can. J. Zool. 87:433–439).

Fig. 1. A) Adult female *Chrysemys picta*, as found near a pond, showing facial abnormality; B) same individual, medial view of head, missing eyes and upper jaw, but no signs of recent trauma.

On 2 April 2018 at 1250 h, we observed an adult female *C. picta* (straight line carapace length = 133 mm, mass = 260 g) on the shore surrounding a small pond on the Wingate University Campus, Wingate, North Carolina, USA (34.9873°N, 80.4283°W; WGS 84) with facial deformities that included both eyes and the majority of the upper jaw missing and recessed (Fig. 1A). This individual *C. picta* was basking within five meters of the water and displayed normal behavior and movement upon our encounter and examination. Initially we assumed the turtle was recently injured but the individual showed no signs of recent trauma or injury and was released. We observed this same individual on two separate occasions (within one week) basking following the initial encounter. Therefore, we conclude this otherwise healthy individual may be able to forage successfully while lacking eyesight, diminished nostrils and the majority of the anterior upper jaw (premaxilla, maxilla, and prefrontal bones) as seen in Fig 1B. The observed anomalies could be developmental or result from previous injury (such as from mammalian predators or possibly even Snapping Turtles, *Chelydra serpentina*, often encountered in this same pond). However, despite these striking facial abnormalities, this turtle was apparently able to survive for an extended (but indeterminate) period of time. We thank North Carolina Wildlife Resources Commission for permits.

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CHRYSEMYS PICTA (Painted Turtle). BASKING BEHAVIOR.

It is not uncommon to find *Chrysemys picta* basking on natural structures extending from the water, including logs, rocks, small islands, sand bars, or the banks of water bodies (Ernst and Lovich 2009. Turtles of the United States and Canada. John Hopkins University Press, Baltimore, Maryland. 840 pp.). *C. picta* have also been observed basking on conspecifics and other turtle species, including *Chelydra serpentina* and *Apalone spinifera* (Moriarty and Hall 2014. Amphibians and Reptiles in Minnesota. University of Minnesota Press, Minneapolis, Minnesota. 370 pp.),

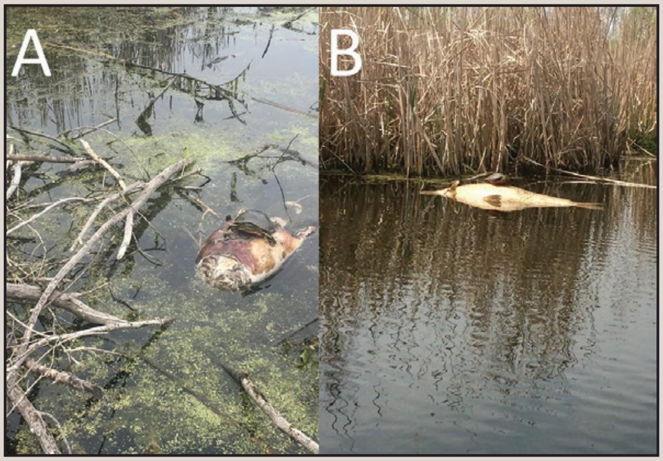

Fig. 1. A) *Chrysemys picta* basking on a dead *Castor canadensis.* B) *C. picta* basking on a dead *Cyprinus carpio.*

and even on the roofs of partially submerged automobiles (Ernst and Lovich 2009, *op. cit.*). At 1200 h on 14 May 2018, we observed *C. picta* basking on a dead *Castor canadensis* (American Beaver; Fig. 1A) and on a dead *Cyprinus carpio* (Common Carp; Fig. 1B) at Medicine Lake in Hennepin County, Minnesota, USA. To our knowledge, this is the first report of *C. picta* basking on animal carcasses.

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***CLEMMYS GUTTATA* (Spotted Turtle). HABITAT USE.** *Clemmys guttata* is known to inhabit a wide variety of wetland types, such as shallow grassy marshes, forest swamps, ponds, and even bays of large lakes (Ernst 1976. J. Herpetol. 10:25–33; Litzgus and Brooks 2000. J. Herpetol. 34:178–185; Stevenson et. al. 2015. Chelon. Conserv. Biol. 14:136–142). *C. guttata* populations inhabiting wetland complexes often display seasonal shifts in habitat use (Ward et. al. 1976. Herpetologica 32:60–64; Beaudry et. al. 2009. J. Herpetol. 43:636–645). Individuals have also been documented moving among wetlands over short time intervals, sometimes remaining within a wetland for only one day (Haxton and Berrill 2001. J. Herpetol. 35:606–614). Previous studies indicate that movement among wetlands typically involves walking through terrestrial habitat, sometimes traveling up to 250 m from water (Ernst, *op. cit.*; Litzgus and Brooks, *op. cit.*; Rasmussen and Litzgus 2010. Copeia 2010:86–96).

Few published studies have investigated the use of streams as habitat or as a factor influencing habitat connectivity for *C. guttata*. Individuals have been documented occupying slow-moving water systems such as drainage ditches and backwater areas of rivers (Stevenson et. al., *op. cit.*), and hibernating on the bottom of shallow (ca. 0.2 m) streams (Ernst 1982. J. Herpetol. 16:112–120). Streams with moderate or rapid flow rates bisecting wetlands could act as barriers to movement, but could also function as travel corridors among wetland patches. Here, we report radiotelemetry-based data showing that individuals in a *C. guttata* population often cross a moderate-flow stream while moving among wetland patches, as well as an observation of apparent use of the stream as a travel corridor.

In spring of 2018, we conducted a radiotelemetry study using six *C. guttata* in a 20-ha wetland complex in Hampshire County, West Virginia, USA (specific location withheld in compliance with state of West Virginia sensitive species data practices). The wetland complex consists of a matrix of seasonally flooded shallow grassy marshes, forest ponds, and dry upland grassland and forest. The wetland complex is bisected by a small, moderate-flow stream (2–4 m wide, 0.2–1.2 m deep, ca. 0.029 cm/s flow rate during the study period). *C. guttata* were outfitted with 3.6-g glue-on radiotransmitters (Advanced Telemetry Systems [ATS], Isanti, Minnesota), and tracked from 5 April to 7 May using a R410 scanning receiver (ATS) and 3-element folding yagi antenna. Each individual was located a minimum of three times per week.

During the study, five of the *C. guttata* made stream crossings to access additional wetlands. The females (N = 2) crossed the stream to access a wetland adjacent to the one previously occupied, whereas the males (N = 3) appeared to use the stream as a corridor to move to other wetlands. For example, on 1 May 2018, a male *C. guttata* was tracked into a shallow grassy marsh. On 3 May 2018, at ca. 1030 h, the same individual was tracked a straight-line distance of 0.04 km into the stream and located among a collection of wood and debris that had been trapped by a tree limb that had fallen across the water (Fig. 1). At ca. 1400 h on the same day, the individual was tracked 0.05 km downstream from its previously tracked location, into another wood and debris collection that had developed among the vegetation growing along the bank of the stream. On 4 May 2018, the individual was located in a temporary shrub-sedge wetland, a straight-line distance of 0.05 km from the previously tracked location. On 7 May 2018, the individual was then tracked back

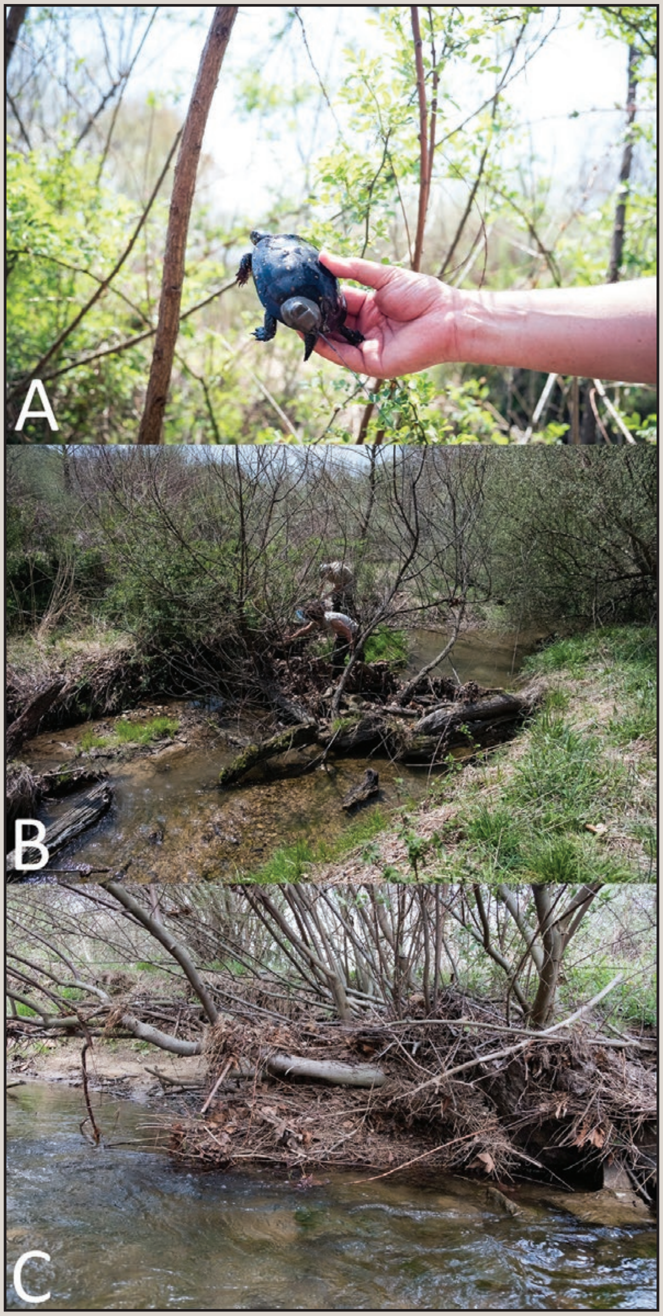

FIG. 1. A) Radiotracked *Clemmys guttata* captured in a stream on 3 May 2018 in Hampshire County, West Virginia, USA; B) Upstream detection location at ca. 1030 h; C) downstream detection location at ca. 1400 h. The individual moved 0.05 stream km between observations.

to the original shallow grassy marsh, 0.13 km from the previous location.

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***EMYDOIDEA BLANDINGII* (Blanding's Turtle). VOCALIZATIONS.** Traditionally it was generally presumed that all turtles were silent and deaf (Pope 1955. The Reptile World. Knopf, New York. 325+xiii pp.), and that any sounds they did produce were likely just noises made during breathing or nesting (Mrosovsky 1972. Herpetologica 28:256–258; Wever 1978. The Reptile Ear: its Structure and Function. Princeton University Press, Princeton, New Jersey. 1038 pp.; Cook and Forrest 2005. Herpetol. Rev. 36: 387–390). In 2009 this paradigm was broken by the description of purposeful vocalizations in *Chelodina colliei* (Giles et al. 2009. J. Acoustic Soc. America 126:434–443). The vocal repetoire of other species of freshwater and marine turtles has now been described, documenting the importance of underwater acoustic communication for both social and reproductive behavior (Ferrara et al. 2012. J. Comp. Psychol. 127:24–32; Ferrara et al. 2014a. Copeia 2014:245–247; Ferrara et al. 2014b. Chelon. Conserv. Biol. 13:110–114; Ferrara et al. 2014c. Herpetologica 70:149–156; Vogt 2014. The Tortoise 1:118–127; Ferrara et al. 2017 Copeia 105:29–32). For example, in *Podocnemis expansa*, postnatal parental care was documented using acoustic communication studies (Ferrara et al. 2012, *op. cit.*). Furthermore, several studies have demonstrated the ability of both marine and freshwater turtles to perceive aerial and underwater low-frequency sounds (Ridgway et al. 1969; Lenhardt et al. 1996. NOAA Technical Memorandum NMFS-SEFSC-387; Christensen-Dalsgaard et al. 2012. Proc. Royal Soc. B doi:10.1098/rspb.2012.0290). Although acoustic communication has been documented in marine turtles (above references) and the Pig-nosed Turtle (*Carettochelys insculpta*) (Ferrara et al. 2017, *op. cit.*), it has yet to be documented in other freshwater cryptodires. The purpose of this note is to document that another freshwater cryptodire, *Emydoidea blandingii,* also emits underwater vocalizations.

We recorded *E. blandingii* for 23 h during April 2009, where M. Pappas has been conducting a long-term population study of this species for the last 40 years, at McCarthy Wildlife Management Area near Weaver Dunes, Minnesota, USA. We initially recorded six individuals (three females and three males) for six hours in captivity to obtain a baseline of the sounds they were emitting (if they were emitting sounds, we needed to know the structure and frequency of the sounds so that we could detect these sounds in nature and distinguish them from the other environmental noises), and to help us adjust the recording equipment for recording these frequencies. We then recorded wild turtles for a total of 17 h during four sessions within the hours of 0900–1200 over a four-day period, in a vernal pool where Blanding's Turtles come year after year to court and copulate in the McCarthy Wildlife Management Area. We chose this area because it is one of the few times and places during the year where we are certain to find the turtles, and, presumably, they are more likely to produce sounds when they are in the presence of other turtles than if they are alone. All sound recordings were made using a Fostex FR-2 recorder adjusted to 48 kHz at 24 bits. The underwater recordings were made with a Reson (TC4043) omnidirecional hydrophone with sensitivity of 2 Hz-100 kHz ± 3 dB. Airborne sounds were recorded using a Sennheiser K6 unidirectional microphone with a Sennheiser ME-66 windscreen. While recording at the surface of the water, the microphone was positioned 30 cm above the water and pointed towards a floating log 40 cm away where the turtles were frequently noted basking (Figs. 1, 2), to capture the sounds as the heads of the turtles were breaking the surface and during basking. We inserted the hydrophone 0.5 m from the bottom of the pool (depths varied from 1 to 1.5 m) and 40 cm from the bank of the pool. We monitored the recordings in real time using Sony MDR-7506 headphones and adjusted the recording level manually to maximize the signal-to-noise ratio and to prevent distortions ("clipping") caused by excess gain.

FIG. 1. Adult *Emydoidea blandingii* on basking log in the breeding vernal pool in Hastings Wildlife area. Note male with inflated throat and elevated head posture.

FIG. 2. Adult *Emydoidea blandingii* on basking log in the breeding vernal pool in Hastings Wildlife area. Note male with open mouth posture.

Raven Pro 1.3 (Cornell Laboratory of Ornithology) was used to analyze the recordings using the following spectrographic parameters: window type—Hamming; window size—512 samples. Sounds with similar characteristics of published turtle sounds (Giles et al. 2009, *op. cit.*; Ferrara et al. 2012, *op. cit.*) and within the hearing range of turtles (Ridgway et al. 1969. Proc. Nat. Acad. Sci. 64:884–890) were detected manually by two experienced researchers using visual and aural inspection of the recordings.

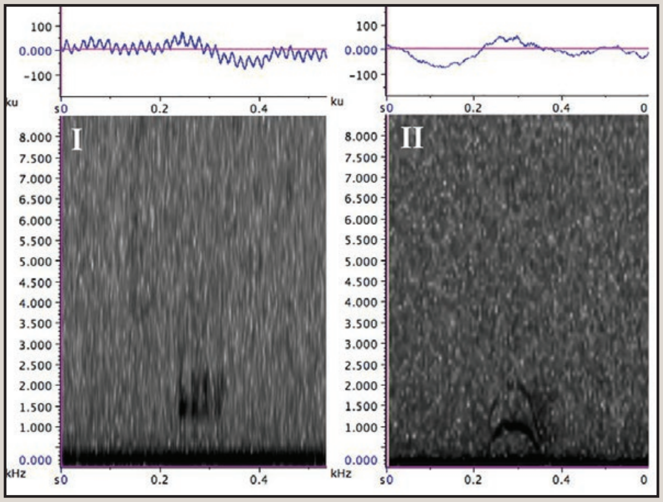

FIG. 3. Waveform and spectrogram views of the sounds produced by *Emydoidea blandingii* (window type: hamming, window size, and FFT = 512).

We detected 12 sounds produced by *E. blandingii* from the pond during the 17 hours we recorded. The lowest value recorded for peak frequency was 140 Hz and the highest was 3,656.2 Hz (mean = 1281.0 Hz and sd = 965.1). It is too early to describe the vocal repertorie of *E. blandingii*, but the sounds we found suggest a preliminary classification into two types according to their acoustic and spectral characteristics (Fig. 3). All sounds detected included sounds with harmonic and non-harmonic frequency bands modulated in amplitude and frequency and noisy aural quality.

Type I (N = 4).—Multiple frequency bands not harmonically related resulting in a noisy aural quality. The mean peak frequency was 2132.8 Hz and varied from 1500 to 3656.2 Hz, and the total duration varied from 0.03 to 0.10 s.

Type II (N = 8).—Is the most common sound in the repertoire of *E. blandingii*. These are harmonic and non-harmonic frequency bands with frequency modulated. Duration of the sound varies from 0.025 to 0.091 s, the peak frequency from 2062.5 to 140.6 Hz, and the number of harmonics from 2–10.

We wondered whether turtles with mouths open and gular region inflated (shown in Figs. 1 and 2), were vocalizing in the air, however we not did detect any airborne vocalizations in our recordings. It is possible they were emitting sounds in the infra- or ultra- sound range, rather than the frequencies we were monitoring, but we have never, to date, found turtles emitting sounds in the infra- or ultra- sound range in any of the marine turtles or pleurodires we have recorded.

Knowledge of the range of the vocal repertoire of species from different taxonomic groups is essential to understanding the evolution of the complexity of animal communication. Recordings of adult *E. blandingii* demonstrate that this species makes vocalizations with different structural characteristics that included harmonic and non-harmonic structures, as has been described for other species of freshwater and marine turtles such as *Dermochelys coriacea* (Ferrara et al. 2014b, *op. cit.*), *Chelonia mydas* (Ferrara et al. 2014a, *op. cit.*), *Carettochelys insculpta* (Ferrara et al. 2017, *op. cit.*), *Chelodina colliei* (Giles et al. 2009, *op. cit.*), and *Podocnemis expansa* (Ferrara et al. 2012, *op. cit.*).

Even though the number of samples and the diversity of sound types in the vocal repertoire of *E. blandingii* noted here was not as extensive as described for other aquatic turtles, it is sufficient to demontrate that this species is vocalizing underwater. Vocal repertoire surveys are important to document the taxonomic breadth of underwater vocalizations for comparative study with other species of turtles. We hope that these results will stimulate more bioacoustic studies of different life history stages of additional species of turtles, to elucidate the significance of underwater vocalizations in turtles.

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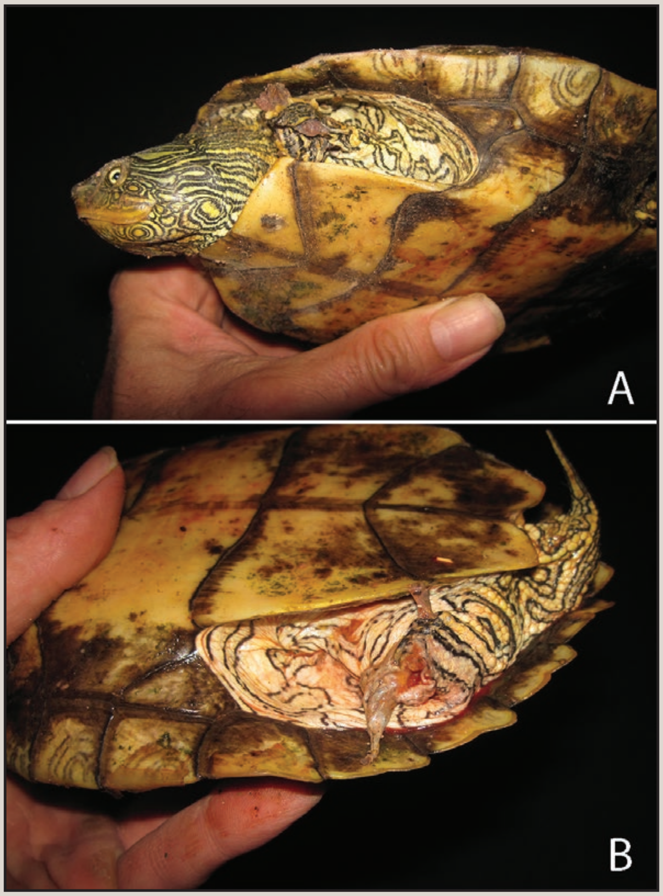
***GRAPTEMYS VERSA* (Texas Map Turtle). PREDATION.** Raccoons (*Procyon lotor*) are known predators for many turtle species and their eggs. Herein I report the first documentation of predation on *Graptemys versa* by a raccoon (Lindeman 2013. The Map Turtle and Sawback Atlas Ecology, Evolution, Distribution and Conservation. Oklahoma University Press, Norman. 460 pp.). On 20 May 2014 at 2247 h in Menard County, Texas, USA, at the San Saba River and Dunagan Road (30.86743°N, 100.02180°W; 616 m elev.), an adult female *G. versa* was out of the water and on the edge of Dunagan Road when it was encountered by a raccoon. I had just waded a short distance into the river from the aforementioned location when I heard a noise coming from the road and saw the raccoon attacking the turtle. Within a span of



PHOTOS BY CARL J. FRANKLIN

FIG. 1. A) Deceased *Graptemys versa* missing left front leg due to raccoon attack; B) showing missing right rear leg.

approximately two minutes the raccoon had already bitten and chewed off the turtle's left front leg and right rear leg before I was able frighten it away, causing it to drop the turtle (Fig. 1). The turtle died minutes later. The turtle and surrounding area on the road was dry suggesting that she was likely returning to the water when captured. Digital probing inside the body cavity post-mortem did not reveal any shelled eggs. Given the lack of palpable eggs and time of terrestrial activity, she may have been returning from nesting. Voucher photographs documenting the trauma were deposited in the digital collection at the University of Texas at Arlington Amphibian and Reptile Diversity Research Center (UTADC 9209, 9210; Fig. 1).

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***MACROCHELYS TEMMINCKII* (Western Alligator Snapping Turtle). DEFENSIVE BEHAVIOR.** Alligator Snapping Turtles are well known for being able to defend themselves with a massive gaping mouth, large sharp beak, and a powerful bite. Adult specimens may occasionally lunge or move towards a perceived nuisance with their mouths wide open (Allen and Neill 1950. Spec. Publ. Ross Allen's Rept. Inst. 4:1–15). They are also renowned for being one of the largest species of freshwater turtles in the western hemisphere, with adult males achieving a mass of 113 kg (Pawley 1987. Bull. Chicago Herpetol. Soc. 22:134) and a straight carapace length of 80 cm (Pritchard 1980. Chelonologica 1:113–123). Herein we describe a previously unreported defensive behavior for a juvenile *Macrochelys temminckii.*

On 1 June 2018 at ca. 0900 h, we captured a juvenile specimen in at the Trinity River at Arlington, Tarrant County, Texas, USA. The specimen's general measurements were: carapace length = 13.7 cm, carapace width = 10.5 cm, shell height = 4.6 cm, plastron length = 8.7 cm, post cloacal tail length = 11 cm, and mass = 366 g. At approximately 0945 h the turtle was being positioned for photographic documentation (UTADC 9202). After the turtle was placed into a photogenic position it attempted to escape, whereupon the assistant repositioned it for the photographer. Upon the third attempt to escape, the assistant grabbed the posterior portion of the carapace and the turtle forcibly snapped its mouth shut while lunging in a counter-clockwise direction resulting in it spinning around and changing its entire body position by 180°. This resulted in the gaping mouth facing the assistant. Carefully, the assistant repositioned the turtle for the camera and once again it performed the previously described movement upon being contacted by the posterior portion of the carapace. Neither of these episodes were timed or captured on video, leaving us to speculate on the precise amount time required for the movement (estimated < 1 sec) and whether or not the turtle's body left the ground during the behavior. This defensive behavior would presumably be limited to young turtles due to their lighter mass. This described action could be an effective deterrent to a predator attempting to contact the turtle from the posterior of the carapace. Field work conducted under TPWD SPR-1017-201.

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***MALACLEMYS TERRAPIN CENTRATA* (Carolina Diamondback Terrapin). NESTING BEHAVIOR.** *Malaclemys terrapin centrata* is the only one of the seven recognized subspecies of *M. terrapin* known to occur along the Georgia, USA, coast. Females emerge in late spring or summer in search of appropriate nesting sites, which include virtually any area with sandy soil. Nesting sites selected by this species include sandbars, spits, dunes, and creek banks provided the site is adjacent to marsh habitat and above the high tide line (Jensen et al. 2008. Amphibians and Reptiles of Georgia. University of Georgia Press, Athens, Georgia. 575 pp.). Here we describe the observation of a female *M. t. centrata* nesting in a Nine-banded Armadillo (*Dasypus novemcinctus*) burrow, which to our knowledge has not been previously documented in the literature.

On 17 June 2018 at 1204 h, an adult female *M. t. centrata* was documented nesting in a burrow via a remote infrared camera (Bushnell®, Overland Park, Kansas, USA) on Little St. Simons Island, Georgia, USA (Fig. 1). Cameras were positioned to capture photos and video of the commensal use of armadillo burrows by other species. The burrow was located in the dune system on the northeast end of the island (31.27179°N, 81.27753°W; WGS 84, 1 m elev.) and was approximately 170 m east from the nearest tidal creek and 370 m landward of the Atlantic Ocean. The camera recorded eight pictures and four 10-sec videos of the *M. t. centrata* using the burrow, illustrating that the individual turtle had spent approximately 37 min nesting inside the burrow (Fig. 1). Photos and video show that the nest was depredated a little over 10 h later by a Northern Raccoon (*Procyon lotor*) at 2257 h. Previous research has shown that terrapin nests are regularly preyed upon within 24 h of oviposition and that raccoons are the most significant predator of these nests (Butler et al. 2004. Am. Midl. Nat. 152:145–155).

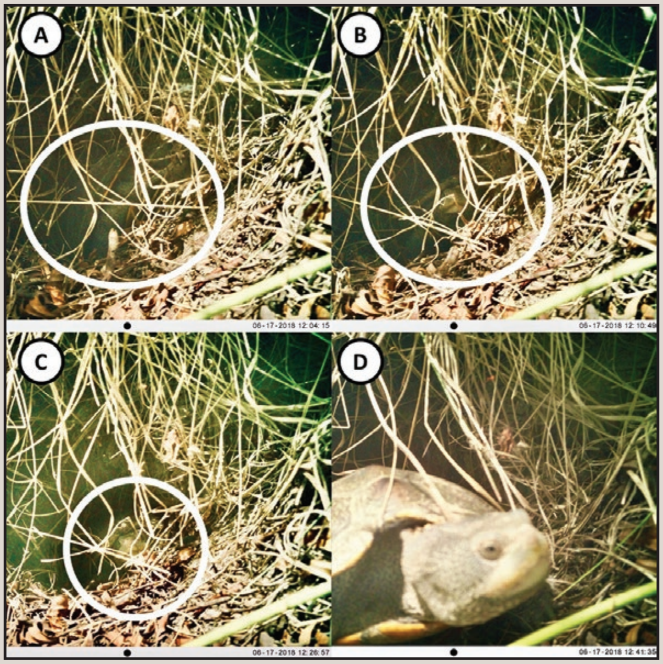

Fig. 1. The sequence of a female *Malaclemys terrapin centrata* nesting in an armadillo burrow on Little St. Simons Island, Georgia, USA on 17 June 2018. A) *M. t. centrata* entering the burrow at 1204 h; B) *M. t. centrata* digging in the burrow at 1210 h; C) *M. t. centrata* presumably nesting in the burrow at 1226 h; D) *M. t. centrata* leaving the burrow at 1241 h.

This observation is the first direct record of *M. t. centrata* using a burrow of a Nine-banded Armadillo. This is significant because armadillo burrows are documented providing shelter and forage for a variety of herpetofauna, including the Eastern Diamondback Rattlesnake (*Crotalus adamanteus*; Means 2017. Diamonds in the Rough: Natural History of the Eastern Diamnondback Rattlesnake. Tall Timber Press, Tallahassee, Florida. 390 pp.), Eastern Kingsnake (*Lampropeltis getula*; Steen et al. 2010. Copeia 2010:227–231), Furrowed Wood Turtle (*Rhinoclemmys areolata*) Neotropical Rattlesnake (*Crotalus durissus*), Tropical Rat Snake (*Spilotes pullatus*), Boa Constrictor (*Boa constrictor*), and Basilisk lizard (*Basiliscus vittatus*) (Platt et al. 2004. Mamm. Biol. 69:217–224). Additional reptile species found using armadillo burrows during this study that have not previously been documented include Coachwhip snakes (*Masticophis flagellum*) and Black Racers (*Coluber constrictor*). These observations indicate that armadillo burrows might function as important refugia for herpetofauna by facilitating protection against temperature extremes, fire, and predation. Furthermore, our observation suggests that these burrows provide suitable soil conditions for *M. t. centrata* nesting when located adjacent to the marsh habitats in which they reside. Additional research is needed to understand whether armadillo burrows could provide ideal nesting habitat for *M. t. centrata* in areas where they co-occur.

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***PSEUDEMYS GORZUGI* (Rio Grande Cooter). KYPHOSIS.** Kyphosis and kyphoscoliosis are deformations of the spine that can cause doming of the carapace in turtles. These two forms of shell deformities have been reported in many chelonian species, including *Trachemys scripta elegans* (Tucker et al. 2007. Herpetol. Rev. 38:337), *Emydura macquarii krefftii* (Trembath 2009. Chelon. Conserv. Biol. 8:94–95), *Podocnemis erythrocephala* (Bernhard et al. 2012. Herpetol. Rev. 43:639), *Graptemys oculifera* (Selman et al. 2012. Chelon. Conserv. Biol. 11:259–261), *Deirochelys reticulara chrysea* (Mitchell et al. 2014. Herpetol. Rev. 45:312), *Graptemys sabinensis* (Louque et al. 2015. Herpetol. Rev. 46:81), *Podocnemis sextuberculata* (Perrone et al. 2016. Herpetol. Rev. 47:287), and *Apalone ferox* (Taylor et al. 2017. Herpetol. Rev. 48:418–419). However, these conditions are generally rare, with only 0.93% of 216 *G. sabinensis* (Louque et al., *op. cit.*) and 0.06% out of 21,786 *T. scripta elegans* (Tucker et al., *op. cit.*) exhibiting kyphosis; kyphoscoliosis is even rarer with very few cases reported, including *Pseudemys suwaniensis* (Mitchell et al. 2016. Herpetol. Rev. 47:127–128) and *D. reticularia chrysea* (Mitchell et al., *op. cit.*). During our long-term population study of *Pseudemys gorzugi* in southeastern New Mexico, a female specimen exhibiting an obvious carapace deformation consistent with kyphosis was captured via snorkeling, representing the first reported case of kyphosis in *P. gorzugi* (Fig. 1A).

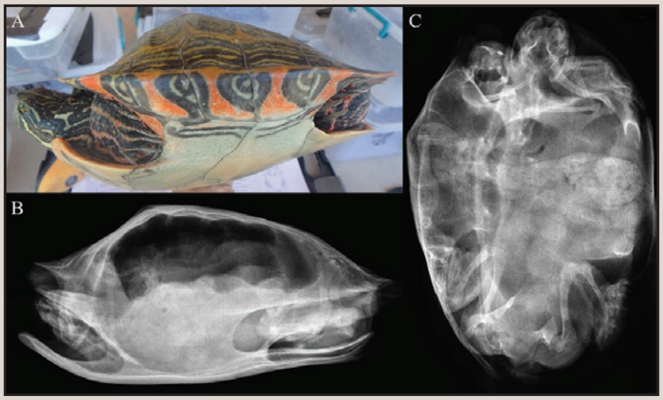

FIG. 1. Female *Pseudemys gorzugi* expressing typical spinal deformation associated with kyphosis: a lateral view photograph (A), a lateral view radiograph (B), and a dorsal view radiograph at 45-degree angle (C).

The turtle was captured on 12 June 2018 in a pond near the Black River, Eddy County, New Mexico, USA (32.11447°N, 104.578°W, WGS 84; 1067 m elev.), with a straight-line carapace length (CL) of 220 mm and a body depth (BD) of 103 mm. The turtle was radiographed at Desert Willow Wildlife Rehabilitation Center, Carlsbad, New Mexico, which confirmed the spinal deformation (Fig. 1B and 1C.). Since 2016, we have marked 420 *P. gorzugi* along the Black River and the lentic water bodies in the surrounding area. To date, this is the only *P. gorzugi* found with kyphosis which is 0.24% of all marked individuals.

We thank the staff at Desert Willow Wildlife Rehabilitation Center for assistance with radiographs. This research was approved by New Mexico Department of Game and Fish (Permit Authorization No. 3621), and Eastern New Mexico University IACUC (Approval #04-27/2018). We thank US Fish and Wildlife Service and New Mexico Department of Game and Fish - Share with Wildlife Program for supporting this project (State Wildlife Grant T-32-5).

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***TERRAPENE CAROLINA CAROLINA* (Woodland Box Turtle). CLAW MORPHOLOGY.** Herein, I report observations of an individual female *Terrapene carolina carolina* with exceptionally long claws on the hind limbs from Columbia County, Pennsylvania, USA (exact locality information withheld due to conservation concerns). On 29 May 2015 at 1100 h, I observed an adult female *T. c. carolina* in the vicinity of a railroad right-of-way with strongly curved hind-limb claws approximately 3 cm in length. This same turtle, identified by color pattern and minor damage on the carapace (Dodd 2001. North American Box Turtles: A Natural History. University of Oklahoma Press, Norman, Oklahoma. 231 pp.), was observed in the same general area again on 1 June 2017 and on 9 June 2018 and retained these exceptionally long, curved claws on the hindlimbs (i.e., these structures did not shorten over an approximately three-year period). The hindlimb claws of other male and female *T. c. carolina* observed in this population typically ranged from approximately 1 to 1.5 cm in length.

Terrapene c. carolina exhibits a number of dimorphic secondary sex characteristics; among these, males typically have longer and more curved claws on the hindlimbs than females (Ernst and Lovich 2009. Turtles of the United States and Canada, 2nd ed. Johns Hopkins University Press, Baltimore, Maryland. 827 pp.). Thus, the length of the hindlimb claws of the individual reported herein appears to be unusual and exceptional, especially considering the gender of the turtle. When in captivity, the claws of box turtles can grow to exceptional lengths (often requiring trimming) because the claws may not experience the typical wear that would occur in the wild. Therefore, perhaps this individual in some way does not encounter the conditions within its home-range that typically wear the claws of conspecifics. However, because other *T. c. carolina* in this population do not display exceptionally long hindlimb claws, and the population occurs within a relatively small (ca. 40 ha) wetland and forest fragment bordered on all sides by developed areas (i.e., other individuals in the population likely have overlapping home ranges), the long, curved hindlimb claws of this female might alternatively be attributed to some unique genetic or developmental factor contributing to exceptional growth.

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***TERRAPENE ORNATA* (Ornate Box Turtle). LONGEVITY.** The captive longevity record for *Terrapene ornata* is 28 yrs for a female (Slavens and Slavens 1999. Reptiles and Amphibians in Captivity Breeding—Longevity and Inventory January 1, 1999. Slaveware, Seattle, Washington. 400 pp.), although another female lived 22 years in captivity and was estimated to be 20 yrs old when first acquired (Ernst, in Ernst and Lovich 2009. Turtles of the United States and Canada. The Johns Hopkins University Press, Baltimore, Maryland. 840 pp.). In the field, Legler (1960. Univ. Kansas Publ. Mus. Nat. Hist. 11:527–669) speculated that *T. ornata* in Kansas might live to 50 yrs, but he lacked long-term recapture data to confirm this. Based on a 23-year study in Texas, Blair (1976. Southwest. Nat. 21:89–104) estimated his three oldest box turtles to be 31–32 yrs of age. Similarly, following 26 years of fieldwork in Kansas, Metcalf and Metcalf (1985. J. Herpetol. 19:157–158) estimated their oldest box turtles to be about 28 yrs, and they explicitly rejected Legler's (1960, *op. cit.)* 50-yr longevity estimate. Finally, at a site in New Mexico, Germano (2014. Chelon. Conserv. Biol. 13:56–64) recaptured three box turtles over a 22-year period that he estimated to be >40 years old. We here report that our field data from Nebraska confirm Legler's speculation.

During our mark-recapture study of turtles at and around Gimlet Lake on the Crescent Lake National Wildlife Refuge (Garden County, Nebraska) from 1981–2018, we individually marked 609 Ornate Box Turtles (plus over 2073 recaptures). We used counts of plastral annuli to estimate the age of each turtle at first capture (following Legler 1960, *op. cit.* and Blair 1976, *op. cit.*), although for turtles with more than ca. 12 annuli, we could only estimate a minimum age. A number of adult box turtles that were so aged in the early years of our study were captured as many as 37 yrs later, allowing us to estimate longevity in our population (Table 1).

Furthermore, of 19 females first captured as adults in 1981 or 1982, five were never seen again (presumably transients: see Kiester et al. 1982. Evolution 36:617–619); however, four were recaptured in 2018, after 37–38 yrs. Similarly, of 10 males first captured as adults in 1981 or 1982, two were never seen again, and one was recaptured in 2015 after 33 yrs. Hence, of 22 "resident" adults present in 1981–1982, at least five (23%) survived at least 33 yrs (an annualized survival rate of 99.2%, and four survived at least 36 yrs (an annual rate of 99.4%; see also Converse et al. 2005. Ecol. Appl. 15:2171–2179). These data clearly suggest that Ornate Box Turtles at the northern end of the species' range live well beyond 50 years, that females may outlive males, and that some box turtles may survive to six decades.

Table 1. Maximum carapace length (CL in mm; following Cagle 1946. Amer. Midl. Nat. 36:685–729), maximum plastron length (PL in mm), and estimated age (in number of winters) of long-term recaptures of *Terrapene ornata* in western Nebraska. Ages at initial capture were estimated from counts of plastral annuli and represent minimum ages. Letters after ID numbers indicate sex (M, male; F, female).

	First capture				Last capture			
ID	Year	CL	PL	Age	Year	CL	PL	Age
2F	1981	111.1	118.1	>20	2018	110.3	119.9	>57
8F	1981	107.5	114.2	>20	2018	107.4	121.3	>57
129F	1981	104.9	115.9	>20	2018	104.9	118.9	>57
29F	1982	111.5	—	>20	2018	110.9	116.2	>56
138F	1983	116.8	120.5	>20	2017	115.2	120.7	>54
81M	1982	107.5	111.4	>20	2015	114.4	117.4	>53
233F	1988	110.5	116.3	>20	2018	110.3	122.0	>50
134F	1986	112.2	119.9	>20	2014	114.0	121.8	>48
132AF	1983	119.0	131.8	>20	2010	—	—	>47
538F	1990	105.2	114.0	>20	2017	109.8	116.3	>47
132BF	1985	119.0	126.4	>20	2010	—	—	>45

We thank the staff of the Crescent Lake National Wildlife Refuge (CLNWR) for allowing us to undertake this research. Turtles were captured and held under annual permits from the CLNWR as well as the Nebraska Game and Parks Commission. Our field methods adhered to the American Society of Ichthyologists and Herpetologists' Guidelines for use of Live Amphibians and Reptiles in Field and Laboratory Research, and in recent years to approved protocols from the Earlham College Institutional Animal Care and Use Committee.

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***TRACHEMYS ORNATA* (Ornate Slider). PREDATION.** *Trachemys ornata* is a large freshwater turtle endemic to the Pacific Coast region of western Mexico from southern Sinaloa and northern Nayarit (Parham et al. 2015. Proc. California Acad. Sci. 62:359–367). This species is listed as Vulnerable by the IUCN Red List (www.iucnredlist.org; 21 June 2018) and little is known about its natural history. Here I provide evidence of natural predation of an adult *T. ornata* by a Jaguar (*Panthera onca*) in Nayarit, western Mexico.

At 2307 h on 8 April 2018, a camera trap (Cuddeback Color C1) set by VHL at "El Pozo Chino" in the Municipality of Santiago Ixcuintla, Nayarit (21.69017°N, 105.45927°W, WGS 84; 8 m elev), captured an image of an adult female *Panthera onca* carrying an adult *Trachemys ornata* in its mouth (Fig. 1) in a seasonally flooded mangrove forest. With the exception of its head, the Jaguar was completely wet, which suggests that she had crossed a nearby body of water where she probably encountered the turtle. The presence of *T. ornata* in the area has been confirmed by VHL visually all across the Santiago River basin, including the wetland (El Pozo Chino) within 50 m of the location where the photo was recorded.

Fig. 1. A camera trap image of a female *Panthera onca* carrying an adult *Trachemys ornata*, Santiago Ixcuintla, Nayarit, western Mexico.

Jaguars are known to prey on turtles, with sea turtles (Veríssimo et al. 2012. Oryx 46:340–347), freshwater and terrestrial turtles (Emmons 1989. J. Herpetol. 23:311–314) having been documented as part of the Jaguar's diet. However, to our knowledge, this is the first evidence of Jaguar predation on *T. ornata*.

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CROCODYLIA — CROCODILIANS

***ALLIGATOR MISSISSIPPIENSIS* (American Alligator). NOVEL NESTING SITE.** *Alligator mississippiensis* occurs throughout Louisiana, USA, with abundant habitat and the highest nest densities being found in coastal marshes (McNease and Joanen 1978. Proc. Ann. Conf. SE Assoc. Fish Wildl. Agencies 32:182–186). In addition to nesting in the marsh proper, *A. mississippiensis* will also nest on levees (6.7% of 315 nests, Joanen 1969. Proc. SE Assoc. Game Fish Comm. Conf. 23:141–151) and spoil banks (9.4% of 53 nests, Platt et al. 1995. Proc. Annu. Conf. SE. Assoc. Fish Wildl. Agencies 49:629–639). In recent years, marsh restoration efforts have included construction of terraces, which are discontinuous narrow strips of created marsh, typically formed of dredge material from pond bottoms, often stabilized with emergent plants (O'Connell and Nyman 2011. Environ. Manage. 48:975–984, and references therein). Terraces might limit marsh losses, as they can increase marsh edge, presumably slow erosion, stimulate production of vegetation, and decrease excessive pond depths (O'Connell and Nyman 2010. Wetlands 30:125–135; O'Connell and Nyman 2011, *op. cit.*).

On 12 June 2107, aerial surveys were conducted by helicopter to locate *A. mississippiensis* nests on Rockefeller Wildlife Refuge in Cameron Parish, Louisiana, USA, to obtain eggs for various research studies. Due to time constraints and expense associated with helicopter surveys, we attempted to locate as many nests as possible in the best habitat, to limit flight time required, and work most efficiently. Thus, neither fixed transects nor systematic grids were flown; we simply focused searches on areas known to have the best quality nesting habitat, in one of the marsh management unit systems (Unit 6 off the Superior canal), and located approximately 150 nests in this and surrounding units. One *A. mississippiensis* nest was observed incidentally on a terrace (Fig. 1) while "deadhead" flying back to the refuge headquarters to refuel the aircraft. Construction of the earthen terrace was completed in November 2015, and it was subsequently planted with *Paspalum vaginatum* (Seashore Paspalum) and *Scirpus californicus* (Giant Bulrush or Bullwhip) in spring of 2016 (Louisiana Department of Wildlife and Fisheries 2015 – 2016 Annual Report. 144 pp.).

On 14 June 2017, we collected *A. mississippiensis* eggs from many of the nests found on 12 June, including the one located on the terrace. This nest contained 26 eggs, all of which were fertile, and approximately 6 days old. The female *A. mississippiensis* actively defended the nest. The eggs were collected and placed in a field incubator. On 20 June we returned to the nest site to collect an adult female *A. mississippiensis* for a research study. The female alligator again defended the nest, was captured, and measured 211 cm total length. The nest was constructed of *P. vaginatum* (Fig. 2) and dimensions were 145 cm x 152 cm across (slightly smaller than the average nest diameter of 182 cm in Joanen, *op. cit.*), and nest height was 43 cm. The surrounding *P. vaginatum* on the terrace measured approximately 53 cm high. The terrace was 6.7 m wide and approximately 290 m long.

The 26 eggs from the nest described were provided to a university researcher on 23 June. Three eggs died during incubation, two eggs were sacrificed, and the remaining 21 hatched successfully (hatch rate at least 80.8%, possibly 88.5% had the two eggs sacrificed hatched successfully).

Due to abundant wetlands habitat in Louisiana (2–3 million acres; Elsey and Kinler 2011. *In* P. S. Soorae [ed.], Global Re-introduction Perspectives: 2011. More Case Studies from around the Globe, pp. 125–129. IUCN/SSC Re-introduction Specialist Group, Gland, Switzerland and Environment Agency-Abu Dhabi, Abu Dhabi, UAE). *Alligator mississippiensis* may not "need" to nest on terraces, but an additional benefit of marsh restoration projects to conserve and protect fragile wetlands might also be to incidentally provide additional alternate nesting habitat sites

Fig. 1. Map illustrating location of *Alligator mississippiensis* nest (yellow circle) on a terrace in Cameron Parish, Louisiana. Note other terraces south and northeast of the one with the nest (white circles).

for *A. mississippiensis*. It is possible we might have observed more *A. mississippiensis* nests on the terraces had we conducted an intensive search of all terraces in the Superior marsh system. A land manager for an adjacent private property also reported seeing an *A. mississippiensis* nest on a similar terrace in the 2017 nesting season (T. Joanen, pers. comm.), but it is unknown if any eggs were present, or if eggs were fertile or viable. *Alligator mississippiensis* sometimes have incomplete nesting attempts that do not contain eggs (Joanen, *op. cit.*; Platt et al., *op. cit.*). Similar to our observation in Louisiana, a recent report noted the finding of an *A. mississippiensis* nest on a constructed tree island on Arthur R. Loxahatchee National Wildlife Refuge in Florida, USA; these tree islands are part of restoration efforts in the Florida Everglades and provide suitable habitat for certain vertebrates (Cline et al. 2016. Herpetol. Rev. 47:455–456).

Terraces have also provided nesting habitat for *Anas fulvigula* (Mottled Duck) in coastal Louisiana (Brasher et al. 2007. Occurrence of mottled duck nests on constructed marsh terraces in Louisiana and Texas – a pilot study. Final Report 14 November 2007. Gulf Coast Joint Venture. National Wetlands Research Center, Lafayette, Louisiana. 8 pp.) and might be used by other marsh birds; future studies are underway to document possible terrace use by other avian species on Rockefeller Wildlife Refuge (J. Marty, pers. comm.).

In addition to the unusual site for the nest described herein, the plant material of which the mound was constructed is also novel. *Paspalum vaginatum* was not noted as vegetation in any of the 315 *A. mississippiensis* nests in a prior study at this site (Joanen, *op. cit.*), nor in other studies in Louisiana (Carbonneau 1987. Nesting Ecology of an American Alligator Population in a Freshwater Coastal Marsh. Master of Science Thesis. Louisiana State University. 53 pp; Platt et al. 1995, *op. cit.*). This species was also not used in nests in other states, including a large series of 111 *A. mississippiensis* nests in Florida (Deitz and Hines 1980. Copeia 1980:249–258) or 767 nests monitored in South Carolina (Wilkinson 1983. Nesting Ecology of the American Alligator in Coastal South Carolina. Study Completion Report. August 1978 – September 1983. S.C. Wildlife and Marine Resources Dept. 114 pp.). A small study in Texas (Hayes-Odum et al. 1993. Texas J. Sci. 45:51–61) documented two nests at an inland site that were composed in part of *Paspalum floridanum* (Florida Paspalum).

Fig. 2. Adult *Alligaor mississippiensis* (211 cm total length) defending nest constructed on a terrace in Cameron Parish, Louisiana.

This finding of an *A. mississippiensis* nest on a narrow terrace illustrates how adaptable the species can be, and that they may use various new plant materials to construct nest mounds. Of particular interest, an extraordinary *A. mississippiensis* nest was found constructed on a sanitary landfill on New Orleans, Louisiana in 1991, and was composed almost entirely of plastic bags (Coulson and Coulson 1993. Herpetol. Rev. 24:58). The nest mound also contained a flip-flop sandal, a plastic baby doll, and a plastic vegetable oil container; and the clutch had 36 *A. mississippiensis* eggs; the following year another *A. mississippiensis* nest was found 4.6 m from the 1991 nest (Coulson and Coulson, *op. cit.*).

Crocodilians can nest on man-made and natural islands (Hayes-Odum, *op. cit.*; Platt et al. 2008. J. Zool. 275:177–189); terraces might serve as functional islands providing potential nesting habitat. Future studies are planned to determine if adverse habitat conditions such as flooding or drought in natural marshes lead to more use of terraces as nesting sites for *A. mississippiensis*.

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***ALLIGATOR MISSISSIPPIENSIS* (American Alligator). TOLERANCE OF POTENTIALLY TOXIC NON-NATIVE PREY.** American Alligators are opportunistic predators and their food habits have been well studied (Elsey et al. 1992. Proc. Southeast. Assoc. Fish Wildl. Agencies 46:57–66, and references therein). Composition of *Alligator mississippiensis* diet often varies due to regional prey availability (Gabrey 2010. Herpetol. Conserv. Biol. 5:241–250; Rosenblatt et al. 2015. Oecologia 178:5–16). Several studies conducted in Florida reported the native *Pomacea paludosa* as important invertebrate prey for *A. mississippiensis* (Fogarty and Albury 1967. Proc. Annu. Conf. SE. Assoc. Game Fish Comm. 21:220–222; Delany and Abercrombie 1986. J. Wildl. Manag. 50:348–353). A recent study conducted in southeastern and southwestern Louisiana on food habits of 448 adult *A. mississippiensis* reported unspecified "snails" were observed in some stomach contents (Gabrey, *op. cit.*). However, differential digestion rates can lead to over-representation of prey items such as keratinized scutes and scales which are resistant to digestion, or under-representation of rapidly digested soft-bodied prey items (Jackson et al. 1974. J. Herpetol. 8:378–381; Garnett 1985. J. Herpetol. 19:303–304; Delany and Abercrombie, *op. cit.*; Platt et al. 1990, Northeast. Gulf Sci. 11:123–130; Barr 1997. Food Habits of the American Alligator, *Alligator mississippiensis*, in

Fig. 1. *Alligator mississippiensis* stomach filled nearly to capacity with 453 *Pomacea maculata* opercula; specimen was a 328-cm male collected in Terrebonne Parish, Louisiana.

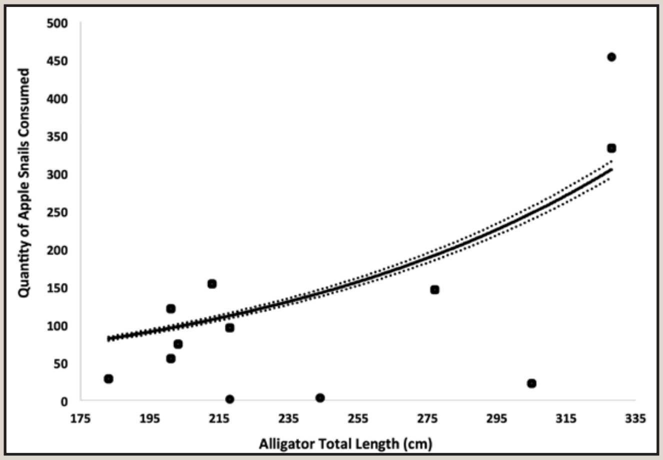

Fig. 2. *Alligator mississippiensis* size (cm) and quantity of opercula recovered in stomach contents.

the Southern Everglades. PhD dissertation. University of Miami, Florida. 244 pp; Nifong et al. 2012. Copeia. 2012:419–423; Rosenblatt et al., *op. cit.*). Fogarty and Albury (1967, *op. cit.*) reported that native *Pomacea paludosa* (Florida Apple Snail) were found to comprise 65.8% of the stomach contents (24 stomachs contained 119 *P. paludosa*) in a small study of 36 immature alligators conducted in a single night with samples collected from a single canal in the Florida Everglades; Rosenblatt et al. (2015, *op. cit.*) further discuss the prevalence of gastropods in the diet of *A. mississippiensis* across a wide range of habitats.

Pomacea maculata (Giant Apple Snail) is an invasive freshwater snail native to South America, and is now established throughout the southeastern United States (Monette et al. 2016. Southeast. Nat. 15:689–696), including Louisiana (Byers et al. 2013. PLoS ONE 8:e56812). This species can pose risks to agricultural crops as well as to human and wildlife health; laboratory studies demonstrated the neurotoxin linked to Avian Vacuolar Myelinopathy (AVM) can be transferred by *P. maculata* to its avian predators (Robertson 2012. Potential Threats of the Exotic Apple Snail *Pomacea insularum* to Aquatic Ecosystems in Georgia and Florida. Master of Science Thesis. University of Georgia, Athens, Georgia. 74 pp.; Byers et. al., *op.cit.*; Dodd et al. 2016. J. Wildl. Dis. 52:335–344). This could be a concern if the neurotoxin is transmittable to alligators by consumption of an affected bird, as *A. mississippiensis* prey on a variety of avian species (Gabrey and Elsey 2017. J. Louisiana Ornithol. 10:1–10); alternately, alligators might be directly susceptible to the neurotoxin.

To deter predation, the egg masses of *P. maculata* contain multiple toxins produced in the albumen glands of females, and when fed to experimental mice, observed effects were lethargy, paralysis, or death depending on the dose (Giglio et al. 2016. Can. J. Zool. 94:777–785). We recently documented an *A. mississippiensis* captured in southeastern Louisiana (in a region with *P. maculata*) with opercula in the stomach (Elsey et al. 2017. Herpetol. Rev. 48:627–628). If *A. mississippiensis* are shown to be susceptible to the toxins from *P. maculata*, it could have important ramifications for *A. mississippiensis* conservation in Louisiana. Thus, we initiated this study to expand our earlier report (Elsey et al. 2017, *op. cit.*) and determine prevalence of *A. mississippiensis* feeding on potentially toxic *P. maculata*, to quantify number of snails consumed, and investigate if snail consumption is related to *A. mississippiensis* size.

We collected stomachs from 12 *A. mississippiensis* from 31 August 2017 through 4 September 2017 from specimens taken in Louisiana's sanctioned alligator harvest. The adult *A. mississippiensis* (183–328 cm total length) were trapped on three properties in Terrebonne Parish and brought to a processing shed in Houma, Louisiana. Efforts were made to select *A. mississippiensis* caught in regions known to harbor *P. maculata*. The viscera or stomach was dissected from carcasses and frozen for later analysis; these were subsequently thawed and stomach contents sorted. When present, *P. maculata* opercula were quantified and each measured to the nearest 0.01 mm with digital calipers. General notes were recorded about other stomach contents present.

All 12 specimens examined had consumed *P. maculata*; one stomach contained only shell fragments, the other stomachs contained numerous (range 3–453) opercula (Fig. 1). Most contained a few opercula with some soft tissue remaining (2.6%, ca. 39 of 1487), indicating recent consumption (Barr 1997, *op. cit.*); that stomach clearance study noted *P. paludosa* opercula remained attached to soft-body tissue for up to 36 h in sub-adult (80–150 cm total length) *A. mississippiensis* stomachs. The size of opercula measured ranged from 10.73–61.59 mm. Other stomach contents included typical prey items found in *A. mississippiensis* stomachs, including insect parts, crustacean remains, fish bones, turtle bones, *A. mississippiensis* parts, bird feathers, fur, parasites, and woody debris and vegetation; as well as some non-food items (fish hooks, bullet fragments, and stones).

There was a statistically significant (albeit a poor fit to the observed data, based on goodness of fit statistic $\hat{c} = 2928.5$) direct relationship between *A. mississippiensis* total length and quantity of snails consumed (general linear model $F_{1,11} = 3139.4$, $P < 0.01$, $\beta = 1.7$ snails mm^{-1} ± 0.03 SE, Fig. 2). When we excluded the two outlier samples with evidence of having consumed only one and only three snails (three rule; Pukelsheim 1994. Am . Statistician 2:88–91), a statistically significantly model with strong explanatory power and a better fit, although underdispersed and potentially exhibiting inaccurately large estimated standard errors ($\hat{c} = 0.66$), resulted for the relationship between *A. mississippiensis* total length and number of snails consumed (generalized linear model with log link and negative binomial probability distribution $F_{1,1028} = 1948.1$, $P < 0.01$, $\beta = 1.6$ snails mm^{-1} ± 0.006 SE). Indeed, this model estimated approximately an increase of one additional snail consumed per 1-mm increase in total length. Also, we found a significant positive relationship between *A. mississippiensis* total length and size of opercula of snails consumed, for those specimens having consumed more than three snails (generalized linear model with a log link and Poisson probability distribution $\hat{c} = 1.53$, $F_{1,1027} = 20.65$, $P < 0.01$, = 0.99 ± 0.009 SE mm size of opercula per change in *A. mississippiensis* length).

Our results in this study are similar to a prior study on *Crocodylus moreletii* (Morelet's Crocodile) food habits in Belize, wherein a significant positive correlation occurred between *C. moreletii* SVL and mean, minimum, and maximum length of opercula of *Pomacea flagellata* (Golden Apple Snail) found in *C. moreletii* stomachs (Platt et al. 2006. Herpetol. J. 16:281–290). Interestingly, Platt et al. (2006, *op. cit.*) noted the occurrence of 618 *P. flagellata* in a single *C. moreletii* stomach; these are a large snail (ca. 60–70 g; Platt et al. 2006, *op. cit.*), although perhaps not as large as *P. maculate*, which have been reported with masses ranging from 55.6–135.3 g (Monette et al., *op. cit.*).

Of interest, contents of one stomach sample (277 cm total length male *A. mississippiensis*) contained a large amount of *Myocastor coypus* (Nutria) remains, in addition to 146 *P. maculata* opercula, vegetation, and roundworms. Thus, this *A. mississippiensis* fed almost entirely upon two non-native species (Nutria and Giant Apple Snail), illustrating its adaptability and utilization of available prey resources.

The current study corroborates our earlier finding (Elsey et al. 2017, *op. cit.*) of *A. mississippiensis* consuming *P. maculata*; we now provide details on large quantities consumed (despite potential toxicity) and correlations with alligator size. Thus far we have not noted nor received any reports of any wild alligator morbidity associated with consumption of potentially toxic Giant Apple Snails in Louisiana.

We recently conducted a short-term experimental feeding trial to determine if direct consumption of female Giant Apple Snails could have an adverse effect on alligators; the evidence thus far does not support cause for concern about alligators becoming poisoned by eating female Giant Apple Snails. (Carter et. al., unpubl.). It would be of interest expand the current study even further, and over a longer duration, to determine to what extent *A. mississippiensis* consumes *P. maculata*, as the snails are spreading across Louisiana. However, it is important to recognize the opercula are likely resistant to digestion and could be over-represented in stomach content analyses; Barr (*op. cit.*) noted opercula of *P. paludosa* remained in *A. mississippiensis* stomachs for up to 200 days after ingestion, after which time observations were discontinued. It might be beneficial to determine if *P. maculata* are energetically advantageous as a prey item, perhaps leading this species to consumed with greater frequency by *A. mississippiensis* and other large adult crocodilians. *P. maculata* might also be consumed in high quantity as gastropods might be considered an "easy-capture" prey item (Rosenblatt et al. 2015., *op. cit.*). Whether consumption of possibly toxic *P. maculata* consumed in larger quantities and for extended time periods has adverse effects on *A. mississippiensis* remains unknown. *A. mississippiensis* may possibly have adaptations to neutralize any toxins in the invasive Giant Apple Snails.

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SQUAMATA — LIZARDS

***AMEIVA FESTIVA* (= *HOLCOSUS FESTIVUS*). (Central American Whiptail Lizard). JUVENILE COLORATION.** A variety of reptiles and amphibians exhibit body coloration changes over maturation. A common ontogenetic trend among many lizard species is the loss of conspicuous tail coloration: juveniles bear bright tails that presumably direct predator attacks to a more expendable body region, whereas adults typically lose conspicuous tails as their activity patterns and habitat use (and concomitant selective pressures) change with age (Hawlena et al. 2006. Behav. Ecol. 17:889–896). *Ameiva festiva* is a neotropical, active-foraging lizard inhabiting a wide range of terrestrial habitats including forest interiors, small clearings, and disturbed areas throughout Central America. Juvenile *A. festiva* have bright blue tails, but adults (males > 85 mm; females > 78 mm) lose this trait (Savage 2002. The Amphibians and Reptiles of Costa Rica: A Herpetofauna between Two Continents, between Two Seas. University of Chicago Press, Chicago, Illinois. 934 pp.). Although the function of *A. festiva*'s ontogenetic color change has not been directly examined, it is plausible that differing predation pressures or escape abilities between life stages contribute to the maintenance of this ontogenetic shift in color.

We used spectrophotometry to document the spectral reflectance of a juvenile *A. festiva*'s tail to determine whether juveniles of this species might use ultraviolet signals. We opportunistically captured a juvenile *A. festiva* on a small stream bank within the pre-montane forests of Las Cruces Biological Station, Coto Brus County, Costa Rica, on 25 July 2016. During capture this individual autotomized its tail, which was in the juvenile-typical blue phase. We temporarily retained the autotomized tail and measured tail color within 60 seconds of tail separation. Spectrophotometric measurements were performed using a hand-held portable JAZ spectrophotometer (Ocean Optics, Dunedin, Florida). The tail was illuminated by a xenon-pulsed light source via a bifurcated optical fiber with a shielded probe held at a 90°-angle to the surface of the tail. Reflectance was recorded relative to a 99% Spectralon WS-2 reflectance standard (Ocean Optics). Four separate points were measured on the tail, and we used the average of the four spectra to characterize tail reflectance within the visible and ultraviolet range (300–700 nm). The spectra were mean-averaged and smoothed (Maia et al. 2013. Methods Ecol. Evol. 4: 906–913) using a LOESS smoother with a span parameter set to 0.1 in R (ver. 3.1.2; R Core Team 2014). The average reflectance spectrum (Fig. 1) shows a single major peak at 475 nm, indicating a high level of reflectance in the visible blue region of the spectrum. No reflectance peak was noted in the ultraviolet region.

Our observation suggests that juvenile *A. festiva* tails do not bear substantial ultraviolet colors; tail coloration appears to be limited to blue wavelengths. The blue coloration is most

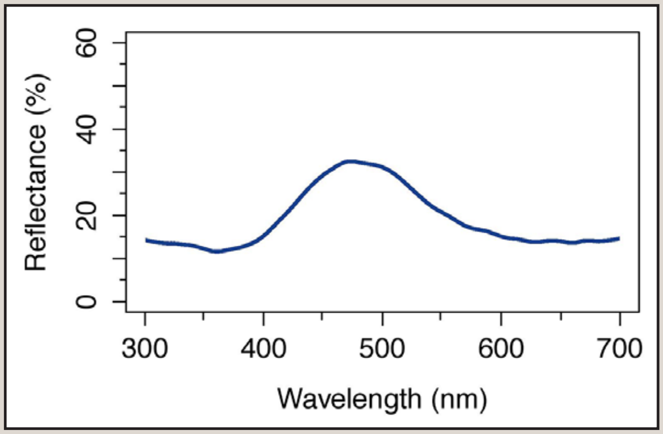

Fig. 1. Average tail reflectance spectrum of a juvenile *Ameiva festiva*.

likely generated via a structural mechanism (through blue light scattering as it passes through skin chromatophore cells). As blue tail coloration is restricted to juveniles, it is unlikely to serve a role in sexual signaling. Information about the primary predators of adult and juvenile *A. festiva* is generally lacking, although some studies report that snakes might be important predators of *A. festiva* (Sorrel 2009. Copeia 2009:105–109) and other co-occurring *Ameiva* species (Hirth 1963. Ecol. Monogr. 33:83–112). Nevertheless, if blue tails do indeed function to misdirect predator strikes in juvenile *A. festiva*, the tail's conspicuous coloration suggests that visual predators (e.g., birds) might also be important during this life stage.

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***ANOLIS UNILOBATUS*. SURFACE TENSION.** *Anolis unilobatus* is a small anoline lizard that ranges from southern Mexico to Costa Rica (Köhler and Vesely 2010. Herpetologica 66:207–228). They can often be found on fences where they are usually perched head down on the posts, especially near bushes. At some locations they can reach high densities (Köhler and Vesely, *op. cit.*). Several species of *Anolis* exhibit aquatic activity, perching near bodies of water to either hunt or escape from predators by swimming or diving (Robinson 1962. Copeia 1962:640–642; Brandon et al. 1966. Herpetologica 22:156–157; Beuttell and Losos 1999. Herpetol. Monogr. 13:1–28; Leal and Losos 2000. J. Herpetol. 34:318–322; Birt et al. 2001. J. Herpetol. 35:161–166). Herein, I report for the first time use of water as an alternative mechanism against threat in *A. unilobatus*.

At 1054 h on 29 November 2017, in ejido Copoya, Municipio de Tuxtla Gutiérrez, Chiapas, México (16.71730°N, 93.12820°W, WGS 84; 868 m elev.), I observed a subadult male *A. unilobatus* perched on a fallen branch at 1 m above ground level. The vegetation in the area is tropical dry forest, with temporary bodies of water during the rainy season. When I approached, the lizard jumped and fell onto the surface of a small pond, remaining suspended by its limbs and tail on the water's surface. After remaining immobile for less than a minute and suspended on the water surface, the lizard swam ca. 60 cm in a straight line on the surface to the shore. Swimming motions consisted of rapid undulatory movements of the body and tail with adpressed limbs. Although *A. unilobatus* is not an inhabitant of aquatic habitats, this could imply that the use of water as a refuge is a facultative phenomenon as Powell and Parmerlee (1993. Herpetol. Rev. 24:59) mention for *A. chlorocyanus*, and the small size at the subadult stage might facilitate suspension on water and surface swimming.

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***CHALCIDES OCELLATUS* (Ocellated Skink). DIET.** A specimen of *Chalcides ocellatus* was collected at Mishor Yamin, Israel (31.00397°N, 35.10713°E) on 22 June 2012. The specimen, an adult male with a total length of 132 mm (snout–vent length 82 mm, tail length 50 mm, mass 11.8 g), was deposited in the Steinhardt Museum of Natural History (Tel Aviv, Israel) as TAU 16357. Dissection of the *C. ocellatus* specimen revealed a *Mesalina guttulata* (Small-spotted Lizard, TAU 17847) within its digestive tract (Fig. 1). The head and tip of the tail of the *M. guttulata* are missing (presumably digested during the intervening period between initial predation and time of collection). The total length of the remaining *M. guttulata* specimen was 44 mm from base of neck to broken tail tip (base of neck to cloaca: 22.7 mm; cloaca to broken tip of tail 21.3 mm). This means the *M. guttulata* was no less than a third of the total length of the *C. ocellatus*, and in all likelihood larger with the inclusion of its head and, potentially, its tail.

In Israel *C. ocellatus* is noted to primarily feed on various arthropods, occasionally supplementing its diet with fruit (Bar and Haimovitch 2011. A Field Guide to Reptiles and Amphibians of Israel. Pazbar LTD, Herzliya. 246 pp.). In other parts of its distribution, instances have been reported of it preying on *Podarcis filfolensis* (Filfola Wall Lizard) and conspecific juveniles (Carretero et al. 2010. Bonn Zool. Bull. 57:111–118). However, saurophagy has not been reported for this species from Israel. This new finding strengthens our understanding of *C. ocellatus* as an opportunistic omnivore.

Fig. 1. *Chalcides ocellatus* (left) with a partially digested *Mesalina guttulata*, removed from its stomach (right).

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***CYRTODACTYLUS PULCHELLUS* (Malayan Forest Gecko). ENDOPARASITE.** *Cyrtodactylus pulchellus* ranges from southern Thailand throughout much of Peninsular Malaysia; it is nocturnal and scansorial, restricting its activity to rocks, trees, or root systems (Grismer 2011. Lizards of Peninsular Malaysia, Singapore and their Adjacent Archipelagos. Edition Chimaira, Frankfurt am Main, Germany. 728 pp.). We know of no reports of endoparasites from *C. pulchellus*. Here we report the presence of one species of Nematoda, thereby establishing the helminth list for this gecko.

One female *C. pulchellus* (SVL = 111 mm), from Peninsular Malaysia, Penang State, Pulau Pinang. Air Terjun Titi, Kerawang (5.40388°N, 100.22333°E, WGS 84; 257 m elev.) and deposited in the herpetological collection of La Sierra University (LSUHC), Riverside, California, USA as LSUHC 10022 was examined. The specimen had been collected in March 2011 by hand, was euthanized within 12 h of capture, preserved in 10% formalin, and stored in 70% ethanol. The body cavity was opened by a longitudinal incision, and the digestive tract was removed and opened. The esophagus, stomach, small intestine, and large intestine were examined for helminths under a dissecting microscope. Only one nematode was found (small intestine) which was placed on a glass slide in a drop of lactophenol, a coverslip added, and identification made from this temporary wet mount utilizing Anderson et al. (2009. Keys to the Nematode Paraasites of Vertebrates, Archival Volume. CAB International, Wallingford, Oxfordshire. 463 pp.) and Gibbons (2010. Keys to the Nematode Parasites of Vertebrates, Supplementary Volume. CAB International, Wallingford, Oxfordshire, UK. 416 pp.). The nematode was identified as a male *Rhabdocona* sp. and subsequently deposited in the Harold W. Manter Parasitology Laboratory (HWML), The University of Nebraska, Lincoln, Nebraska, USA as HWML 92091.

We have assigned our specimen to *Rhabdochona* because the cylindrical, elongated pharynx is dilated anteriorly to form a well-defined funnel-shaped buccal cavity armed with sclerotized rods projecting anteriorly as teeth. In addition, caudal alae are absent, gubernaculum is absent, and spicules are unequal and dissimilar.

Rhabdochona is a speciose genus considered by Asmatullah-Kakar et al. (2012. Pakistan J. Zool. 44:95–99) to contain over 160 species. Members of *Rhabdochona* are commonly found as parasites of freshwater fishes, less frequently in marine fish from all zoogeographical realms (Bilqees 1979. Zool. Scripta 88:107–110; Lakshmi 2001. Bol. Chileno Parasitol. 57:3–4; Moravec 2007. Folia Parasitol. 55:144–160; Moravec 2010. Acta Parasitol. 55:144–160).

Rhabdochona sp. in *C. pulchellus* is a new host record and the first report of this genus from a lizard.

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***HOLBROOKIA LACERATA* (Spot-tailed Earless Lizard). BURYING BEHAVIOR.** Burying behavior is well documented within the phrynosomatid sand lizards, but no literature exists on the burying habits of *Holbrookia lacerata*. Other members of this clade prefer sandy soils and are known to bury in soft soils to avoid extreme temperatures and predation, and to lay eggs (Axtell 1956. Bull. Chicago Acad. Sci. 10:163–179; Brennan and Holycross 2009. A Field Guide to the Amphibians and Reptiles in Arizona. Arizona Game and Fish Department, Phoenix. 150 pp.; Hibbitts and Hibbitts 2015. Texas Lizards: A Field Guide. University of Texas Press, Austin. 351 pp.). *Uma notata* (Colorado Desert Fringe-toed Lizard) have been documented burying themselves in coarse pebbly sand (Pough 1970. Copeia 1970:145). However, clay soils are preferred by *H. lacerata* (Hibbitts and Hibbitts, *op. cit.*).

Over the course of a telemetry study on *H. lacerata* from May to July 2017, a number of individuals were discovered buried in multiple substrates and under varied weather conditions. All lizards used in the study were adults. Two sites were used for this study: one located in Crockett County, Texas, USA (ca. 30.9300°N, 101.1916°W; WGS 84) and another located in Val Verde County, Texas, USA (ca. 29.3712°N, 100.7722°W; WGS 84). Lizards from these sites represent two separate subspecies: *H. l. lacerata* (Northern Spot-tailed Earless Lizard) at the Crockett County site and *H. l. subcaudalis* (Southern Spot-tailed Earless Lizard) at the Val Verde County site. The Crockett County site consists of a mixture of Chihuahuan thornscrub and arid grasslands. The Val Verde County site is heavily modified and consists of a mowed airfield surrounded by Chihuahuan thornscrub. Both sites are primarily clay soils intermixed with varied amounts of limestone.

In Val Verde County, eight individual lizards were observed at least partially buried a combined total of 37 times. Many of these events were sequential encounters in the exact same location. Assuming these represented times the lizards did not become active and then rebury themselves at the same location, lizards were discovered buried 17 times. Five lizards were female, two of which were gravid during and after our observations. With respect to weather, 78% of encounters with buried lizards occurred during overcast or rainy conditions, while the remaining observations were made during sunny conditions. Lizards never buried more than 1 cm deep, and were occasionally partially exposed. One female lizard was documented twice buried into a harvester ant (*Pogonomyrmex* sp.) mound. Four lizards were recorded buried along caliche roads in shallow, relatively loose gravel. Two lizards were discovered buried in the detritus and shallow soil occupying cracks in an abandoned asphalt runway. Additionally, 325 of 578 total observations (56.2%) of lizards were found completely hidden underneath thick forbs or grass bunches but not buried. Most of these lizards were hiding in detritus, primarily dead grass, beneath the plants. In total, lizards were hidden 62.6% of encounters.

In Crockett County, 10 individual lizards were observed at least partially buried a combined total of 82 times. Excluding sequential encounters in the same exact location, lizards were observed buried 40 times. Six of these lizards were female, and four were male. Five of the females were gravid during burying observations. In contrast to Val Verde County, only 25% of encounters with buried lizards in Crockett County occurred under overcast or rainy conditions. Lizards were recorded buried, or actively burying, in caliche roads 13 times. Additionally, 144 of 475 total observations (30.3%) of lizards were found completely hidden beneath thick forbs or grass bunches, and under dry cattle feces in three cases. Similar to the Val Verde County site, detritus beneath the plants were used as cover. In total, lizards were hidden 47.6% of encounters. Burying behavior at both sites seemed to coincide with longer periods of inactivity (i.e. cool, overcast days). Short-term refuge use was most often just the cover of vegetation or detritus.

Other members of this genus have been shown to be extremely wary, readily sprinting away when approached (Cooper 2000. Behaviour 137:1299–1315). This wariness, in conjunction with their cryptic pattern and the burying habits described here, suggest that the detection probability of this species could be extremely low.

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***HOLBROOKIA LACERATA* (Spot-tailed Earless Lizard). PREDATION.** *Holbrookia lacerata* is a small phrynosomatid lizard that inhabits short-grass prairies in central and south Texas (USA) and adjacent Mexico. Populations of this species are thought to be in decline, but little is known about its natural history, especially sources of predation. Here we report an observation of predation on *H. lacerata* by a Rio Grande Ground Squirrel, *Ictidomys parvidens.*

At 1325 h on 4 June 2017, during a telemetry study of *H. lacerata* on Laughlin Air Force Base in Val Verde County, Texas, USA, while attempting to locate a study lizard on the airfield, we received a signal, and approximately 30 m ahead a ground squirrel was eating a food item. Upon further inspection through binoculars, the food item was determined to be the telemetered study lizard in question (Fig. 1). The lizard was still alive when first sighted. The squirrel shook the lizard a few times, and then began eating it, headfirst. When approached, the squirrel retreated to a nearby burrow with the lizard. The lizard was a gravid female (SVL = 58 mm, mass = 6.4 g). The lizard had two missing toes and a partially regrown tail, perhaps contributing to its capture.

Sciurid consumption of animals, particularly small vertebrates, has been well documented (Callahan 1993. Great Basin Nat. 53:137–144) but little literature exists on the specific predatory habits of *Ictidomys parvidens.* This squirrel is native to southern and western Texas, southeast New Mexico, and adjacent Mexico. It occupies grass and shrublands, and shares much of this habitat with *H. lacerata.* These two species are commonly encountered on the airfield portion of the base. Both also exist in above-average numbers (as compared to surrounding habitat) and interactions between species are likely not uncommon.

Fig. 1. *Ictidomys parvidens* preying upon *Holbrookia lacerata.* The radiotransmitter can be seen affixed to the back of the lizard.

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***LYGODACTYLUS KLUGEI* and *TROPIDURUS COCOROBENSIS*. DEATH FEIGNING.** *Lygodactylus klugeii* is a small, diurnal, and arboreal lizard occurring in the areas of Caatinga domains, in northeastern Brazil. *Tropidurus cocorobensis* is a medium-sized and diurnal lizard with a relictual distribution in the northeastern semi-arid zone, occurring in the states of Bahia, Alagoas, and Pernambuco (Uetz et al. 2018. The Reptile Database; http://www.reptile-database.org; accessed 8 January 2018). Information about its natural history remains scarce. Here, we describe defensive behavior of *L. klugei* and *T. cocorobensis* in an area of caatinga, Brazil.

At 0953 h on 18 October 2017, during fieldwork in the Catimbau National Park, Pernambuco, Brazil (8.34150°S, 37.14385°W, WGS 84; 764 m elev.), a *T. cocorobensis* was captured by hand. Immediately after capture the lizard displayed death feigning behavior, remaining immobile for about three minutes.

The second observation occurred at 0923 h on 21 December 2017, also in Catimbau National Park. Here we captured two *L. klugei*; each displayed similar death feigning behavior. The lizards remained on their back, with eyes open and feet up, both for about a minute, after which time each returned to its usual position.

Our observations are consistent with thanatosis in response to the percevied threat of predation. Thanatosis has been reported in lizards including species of Tropiduridae: *Tropidurus montanus* (Machado et al. 2007. South Am. J. Herpetol. 4:136–140), *Eurolophosaurus nanuzae* (Galdino and Pereira 2002. Herpetol. Rev. 33:54), *E. divaricatus* (Gomes et al. 2004. Amphibia-Reptilia 25:321–325), *T. torquatus*, *T. hispidus*, and *T. cocorobensis* (Bertoluci et al. 2006. Herpetol. Rev. 37:472–473). Our observations are the first record of this defensive behavior for *L. klugei* and the second for *T. cocorobensis.*

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***MICROLOPHUS ATACAMENSIS* (Atacamen Pacific Iguana). DIET.** *Microlophus atacamensis* (Tropiduridae) lives in the intertidal zone of the Pacific Coast in the Atacama Desert region of northern Chile (Farina et al. 2008. J. Anim. Ecol. 77:458–468). Although it has a catholic diet that includes conspecifics, as well as insects, marine algae, crustaceans, and mollusks, there are no published records of capturing or consuming fish (Donoso-Barros 1948. Bull. Mus. Nac. Hist. Nat. Chil. 24:213–216; Farina et al., *op. cit.*; González et al. 2011. Oikos 120:1247–1255). At 1450 h on 15 January 2018, when conducting a separate study along the coast of northern Chile, approximately 9 km N of Chañaral, Region III (Atacama), Chañaral Province (26.29549°S, 70.67402°W; WGS84), we observed a medium-sized *M. atacamensis* (subadult, SVL ca. 85 mm; Vidal et al. 2002. Rev. Chil. Hist. Nat. 75:283–292) carrying a small fish (a "blenny" or "borrachilla") from a tide pool up onto the rocks. As we approached, the lizard dropped the fish and fled; we did not witness actual consumption of the fish. The fish was dead when we inspected it. We also do not know if the lizard captured the fish alive or scavenged it. This species of fish is known to cause gastric distress and drowsiness in humans that consume them (Méndez-Abarca and Mundaca 2016. Rev. Biol. Mar. Oceanogr. 51:475–481).

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***PINOYSCINCUS JAGORI* (Jagor's Sphenomorphus). PARASITES.** *Pinoyscincus jagori* is widely distributed in the Philippines (Uetz et al. 2018. The Reptile Database. http://www.reptile-database.org, accessed 29 June 2018) where it is endemic (Gaulke 2011. The Herpetofauna of Panay Island, Philippines. Edition Chimaira, Frankfurt. 390 pp.). We know of no reports of helminths from *P. jagori* and herein establish the helminth list for this skink.

Eight *P. jagori*, collected March 2017 from the Philippines, Albay Province, Municipality of Tabaco, Barangay Mariroc, Sitio Nagsipit (13.32471°N, 123.70018°E; WGS 84) and deposited in the herpetology collection of the Sam Noble Natural History Museum (OMNH), University of Oklahoma, Norman, USA (as OMNH 46152, 46306, 46307, 46310, 46311, 46314, 46315, 46318), were examined. The skinks were fixed in neutral buffered formalin and stored in 70% ethanol. The body cavity was opened by a longitudinal incision and the digestive tract was removed and opened. The esophagus, stomach, and small and large intestine were examined for helminths utilizing a dissecting microscope. Nematodes were placed on a glass slide in a drop of lactophenol, a coverslip was added and identification was made from these temporary wet mounts. Identification was made utilizing Anderson et al. (2009. Keys to the Nematode Parasites, Archival Volume. CAB International, Wallingford, Oxfordshire. 463 pp) and Gibbons (2010. Keys to the Nematode Parasites of Vertebrates, Supplementary Volume. CAB International, Wallingford, Oxfordshire, UK. 416 pp.).

We found cysts containing larvae of two species of Nematoda (*Physocephalus* sp. and larvae assignable to the Ascaridiidae) as well as adult female individuals assignable to the Superfamily Trichostrongyloidea (perhaps *Oswaldocruzia* or *Bakeria* [males necessary for determination]); 4/8 lizards had stomach cysts containing *Physocephalus* sp. (55 total cysts); 2/8 lizards had stomach cysts containing 17 ascaridiid larvae; 3/8 lizards had 4 female trichostrongyloid adults in the stomach or small intestine.

Ascaridiid nematodes are common intestinal parasites of gallinaceous birds (Anderson 2000. Nematode Parasites of Vertebrates Their Development and Transmission, 2nd ed., CABI Publishing, Oxfordshire, U.K. 650 pp.). *Pinoyscincus jagori* are presumably infected by consuming ascaridiid eggs while feeding in fecal contaminated soil. Since development beyond ascaridiid larval stages does not occur in *P. jagori*, they are best considered as paratenic (= transport) hosts. Adults of *Physocephalus* sp. occur in the stomachs of wild and domestic pigs and less commonly in tapirs, horses, cattle, and rabbits; beetles serve as intermediate hosts (Anderson, *op. cit.*). *Pinoyscincus jagori* presumably acquires *Physocephalus* sp. by feeding on infected beetles. As development beyond larval stages of *Physocephalus* does not occur in lizards, we consider them to be paratenic hosts. Goldberg et al. (1994. J. Wild. Dis. 30:274–276) described the pathology caused by *Physocephalus* larvae in the stomachs of *Sceloporus serrifer*.

Members of the Superfamily Trichostrongyloidea are found as parasites of the stomach and intestine in all terrestrial vertebrate groups; infection is acquired by ingestion of larval stages (Anderson, *op. cit.*). Ascaridiidae cysts, *Physocephalus* sp. cysts, and adults of Trichostrongyloidea in *P. jagori* are new host records. Voucher helminths were deposited in the Harold W. Manter Laboratory (HWML), University of Nebraska, Lincoln, USA as: Ascaridiidae cysts (HWML 110422), *Physocephalus* sp. cysts (HWML 110421), and Trichostrongyloidea (HWML 110420).

We thank Cameron D. Siler (OMNH) for permission to examine *P. jagori* and Jessa Watters (OMNH) for facilitating the loan. *Pinoyscincus jagori* were collected under the following permit to CDS: Republic of the Philippines, Department of Environment and Natural Resources, Biodiversity Management Bureau, Gratuitous Permit #260.

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***PLESTIODON LATICEPS* (Broad-headed Skink). REPRODUCTION.** *Plestiodon laticeps* is an arboreal skink that occupies deciduous forests of the southeastern United States. Eastern Kansas comprises the northwestern periphery of its range. Only a single nest has been documented in this portion of the range (Miller and Collins 1993. History, Distribution, and Habitat Requirements for Three Species of Threatened Reptiles in Eastern Kansas. Kansas Department of Wildlife, Parks and Tourism, final report. 30 pp.). Here we report three new nest observations that are the result of a three-year survey in forests of eastern Kansas.

One nest was found on 6 July 2017 at Marais des Cygnes Wildlife Area (38.24080°N, 94.70074°W; WGS 84) in a decayed oak log (*Quercus palustris*). The nest was under bark approximately 0.75 m above the soil surface and consisted of a clutch of 12 eggs. No adults were present at the time of the observation. The dimensions of one egg were 16 mm × 11.5 mm. The nest was checked on 10 July 2017, revealing hatched eggshells.

Two nests were found in Bourbon County (37.71031°N, 94.63465°W; WGS 84) on 12 July 2017 (Fig. 1). The nests were in the same decayed log (*Q. palustris*). Both clutches had 19 eggs and each had an adult female in attendance. The first female weighed 19.5 g and had an SVL of 9.7 cm. The dimensions of one egg from her clutch were 17.5 mm × 12 mm. The second female weighed 19 g and had an SVL of 8.6 cm. The dimensions of one egg from

FIG. 1. Two nests of *Plestiodon laticeps* on a decayed log (*Quercus palustris*).

her clutch were 17 mm × 13 mm. These nests reflect communal nesting at the level of the log, which might or might not be due to limited nest sites. Communal nesting in *P. laticeps* has not been reported in the literature (but see Vitt, pers. comm. in Doody et al. 2009. Quart. Rev. Biol. 84:229–252). The nest reported in 1993 was found under a railroad tie and surrounded by tall trees with thick understory. The nests found in 2017 were in mature forest with little understory and within 60 m of the forest edge.

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***PRISTIDACTYLUS CF. SCAPULATUS* (Burmeister's Anole). SAUROPHAGY.** *Pristidactylus cf. scapulatus* is a poorly known lizard that inhabits rocky environments of the Andean Cordillera of San Juan, Argentina (Etheridge and Williams 1985. Breviora 483:1–18). This species feeds on invertebrates (Cei 1993. Reptiles del Noroeste y Este de la Argentina Herpetofauna de las Selvas Subtropicales, Puna y Pampas. Museo Regionale di Scienze Naturali. Torino. 949 pp.) including scarabs (Scarabaeidae), darkling beetles (Tenebrionidae), fruits of *Lycium chañar* and *Ephedra breana* (Acosta et al. 2004. Herpetol. Rev. 35:171–172), and occasionally other lizards (Villavicencio et al. 2009. Herpetol. Rev. 40:225–226; Sanabria and Quiroga 2009. Herpetol. Rev. 40:349–350). Although saurophagy has been documented for *P. cf. scapulatus*, details of those events remain poorly known. Herein, we expand the knowledge of saurophagy by providing a record of predation of *Liolaemus parvus* by *P. cf. scapulatus*.

In December 2010, during a diet study of *P. cf. scapulatus*, we collected 10 adults (mean SVL = 100.2 mm), from Quebrada Vallecito (31.1791°S, 69.7092°W, WGS84; 2860 m elev.), Calingasta Department (San Juan Province, Argentina). We then collected lizard feces until the intestines were completely empty. After feces collection, the animals were released at their original points of capture. We obtained two types of samples: 49 complete fecal boli and a group of disintegrated feces. Samples were preserved in 75% isopropyl alcohol, and analyzed with a dissecting binocular microscope.

We found remnants of *L. parvus* in 30.6% of the analyzed feces (bones, skin, scales, etc.). It was possible to identify four *L. parvus* jaws, belonging to one juvenile (SVL ca. 31 mm) and three adults (SVL ca. 63 mm, 62 mm, and 58 mm). We were able to identify the prey species by comparing jaws with voucher material of *L. parvus* from the herpetological collection of the Facultad de Ciencias Exactas, Físicas y Naturales, Universidad Nacional de San Juan. The remnants of *L. parvus* found were deposited in this collection.

To our knowledge this is the first record of predation of *L. parvus* by *P. cf. scapulatus*. We thank the Secretaría de Ambiente y Desarrollo Sustentable of San Juan for granting us permission to conduct research.

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***SALVATOR RUFESCENS* (Argentine Red Tegu). DIET.** *Salvator refescens* is one of the largest lizards in South America, distributed in Argentina, Bolivia, Paraguay, and Brazil (Montero et al. 2004. Cuad. Herpetol. 18:17–32; Cabrera 2009. Lagartos del Centro de la Argentina. Fundación de Historia Natural, Córdoba, Argentina. 120 pp.). It is mainly omnivorous, feeding on a diversity of prey (Williams et al. 1993. Neotrópica 39:45–41; Lopez Juri et al. 2015. South Am. J. Herpetol. 10:132–142). Herein we describe new dietary items for *S. rufescens*.

In January 2018 a juvenile *S. rufescens* (SVL = 24 cm; mass = 1.5 kg) was found dead on the road at Encón (32.18283°S, 67.82437°W), 25 de Mayo Department, Province of San Juan, Argentina, with the Monte phytogeographic formation. It was deposited in the Herpetology Collection, Universidad Nacional de San Juan (UNSJ 4309). The body cavity was opened by a mid-ventral incision and the digestive tract was removed. The stomach and intestines were longitudinally slit and their contents were examined using a microscope. The dissection revealed four types of prey items: two classes of native seeds—*Prosopis flexuosa* (69%, by number) and *Ximenia americana* (6%)—as well as Coleoptera (Scarabaeidae) (24%) and an adult *Pleurodema nebulosum* (Anura: Leptodactylidae) (1%).

Dietary records of *S. rufescens* are detailed in reports by Williams et al. (1993. Neotrópica 39:45–41), Donadio and Gallardo (1994. Rev. Mus. Arg. Cs. Nats. B. Rivadavia Zool.

13:117–127), and Lopez Juri et al. (2015. South Am. J. Herpetol. 10:132–142). Our findings include the first records of *Pleurodema nebulosum* and *Prosopis flexuosa* seeds in the diet of *S. rufescens.*

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***TRACHYLEPIS SECHELLENSIS* (Seychelles Skink). DIET.** *Trachylepis* (previously *Mabuya*) *sechellensis* is a species of skink endemic to the Republic of Seychelles whose diet is known to include arthropods, fruit, bird feces, and dead seabird chicks (Le Maitre 1998. M.Sc. Thesis, University of Natal, South Africa). Here we present the first definitive record of predation of Gekkota by *T. sechellensis.*

At 0856 h on 6 June 2016, we observed an adult *T. sechellensis* consuming a Pacific Gecko (*Gehyra mutilata*) inside a private residence on Denis Island, Seychelles (3.804167°S, 55.6625°E). The *G. mutilata*—which was large in comparison to the skink—was alive and struggling through much of the process, which lasted for several minutes (Fig. 1). A videographic record of the event is also available at https://tinyurl.com/yc32ey9q. Although records exist of *T. sechellensis* preying upon conspecifics and the congener *T. wrightii* (Brooke and Houston 1983. J. Zool., Lond. 200:179–95), we believe this to be the first conclusive record of *T. sechellensis* preying upon a species of gecko.

Fig. 1. Seychelles Skink (*Trachylepis sechellensis*) preying upon a Pacific Gecko (*Gehyra mutilata*).

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***UTA STANSBURIANA* (Common Side-blotched Lizard). INTERSPECIFIC AGONISTIC BEHAVIOR.** *Uta stansburiana* has a large range within the deserts of North America, extending from the Pacific Coast to Texas, and from the northern reaches of the Great Basin south into Mexico. This species is also one of the most commonly encountered lizards within that geographic area. Consequently, Side-blotched Lizards have been used extensively as research subjects to study phenomena as diverse as thermal biology (e.g., Waldschmidt and Tracy 1983. Ecology 64:476–484) and reproductive physiology (e.g., Sinervo and Licht 1991. Repro. Biol. 257:252–264). They have been described as one of the best understood lizards in the world with regards to their ecology (Parker and Pianka 1975. Copeia 1975:615–632). Side-blotched Lizards have been noted for their territoriality and their frequent aggressive intraspecific interactions (Irwin 1965. Copeia 1965:99–101). Herein we report an apparent interspecific aggressive interaction between a *U. stansburiana* and a galliform bird.

Fig. 1. *Uta stansburiana* having righted itself after an attempted attack of a Gambel's Quail.

At 0942 h on 23 July 2017, CH was observing birds in the vicinity of Ivins, Utah, USA (37.16860°N, 113.67501°W, WGS 84; 939 m elev.). While photographing a pair of Gambel's Quail (*Callipepla gambelii*), CH observed an adult male *U. stansburiana* emerged onto a rock near the subjects. Seemingly unprovoked, the lizard lept from its position on the adjacent rock toward the cervical region of the male quail. The lizard then slipped from the back of the bird and fell near the bird's feet. Although unable to photograph the jumping component of the interaction, Fig. 1 shows the lizard righting itself at the bird's feet. The quail seemed unphased by the encounter.

There are three apparent hypotheses regarding the motivation for this behavior, with varying levels of viability. First, the lizard may have been attempting to attack the bird as a potential prey item. This seems unlikely given the dramatic size difference between the two animals and the typical diet (insects) for *U. stansburiana*. Second, the lizard may have been jumping to catch an insect on or near the quail. While this seems to be a plausible hypothesis, the observer did not note the presence of any insects near the birds. Third, the lizard may have been attempting to intimidate the bird in defense of its territory. Given the territorial nature of *U. stansburiana*, and the fact that this was a mature male during breeding season (Medica and Turner 1976. J. Herpetol. 1976:123–128), this third hypothesis seems the most plausible. We are not aware of any other reported accounts of *U. stansburiana* exhibiting agonistic behavior toward an avian species.

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SQUAMATA — SNAKES

AGKISTRODON PISCIVORUS **(Western Cottonmouth). PREDATION.** Previous to the following observation, the known natural predators of *Agkistrodon piscivorus* include wading birds, birds of prey, alligators, and other ophiophagous snake species including *Lampropeltis getula* (Common Kingsnake) and *A. piscivorus* (Gloyd and Conant 1990. Snakes of the *Agkistrodon* Complex: A Monographic Review. Society for the Study of Amphibians and Reptiles, Oxford, Ohio. 614 pp.). On 16 October 2017, at 1139 h, we radio tracked a large (SVL = 103.4 cm, tail length = 16.0 cm, 1490 g) male *A. piscivorus* to a mammal burrow (33.26849°N, 95.801719°W; WGS 84) 120 m away from a small tributary of the South Sulphur River located on the Cooper Lake Wildlife Management Area in northeastern Texas, USA. The individual was in ecdysis (eyes completely opaque) and was basking ca. 60 cm from the burrow's entrance. On the following day (1226 h), we radio tracked the individual to the same location. Upon approaching the immediate area, we observed a single *Canis latrans* (Coyote) depredating the snake prior to retreating into the woods and carrying off most of the carcass.

Canis latrans is known to prey on venomous snakes (Rubio 1998. Rattlesnake: Portrait of a Predator. Smithsonian Institution Press, Washington DC. 240 pp.); however, to our knowledge this is the first detailed account of predation on *A. piscivorus* by *C. latrans*. We speculate that the successful predation event on such a large venomous snake may be attributed to the snake's ecdysis state. During ecdysis snakes lose visual acuity (King and Turmo 1997. J. Herpetol. 31:310–312) and increase basking behavior for thermoregulation which may incur greater risks for predation (Gibson et al. 1989. Can. J. Zool. 67:19–23).

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BOIRUNA MACULATA **(Mussurana). COLORATION.** *Boiruna maculata* is a medium-sized dipsadine colubrid found in southern Brazil, southern Bolivia, Paraguay, Uruguay, and northwestern Argentina. Some specimens of *Boiruna maculata* (as well as *Clelia clelia*, *C. plumbea*, and species of *Pseudoboa*) are irregularly spotted with white, in some cases being almost completely white with some black patches (Boulenger 1896. Catalogue of the Snakes in the British Museum. Vol. III. xiv+727 pp.; Cei 1993. Mon. Mus. Reg. Sci. Nat. Torino. 14:1–949; Giraudo 2001. Serpientes de la Selva Paranaense y del Chaco Húmedo. Literatura of Latin América, Buenos Aires. xiv+289 pp.; Scott et al. 2006. Pap. Avul. Zool. 46:77–105). In the literature, just one specimen of *B. maculata* with white is known from Uruguay (Carreira et al. 2005. Reptiles de Uruguay. DIRAC. Fac. Ciencias. 639 pp.), deposited in the vertebrate collection of the Faculty of Sciences (State University) as ZVC-R 5459 (Fig. 1). It was collected by A. Olmos on Route 3, km 415, Dpto. Paysandú (31.85000°S, 57.86666°W; WGS 84) on 19 December 1997. This specimen was found dead on the road and parts of its body are partially destroyed. It is an adult male measuring 885 mm in SVL (head = 31.82 mm; tail = 193 mm), with 223 ventrals, 63 subcaudals, dorsal scales in –/19/15 rows.

FIG. 1. First individual of *Boiruna maculata* with white color reported for Uruguay (ZVC-R 5459).

FIG. 2. New specimen of *Boiruna maculata* with white spots collected in 2017 (MNHN 9531).

On 25 August 2017 we collected an additional specimen, killed by local people 5 km NE of Lorenzo Geyres (32.04530°S, 57.88955°W; WGS 84), Dpto. Paysandú, Uruguay. This is the second known specimen with white patches for the country (Fig. 2), and deposited in the herpetology collection of the Natural History Museum as MNHN 9531. It is an adult male measuring 986 mm in SVL (head = 25.23 mm; tail = 183 mm), with 211 ventrals, 53 subcaudals, and dorsal scales in 19/19/15 rows.

There are now 24 specimens of *Boiruna maculata* in reference collections in Uruguay (Natural History Museum and Faculty of Sciences). White-spotted patterns are scarce in Uruguay and are not taken into account in some published descriptions of the species (Meneghel et al. 2001. Clave para la Determinación de los Reptiles del Uruguay. 56 pp.; Achaval and Olmos 2007. Anfibios y Reptiles del Uruguay. Biophoto, Montevideo. 160 pp.; Carreira and Maneyro 2013. Guía de Reptiles del Uruguay. Ediciones de la Fuga, Montevideo. 285 pp.). This new specimen shows that this variation exists at least in the west-central part of Uruguay, where the two reports are ca. 20 km away from each other.

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***BOTHROCOPHIAS MICROPHTHALMUS* (Small-eyed Toad-headed Pitviper). HABITAT USE and DIET.** *Bothrocophias microphthalmus* is a medium-sized, heavy-bodied terrestrial pitviper that occurs mainly on the Amazonian slopes of the Andes in Colombia, Ecuador, and Perú (Campbell and Lamar 2004. Venomous Reptiles of the Western Hemisphere. Vol. I. Comstock Publishing Associates, Ithaca, New York. 475 pp.). It is thought to feed on anurans, lizards, and small rodents (Prado and Hoge 1948. Mem. Inst. Butantan 20:283–296; Cisneros-Heredia et al. 2006. Herpetozoa 19:17–26). Here we report the use of arboreal habitats by *B. microphthalmus*, as well as predation on two new prey items.

At 2135 h on 2 November 2011, we observed an adult *B. microphthalmus* (FHGO 9983) perched on a tree ca. 3 m above the ground (Fig. 1A). Another juvenile specimen (not collected) was observed at 1943 h on 9 November 2017 moving through a shrub ca. 1.9 m above ground level (Fig. 1B). Both specimens were found at La Zarza (3.75964°S, 78.53747°W, WGS84; 1447 m elev.), province of Zamora Chinchipe, Cordillera del Cóndor, Ecuador.

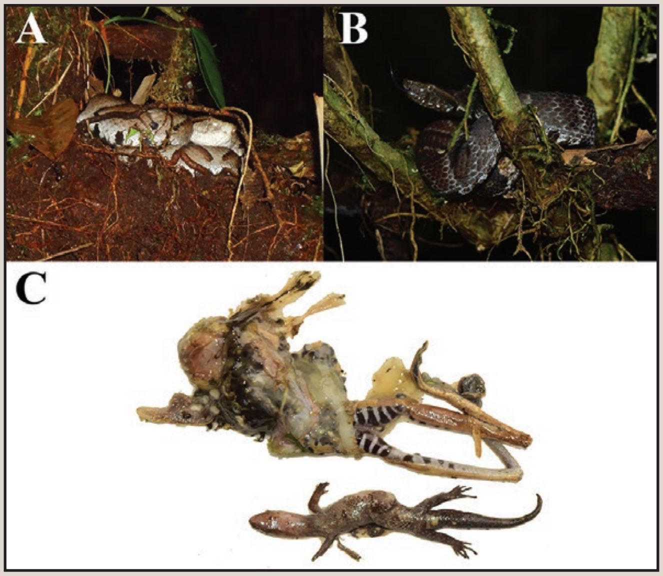

Fig. 1. A–B) Arboreal habitat use by *Bothrocophias microphthalmus*; C) regurgitated prey *Boana fasciata* and *Lepidoblepharis festae* from a juvenile *B. microphthalmus.*

Additionally, we collected a juvenile *B. microphthalmus* at 2023 h on 25 January 2017 (Fig. C; SVL = 510 mm, tail length [TL] = 72 mm), also at La Zarza (3.75600°S, 78.51041°W, WGS84; 1554 m elev.), province of Zamora Chinchipe, Cordillera del Cóndor, Ecuador, which regurgitated two prey items. The first prey item was an adult *Lepidoblepharis festae* (Brown Dwarf Gecko; SVL = 38 mm, TL = 28 mm) and the second was a gravid adult female *Boana fasciata* (Gunther's Banded Treefrog; SVL = 45 mm; Fig. 1C). All specimens (snakes and prey) were deposited at the Herpetological Collection, Fundación Herpetológica Gustavo Orcés, Quito, Ecuador.

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***BOTHROPS ERYTHROMELAS* (Caatinga Lancehead). ENDOPARASITES.** Snakes are parasitized by a wide variety of endoparasites (Silva et al. 2001. Rev. Bras. Parasitol. Veter. 10:91–93). *Bothrops erythromelas* is endemic to the Caatinga biome in eastern Brazil (Nery et al. 2016. Rev. Soc. Bras. Med. Trop. 49:680–686). In October 2015, a female *B. erythromelas* (SVL = 230 mm, tail length = 45 mm, mass = 15 g) was found dead on the road at Colonia, Exu, Pernambuco, Brazil (7.5119°S, 39.7241°W; WGS 84). The specimen was taken to the laboratory of Zoology of the Universidade Regional do Cariri-URCA, where it was measured, fixed in 10% formaldehyde, and preserved in 70% alcohol. The gastrointestinal tract was removed for dissection and examined for endoparasites using a stereomicroscope. A parasite identified as *Physaloptera* sp., larval stage, was found in the large intestine.

The only known parasites of *B. erythromelas* are pentastomids (*Cephalobaena tetrapoda*; Oliveira et al. 2015. Herpetol. Rev. 46:444); thus, our finding is the first record of the nematode *Physaloptera* sp. parasitizing *B. erythomelas*. Nematodes of the genus *Physaloptera* have been recorded in amphibians (Teles et al. 2017. Herpetol. Notes. 10:525–527), birds (Dixon and Roberson 1967. Avian Dis. 11:41–44), mammals (Saad and Nour 2012. J. Egypt. Soc. Parasitol. 42:675–690), and reptiles (Pereira et al. 2012. J. Parasitol. 98:1227–1235). Nematodes of the genus *Physaloptera* rely on insects (e.g., crickets, grasshoppers, cockroaches, and beetles) as intermediate hosts (Gray and Anderson 1982. Can. J. Zool. 60:2134–2142), which likely require subsequent consumption by a lizard or amphibian, before ultimate consumption by *Bothrops* (Costa et al. 2015. Herpetol. Notes. 8:69–98; Oliveira et al., *in press*. Herpetol. Rev; Rodrigues et al. 2016. Zool. 33:1–13; Santos-Costa-Pereira et al. 2016. Herpetol. Rev. 47:142).

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***BUNGARUS FASCIATUS* (Banded Krait). DIET.** The diet of *Bungarus fasciatus* is fairly well known. The species feeds mainly on other snakes, but it is also known to eat fish, frogs, skinks, and snake eggs. Among the snakes taken by *B. fasciatus* are *Xenopeltis unicolor* (Sunbeam Snake), *Xenochrophis piscator* (Checkered Keelback), *Amphiesma stolatum* (Buff Striped Keelback), *Ptyas mucosa* (Dhaman), *P. korros* (Indo-Chinese Rat Snake), *Boiga trigonata* (Common Cat Snake), *Daboia russelii* (Russel's Viper), *Enhydris enhydris* (Rainbow Water Snake), and *Cylindrophis ruffus* (Red-tailed Pipe Snake) (Daniels 2002. Book of Indian Reptiles and Amphibians. Oxford University Press, London. 238 pp; Tyler Knierim et al. 2017. Herpetol. Rev. 48:204–205). Here we report a new viperid snake species in the diet of *B. fasciatus*.

At 2000 h on 13 June 2017, in a small stream of Tam Thanh Commune, Quan Son District, Thanh Hoa Province, Vietnam, very close to boundary of Vietnam and Laos (20.18322°N, 104.80339°E, WGS 84; 979 m elev.), we observed an adult female *B. fasciatus* (SVL = 119.1 cm; tail length = 12.5 cm) in the process of consuming an adult female *Ovophis tonkinensis* (Tonkin Pitviper) (SVL = 68.4 cm; tail length = 12.1 cm; Fig. 1). When we found them, the *B. fasciatus* was swallowing the *O. tonkinensis* headfirst and had consumed one-fourth of it body length. The *O. tonkinensis* was dead with many bites on the body and eggs were exposed from one of the bite wounds (Fig. 2). Such severe wounds are uncommon as a result of snake predation; it is unclear if the wounds reflect a prolonged struggle between the two snakes, or perhaps are an indication that the *O. tonkinensis* was previously injured or was scavenged after being killed by a different predator. When we approached, the *B. fasciatus* quickly released the *O. tonkinensis* and began to crawl away. We collected both animals and deposited them in the collections of the Vietnam National University of Forestry (VNUF), Hanoi, Vietnam (*B. fasciatus* [VNUF RTH 2017.25]; *O. tonkinensis* [VNUF RTH 2017.24]).

Fig. 1. An adult female *Bungarus fasciatus* consuming an adult female *Ovophis tonkinensis* in a small stream of Tam Thanh Commune, Quan Son District, Thanh Hoa Province, Vietnam.

Fig. 2. Dead *Ovophis tonkinensis* with many bites and its exposed eggs.

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***CEMOPHORA COCCINEA* (Scarletsnake). PREDATION.** At 1750 h on 14 June 2016, CWC discovered a juvenile *Cemophora coccinea* (total length ca. 16 cm) in the web of an adult female Brown Widow (*Latrodectus geometricus*) at a private residence on Whitemarsh Island, Chatham County, Georgia, USA (32.03390°N, 81.01440°W; WGS84). The snake was hanging in the spider's web, which was located ca. 1.5 m below a porch and 1.2 m off the ground. The snake didn't appear to have been dead long and the spider appeared to be feeding on it. Insofar as we are aware, this is the first report of *L. geometricus*, a spider native to Africa that has become widely established around dwellings in the southeastern United States, feeding on *C. coccinea*. Although most of their diet consists of other arthropods, widow spiders (Family Theridiidae, genus *Latrodectus*) are known predators of small vertebrates, including amphibians and reptiles (McCormick and Polis 1982. Biol. Rev. 57:29–58; Tinoco et al. 2016. Herpetol. Rev. 47:641–642).

PHOTO DANIEL R. CROOK

Fig. 1. An adult female Brown Widow spider (*Latrodectus geometricus*) with a juvenile *Cemophora coccinea*.

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***CHIRONIUS SEPTENTRIONALIS* (South American Sipo). DIET.** On 8 January 2010, while conducting daytime fieldwork in the Heights of Guanapo watershed on the south slope of Trinidad's Northern Range Mountains, we encountered an adult *Chironius septentrionalis* along a first-order stream (Taylor River) in closed canopy second growth forest (10.70831°N, 61.271583°W; WGS84). The snake was on the ground attempting to swallow an adult *Hypsiboas boans* (Giant Gladiator Treefrog) headfirst. In response, the frog had inflated its body and was apparently grasping a corner of the snake's jaw in each forefoot (Fig. 1). After several minutes of unsuccessful swallowing attempts, the snake worked its left mandible around the frog's right forelimb, at which point the other limb quickly followed. Immediately, the frog emitted distress vocalizations and gave two powerful kicks with its hind-limbs that elevated the snake's fore-body off the substrate. The snake completed swallowing in a matter of minutes. We thank John Murphy for helping us identify predator and prey.

FIG. 1. An adult *Chironius septentrionalis* in the process of consuming an adult *Hypsiboas boans* along a first-order stream in Trinidad's Northern Range Mountains.

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FIG. 1. *Chrysopelea ornata* feeding on *Polypedates leucomystax* at Satchari National Park, Bangladesh.

FIG. 2. *Chrysopelea ornata* feeding on *Gekko gecko* at Satchari National Park, Bangladesh.

***CHRYSOPELEA ORNATA* (Ornate Flying Snake). DIET.** *Chrysopelea ornata* is a diurnal snake that is distributed in and around forests and human settlements from India to the Philippines. It is capable of gliding long distances from tree to tree (Socha 2011. Integr. Comp. Biol. 51:969–982) and feeds chiefly on lizards, especially geckos, but also on frogs, small birds, rodents, small bats, fish, and small snakes (Das 2010. A Field Guide to the Reptiles of Southeast Asia. New Holland Publishers Ltd., London. 376 pp.; Hasan et al. 2014. Amphibians and Reptiles of Bangladesh—A Field Guide. Arannayk Foundation, Dhaka, Bangladesh.144 pp.; Melvinselvan and Nibedita 2016. Russ. J. Herpetol. 23:311–314), which they subdue using a combination of constriction and venom (Murphy 1977. Copeia 1977:182–184). Here we present records of novel prey items and foraging behaviors for *C. ornata.*

At 1330 h on 26 August 2017, we observed a *C. ornata* (Fig. 1) feeding on a *Polypedates leucomystax* (Four-lined Tree Frog) at a general store near Satchari National Park, Bangladesh (24.12529°N, 91.44194°E, WGS84; 850 m elev.). The *C. ornata* was hanging from a wire and consumed the *P. leucomystax* headfirst over a period of 34 min.

At 1714 h on 31 July 2016, we observed a *C. ornata* (Fig. 2) feeding on a *Gekko gecko* (Tokay Gecko) at Satchari National Park (24.12661°N, 91.44240°E, WGS84; 800 m elev.) in a clay house. Presumably the *G. gecko* was attracted to the insects around the electrical light, although this observation took place ca. 1.5 h before sunset. We observed the *C. ornata* chase, catch, and constrict the *G. gecko*, which it then swallowed over a period of 41 min. There are several reports of other species of *Chrysopelea* preying or attempting to prey on oversized prey items that can take a long time to consume (Lim and Peral 1959. Malayan Nat. J. 14:33–34; Leong and Foo 2009. Nature in Singapore 2:311–316; Grossmann 1999. Sauria 21:3–6).

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***CROTALUS VIRIDIS* (Prairie Rattlesnake). REPRODUCTION: MALE-MALE-MALE COMBAT.** Male-male combat is an important component of rattlesnake reproductive behavior and has been documented in numerous species, including *Crotalus atrox, C. horridus* (*atricaudatus*), *C. viridis*, and *Sistrurus catenatus edwardsii* (Gillingham et al. 1983. J. Herpetol. 17: 265–270; Klauber 1972. Rattlesnakes: Their Habits, Life Histories, & Influence on Mankind. University of California Press, Berkeley. 1553 pp.; Wastell and Mackessy 2016. J. Herpetol. 50:594–603; Senter et al. 2014. PLoS ONE e107528.). In all of these species, male combat shows a conserved, stereotypic pattern: two males will lift their heads high above the ground, approximate one another and then attempt to pin the other individual's head to the ground by hooking the second individual's neck and rapidly forcing it downward, attempting to topple the other individual (Klauber, *op. cit.*; Carpenter 1979. Copeia 1979:638–642; Gillingham et al., *op. cit.*). The winner of this combat will then typically gain access to a female for courtship and mating. Lab studies have demonstrated that after combat, the losing male will not mate with the female, even if the winning male is removed (Gillingham et al., *op. cit.*); a similar suppressive effect on mating was observed with *Agkistrodon contortrix* (Schuett 1996. Zoo Biol. 15:209–221). However, published observations of male combat have only recorded two males in combat simultaneously. Here we report an incidence of three male *Crotalus v. viridis* in concurrent combat at the entrance to a den site utilized by hundreds of individuals.

Fig. 1. Three male *Crotalus viridis* in combat at the entrance of a hibernaculum.

Fig. 2. Three *Crotalus viridis* in combat one hour after the initial image was captured.

Crotalus viridis is a wide-ranging species found in a variety of habitats throughout the western United States, and there have been multiple observations of *C. viridis* combat in the wild (e.g., Gloyd 1947. Nat. Hist. Misc. 12:1–4; Holycross 1995. Herpetol. Rev. 26:37–38). Our photographs were captured as a result of an unrelated study where two time-lapse cameras were placed outside of a known *C. viridis* hibernaculum in Weld County, Colorado, USA. Cameras were set to take a photograph every minute from two angles at the entrance of the hibernaculum. On 14 April 2016, eight photographs were captured showing male-male combat, three of which have three individuals clearly in combat. The first two photographs, taken at 0801 and 0804 h (air temperature = 21°C), show three individuals in combat (Fig. 1). The next two photographs that capture this combat bout were at 0805 h and 0807 h, and both showed two individuals. A final photo was captured at 0855 h (26°C), and showed three individuals intertwined again (Fig. 2); between these photographs there was an additional image of three individuals intertwined; however, it cannot be clearly distinguished as combat. We believe that this is the first evidence of triple-male combat, based on the ritualized nature of the combat, in which only males are known to engage. There has only been one recorded case of a female raising her head in a manner similar to that in male combat, but this was in *Crotalus atrox* and she was the only individual present (Gillingham et al., *op. cit.*). In retrospect, it seems unusual that 2+ male combat has not been observed in species with massive denning aggregations, such as *C. viridis*, as competition for mates under such conditions should be intense. However, we have not observed copulation at this den site, in spite of hundreds of hours of observation over 15 years, so the interaction observed may simply be male dominance displays, rather than courtship-related dominance.

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***DRYMARCHON MELANURUS* (Central American Indigo Snake). DIET.** *Drymarchon melanurus* is distributed from southern Texas, USA, through Mexico in the states of Sonora, Sinaloa, Nayarit, Tamaulipas, Hidalgo, Guerrero, Oaxaca, Veracruz, Tabasco, Yucatan, and Quintana Roo, to Guatemala (Ramírez-Bautista et al. 2014. Los Anfibios y Reptiles de Hidalgo, México: Diversidad, Biogeografía y Conservación. Sociedad Herpetológica Mexicana. Pachuca, México. 387 pp.). This snake is recognized for its generalist and opportunistic diet, which includes eggs, fish, frogs, turtles, snakes, birds, and small mammals (e.g. Costa et al. 2014. Herpetol. Notes 7: 99–108; Irwin et al. 2003. J. Kansas Herpetol. 7:13–18). Nevertheless, the species of fishes preyed upon by *D. melanurus* have seldom been identified.

On 13 December 2017, at 1504 h, we encountered a *D. melanurus* (apparently an adult female) that had caught a *Rhamdia guatemalensis* (Pale Catfish; Fig. 1) in a temporary stream in the agricultural fields of the Colegio de Postgraduados campus Córdoba (18.85856°N, 96.86268°W, WGS84; 649 m elev.), Municipality of Amatlán de los Reyes, in the region of Altas Montañas of Veracruz, México. Upon noticing our presence, the snake abandoned its prey and later moved away from the site. To our knowledge this represents the first record of *R. guatemalensis* in the natural diet of *D. melanurus.*

Fig. 1. *Drymarchon melanurus* and its prey, *Rhamdia guatemalensis*, from Veracruz, México.

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***DRYMOLUBER DICHROUS* (Northern Glossy Racer) and *TANTILLA MELANOCEPHALA* (Black-headed Centipede Snake). PREDATOR-PREY RELATIONSHIP.** *Drymoluber dichrous* is a widespread colubrid snake that occurs along the eastern versant of the Andes, in the Amazon forest, on the Guiana Shield, in the Atlantic forest, and transitional areas between the Caatinga and Cerrado in northern South America (Costa et al. 2013. Zootaxa 3716:349–394). Although widely distributed, its biology remains poorly known. The species is diurnal and terrestrial, sleeping on vegetation at night (Cunha and Nascimento 1978. Publ. Avul. Mus. Par. Emílio Goeldi 31. 218 pp.; Dixon and Soini 1986. The Reptiles of the Upper Amazon Basin, Iquitos Region, Peru. Milwaukee Public Museum, Milwaukee, 154 pp.; Duellman 1978. Univ. Kansas Mus. Nat. Hist. Misc. Publ. 65:1–352). The species is known to feed mainly on lizards; amphibians and snakes (including a case of cannibalism) are rarely recorded (Cunha et al. 1985. Bol. Mus. Par. Emilio Goeldi 40:9–17; Martins and Oliveira 1998. Herpetol. Nat. Hist. 6:78–150; Borges-Nojosa and Lima 2001. Bol. Mus. Nac. Rio de Janeiro 7:1–5; Abbeg et al. 2015. Herpetol. Brasil. 4:60–63). Because of the diurnal and terrestrial habits of most of its lizard prey, it is likely that *D. dichrous* feeds mainly on the ground or at most in the lower strata of the forest.

On 8 September 2012 at the Floresta Nacional de Saracá-Taquera, central Amazonia, Pará state, Brazil (1.5186°S; 56.3750°W, WGS 84; 85 m elev.), a *D. dichrous* was found dead on a road. The specimen was collected and deposited in the herpetological collection of Museu de História Natural Capão da Imbuia in Curitiba, Paraná state (MHNCI.14248). In its stomach, we found one specimen of *Tantilla melanocephala*, a fossorial colubrid snake that lives most of time under the soil or in the leaf-litter, coming to the surface at night (Fraga et al. 2013. Guide to the Snakes of the Manaus Region-Central Amazonia. Editora INPA, Manaus. 303 pp.). This is the first record of predation of *T. melanocephala* by *D. dichorus*.

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***EUNECTES MURINUS* (Green Anaconda). DRY SEASON HOME RANGE.** Studies on *Eunectes murinus* have focused mostly on populations in the Venezuelan Llanos (Rivas et al. 2007. *In* Henderson and Powell [eds.], Biology of the Boas and Pythons, pp. 128–138. Eagle Mountain Publishing, Eagle Mountain, Utah; Rivas et al. 2016 Copeia 104:402–410), but little is known about this species in other habitats. In a study of the natural history of *E. beniensis* within the Sirionó Indigenous Territory in Bolivia (14.8031°S, 64.4352°W; WGS 84), we captured a single female *E. murinus* (total length = 205 cm). We equipped the snake with a radio transmitter (~27 g; Model F1850B, Advanced Telemetry Systems, Inc.), implanted subcutaneously using standardized procedures (Raphael et al. 1996. Proc. Wildl. Dis. Assoc. 1996:82), and radio-tracked it for 90 days (September to December 2010). We located the snake daily by foot until we either saw the animal or located it via triangulation within 2 m. We obtained 50 locations and calculated its home range using Minimum Convex Polygon (MCP) with 95% to avoid the effect of extreme data (Bath et al. 2006 J. Wildl. Manage. 70:422–434). The total home range was 0.091 ha and the core area was 0.006 ha. This home range size is much smaller than the average reported for *E. murinus* during the dry season in the Venezuelan Llanos (25.1 ha; N = 48 snakes; Rivas 2015. Natural History of the Green Anaconda with Emphasis on its Reproductive Biology. CreateSpace Independent Publishing Platform, North Charleston, South Carolina. 205 pp.), but similar to the average dry season home range of similarly sized *E. beniensis* at the same study site in Bolivia (0.29 ha, N = 4; De la Quintana et al. 2017. Amphibia-Reptilia 38:547–553).

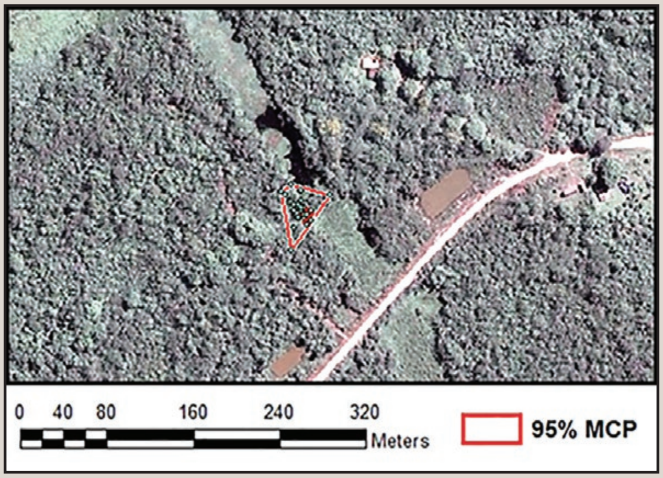

Fig. 1. Home range of *Eunectes murinus* (95% Minimum Convex Polygon) during the dry season in Bolivia.

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***EPICRATES MAURUS* (Brown Rainbow Boa). DICEPHALISM.** Dicephalism (axial bifurcation) has been documented in *Epicrates maurus* previously (Wallach 2007. Bull. Maryland Herpetol. Soc. 43:57–95), but it is unclear if a specimen exists and whether it was captive bred or wild-caught. Here we present an overlooked prodichotomous *E. maurus* neonate that was collected by W. L. Burger on 24 June 1950 in Cumanocoa, Sucre, Venezuela (10.25°N, 63.92°W, WGS 84; Figs. 1, 2). The specimen (UIMNH 63587, University of Illinois Museum of Natural History Herpetology Collection) measures 362 mm in total length (SVL = 319 mm; ventrals = 124; subcaudals = 55) and exhibits remarkably similar scalation between both heads as well as numerous partial ventral scales.

FIG. 1. Dorsal view of the dicephalic *Epicrates maurus* specimen (UIMNH 63587).

FIG. 2. Lateral (top), dorsal (middle), and ventral (bottom) views of the dicephalic anomaly in the *Epicrates maurus* specimen (UIMNH 63587).

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***HETERODON NASICUS* (Western Hognose Snake). EARLY ACTIVITY.** On 9 February 2018 at 1130 h, in Otero County, New Mexico, USA (32.09998°N, 105.66660°W, WGS 84; 1523 m elev.), we found a sub-adult male *H. nasicus* (SVL = 263 mm; tail length = 59 mm) underneath a large rock. One week prior to this encounter, on 2 February 2018, one of us (FP) inspected beneath this same rock and did not find any evidence of snake activity. Further, we did not observe burrows or tunnels under the rock that the snake could have used as a hibernaculum. As a result, the snake we encountered was likely active on the surface and sought the rock as a refuge. Average daytime air temperatures between these two observations from the nearby town of Chaparral, New Mexico, ranged 18.3–24.4°C, including six days with averages > 20°C. The seasonal activity of *H. nasicus* varies predictably along a latitudinal gradient across its distribution: 10 May–20 September in Alberta, Canada (Ernst and Ernst 2003. Snakes of the United States and Canada. Smithsonian Press, Washington, D.C. 668 pp.); 25 April–31 October in Kansas; 25 March–23 October in south-central Texas (Werler and Dixon 2000. Texas Snakes: Identification, Distribution, and Natural History. University of Texas Press, Austin. 437 pp.); and March–October in New Mexico (Degenhardt et al. 1996. Amphibians and Reptiles of New Mexico. University of New Mexico Press, Albuquerque. 431 pp.). November to February is considered a dormant period for *H. nasicus*, and for Chihuahuan Desert snake activity in general (Degenhardt et al., *op. cit.*; Werler and Dixon, *op. cit.*). To the best of our knowledge, this observation of a presumably surface active *H. nasicus* in early February from southern New Mexico is the earliest reported activity for this species.

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***HETERODON SIMUS* (Southern Hog-nosed Snake). USE OF POCKET GOPHER MOUNDS AND GOPHER TORTOISE BURROWS.** *Heterodon simus* is endemic to sandy habitats (e.g., xeric longleaf pine sandhills) of the southeastern Coastal Plain, USA, where it is strongly fossorial and adept at digging burrows and retreats in friable soils. Here, we report the use of *Geomys pinetis* (Southeastern Pocket Gopher) mounds and *Gopherus polyphemus* (Gopher Tortoise) burrows by *H. simus.*

On 25 September 1991, DJS unearthed a hatchling *H. simus* while raking by hand the sand of a newly created *Geomys pinetis* mound in intact sandhill habitat on Lake Panasoffkee Wildlife Management Area, Sumter County, Florida, USA. Similarly, on

7 January 2016, WSK found two juvenile *H. simus* while raking *G. pinetis* mounds (the snakes were in different mounds) in disturbed sandhill habitat near Lecanto, Citrus County, Florida. On 14 January 2016, WSK found an adult female (ca. 38 cm total length [TL]; UF 181095) in a *G. pinetis* mound at the same site.

Heterodon simus has not been previously reported using *Geomys* mounds. Funderburg and Lee (1968. J. Herpetol. 1:99–100) reported five species of snake, including *Heterodon platirhinos* (Eastern Hog-nosed Snake), in *Geomys* mounds in Florida, but the three sites searched were outside the range of *H. simus*. Mount (1963. Am. Midl. Nat. 70:356–385) reported finding *Tantilla relicta* (Florida Crowned Snake) and *Lampropeltis elapsoides* (Scarlet Kingsnake) in *Geomys* mounds in sandhill habitat while searching for *Plestiodon egregius* (Mole Skink). Like *P. egregius*, we suspect *H. simus* occasionally uses *Geomys* mounds for subsurface thermoregulation, particularly on cool, sunny days (Mount, *op. cit.*).

In the morning on 14 April 1990, DJS observed an *H. simus* (ca. 30 cm TL) enter an adult *G. polyphemus* burrow in disturbed sandhill habitat near Mount Dora, Lake County, Florida. In May 2011, TWH observed an adult male *H. simus* (UF 170515) ca. 1 m from the entrance of a *G. polyphemus* burrow in an active cattle pasture in Pasco County, Florida. The snake retreated toward the mouth of the burrow when approached. At 1845 h on 15 July 2017, TD and CP observed an adult *H. simus* (ca. 40 cm TL; UF 181111) at the mouth of an abandoned tortoise burrow in sandhill habitat on the Ashton Biological Preserve, 6 km W of Archer, Alachua County, Florida. The snake immediately crawled into the burrow when approached. At the same locality, an *H. simus* (ca. 25 cm TL) was photographed by a motion-activated camera at 1330 h on 21 October 2017 as it crawled across the apron of a subadult tortoise burrow toward its entrance.

Heterodon simus has not been listed as one of the many species using *G. polyphemus* burrows (e.g., Jackson and Milstrey 1989. *In* Diemer et al. [eds.], Gopher Tortoise Relocation Symposium Proceedings Technical Report No. 5, pp. 86–98. Florida Game and Fresh Water Fish Commission, Tallahassee, Florida). In addition to our observations, two records exist from Georgia of *H. simus* found at *G. polyphemus* burrows (Williamson and Moulis 1994. Herpetological Specimens in the Savannah Science Museum Collection: Volume 2 – Reptiles. Savannah Science Museum Special Publication No. 2, Savannah, Georgia). An adult female (GSU 5840) and an adult male (GSU 5841) *H. simus* were found on the surface at *G. polyphemus* burrow entrances on 27 April 1975 in intact sandhill habitat in Effingham County, Georgia (Williamson and Moulis 1994, *op. cit.*). We do not know whether these snakes were using tortoise burrows as refuges. We suspect that *H. simus* uses *G. polyphemus* burrows for refugia and to forage for *Anaxyrus* spp. and *Scaphiopus holbrookii* (Eastern Spadefoot), which are commonly found in tortoise burrows (Jackson and Milstrey 1989, *op. cit.*). *Heterodon simus* has undergone significant declines in portions of its range (Tuberville et al. 2000. J. Elisha Mitchell Sci. Soc. 116:19–40). To what extent these declines may be related to declining *G. polyphemus* and *G. pinetis* populations in areas of sympatry is unknown.

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HYPSIGLENA OCHRORHYNCHA (Coast Nightsnake). DIET. *Hypsiglena ochrorhyncha* (formerly part of *H. torquata*) is a small (< 60 cm total body length), secretive dipsadine occurring from around the Central Valley of northern California, USA, south to the Cape Region of Baja California, Mexico (Mulcahy 2008. Mol. Phylogenet. Evol. 46:1095–1115). This species is considered a habitat generalist and is reported to be both crepuscular and nocturnal (Stebbins 2012. Field Guide to Amphibians and Reptiles of California, University of California Press, Berkeley, California. 538 pp.). The diet of *H. torquata* includes mainly phrynosomatid lizards and squamate eggs and occasionally insects, salamanders, anurans, other lizards, and small snakes (Rodríguez-Robles et al. 1999. Copeia 1999:93–100; Ernst and Ernst 2003. Snakes of the United States and Canada. Smithsonian Books, Washington, DC. 668 pp.). Although the majority of specimens examined by Rodríguez-Robles et al. (1999, *op. cit.*) are from the range of what is now *H. ochrorhyncha*, the only *Sceloporus* identified to species in this study was *S. graciosus*. Weaver (2010. J. Herpetol. 44:148–152) reported *S. occidentalis* in the diet of *H. chlorophaea*.

On 13 September 2017, at approximately 1845 h, an adult *H. ochrorhyncha* (ca. 35 cm total length) was observed in the early stage of consuming a freshly killed adult *Sceloporus occidentalis* (ca. 7 cm SVL) head first (Fig. 1). The event occurred near a rubble rock wall in the front yard of the home of one of us (BMA) located in the rural community of Del Dios, San Diego County, California, USA (33.06656°N, 117.02027°W; WGS84; elev. 104 m). After 5 min, the snake, still gripping the lizard, was removed from the front yard and relocated about 25 m to a lot consisting of native coastal sage scrub habitat. Despite being handled, the snake completely consumed the *S. occidentalis* in about 30 min. To our knowledge, this is the first report of *S. occidentalis* in the diet of *H. ochrorhyncha* and represents a diurnal-crepuscular predation event by *Hypsiglena* (Rodríguez-Robles, et al. 1999, *op. cit.*; Lance 2012. Son. Herpetol. 25:99–100). Photographic vouchers were deposited at the San Diego Natural History Museum herpetology photographic collection (SDNHM-HerpPC 5360–5364).

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Fig. 1. Adult *Hypsiglena ochrorhyncha* consuming an adult *Sceloporus occidentalis* in the community of Del Dios, San Diego County, California, USA.

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***INDOTYPHLOPS BRAMINUS* (Brahminy Blindsnake) and *DINODON RUFOZONATUM* (= *LYCODON RUFOZONATUS*) (Red-banded Snake). PREDATION and DIET.** *Indotyphlops braminus* and *Dinodon rufozonatum* are both native species that are widely distributed in Taiwan. *Indotyphlops braminus* is known to be a ground-dwelling species and *D. rufozonatum* is considered a terrestrial generalist, preying on insects, fish, frogs, toads, snakes, lizards, and birds (Kidera and Ota 2008. Current Herpetol. 27:23–27; Tu 2004. Big Surprise of Snakes. Yuan-Liou Publishing Co. Ltd., Taipei. 279 pp.). Here we report an unusual case of predation by *D. rufozonatum* on *I. braminus* and excretion of the *I. braminus* through the cloaca without digestion.

The *Dinodon rufozonatum* (female; SVL = 582 mm; 47 g) was captured at Chinyang Farm, Shoufeng Township, Hualien County, Taiwan (23.90632°N, 121.50896°E; WGS84) at 1910 h on 20 August 2017 while crawling across a cement floor. When we palpated its stomach, we noticed a small item that seemed like the head of a blindsnake emerging from its cloaca. After confirming that the item was not a part of an organ of *D. rufozonatum* and was not moving, we used forceps to gently remove the item. The item was identified as an adult *I. braminus*, which was ca. 155 mm in total length (Fig. 1). In addition to the *I. braminus*, two leathery egg shells fell out of the cloaca. Although the *I. braminus* was already dead before being pulled out, the individual was almost uninjured except for a few body parts that seemed to be compressed, causing slight damage. The evidence suggests that *I. braminus* passed through the stomach and intestine of *D. rufozonatum* without digestion.

As far as we know, our observation is the first case of this phenomenon confirmed in a snake that had fed on an *I. braminus*. Amazingly, similar cases have been reported in which *I. braminus* have been swallowed by a predator and remained intact after passing through the digestive system. Two species of toads, *Duttaphrynus melanostictus* (O'Shea et al. 2013. Herpetol. Notes 6:467–470) and *Rhinella marina* (Zlotnik et al. 2017. Herpetol. Rev. 48:675), excreted intact *I. braminus*. In the *D. melanostictus* case, the *I. braminus* remained alive for a while after struggling out of the cloaca under its own power. Based on these cases, we assume that *I. braminus* may have specialized scale structures that delay digestion by predators, which gives it limited time to escape the predator's digestive system. Further work on the morphological and physiological adaptations of *I. braminus* are required to test this hypothesis.

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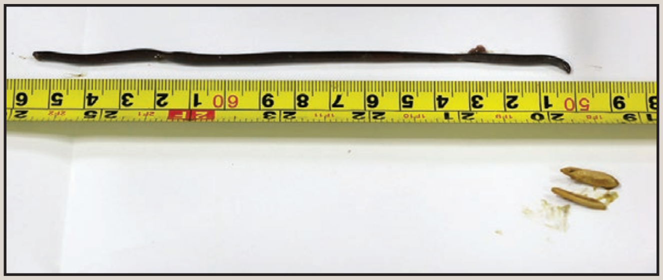

FIG. 1. *Indotyphlops braminus* excreted from the cloaca of *Dinodon rufozonatum*. Two leathery egg shells were also excreted from the cloaca after pulling out the *I. braminus*.

***LAMPROPELTIS MICROPHOLIS* (Ecuadorian Milksnake, Falsa Coral Interandina). DIET.** *Lampropeltis micropholis* is a member of the *L. triangulum* complex and is distributed from eastern Costa Rica, throughout Panama, and south to Ecuador (Ruane et al. 2014. Syst. Biol. 63:231–250). In Colombia, *L. micropholis* occurs between the Caribbean Coast and the western flank of the Cordillera Occidental (Western Cordillera), occupying the inter-Andean valleys of the Cauca and Magdalena rivers basin, and both Occidental and Central cordilleras (Dunn 1944. Caldasia 3:155–224; Rojas-Morales 2012. Phyllomedusa 11:135–154). Species in the *L. triangulum* complex in Mexico and the United States are known to consume a wide array of small mammals, lizards, snakes, birds and their eggs, and occasionally amphibians, fish, and invertebrates (Ernst and Ernst 2003. Snake of the United States and Canada. Smithsonian Books, Washington, D.C. 668 pp.; Rodríguez and Drummond 2000. J. Herpetol. 34:139–142; Aguilar-López and Pineda 2013. Herpetol. Notes. 6:89–90), but information on the ecology and diet of *L. micropholis* is lacking. Herein we present the first record of consumption of *Mus musculus* (House Mouse; Rodentia: Muridae) by *L. micropholis*.

On 15 April 2016, a female *L. micropholis* (total length = 730 mm; SVL = 630 mm; Fig. 1), was killed by a farmer at the Tesorito farm (5.03156°N, 75.44865°W, WGS 84; elev. 2164 m), Manizales, Cordillera Central of Colombia. The specimen was deposited at the Museo de Historia Natural de la Universidad de Caldas (MHNUC-0302). There, MSCO and JMHL opened the specimen and found a mouse consumed headfirst. The mouse was identified by HERC as a juvenile (last molars not erupted) *Mus musculus*, based on the presence of molars with cusps organized in three longitudinal rows, and small body size (head and body length = 60 mm; tail length = 63 mm). *Mus musculus* is an exotic species in Andean ecosystems of the Cordillera Central of Colombia.

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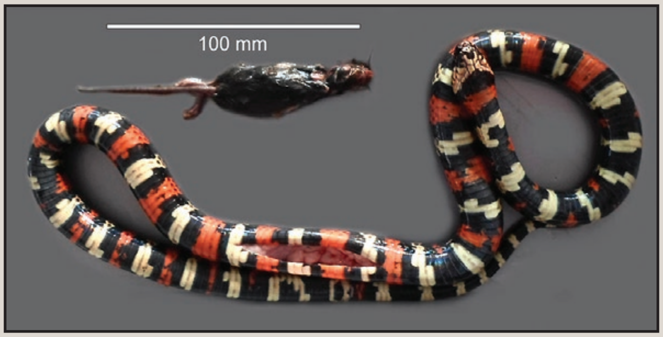

FIG. 1. House mouse (*Mus musculus*) preyed upon by a female *Lampropeltis micropholis* from the Cordillera Central of Colombia.

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***MASTICOPHIS FLAGELLUM* (Coachwhip). DIET.** *Masticophis flagellum* feeds on a variety of prey, including invertebrates, frogs, turtles, snakes, lizards, birds, and small mammals (Ernst and Ernst 2003. Snakes of the United States and Canada. Smithsonian Institution Press, Washington, D.C. 668 pp.). We dissected specimens of *M. flagellum* and noted two novel prey items for this species. SRSU 5641, a 104.3 cm SVL *M. flagellum* collected 29 April 1985 on Hwy 385, 29 km S Marathon in Brewster County, Texas, USA (approximate coordinates: 29.96246°N, 103.25664°W; WGS 84), contained an individual of *Aspidoscelis gularis* (Texas Spotted Whiptail). SRSU 6799, a 95.6 cm SVL *M. flagellum* collected DOR on 15 April 2016 in Brewster County, Texas, USA (29.74698°N, 103.16012°W; WGS 84), contained a small *Bogertophis subocularis* (Trans Pecos Rat Snake) (SVL = 31.4 cm). To the best of our knowledge this represents the first record of *M. flagellum* feeding upon *A. gularis* or *B. subocularis* (Ernst and Ernst, *op. cit.*).

Specimens examined for this study were from the James F. Scudday Vertebrate Collections at Sul Ross State University. Stomach contents were retained and stored in 70% EtOH. This research was supported by the Ronald E. McNair Post-Baccalaurete Achievement Program at Sul Ross State University.

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***MICRUROIDES EURYXANTHUS* (Sonoran Coralsnake). BEHAVIOR.** *Micruroides euryxanthus* is presumably common, but secretive, and therefore infrequently encountered. Though it is occasionally seen abroad during the day, it is primarily nocturnal in activity. *Micruroides euryxanthus* is found at elevations ranging from ca. 58 m to > 1500 m encompassing habitats from low deserts to oak/pine woodlands. Drainages including dry washes are frequented by this species. Here, we report three observations of *M. euryxanthus* entering harvester ant (*Pogonomyrmex* sp.) nests. All observations occurred within a few hundred meters of each other in the same dry wash traversing a bajada in Arizona Upland Subdivision Sonoran Desert (Brown 1994. Biotic Communities: Southwestern United States and Northwestern Mexico. University of Utah Press, Salt Lake City, Utah. 342 pp.), Superstition Mountains, Pinal Conty, Arizona, USA.

At 2047 h MST on 1 July 2016, we observed an *M. euryxanthus* (total length [TL] ca. 380 mm) with the posterior two thirds of its body protruding from a harvester ant nest. The snake was lifted from the nest; it appeared alert and healthy and was released. Interestingly ants were neither attacking the snake, nor did they appear agitated by the snake's presence.

At 2024 h MST on 11 August 2017, a large adult *M. euryxanthus* (SVL = 422 mm; 23 g) was observed exiting a harvester ant nest. The posterior few cm were still in the nest when sighted. The snake was disturbed by the light from a headlamp and retreated into the ant nest. It emerged again 2 min later but again retreated into the nest when disturbed by the light of the headlamp. The snake exited the nest 3 min later and was captured. As before, none of the ants attacked the snake or appeared to be agitated by its presence.

At 2019 h MST on 26 August 2017, a presumably recently hatched (TL ca. 127 mm) *M. euryxanthus* was observed outstretched near (ca. 24 cm) a harvester ant nest. The snake was positioned in a manner that gave the impression it had just exited the nest. We watched the snake for about 5 min. It moved slowly, circling the ant nest's entrance. The snake paused each time it contacted an ant but as before, the ants paid no attention to the snake. The snake soon entered the nest, passing among numerous ants moving the opposite direction, none of which molested the snake.

These are the first documented observations of *M. euryxanthus* entering ant nests. *Micruroides* preys primarily on small snakes and threadsnakes (*Rena* spp.) are favored among these (Lowe et al. 1986. The Venomous Reptiles of Arizona. Arizona Game and Fish Dept., Phoenix, Arizona. 113 pp.; Vitt and Hulse 1973. Herpetologica 29:301–304). Approximately a third of the diet of *Rena humilis* (30.1%) and *R. dulcis* (29.8 %) is comprised of ant larvae and pupae (Punzo 1974. J. Herpetol. 8:153–156). Both of these *Rena* species are sympatric with *M. euryxanthus* in southeast Arizona (Brennan and Holycross 2009. A Field Guide to the Amphibians and Reptiles in Arizona. Arizona Game and Fish Dept., Phoenix, Arizona. 150 pp.). *Micruroides euryxanthus* may enter ant nests in search of foraging *Rena* sp.

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***MICRURUS DUMERILII* (Dumeril's Coralsnake, Coral de Dumeril). DIET.** *Micrurus dumerilii* is a medium-sized coralsnake (maximum total length = 954 mm; Meneses-Pelayo and Caicedo-Portilla 2015. Herpetol. Rev. 46:647) that inhabits lowland (0–600 m) wet/moist forest in northwestern Venezuela, northern, central, and eastern Colombia, and the Pacific Coast from southeastern Panama to northern Ecuador (Campbell and Lamar 2004. The Venomous Reptiles of the Western Hemisphere. Comstock Publishing Associates, Cornell University Press, Ithaca, New York. 976 pp.; Praire et al. 2015. Mesoam. Herpetol. 2:253–259). The species is poorly studied, but is thought to feed on small vertebrates like lizards and fishes (Campbell and Lamar 2004, *op. cit.*). Here we present the first record of predation of *Caecilia thompsoni* (Gymnophiona: Caecilidae) by *M. dumerilii.*

At 1051 h, on 03 of May of 2017, in the Reserva Río Manso (5.67169°N, 74.77786°W, WGS 84; elev. 217 m) in the department of Caldas, Colombia, we found a *M. dumerilii* (total length = 720 mm) ingesting a specimen of *C. thompsoni* (SVL = 923 mm) headfirst (Fig. 1). The snake was released, but the *C. thompsoni* was deposited in the Museo de Historia Natural de la Universidad de Caldas (MHN-UC 0835). This is the first record of predation of a caecilian by *M. dumerilii.*

We thank the Vertebrate Zoology course (2017-1 period) of the Biology program of the Universidad de Caldas for support in the field.

FIG. 1. *Micrurus dumerilii* ingesting a *Caecilia thompsoni* in the Reserva Río Manso, Colombia.

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***MICRURUS IBIBOBOCA*. ENDOPARASITES.** The genus *Micrurus* occurs from Argentina to the southern United States (Campbell and Lamar 2004. The Venomous Reptiles of the Western Hemisphere. Cornell University Press, Ithaca, New York. 774 pp.). *Micrurus ibiboboca* is widely distributed in Brazil (south of the Amazon, Bahia, Sergipe, S Ceará, Alagoas, Maranhão, Paraíba, Pernambuco, Piauí, Sergipe, Rio de Janeiro, Rio Grande do Norte) (Uetz et al. 2017. http://www.reptile-database.org. Accessed on 21 February 2018). Several studies have examined helminthofauna of snakes from northeastern Brazil (Almeida et al. 2006. Brazil. J. Biol. 66:559–564; Almeida et al. 2007. Brazil. J. Biol. 67:759–763; Almeida et al. 2008. Brazil. J. Biol. 68:193–197; Araujo Filho et al 2013. Herpetol. Rev. 44:43–43; Oliveira et al 2015. Herpetol. Rev. 46:444–444), but the only known infection for *M. ibiboboca* is Pentastomida: *Raillietiella* sp. (Almeida et al. 2007, *op. cit.*).

In December 2015, a female *M. ibiboca* (SVL = 94 mm, TL = 30 mm, 130 g) was found dead on the road in the municipality of Farias Brito (39.533194°W, 06.783278°S, WGS84; 309 m elev.), Ceará, Brazil. The specimen was deposited in the collection of the laboratory of Zoology of the Regional University of Cariri-URCA. The gastrointestinal tract was removed and endoparasites were examined using a stereomicroscope. A parasite identified as a larval stage *Physaloptera* sp. was found in the stomach of the *M. ibiboboca*.

Nematodes are the major endoparasites of the digestive tract of snakes; most commonly found in snakes are those of the genus *Physaloptera* (Barbosa et al. 2006. Revista Biología Ciências da Terra 6:1–19). The intermediate hosts of *Physaloptera* are invertebrates, including crickets (Orthoptera), locusts (Orthoptera), cockroaches (blattodea), and beetles (Coleoptera) (Gray and Anderson 1982. Can. J. Zool. 60:2134–2142). The present study establishes the first record of the nematode *Physaloptera* sp. parasitizing *M. ibiboboca*.

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***MICRURUS NARDUCCII MELANOTUS* (Andean Black-backed Coralsnake). MAXIMUM LENGTH.** On 26 July 1964, J. Bowerman collected a large female *Micrurus narduccii melanotus* from Limoncocha, Sucumbíos Province, Ecuador (0.41°S, 76.63°W; WGS 84). The specimen (UIMNH 61058, University of Illinois Museum of Natural History Herpetology Collection) measures 1173 mm total length (SVL = 1131 mm). The previous reported maximum length for *M. n. melanotus* was 1157 mm total length (SVL = 1117 mm) and belongs to USNM 232473 (National Museum of Natural History, Smithsonian Institution, Department of Vertebrate Zoology, Washington D.C.) collected from Río Corrientes, Pastaza Province, Ecuador (Roze and Bernal-Carlo 1987. Boll. Mus. Reg. Sci. Nat. Torino 5:573–608). Accordingly, UIMNH 61058 represents a new maximum length record for *M. n. melanotus*, the larger of the two allopatric subspecies (Campbell and Lamar 2004. The Venomous Reptiles of the Western Hemisphere. Comstock Publishing Associates, Ithaca, New York. 962 pp.; Valencia et al. 2016. Serpientes Venenosas del Ecuador. Fundación Herpetológica Gustavo Orcés, Quito, Ecuador. 653 pp.).

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***OXYRHOPUS GUIBEI* (False Coralsnake). DEFENSIVE BEHAVIOR.** Thanatosis (death feigning) is one of the most well-known and widespread defensive tactics in different animal groups (Mendonza 2009. Herpetotropicos 5:67; Miyatake et al. 2009. Proc. R. Soc. Lond. B 276:2763–2767; Toledo et al. 2010. J. Nat. Hist. 44:31–32; Brauder et al. 2015. Herpetol. Conserv. Biol 10:559–571) that depresses predatory behavior and stimuli (Pasteur 1982. Annu. Rev. Ecol. Syst. 13:169–199). However, its effectiveness is still controversial and debated in the literature (Gregory et al. 2007. J. Comp. Psychol. 121:123–129). Thanatosis occurs most frequently after an animal is manipulated or disturbed (Muscat et al. 2016. Herpetol. Notes 9:95–97), and has been recorded in a variety of snakes, mostly within the Colubridae and Dipsadidae (e.g., Mendonza, *op. cit.*; Brauder et al., *op. cit.*; Muscat et al., *op. cit.*; Costa-Expósito et al. 2017. Bol. Asoc. Herpetol. Esp. 28:2017). Herein, we provide the first report of thanatosis in *O. guibei*.

On 6 October 2017, in a disturbed area of Cerrado (17.1325°S, 46.5447°W, WGS84; 542 m elev.), one of us (FDS) captured an *O. guibei* (total length = 96.3 cm; SVL = 78 cm) in the Municipality of Paracatú, state of Minas Gerais, Brazil (Fig. 1). When the animal was manipulated to be released, it kept its body extremely rigid and when placed on the ground, it remained motionless with its ventral region facing upward while keeping its mouth open. The animal was then handled again, but the behavior continued. The

Fig. 1. Death-feigning behavior in an *Oxyrhopus guibei* from Paracatu, Minas Gerais, Brazil.

behavior was maintained for approximately 3 min, thereafter the animal quickly restored to the upright position.

The defensive behaviors previously reported in the literature for *O. guibei* include struggling and discharging cloacal secretions (Gaiarsa et al. 2013. Pap. Avul. Zool. 53:261–283). Additionally, defensive behaviors previously reported for the genus *Oxyrhopus* include body thrashing and lateral compression, biting, tail vibration, and cloacal secretion (Martins and Oliveira 1998. Herpetol. Nat. Hist. 6:78–150; Sawaya et al. 2008. Biota Neotrop. 8:127–149; Gaiarsa et al., *op. cit.*). Thus, this is the first study to report thanatosis in the genus *Oxyrhopus*.

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***OXYURANUS MICROLEPIDOTUS* (Inland Taipan). DIET.** The Australasian elapids of the genus *Oxyuranus* (taipans) are well-documented for their partiality to mammalian prey (Shine 1993. Australian Snakes: A Natural History. Reed Publishers, Chatswood, New South Wales. 210 pp.). *Oxyuranus microlepidotus* is a large (total length [TL] = 2 m), diurnal, terrestrial, elapid endemic to arid Australia, inhabiting sparsely-vegetated cracking plains associated with drainage systems in south-western Queensland and north-eastern South Australia (SA); and with an isolated population in central SA (Read 1994. Trans. R. Soc. S. Aust. 118:143–145; Wilson and Swan 2017. A Complete Guide to Reptiles of Australia. Reed New Holland, Sydney. 518 pp.). *Oxyuranus microlepidotus* appears to be a specialized feeder on *Rattus villosissimus* (Long-haired Plains Rat) and as such, the distribution and population ecology of both predator and prey are intimately correlated (Read, *op. cit.*). Although the diet of *O. microlepidotus* is widely documented, observations of foraging or predator-prey interactions appear unrecorded in the field due to the remote distribution and secretive nature of the species. Herein, we report a predator-prey interaction and a novel dietary item of *O. microlepidotus*, in an isolated population of the species' range.

On 16 December 2015 at 0725 h (light cloud cover; road surface temperature = 29°C), a large (TL = ca. 170 cm, taken from track markings) *O. microlepidotus* was located active on the Mt Barry Road to Oodnadatta northeast of Coober Pedy, South Australia, Australia (28.8781°S, 134.8539°E, WGS84; 144 m elev.). The individual was observed on the side of the road beginning to consume a deceased adult (SVL = ca. 6.0 cm) female *Sminthopsis crassicaudata* (Fat-tailed Dunnart). The snake released the prey and fled as our vehicle approached. The *S. crassicaudata* appeared to have died very recently, as indicated by the five live joeys still attached to it. We suspect that the *S. crassicaudata* was bitten by the *O. microlepidotus* shortly before we arrived and perished on the road following envenomation. There appeared to be no physical injury to suggest death from an alternate cause.

Being nocturnal, it is likely that the *S. crassicaudata* was sheltering in a nearby soil crack at the time it was bitten, and later succumbed to the effects of envenomation after fleeing to the road. This is plausible given that *Oxyuranus* are known to strike-and-release their prey, allowing it to escape and succumb to the venom, and the corpse later tracked through chemoreception (Shine and Covacevich 1983. J. Herpetol. 17:60–69). Previous reports indicate the absence of *R. villosissimus* at this locality, and postulate that *Pseudomys australis* (Plains Rat) constitutes a primary prey item substitute (Read, *op. cit.*). Given that *S. crassicaudata* extensively utilize the same soil cracks as *O. microlepidotus* (Read, *op. cit.*; Waudby and Petit 2017. Integr. Zool. 12:237–249), it is likely that *S. crassicaudata* constitutes a major, if not opportunistic, food source for *O. microlepidotus* at this locality. To the best of our knowledge, this record provides the first account of foraging behavior in a wild *O. microlepidotus* and further documents interest in prey items other than *R. villosissimus*.

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***PSAMMOPHIS PUNCTULATUS TRIVIRGATUS* (Southern Speckled Sand Snake). DIET.** Although *Psammophis punctulatus* is a very common species in East Africa, its natural history is poorly known (Cottone and Bauer 2009. Afr. J. Herpetol. 58:126–130). The distribution of the eastern African subspecies, *P. p. trivirgatus*, is reported to comprise Somalia, Tanzania, and Kenya, where it occurs in dry savanna and semi-desert from sea level up to 1400 m. The nominate form occurs in Ethiopia, Eritrea, South Sudan, Sudan, Uganda, Egypt, and Djibouti (Spawls et al. 2004. A Field Guide to the Reptiles of East Africa. A & C Black Publishers Ltd., London, Great Britain. 543 pp.; Baha El Din 2006. A Guide to the Reptiles and Amphibians of Egypt. The American University in Cairo Press. Cairo, Egypt. 359 pp.). Parker (1949. Zool. Verh. 6:1–115) suggested that the nominate form might meet *trivirgatus* near Lake Turkana based on a specimen from northwestern Kenya. Morphologically and genetically, *P. punctulatus* sensu lato (s.l.) has been placed into the *P. schokari* group (Broadley 1977. Arnoldia 8:1–29; Broadley 2002. Afr. J. Herpetol. 51:83–119, Kelly et al. 2008. Mol. Phylogenet. Evol. 47:1045–1060). Nevertheless, a thorough reassessment of *P. punctulatus* (s.l.) is needed to clarify whether *P. p. trivirgatus* deserves full species status. The number of subcaudal scales (Largen and Rasmussen 1993, *op. cit.*; *P. p. punctulatus* 158–178, *P. p. trivirgatus* 143–163) is unreliable since *P. punctulatus* can autotomize its tail to escape from predators and subsequently regenerate a terminal point at the truncated tip (Broadley 1987. J. Herpetol. Assoc. Afr. 33:18–19).

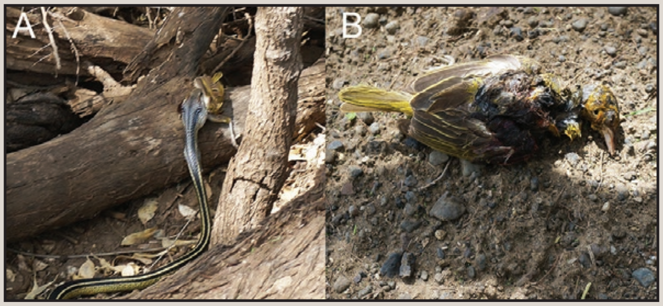

FIG. 1. A) *Psammophis punctulatus trivirgatus* in the process of swallowing a *Ploceus intermedius* (Lesser Masked Weaver); B) bird after it was dropped by the snake.

At 1015 h on 04 April 2017, during a survey of the fauna and flora of the Sibiloi National Park on the eastern shore of Lake Turkana, a medium-sized *Psammophis punctulatus trivirgatus* (SVL = 103.8 cm; tail length = 41 cm; subcaudal scales = 105 [tail mutilated and regenerated], ventral scale rows = 194, midbody scale rows = 17) was observed preying upon an adult *Ploceus intermedius* (Lesser Masked Weaver) in a riverine gallery forest near Alia Bay, Lake Turkana, Marsabit County, northern Kenya (3.68374°N, 36.29167°E, WGS 84; 387 m elev.). When we detected the snake it had already begun to swallow the bird headfirst. When the snake saw us approach, it dropped the freshly dead bird and tried to hide under the fallen tree stem it was sitting on. We captured the snake and collected the *P. intermedius* (Fig. 1). The voucher specimen of the *Psammophis punctulatus trivirgatus* was accessioned into the collection of the National Museums of Kenya, Nairobi (NMK S4604). Our observations match with previous publications stating that *P. punctulatus* (s.l.) is diurnally active and partially arboreal (Spawls et al. 2004, *op. cit.*). However, *P. punctulatus* (s.l.) preying on a bird has never been documented before.

There is only limited information regarding the diet of *Psammophis* snakes in general and *Psammophis punctulatus trivirgatus* in particular. *Psammophis punctulatus* (s.l.) is known to feed on lizards (lacertids such as *Heliobolus* or *Latastia* spp., agamids, scincids) or other snakes, and it is big enough to be able to capture other vertebrate prey (Loveridge 1936. Bull. Mus. Comp. Zool. 79: 207–337, Spawls et al. 2004, *op. cit.*, Largen and Spawls 2010. The Amphibians and Reptiles of Ethiopia and Eritrea. Edition Chimaira. Frankfurt am Main, Germany. 693 pp.). Birds are rarely reported to be prey of *Psammophis* species. An analysis of the stomach contents of nine *Psammophis* (and one *Psammophylax*) species from southern Africa (700 specimens) revealed that prey consists mainly of lizards and other snakes as well as rodents (Shine et al. 2006. Copeia 2006:650–664). Luiselli et al. (2004. Amphibia-Reptilia *25*:415–423) only found reptiles, small mammals, and sporadic arthropods in the diet of two taxa of the *Psammophis 'phillipsi'* complex from southern Nigeria. Stomach contents of 242 dissected *Psammophis crucifer* specimens from southern Africa also exclusively contained reptiles and very few arthropods (Cottone and Bauer 2010. Copeia 2010:578–590). Schmidt and Branch (2005. Ostrich 76:80–81) report a specimen of *Psammophis* cf. *phillipsii* with an egg presumably from a bee-eater (*Merops breweri*) in its stomach. Various field guides state that birds are prey of *Psammophis mossambicus* (Spawls et al. 2004, *op. cit.*), *Psammophis orientalis* (Branch 2005. A Photographic Guide to the Snakes and Other Reptiles and Amphibians of East Africa. Struik Nature, Cape Town, South Africa. 144 pp.), *Psammophis schokari* (Baha El Din 2006, *op. cit.*), *Psammophis subtaeniatus* (Branch 1998. Field Guide to the Snakes and Other Reptiles of Southern Africa. Struik Nature, Cape Town, South Africa. 399 pp.; Marais 2004. A Complete Guide to the Snakes of Southern Africa. Struik Publishers, Cape Town, South Africa. 312 pp.; Branch 2016. Pocket Guide: Snakes and Other Reptiles of Southern Africa. Third reworked edition. Struik Nature, Cape Town, South Africa. 160 pp.), *Psammophis brevirostris*, and *Psammophis leopardinus* (Marais 2004, *op. cit.*), but we cannot say whether this information is anecdotal or based upon published sources. Although we did not observe how the bird was caught it would be most unusual for a *Psammophis* to scavenge on a dead bird. To our knowledge, our observation represents the first verified documentation for *P. punctulatus trivirgatus* preying upon an adult bird.

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PSEUDOERYX PLICATILIS (South American Pond Snake). DIET. The aquatic snake *Pseudoeryx plicatilis* (Colubridae, Dipsadinae) is associated with flooded forests, forest lakes, and streams in South America (Dixon and Soini 1986. The Reptiles of the Upper Amazon Basin, Iquitos Region, Perú. Milwaukee Public Museum, WI, USA. 154 pp.; Mertens 1965. Senckenbergiana Biologica 46:279–285). Previous diet items reported for this snake include frogs and fishes (Dixon and Soini, *op. cit.*; Kaefer and Montanarin 2010. Herpetol. Rev. 41:372).

On 30 January 2007, at 1500 h, I observed an adult female *P. plicatilis* (SVL = 1070 mm; tail length = 130 mm) ingesting an eel-like freshwater fish *Synbranchus marmoratus* (Teleostei: Synbranchidae; total length = 900 mm) in a flooded area in a palm swamp forest dominated by *Mauritia flexuosa* (locally known as *Aguaje*). The snake had captured the fish headfirst and had ingested approximately 1/4 of the fish body. Both individuals were placed in a large snake bag, and the snake regurgitated the prey 15–20 min later. The site was located 1.5 k NW of Los Amigos Biological Station, Manu Province, Madre de Dios Region, Perú (12.5597°S, 70.1103°W, WGS 84; 244 m elev.). Both specimens were deposited in the Museo de Historia Natural Universidad Ncional Mayor de San Marcos, Lima, Peru (*P. plicatilis* = MUSM 24359; catalog number not available for *S. marmoratus*). This is the first record of a large prey item for *P. plicatilis*; in this case, measuring almost the size of its adult predator. A previous diet record of *P. plicatilis* included a smaller individual of *Synbranchus* sp. (total length = 349 mm) in Brazil (Kaefer and Montanarin, *op. cit.*). Also, this record extends the known geographic distribution of *P. plicatilis* in the Madre de Dios Region by approximately 98 km to the west; only one locality, Puerto Maldonado, was included in the most recent geographic distribution map of this species (Scartozzoni et al. 2010. Check List 6:534–537), even though additional localities east and south from Puerto

Fig. 1. Dorsal (A) and ventral (B) views of adult female of *Pseudoeryx plicatilis* (MUSM 24359) and dorsal (C) and ventral (D) views of its prey, *Synbranchus marmoratus*.

Maldonado have been reported in the literature (Duellman 2005. Cusco Amazónico: The Lives of Amphibians and Reptiles in an Amazonian Rainforest. Comstock Publishing Associates. Ithaca, New York. 433 pp.).

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***PTYAS MUCOSA* (Oriental Ratsnake). COLORATION / ALBINISM.** *Ptyas mucosa* is the most widely distributed diurnal snake species found in and around human habitation through South and Southeast Asia (Auliya 2010. Conservation status and impact of trade on the Oriental Rat Snake *Ptyas mucosa* in Java, Indonesia. TRAFFIC Southeast Asia, Petaling Jaya, Selangor, Malaysia. 39 pp.).The body color of this species may vary from yellow, olive, or brown to black with brown or black reticulated markings on the dorsal side and dark crossbars on the ventral side. A single orange-colored morph of the species has been reported from Gujarat, India (Vyas 2013. Reptile Rap 15:43–45).

At 1800 h on 18 September 2015, an albino *P. mucosa* (SVL = 38.8 cm; tail length = 15.2 cm) was rescued by one of us (BM) from a residential area at Nalconagar in Angul District of Odisha, India (20.8519°N, 85.1622°E; WGS84). The specimen was a subadult female, approximately two months old, with a slight bruise on the anterior part of the body. The snake had pink eyes and white and yellow body scales with no dark pigment (Fig. 1).

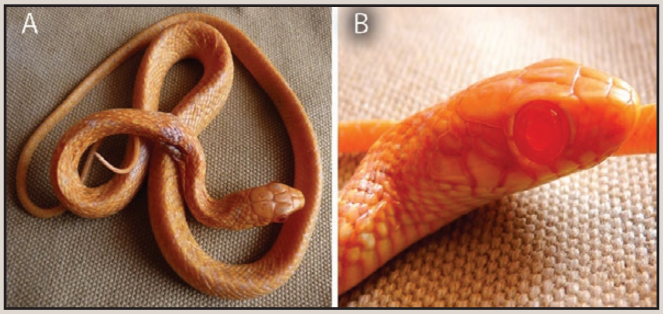

Fig. 1. Albino *Ptyas mucosa* showing lack of melanin pigmentation in the body (A) and iris and cornea of the eye (B).

Even though there are several records of albinism in snakes from India, there is so far no mention of an albino *P. mucosa* in available scientific publications (Mahabal and Thakur 2014. Russ. J. Herpetol. 21:80–88; Vyas and Thakur 2015. Sauria 37:59–61; Kumbaret al. 2017. Reptile Rap 32:29–31).

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***PYTHON BIVITTATUS* (Burmese Python). DISPERSAL / MARINE INCURSION.** *Python bivittatus* is an established invader in southern Florida, USA (Snow et al. 2007. *In* Henderson and Powell [eds.], Biology of the Boas and Pythons, pp. 416–438. Eagle Mountain Publishing, Eagle Mountain, Utah). Appreciation of the dispersal abilities and ecological tolerances of *P. bivittatus* will help conservationists to better anticipate future expansions. Although they inhabit terrestrial and freshwater ecosystems in South Asia, Southeast Asia, and the East Indies, *P. bivittatus* likely have some capacity for marine dispersal. The recently proposed subspecies *P. bivittatus progschai*, an insular endemic of Sulawesi (Jacobs et al. 2009. Sauria 31:5–16), seems to have colonized that island via transoceanic dispersal in the Pleistocene. The Kinmen Islands, lying at minimum 2.1 km off the coast of China, have been naturally recolonized by *P. bivittatus* after the species was

Fig. 1. Photograph of *Python bivittatus* found 25.1 km offshore in Florida on 17 October 2017.

Fig. 2. Photograph of *Python bivittatus* found swimming through open water 10.3 km off the shore of southwest Florida on 8 November 2017.

apparently extirpated there by Cold War military campaigns (You et al. 2013. Zool. Stud. 52:8). A python actively swimming between the Kinmen Islands has also been reported (Chung et al. 2016. Herpetol. Rev. 47:153). Here, we report on two lengthy marine incursions by *P. bivittatus* in Florida. These instances are the furthermost offshore marine observations for this species to date.

At 1000 h EST on 17 October 2017, a boat captain in the Gulf of Mexico observed a *P. bivittatus* tightly coiled around the buoy of a crab trap. The trap location (25.4250°N, 81.4417°W; WGS 84) is 25.1 km from the closest landfall at Highlands Beach, off the west coast of Everglades National Park. The snake, which was killed by the captain and used for crab bait, was estimated to be approximately 2.5 m in total length (Fig. 1). On 8 November 2017, a *P. bivittatus* was collected by another crab boat captain in the Gulf of Mexico, 10.3 km off the west coast of Everglades National Park (25.2928°N, 81.2647°W; WGS 84). The captain observed this second python swimming in open water (Fig. 2), and noted that the snake appeared to be near exhaustion when captured. The latter snake was provided for our examination, and found to be an adult male (SVL = 193 cm; 5 kg).

Three previous records of *P. bivittatus* in offshore locations of coastal Florida have been deposited in an online invasive species location database (https://www.eddmaps.org/distribution/viewmap.cfm?sub=20461; 15 Dec 2017). The database records are all within 1.3 km of the closest land, and within 5.8 km of the mainland; these snakes ranged in size from 0.46–2.74 m total length. Therefore, the present observations greatly increase the oceanic distance that pythons have been documented to traverse. Further, Hart et al. (2012. J. Exp. Mar. Biol. Ecol. 413:56–59) reported that hatchling *P. bivittatus* could survive up to one month when supplied only with seawater for drinking. *Malayopython reticulatus* (Reticulated Python) are found on numerous Indonesian islands, and have been known to cross large stretches of seawater (Murray-Dickson et al. 2017. PLoS ONE 12:e0182049).

One possible explanation for how snakes reached these locations is by being transported as stowaways on boats. However, to the extent that the present observations represent unaided dispersal, a meteorological event might be the underlying cause. One of the most destructive Atlantic hurricanes on record, Hurricane Irma, crossed the Florida Keys with a westerly track, turned northward, then made landfall near Marco Island on 10 September 2017 (37 and 59 d prior to the present observations), bringing sustained winds of 185 kph. Most coastal areas of southern Florida experienced major rain and storm surge flooding. After the storm, river outflows to the sea were increased dramatically and took several weeks to return to normal levels. It is possible that this increased outflow and the subsequently stronger coastal currents may have driven pythons further seaward than they may have swum otherwise. The capacity for *P. bivittatus* to disperse across saltwater should be noted by land managers of Florida's offshore islands, including in the Florida Keys.

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***RHABDOPHIS NIGROCINCTUS* (Black-banded Keelback). DIET.** *Rhabdophis nigrocinctus* is a semi-aquatic, diurnal snake that is often detected near streams (Stuart et al. 2012. The IUCN Red List of Threatened Species 2012: e. T192024A2029525). The main diet of *Rhabdophis nigrocinctus* is thought to be anurans. Here we report the predation of an adult *Fejervarya limnocharis* (Asian Grass Frog) by an adult *R. nigrocinctus.*

At 1300 h on 11 March 2017, in dry season, on pebbles near a forest stream in Pha Xong region (17.599°N, 105.832°E, WGS 84; 280 m elev.) within Hin Nam No National Protected Area, Khammouane Province, central Laos, we witnessed an adult *R. nigrocinctus* eating an adult *F. limnocharis.* When found, the *R. nigrocinctus* was capturing the *F. limnocharis* by grasping its lower body and two legs. Next, the snake continued moving across a fallen tree trunk and swallowed the frog vertically from the legs to the head in about 3 min.

We thank the German International Cooperation (GIZ) Hin Nam No National Project for giving the financial support.

PHOTO BY KHIEUSOMPHONE VANHNABUAOSY

Fig. 1. Adult *Rhabdophis nigrocinctus* consuming an adult *Fejervarya limnocharis* in Hin Nam No National Protected Area, Laos.

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***RHADINAEA FLAVILATA* (Pinewoods Snake). DIET and DEFENSIVE BEHAVIOR.** *Rhadinaea flavilata* is a small dipsadine snake found in the Coastal Plain of the southeastern United States (Ernst and Ernst 2002. Snakes of the United States and Canada. Smithsonian Institution Press, Washington D.C. 661 pp.). Prey of *R. flavilata* include amphibians, small mammals, earthworms, other snakes, and lizards, including skinks, anguids, and *Anolis carolinensis* (Ernst and Ernst, *op. cit.*). Here we report predation on *Anolis sagrei* (Brown Anole) by *R. flavilata.*

At ca. 1600 h on 2 December 2017, two of us (LS, SW) found an adult *R. flavilata* (SVL ca. 28 cm; HM 208107) that had eaten an adult male *A. sagrei* (SVL = 14 cm; HM 208108) under a coverboard at a private residence in Alachua County, Florida (29.65892°N, 82.37920°W; WGS84). The *R. flavilata* had a large food bolus and an odd "kinked" appearance to the lower neck (the tail of the *A. sagrei* was kinked side to side and was visible as separate from the food bolus; see also Jackson et al. 2004. Zoology 107:191–200). Immediately after capture, the *R. flavilata* held its body in a fixed posture and extended its tongue from its mouth for ca. 5 seconds. This is the only time any of us have seen behavior resembling thanatosis in *R. flavilata*, and could have been influenced by the limited mobility conferred by the large food bolus (Ford and Shuttlesworth 1986. Copeia 1986:999–1001; Mehta 2006. Ethology 112:649–656). Defensive behavior in this species is poorly known; Brode and Allison (1958. Herpetologica 14:37–40) mentioned the "offensive" odor of the anal gland secretions, and the species has a tail that is easily broken (Myers 1967. Bull. Florida Mus. Nat. Hist. 11:47–97).

This observation represents further documentation that *R. flavilata* can prey on non-native species (Durso and Smith 2017. Herpetol. Rev. 48:606), and native snakes are also known to prey on *A. sagrei* in Taiwan (Norval et al. 2007. Herpetol. Bull. 101:13–17), despite the observation that some species of snakes respond more strongly to the smell of sympatric than allopatric *Anolis* (Cooper et al. 2000. Amphibia-Reptilia 21:103–115). Other predators of *A. sagrei* in its non-native range include birds (Franz 2001. Herpetol. Rev. 32:253; Bartareau and Leblanc 2006. Herpetol. Rev. 37:462; White and Cove 2016. Herpetol. Rev. 47:460), cats (Bateman and Fleming 2011. Biol. J. Linn. Soc. 103:648–656), crabs (Cates et al. 2014. Herpetol. Rev. 45:491–492), and other *A. sagrei* (Cates et al. 2014. Herpetol. Rev. 45:491).

Finally, Myers (*op. cit.*; N = 123) found that just 0.8% of *R. flavilata* specimens had been collected in December. Comparable modern data from HerpMapper (2.7% of records from December, N = 184; accessed 16 March 2018) also suggest limited but not non-existent winter activity. Thus, our observation of an individual feeding in December, especially on such a large prey item, is noteworthy.

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***SALVADORA GRAHAMIAE* (Eastern Patch-nosed Snake). DIET.** *Salvadora grahamiae* is a small diurnal colubrid of the southwestern U.S. and adjacent Mexico. Previous knowledge of its diet suggests it is a lizard specialist, however, few specific prey items have been reported (Ernst and Ernst 2003. Snakes of the United States and Canada. Smithsonian Institution Press, Washington, D.C., 668 pp.). We dissected specimens of *S. grahamiae* and noted two novel prey items for this species. SRSU 3226, a 33.3 cm SVL specimen collected on 30 May 1973, 27.4 km S of Alpine in Brewster County, Texas, USA (approximate coordinates: 30.17523°N 103.58511°W; WGS 84), contained an *Aspidoscelis exanguis* (Chihuahuan Spotted Whiptail) (SVL = 10.4 cm). SRSU 4732, a 48.5 cm SVL specimen collected 2 November 1977 in Black Gap Wildlife Management Area, Brewster County, Texas, USA, 16.1 km N La Linda (approximate coordinates: 29.55507°N 102.81907°W; WGS 84), contained an *A. marmorata* (Marbled Whiptail) (SVL = 23.5 cm). Both of these lizards constitute new prey records for *S. grahamiae* (Ernst and Ernst, *op. cit.*).

Specimens examined for this study were from the James F. Scudday Vertebrate Collections at Sul Ross State University. All stomach contents were retained and stored in 70% EtOH. This research was supported by the Ronald E. McNair Post-Baccalaurete Achievement Program at Sul Ross State University.

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***SALVADORA INTERMEDIA* (Oaxacan Patch-nosed Snake). DIET.** Snakes in the genus *Salvadora* are diurnal active foragers that feed on small rodents (*S. bairdi*: Setser et al. 2009. Herpetol. Rev. 40:442; Carbajal-Márquez et al. 2014. Herpetol. Rev. 45:344), birds (*S. grahamiae*: Lemos-Espinal and Dixon 2013. Amphibians and Reptiles of San Luis Potosí. Eagle Mountain Publishing, Eagle Mountain, Utah. 300 pp.), and diverse lizard genera (*S. deserticola*: Barker and Sawyer 2011. Herpetol. Rev. 42:304; Gatica-Colima and Córdova-Reza 2012. Herpetol. Rev. 43:350–351; *S. grahamiae*: Ramírez-Bautista et al. 2000. Herpetol. Rev. 31:180). For *S. intermedia*, only one record of the diet is known (*Plestiodon brevirostris*: Santos-Bibiano et al. 2016. Mesoam. Herpetol. 3:159–160).

PHOTOS BY VÍCTOR H. JIMÉNEZ ARCOS

Fig. 1. *Salvadora intermedia* with a *Sceloporus jalapae* captured at the time of observation (A) and finishing ingesting the prey (B). Image of the full body to confirm species identification (C).

On 28 January 2018, at 1318 h, we observed a *S. intermedia* (SVL = 382 mm) in desert-scrub habitat in Tehuacan Valley, Municipio de Atexcal, Puebla, Mexico (18.38647°N, 97.68616°W, WGS 84; elev. 1920 m). At the time of the encounter, the snake had captured a male *Sceloporus jalapae* (Jalapa Spiny Lizard; Fig. 1A and B; SVL ca. 50 mm). The snake was subsequently collected and photographed to confirm its identity (Fig. 1C) before being released. This record represents a new prey item for *S. intermedia*, and the first record of predation on *S. jalapae*.

This work was supported by the Mohamed Bin Zayed Species Conservation Fund, grant number 172516436 to VHJA. The snake was photographed under permit FAUT-0322 issued by the Secretaría de Medio Ambiente y Recursos Naturales. We thank the communal property authorities of San Nicolas Tepoxtitlan for allowing us to make observations within their territory.

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***TAENIOPHALLUS POECILOPOGON* (Cope's Forest Snake). REPRODUCTION.** *Taeniophallus poecilopogon* is a small dipsadid snake, distributed in the Pampa biome, from Uruguay and Argentina, to southern Brazil (Etchepare and Zaracho 2009. Check List 5:770–772). It presents terrestrial and diurnal habits, preying on lizards (Carreira et al. 2005. Reptiles de Uruguay. DI.R.A.C., Facultad de Ciencias, Universidad de la República, Montevideo, Montevideo. 640 pp.). Natural history data for this species are scarce; there are no records regarding reproduction. Here we present the first data on morphometry and clutch size of *T. poecilopogon*.

Fig. 1. *Taeniophallus poecilopogon* (CHFURG 2308) with four developing eggs.

While examining specimens at the Coleção Herpetológica Universidade Federal do Rio Grande (CHFURG), we encountered an adult female (Fig. 1; CHFURG 2308; SVL= 23.5 cm, tail length = 8.83 cm, 6.52 g) specimen of *T. poecilopogon*, collected on 19 October 2012, at Estação Ecológica do Taim (32.7425°S, 52.5744°W; WGS 84). During necropsy, we encountered four elliptical ova that were early in development. Ova measured 20.9, 22.8, 24.9, 25.1 mm in length, 5.27, 5.28, 5.88, 5.61 mm in width, and 0.53, 0.54, 0.56, 0.65 g in weight.

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***THAMNODYNASTES* AFF. *NATTERERI* (Jararaca-de-tapete). DIET.** *Thamnodynastes* is a group of dipsadid snakes associated with forested areas and occasionally edge habitats. They are nocturnal and crepuscular, although they can also be active during daytime (Hartmann et al. 2009. Biota Neotrop. 9:173–184). The genus *Thamnodynastes* has a confusing taxonomic history (Bailey et al. 2005. Phyllomedusa 4:83–102; Bailey and Thomas 2007. Mem. Fund. La Salle Cien. Nat. 166:7–27; Bellini et al. 2013. Herpetologica 69:67–79) and literature on the distribution, ecology, and natural history of species in this genus is likewise confused (Barbosa et al. 2006. Rev. Biol. Cienc. Terra 6:73–82; Franco and Ferreira 2002. Phyllomedusa 1:57–74). Trevine (2017. Sistemática da tribo Tachymenini Bailey, 1967 [Serpentes, Dipsadidae, Xenodontinae]. PhD dissertation. Universidade de São Paulo) found that *Thamnodynastes* is paraphyletic and proposed taxonomic changes that should clarify the status of the species in this genus in the near future. Here we use the name *T. nattereri sensu* Costa and Bérnils (2015. Herpetol. Brasil. 4:75–93).

Thamnodynastes aff. *nattereri* is frequently found near aquatic environments, and feeds mostly on anurans (Marques and Sazima 2004. *In* Marques and Duleba [eds.], Estação Ecológica Juréia-Itatins: Ambiente Físico, Flora e Fauna, pp. 257–277. Holos, São Paulo, Ribeirão Preto). Both arboreal and terrestrial frogs are eaten (Marques et al. 2001. Serpentes da Mata Atlântica: Guia para a Serra do Mar. Holos, São Paulo, Ribeirão Preto, 184 pp.; Dorigo et al. 2014. Herpetol. Notes 7:261–264).

At 1700 h on 18 October 2016, an individual *T. nattereri* (total length = 47.0 cm; Fig. 1A) was collected at the Parque Estadual do Forno Grande, Municipality of Castelo, State of Espírito Santo, southeastern Brazil (20.51644°S, 41.091846°W; WGS84; 1345 m elev.). In the laboratory, the snake was dissected and an adult *Scinax* aff. *hayii* (SVL = 2.8 cm; Fig. 1B) was found in its stomach that had been swallowed legs first. *Scinax* aff. *hayii* is an arboreal

Fig. 1. *Thamnodynastes* aff. *nattereri* (A) and anuran prey *Scinax* aff. *hayii* (B) found in its stomach. Scale bar = 1 cm.

anuran endemic to the Brazilian Atlantic Forest, where it inhabits forested swamps and fragment edges (Haddad et al. 2013. Guia dos Anfíbios da Mata Atlântica: Diversidade e Biologia. Anolis Books, São Paulo, São Paulo. 544 pp.). *Thamnodynastes* aff. *nattereri* has been reported to prey on other *Scinax* species, such as *S. cardosoi* and *S. alter*, as well as on other frogs in the subfamily Scinaxinae, such as *Ololygon trapicheiroi* (Dorigo et al. *op. cit.*). Our finding supports the idea that semi-arboreal *T. nattereri* often prey on arboreal anurans, such as scinaxine tree frogs.

The specimen of *T.* aff. *nattereri* (MBML 3960) was deposited, with prey, at the Zoological Collection of Museu de Biologia Prof. Mello Leitão, Instituto Nacional da Mata Atlântica, Municipality of Santa Teresa, State of Espírito Santo, southeastern Brazil.

Instituto Chico Mendes de Conservação da Biodiversidade (n° 50.402-1) and Instituto Estadual de Meio Ambiente e Recursos Hídricos (nº 005-2016) issued permits for this study. ATM and TSS thank Conselho Nacional de Desenvolvimento Científico e Tecnólogico (CNPq).

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THAMNOPHIS PROXIMUS ORARIUS **(Gulf Coast Ribbonsnake). MAXIMUM SIZE.** *Thamnophis proximus orarius* has a geographic distribution that includes coastal regions of Louisiana and Texas (USA) southward to Tamaulipas, Mexico. Werler and Dixon (2000. Texas Snakes: Identification, Distribution, and Natural History. University of Texas Press, Austin. 437 pp.) report a maximum total body length of 123.2 cm for *T. p. orarius*. Ernst and Ernst (2003. Snakes of the United States and Canada. Smithsonian Books, Washington, D.C. 668 pp.) report a maximum total body length of 126.8 cm for *T. proximus*.

On 27 July 2016, at 0250 h, on FM 490 near Hargill, Hidalgo County, Texas, USA (26.44829°N, 98.05131°W; WGS 84) a road-killed adult female *T. p. orarius* was found that measured 115 cm (SVL), with a tail length of 33 cm (total length = 148 cm). The specimen was collected and deposited in the Amphibian and Reptile Diversity Research Center at the University of Texas at Arlington (UTA R63535). This represents the largest documented specimen of *T. proximus*.

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THAMNOPHIS RADIX **(Plains Gartersnake). DIET / OPHIOPHAGY.** On 27 June 2017, a gravid adult female *Thamnophis radix* (SVL = 41.3 cm; tail length = 9.6 cm; 50.5 g) was captured under a rubber artificial cover object in a remnant mesic sand prairie in Green River State Wildlife Management Area, Lee County, Illinois, USA. This was the initial and only capture of this individual during a mark-recapture study. Upon handling and marking with a unique identification code, the female regurgitated a partially digested segment (5.5 cm) of an adult *Storeria dekayi* (Fig. 1). To our knowledge, ophiophagy has not been reported in *T. radix*, although it has been documented in the genus *Thamnophis* (Mitchell 1986. Cannibalism in Reptiles: A Worldwide Review. SSAR Herpetol. Circ. 15. 37 pp.; Ernst and Ernst 2003. Snakes of the United States and Canada. Smithsonian Books, Washington, D.C. xi + 668 pp.). Environmental stress may be to blame (Polis and Myers 1985. J. Herpetol. 19:99–107). Conceivably harboring lower abundances of more typical prey items (e.g., annelids and amphibians), arid sand prairies may present a unique set of conditions that elicit trophic niche expansion in *T. radix*.

Fig. 1. Remains of a *Storeria dekayi* recovered from a *Thamnophis radix* from Illinois, USA.

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TROPIDOCLONION LINEATUM **(Lined Snake). HABITAT / ELEVATION.** *Tropidoclonion lineatum* is primarily a species of the Great Plains states of the USA where it occurs widely in grassland and sparsely wooded habitats below ca. 1500 m. However, at the western limits of its geographic range, it occurs at higher elevations along the eastern versant of the southern Rocky Mountains and other disjunct mountain ranges farther south. In Colorado, Hammerson (1999. Amphibians and Reptiles in Colorado. 2nd ed. University Press of Colorado, Niwot. 484 pp.) reported *T. lineatum* from the eastern part of the state below 1830 m. In New Mexico, Williamson and Degenhardt (1984. Herpetol. Rev. 15:21) reported a record from "above 2000 m," whereas Degenhardt et al. (1996. Amphibians and Reptiles of New Mexico. University of New Mexico Press, Albuquerque. 431 pp.) identified the known elevation range in the state as 1200–2000 m. Herein we report a high-elevation record for *T. lineatum* in central New Mexico, near the extreme western edge of its known geographic range.

On 12 October 2009, an adult *T. lineatum* (unsexed) was captured by S. Cox at midday as it was crawling on the surface in a grassy area just below the summit of Capilla Peak at ca. 2810 m elevation in the Manzano Mountains, Torrance County, New Mexico (34.69933°N, 106.40445°E; WGS84). The specimen was photographed by C. Hathcock immediately after it was captured and then released. The location was a grazed montane meadow (ca. 4.3 ha) straddling the ridge just south of Capilla Peak. Dominant grasses and forbs of the meadow were: Blue Grama (*Bouteloua gracilis*), Spidergrass (*Aristida ternipes*), Penstemons (*Penstemon* spp.), and Fleabane (*Erigeron* sp.). Surrounding the meadow were stands of trees and shrubs, primarily Ponderosa Pine (*Pinus ponderosa*), Gambel Oak (*Quercus gambelii*), Quaking Aspen (*Populus tremuloides*), Southwestern White Pine (*Pinus strobiformis*), and Douglas-fir (*Pseudotsuga menziesii*).

The species has been previously documented from sites on the east side of the Manzano Mountains (Williamson and Degenhardt 1984, *op. cit.*; Degenhardt et al. 1996, *op. cit.*), although all such records are from lower elevations. The nearest other record to the Capilla Peak site that we are aware of is from ca. 24 km to the NE at "NM 14 [= NM Hwy 337], 3.5 mi [5.6 km] S of Chilili," Torrance County (Museum of Southwestern Biology, University of New Mexico, MSB 39935) at an elevation of ca. 2035 m. Our record indicates that *T. lineatum* occurs at higher elevations than previously reported and suitable habitat for the species in New Mexico apparently extends into montane areas from more typical habitat at lower elevations.

A voucher photograph of the *T. lineatum* was verified by Charles W. Painter and is deposited in the University of Kansas Digital Archives (KUDA 012535). We thank J. Tomasz Giermakowski at the Museum of Southwestern Biology, University of New Mexico for support with museum queries.

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VIPERA BERUS (Common Adder). EARLY SPRING ACTIVITY. *Vipera berus* has the most northerly distribution of any snake in the world, with a comparatively prolonged activity period. In Somero, southern Finland (60.6299°N), the earliest spring record was on 27 March (Viitanen 1967. Ann. Zool. Fen. 4:472–546). In northwestern Russia, at a latitude of 60°N, in the vicinity of St. Petersburg, *V. berus* usually start to emerge from hibernation in the middle of April. In the Vologda Region, in Darwin Natural Reserve (between 58.5647°N and 58.8834°N) the date of earliest activity is 30 March (Kaletskaya 1953. Rybinskoe Water Reservoir, vol. 1, MOIP, Moscow. 214 pp.). In southern Karelia, on the Kizhi Archipelago, the earliest emergence recorded is 24 April (Korosov 2010. Ecology of Common Adder in the North. PetrGU, Petrozavodsk, Russia. 262 pp.).

FIG. 1. Male *Vipera berus* basking in the sun, 12 March 2017.

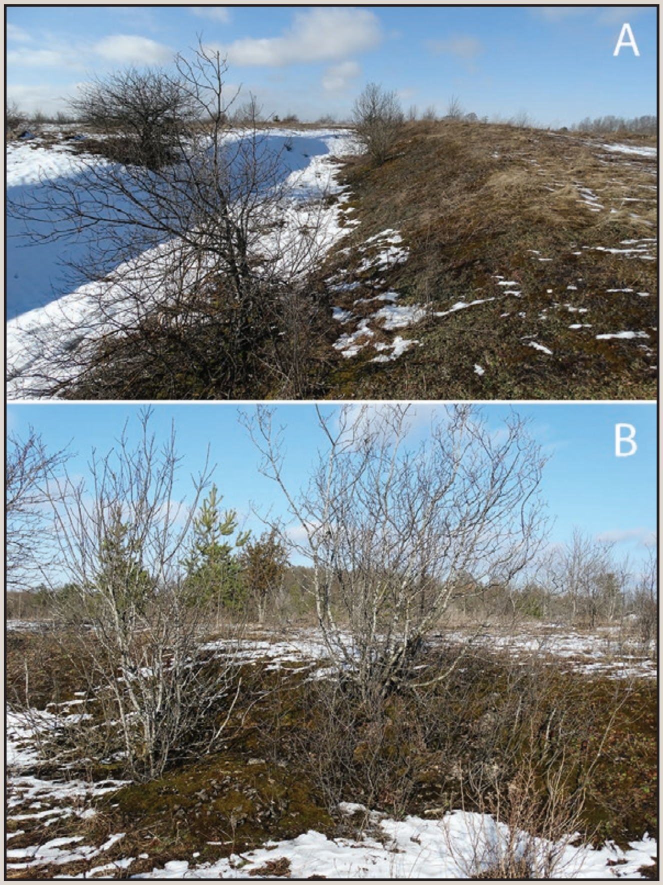

FIG. 2. A-B) Hibernation and basking sites of *Vipera berus* near Petrovschina Village, Russia, 12 March 2017.

On 12 March 2017, at 1425 h, active *V. berus* were recorded in abandoned quarries near Petrovschina Village, Kirovsk District, St. Petersburg Region, Russia. Two adult males (Fig. 1) were basking 2 m from each other on the thawed southern slope of a small ravine (59.8744°N, 31.5025°E, WGS84; Fig. 2A). The air temperature was 4.0°C, and the surface temperature of the soil in the sun was 7.5°C. One additional male was found 300 m from others, also on a south-facing slope under a small bush (59.8761°N, 31.5066°E, WGS84; Fig. 2B), where it was actively moving. The air temperature was 4.5°C, and the surface temperature of the soil in the sun was 9.0°C.

These abandoned limestone quarries were dug in the 19th century into the Putilovo Ridge, a geologic formation stretching along the northern edge of the Baltic-Ladoga coastal escarpment characterized by local outcrops of Ordovician solids. The Putilovo Ridge is a well-drained plateau formed by limestone dumps and covered by meadows and scrubby bushes with junipers, apple trees, pines, and others. The dissected landscape, dry soil, and the abundance of cavities in gravelly ground provide ideal habitat for overwintering *V. berus*. This species is present here during the whole year in high densities, and numerous wintering dens are present. Dryness and high soil temperatures are conducive to early snowmelt on the southern slopes in the spring and very early emergences of snakes. This is the earliest date reported for this latitude and location, earlier than the previous record by 12 days (on 24 March 2016, unpubl. data) and earlier than the previous published record from southern Finland by 15 days (on 27 March 1967).

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***XENOPELTIS UNICOLOR* (Asian Sunbeam Snake). DIET.** *Xenopeltis unicolor* is known to feed on a variety of prey, including frogs, lizards, snakes, small mammals, birds, and even reptile eggs (Rooij 1917. The Reptiles of the Indo-Australian Archipelago, vol. II. Ophidia. E. J. Brill, Leiden, Holland. 334 pp.; Bergman 1955. Zool. Mededelingen, XXXIII, No. 22:209–225; Martins and Rosa 2012. Taprobanica 4:48–51; Milto 2014. Herpetol. Rev. 45:522). In this note we report the predation of an adult *Hypsiscopus plumbea* (Boie's Mud Snake) by an adult *X. unicolor.*

At 2130 h on 19 March 2017, on a trail near a stream of Vang Khon Village, Bualapha District, Khammouane Province, Lao PDR (17.5046°N, 105.7247°E, WGS 84; 166 m elev.) within Hin Nam No National Protected Area, we observed an adult *H. plumbea* being eaten by an adult female *X. unicolor* (SVL = 66.3 cm; tail length = 7.2 cm; Fig. 1). When found, the *X. unicolor* was coiled around the *H. plumbea*. Remarkably, even though we collected both specimens, the *X. unicolor* continued to constrict its prey and had no response to our presence. Upon arrival at the lodge, the *X. unicolor* had completely swallowed the *H. plumbea*. Specimens were deposited in the collections of the Vietnam National University of Forestry (VNUF), Hanoi, Vietnam (VNUF RL.2017.08).

Fig. 1. An adult female *Xenopeltis unicolor* consuming an adult *Hypsiscopus plumbea* near a stream of Vang Khon Village, Boualapha District, Khammouane Province, Laos.

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Erratum

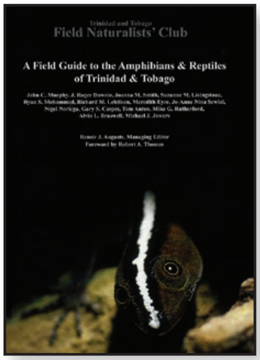
In a recent issue of *Herpetological Review*, a natural history note by Jarvie et al. on *Sphenodon punctatus* (Tuatara)(2017. Herpetol. Rev. 48:840–841), contained typographical errors introduced during print production to the printed and online versions. The typographical errors include the deletion of letters that have macrons, which are used in the Māori language to indicate long vowels: the correct spelling of rokonui is Ōrokonui, Ngti Koata is *Ngāti Koata*, and Kti Huirapa Rnaka ki Puketeraki is *Kāti Huirapa Rūnakaki Puketeraki*. In addition, the plural for *tuatara* was changed to tuataras in one place (tuatara, a Māori name, is the same in both singular and plural).

BOOK REVIEWS

Herpetological Review, 2018, 49(3), 561–563.

A Field Guide to the Amphibians & Reptiles of Trinidad & Tobago

John C. Murphy, J. Roger Downie, Joanna M. Smith, Suzanne M. Livingstone, Ryan S. Mohammed, Richard M. Lehtinen, Meredith Eyre, Jo-Anne Nina Dewlal, Nigel Noriega, Gary S. Casper, Tom Anton, Mike G. Rutherford, Alvin L. Braswell, and Michael J. Jowers. 2018. Trinidad & Tobago Field Naturalists' Club, P.O. Box 642, Port of Spain, Republic of Trinidad and Tobago (www.ttfnc.org). vii + 336 pp. Softcover. US $43.00 (approx.), an international order form is available at http://ttfnc.org/product/field-guide-amphibians-reptiles-trinidad-tobago/. ISBN-978-976-8255-47-1.



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I really like field guides. Like many herpetologists, my first introduction to the relevant literature was a field guide. Since then, however, as I've exceeded the age when it's permissible (maybe even expected) to be a curmudgeon, I've become much more critical. I'm no longer satisfied with brief descriptions, maps, and mere pretty pictures (although, like even the most hardened "professionals," I enjoy pictures that tell stories or provide new or interesting information). I also demand insights into the natural history of a species and I want more than a passing mention of habitats and conservation status. This most recent volume on the herpetofauna of Trinidad & Tobago, the product of an impressive collaboration led by John Murphy, lived up to my expectations.

A number of works have chronicled the extensive diversity of amphibians and reptiles on Trinidad & Tobago. Relatively recent comprehensive efforts include Kenny (1969, 1977), who addressed amphibians, and Boos (2001), who recorded the snakes. The entire herpetofauna has been covered in a previous book by Murphy (1997) as well as in an ongoing blog (https://herpetologytt.blogspot.com/), also by Murphy, and an online checklist maintained by the Department of Life Sciences, University of the West Indies at St. Augustine (https://sta.uwi.edu/fst/lifesciences/amphibians-and-reptiles#Amphibians%20and%20Reptiles). The most recent of these are updated in the current volume.

Although often considered an extension or even part of the Lesser Antilles (the chain of islands ranging from Sombrero in the north to Grenada in the south), the biota of Trinidad & Tobago is much more closely related to that of the South American continent than to those on the oceanic islands to the north. In fact, during the past 10 million years, the islands of Trinidad & Tobago "have been isolated, connected, and re-isolated from the mainland many times" (p. 6).

Because of small size, limited habitat diversity, and isolation from the nearest mainland and often from other islands, insular herpetofaunas often are considered depauperate when compared to those on continents. However, despite relatively small size—Trinidad (4828 km^2) is larger than any of the Lesser Antilles, whereas Tobago (300 km^2) is slightly smaller than Grenada—a diversity of habitats, at least in part attributable to the varied past relationships with South America, support a rather spectacular diversity of amphibians and reptiles. In addition to South American species that presumably established populations during periods when the islands were contiguous with the mainland, this island nation sports a surprising number of endemic species. Furthermore, like tropical islands throughout the world, the herpetofauna of Trinidad & Tobago has and continues to be augmented by human-mediated introductions from near and far.

This book provides accounts of 35 species of anurans in 12 families, four of which (*Eleutherodactylus johnstonei*; *Dendropsophus minusculus*; *Adenomera* sp., previously known as *A. hylaedactyla*; *Leptodactylus latrans*) are apparently human-mediated introductions. Ten species of native turtles (five of them sea turtles) in six families are described. In addition, records of adult *Trachemys scripta elegans* are indicative of multiple introductions, but no evidence of sustainable populations exists. Also, whether *Chelonoidis carbonarius* (listed as *C. carbonaria* in the book) occurs naturally on Trinidad is uncertain. Two species of caimans, including the recently discovered *Paleosuchus palpebrosus*, are the only known native crocodilians. Lizards are represented by 32 species in 11 major lineages, including two species of legless amphisbaenians. Introduced species are *Hemidactylus mabouia*, although trans-Atlantic rafting cannot be ruled out, *Gymnophthalmus underwoodi*, a unisexual species with an ever-expanding range associated with human-mediated dispersal that might have arrived naturally on Trinidad, and six species of anoles (*Anolis aeneus*, *A. extremus*, *A. richardii*, *A. sagrei*, *A. trinitatis*, and *A. wattsi*), with all but the increasingly ubiquitous *A. sagrei* emanating from the Lesser Antilles. Fifty-one species of snakes (four of them venomous) in 10 major lineages are known to occur on Trinidad & Tobago. Surprisingly, *Indotyphlops braminus*, which has an almost circumtropical distribution, including a number of Lesser Antillean islands, apparently is not yet present on the archipelago.

A section titled "waifs & questionable species" (interestingly placed ahead of the accounts of taxa known to occur on the islands) includes brief mention of 13 species (one turtle, six lizards, six snakes) "erroneously reported" from Trinidad &

Tobago and a table lists an additional 16 species (one caecilian, three anurans, three turtles, two crocodilians, three lizards, four snakes) with “some support for their presence in the islands”; these include waifs, species known from museum specimens but not encountered in decades (or longer), museum specimens with questionable data or which have been lost or destroyed, and species reported but not documented as present. Some additional species purportedly from the islands but almost certainly mislabeled are not included. Interestingly, one species (*Anolis* cf. *lemurinus*) is included in both lists but is considered an “improbable member of the fauna.”

The book begins with a list of the coauthors detailing the affiliations and contributions of each. This is followed by a foreword by Robert Thomas, who characterized the pursuit of reptiles and amphibians on Trinidad & Tobago as “never a dull moment and always a challenge.” A preface emphasizes curiosity and mentions stories that engage the inquisitive. An anaconda known as “Big Annie,” a bioluminescent (?) lizard, and the “paradox” of the Paradox Frog (*Pseudis paradoxa*) do serve to capture the imagination—but I wanted more details and even more stories; what was included was too good for a mere mention in a section of the book many readers will skip. Acknowledgments precede a section on “how to use this book,” most of which is devoted to the organization of the species accounts.

The subsequent introduction addresses biodiversity, noting that “no one is sure how many forms inhabit the planet, but humans have applied about 1.9 million scientific names to species in the past 260 years, and the number of named species increases daily.” This in turn was followed by a fascinating calculation stating that “on average in 2014, one new species of frog was described every 2.1 days, one new species of lizard was described every 3.4 days, and one new species of snake was described every 17.3 days.”

The introduction continues with an overview of amphibians and reptiles, which includes some general information on diversity and natural history, and an all-too-brief section labeled “the extinction crisis & hidden diversity.” Although it lists all of the frightening statistics and essentially ends with a comment that “it will take more than legislation to slow the extinction crisis. The situation demands changes in human behaviour.” “Challenges” are included later in the introduction and many species-specific risks and some necessary “changes in behaviour” are detailed in the species accounts, but a topic this important deserves considerably more attention.

The next section on “the environment” is outstanding. Although I might have wanted a somewhat more detailed accounting in some instances, the authors provide an excellent (albeit brief) overview of “physiographic features & geological history” before describing the coasts (broken down into the Trinidad coasts and the Tobago coasts), savanna, freshwater habitats, and forests before moving on to “a tropical urban herpetofauna,” which increasingly characterizes such a large portion of today’s tropical biotas and arguably deserved more attention than given here, and “environmental challenges.” The latter include the ongoing conflict between development and sustainability. In Trinidad & Tobago this is aggravated by the economic reliance on fossil-fuel extraction, increasing demands of ecotourism (providing some hope of preserving at least bits of nature while simultaneously threatening it with too much attention), litter (although progress is evident in increased recycling efforts), and non-sustainable hunting (especially of tegus, iguanas, and sea turtles). The last in particular benefit from legislation and the action of NGOs and local communities who patrol the beaches and provide some protection for nests and hatchlings. Next is another very brief but pointed discussion of the “climate issue,” followed in turn by sections on “conservation” (although much of the relevant information is in Hailey and Cazabon-Mannette [2011]; for the sake of emphasis, I would have liked to see all of the conservation-related content consolidated into a single section) and folklore (which might have been expanded in light of the statement that “amphibians and reptiles have an integral role in culture, folklore, and ecology of the island archipelago”).

Next is a section, critical for a field guide, on “measurements & identifying features” that includes a series of plates featuring excellent line drawings that illustrate the principle characteristics of frogs and toads, turtles, crocodilians, lizards, and snakes. This leads into the previously mentioned section on “waifs & questionable species,” which precedes the 255 pages devoted to the species accounts. These are followed by appendices on anuran reproductive modes (including line drawings of tadpoles), handling amphibians and reptiles (mostly a clear admonition to avoid contact and the reasons why), amphibian chytridiomycosis, snakebite, herpetological collections in Trinidad & Tobago, and a list of research stations and lodging for ecotourists. Unfortunately, the choice to move these sections to appendices will mean that many readers will never see them. I would have preferred to see the excellent section on anuran reproduction incorporated into the species accounts, that on the chytrid fungus into the conservation section (along with a discussion of other diseases affecting tropical herpetofaunas), and that on snakebite into the accounts of venomous snakes along with a reminder that most snakes are not capable of delivering a dangerous (although sometimes painful and bloody) bite that might have been prominently featured at the very beginning of the snake accounts. The book ends with a glossary, a list of references, and an index to common and scientific names.

The critical components of any field guide are the species accounts and the illustrations. Each species account begins with a common name, many of them based on local usage and consequently differ from names applied to those same species elsewhere, and the scientific name, some of which include trinomials, followed by the author of the name and the date of the original description. Because scientific names change, some are followed by previously used names and a few are very recent innovations (e.g., *Ameiva atrigularis*, which was until recently considered a subspecies of *A. ameiva*). These are not and were not intended to be complete synonymies, but explanations are provided for new combinations (e.g., *Adenomera* sp. instead of *A. hylaedactyla*, the name previously applied to what appear to be multiple species on Trinidad). Both snout–vent length (SVL) and total length (TL) are provided, when appropriate, plus sizes at hatching or birth, when sexual maturity is attained, and the maximum known. Brief but diagnostic descriptions are sometimes supplemented with more detailed information (e.g., scale counts or arrangements) when necessary to distinguish similar taxa. Similar species, those with which the species in question is most likely to be confused, also are listed and distinctive characters are noted.

Habitat is described briefly and lifestyles (e.g., arboreal, terrestrial, fossorial) often are included. This precedes a summary of what is known about the species’ life history;

these include activity, prey and predators, reproductive mode, breeding season, parental care, abundance, vocalization, and some aspects of behavior. Maps accompany the vast majority of accounts; localities are marked by colored dots indicating vouchered and unvouchered records since and before 1985 (the choice of 1985 is not explained). Accounts without maps address species for which locality data are not known, for species known from only a few sites the authors chose to protect, and for marine species.

Illustrations in species accounts and elsewhere are arranged in 180 plates, each containing one to several figures or photographs. I particularly liked the photo of a rainstorm in Caroni Swamp (plate 7) adjacent to an outline map of Trinidad & Tobago (plate 8) with graphics showing mean monthly rainfall at various locations throughout the islands. The authors have strived to illustrate at least some of the considerable variation in many species and plates dealing with taxa often include multiple life history stages, including eggs and larvae, although some photographs and detailed drawings of tadpoles are relegated to the introduction or Appendix I instead of being included in the relevant species account. In addition to the aforementioned line drawings (plates 17–21) included in the introductory section on measurements and identifying features, drawings and photographs showing diagnostic features are included in many accounts and, when appropriate, the introductory sections to families or sometimes genera (e.g., plate 96 with colored drawings illustrating the introduced species of anoles).

Other than my previously stated quibbles regarding organization, I have few complaints. Photographs range from excellent to adequate (those in the latter category frequently appear to be scanned slides, which often lack the sharp definition and contrast readers have come to expect from modern digital images). In plates comprised of multiple photographs, I would have preferred a distinct delineation between images (such as in plates 106 and 113, for example), rather than those in some plates that have quite similar color palates and tend to run together (e.g., plates 81 and 132a). As personal preferences, I would have enjoyed an overview of the history of herpetological research in the archipelago, the inclusion of a key, and a more detailed table of contents directing one to specific accounts (although that can be accomplished by consulting the index). I also would have liked more detailed natural history information, but I acknowledge that too much (as much as I wanted more) can detract from the utility of a field guide. I found only two errors, both rather technical (and possibly attributable to the fact that the collaborative efforts that resulted in this book began at least a decade prior to its publication). As mentioned previously, *Chelonoidis carbonarius* is listed as *C. carbonaria*, although Olson and David (2014) clearly demonstrated that the generic name is masculine. Also, the range of *Rhinella marina* is described as extending from the "southern USA through Central and South America," overlooking the partitioning of that taxon into at least two and more likely three species (with the name *R. horribilis* applied to Mexican and Central American populations; e.g., Maciel et al. 2010; Vallinoto et al. 2010).

In summary (and despite my quibbles), this is an outstanding, educational, and entertaining overview of a complex insular herpetofauna. Sufficiently detailed and documented to meet the needs of professionals but adequately straight-forward and not too technical to serve readers using it solely as a means to identify species they encounter during a visit to the islands. This excellent guide is a must for the libraries of herpetologists with an interest in insular herpetofaunas and for the backpacks of any naturalist fortunate enough to experience Trinidad & Tobago.

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American Snakes

Sean P. Graham. 2018. Johns Hopkins University Press, Baltimore, Maryland (https://www.press.jhu.edu/). xiii + 293 pp. Hardcover. US $29.95. ISBN 978-1-4214-2359-3. E-Book, US $29.95. ISBN 978-1-4214-2360-9.

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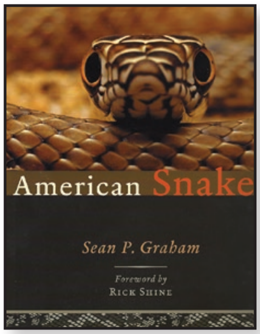

Books on the behavior and natural history of snakes have a long and distinguished history in herpetology. Texts such as "Snakes and Snake Hunting (Kauffeld 1957) and "Snakes: The Evolution of Mystery in Nature" (Greene 2000) have helped introduce generations of readers to the wonders of the natural history of snakes. To the author of the present book, however, the masterpiece by Rick Shine, "Australian Snakes," was the inspiration for his attempt to summarize the "everyday lives" of the snakes that call the United States home (apparently the "American" in the title does not refer to Central or South America).

We reviewed this book from two rather different perspectives. One of us (NAS) can be described as an "interested amateur" and not a professional herpetologist. As such, she is likely closest to the target audience for this book. RAS is an academic herpetologist and was interested in this book as a possible resource for undergraduates first becoming interested in working with or learning about snakes.

The text is divided into 11 chapters of 20–33 pages each. These include an Introduction that centers on taxonomic diversity, a chapter on snake functional morphology and physiology ("Form and Function"), two chapters on snake activity cycles ("A Day in a Life of a Snake" and "A Year in the Life of a Snake"), and then seven chapters on snake sex, food, predators ("eaters"), defense, dangerous snakes, snake invaders, and snake conservation. Each chapter contains numerous color photos, maps, and line art. The quality of the photos ranges from mediocre (e.g., the illustration of lateral undulations on p. 53, the thermal profile graph on p. 76) to superb (e.g., the Rough Green Snake photo on p. 178, the photo of the Diamondback Rattlesnake on p. 127, and the sequenced photos of a Bobcat attacking a Western Diamondback Rattlesnake on p. 161 were especially striking). Finally, most chapters have a 1–2 page profile of the background of snake biologists, including such notables as Henry Fitch, Harry Greene, and Rick Shine. The text also has a useful subject and taxonomic index.

Overall, both of us felt that the book achieved its goal of being an accessible, interesting, and entertaining book about snakes. The author has an engaging writing style and strikes a reasonable balance between being "chatty" and overly technical. We especially liked the story of Rick Shine's first encounter with the famous garter snake dens in Manitoba (pp. 100–102), which combined the excitement snake scientists feel when going to a new study site with some solid information about snake biology, especially snake sex. The author, does, however, occasionally use colloquiums that we found questionable, especially the division of animals into "bad" and "good" parents (p. 122) based on their degree of parental care.

From the perspective of the interested amateur, we thought the book worked fairly well. This would be a great book to buy for interested high school students, early undergraduates, and nature enthusiasts. The author's personal stories make for great entertainment and are quite memorable. The tongue-in-cheek advice on snake-proofing your yard (complete with cost estimates) was especially enjoyable. The insert regarding snake bites and how to handle them was very well written and should be mandatory reading for those who venture into the field, especially since so many myths about "dangerous snakes" and snake bite remedies continue to proliferate even though they have been disproven. Finally, the story about the founding of the Orianne Society and its impact on snake conservation was an upbeat way of ending the book and showing readers what one individual's efforts can accomplish.

NAS was less taken with the short personal biographies of snake biologists, finding their placement in the middle of chapters distracting. While they lend an interesting twist to the text, neither of us felt their loss would have detracted from the book.

Although we clearly enjoyed the book quite a bit, we would be remiss to not point out that there were some errors and inconsistences that detracted from its usefulness. Some of the more important examples are indicated below:

1. The discussion of the difficulties in getting venomous snakes placed on the US Endangered Species list is interesting reading but contains a serious error. On p. 251 the author states that the New Mexico Ridge-Nosed Rattlesnake is the only venomous snake protected by ESA. This is, however, incorrect, as Eastern Massasauga Rattlesnakes were listed as Threatened in 2016 (https://www.fws.gov/midwest/endangered/reptiles/eama/faqFinalListEMR.html). Perhaps the Eastern Massasauga had not been listed when the book was submitted for publication, but then this should have been corrected in page proofs.
2. In the Preface, the author notes that the scientific literature is "dense, snooty stuff…off limits to general readers" (pp. xi). One has to wonder then why there is a 25-page reference section that is filled with this "dense, snooty stuff." While this would be valuable information for undergraduates and beginning graduate students, it seems out of place in a text directed at the audience discussed in the Preface to the book.
3. The SSAR Common Names list (which the author indicates he is following for the text) mandates that the English common names of reptiles and amphibians should be capitalized, but that was not done in the text.

These issues aside, Sean Graham has produced a handsome, useful book that can be recommended to both interested amateurs and undergraduate students alike. What's more, the price is an absolute stunner—US $22.00 on Amazon.com as of this writing is nothing short of remarkable in an age when most books with extensive color plates go for over US $100. At this price, this is an easy book to recommend.

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The Ark and Beyond. The Evolution of Zoo and Aquarium Conservation

Ben A. Mintner, Jane Maienschein, and James P. Collins (editors). 2018. The University of Chicago Press, New York and London (http://press.uchicago.edu). x + 454 pp. Softcover US $35.00. ISBN: 978-0-226-53846-4; Hardcover US $105.00. ISBN: 978-0-226-53832-7.

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Although zoos have had some involvement with species recovery efforts since at least the late 18th and early 19th centuries, it was not until the latter half of the 1980s that zoos (and some aquariums) began viewing conservation as a major component of their diverse programming. Faced with the reality of regulatory barriers preventing collection and importation of many species identified as being at risk, and a new altruistic realization of the shocking decline in many wild populations of animals, the questions of zoo conservation rapidly became existential ones: Why are we here and what is our purpose? *The Ark and Beyond* explores the evolution of zoos and aquariums from postage-stamp menageries to modern institutions with multifaceted programming designed to foster educational outreach, scientific endeavor, recovery of species at risk, and (of course) recreation.

The editors of this volume, Ben A. Minteer, Jane Maienschein, and James P. Collins, all faculty members of Arizona State University with extensive backgrounds in wildlife conservation, attempt to explore the evolution of zoo conservation programs in six sections ranging from the historical antecedents of zoo collections, the science and challenge of recent zoo conservation efforts, and alternative models and futures for zoo conservation programs. The book is an outgrowth of two symposia hosted by Arizona State University's Marine Biological Laboratory and the Phoenix Zoo in 2014 and 2015 funded by the National Science Foundation and a Carnegie Investment Fund Grant. The contributions of more than two dozen authors who were participants in these events make up the 30 chapters in this volume. The contributors represented a wide range of institutions ranging from zoos and aquariums, museums, and universities. The wide diversity of the contributions is both a strength and weakness of this 454-page book.

The first section, entitled "Protoconservation in Early European Zoos," traces the history of zoos from the Roman arena through the improvements to animal care and welfare up to the *fin de siècle* of the 20th century. By the authors' own admissions, there was *no* zoo conservation during this period, and the inclusion of this section adds very little to our understanding of the evolution of zoo and aquarium conservation programs. Happily, in Part 2 entitled "The Rise of Zoo and Aquarium Conservation in the Nineteenth and Twentieth Centuries," the historical and cultural foundations of zoo conservation are explored. For those not familiar with the stories, the attempts at species recovery of the Passenger Pigeon by the Cincinnati Zoo and the American Bison by the New York Zoological Park make for some interesting reading. Not only are the zoo efforts described, but the historical events that led to their imperilment are related in scholarly detail. This section would have been even more engaging for the herpetologist if the renaissance in herpetological husbandry and conservation breeding in the 1970s would have been explored a bit. Fueled in no small part by the European journals *Salamandra* and *Lacerta,* there was an explosion of breeding—primarily in Texas zoos—of many exotic reptile species as curators (Jim Murphy, Rick Hudson, and the late Joe Laszlo come to mind) switched to naturalistic exhibits and appropriate thermal regimes for species in their care. Another apparent success story in Chapter 6, "Reintroducing the Przwalski's Horse," describes the salvage of the only remaining wild horse species from the Mongolian Steppes. Although the account relates many historical facts regarding the initial recovery efforts at the New York Zoological Park, there is scant mention of the considerable hurdles this species faces before real recovery is secure: explosive population growth of Mongolian herdsmen and the subsequent overgrazing by domesticated animals, increased desertification, and increased frequency of extreme weather events. More positive news also could have been included, such as the increased participation in the recovery effort by foreign zoos, such as the Prague Zoo, the Cologne Zoo, and the Gansu National Breeding Center near Wuwei, China. The last chapter in this section gives some interesting historical background on the evolution of aquarium conservation from cooperative hatchery projects to participation in stranding networks and marine mammal rehabilitation.

Part 3 has the greatest fidelity to the title of the volume. There is an overview of current cooperative Aquarium and Zoo Association (AZA) programs paired with some impressive examples of AZA programs that have coupled *in situ* efforts with field conservation efforts. How wonderful it would have been to have had some background history on the many zoo conservation programs—Taxon Advisory Groups (TAGs), Species Survival Plans (SSPs), Faunal Interest Groups and several others—that blossomed under the leadership of William G. Conway and the late Michael Hutchins during the 1980s and 1990s. A candid discussion of the problems faced by TAGs and SSPs in space limitations and lack of cooperation in animal transfers would have been welcome, as well as some of the more recent accomplishments of aquariums: freshwater mussel propagation at the Columbus Zoo, hellbender and lake sturgeon head-starting at the Toledo Zoo, and many others. Unfortunately, within Part 3 there is scant mention of any conservation programs dealing with amphibians and reptiles—except for a brief mention of the Kihansi Spray Toad and the Western Pond Turtle's inclusion within the SAFE (Saving Animals from Extinction) program. The most thought-provoking essay within this section is by Rick Barongi who, in a "come to Jesus"-style admonition, exhorts zoos to commit more resources to conservation programs that have a connection to wild nature. He makes an interesting observation—all too true—that nearly every successful zoo conservation program has had a champion that shepherded it to success.

The editors decided to include several submissions on "wellness" and "animal welfare." These are presented in Part 4—"Caring for Nature: Welfare, Wellness, and Natural Connections." As lofty as the principles behind these submissions are, they have little relevance to species recovery unless they in some way enhance the *fitness* of the populations we are seeking to recover. Too often animal transfers necessary to preserve genetic diversity in the captive population have been thwarted by well-meaning zoo staff or animal rights groups seeking to place the "welfare" of individual zoo animals above the genetic needs of a managed population. Instead, I would have liked to have seen a section of submissions about the outstanding contributions zoo veterinarians have made to true conservation initiatives, many of them involving endangered reptiles and amphibians.

Some of the most outstanding zoo conservation work has been in the fields of molecular and population genetics and assisted reproductive technologies such as gamete banking, cloning, and *in vitro* fertilization. Fortunately, some of these efforts are showcased in Part 5 that explores the "Science and Challenge of the Conservation Ark." There is a brief nod to herpetology as well. Chapter 24 by Joseph Mendelson examines the zoo response to the global amphibian crisis.

The last section of this book examines alternative trajectories for zoos in an age of unprecedented threats to wild animal populations directly traceable to human overpopulation and activity. How can zoos effectively contribute to conservation when they consistently struggle to genetically manage their own populations? Some suggestions are forthcoming in this section: expansion of management expertise and scientific endeavor, augmentation of exhibit and holding space to facilitate genetic and reproductive management of captive populations, embracing a regional focus, and enhancement of "wildness" in management activities. All of these deserve thoughtful consideration by zoo directors and curatorial staff. One contribution urges zoos to embrace "sustainable development"—a fantasy if there ever was one—in our conservation programs.

The price of this volume—US $35.00 paperback—is a value for those who wish to explore *some* of the historical aspects of zoo conservation. With the lower price, however, the reader gets a coarser grade of paper and grainy black and white photographs. After reading this book, however, one wonders—as the late Kevin Wright (a wonderful zoo amphibian and reptile veterinarian) did so many years ago, "If it don't have hair, do people care?" Numerous zoo amphibian and reptile conservation projects—many of them involving successful reintroductions—have been ignored. As examples I could point to the rescue of several endangered populations of West Indian rock iguanas (*Cyclura carinata, collei, lewisi,* and *pinguis*) by zoo members of the IUCN Iguana Specialist Group, the successful reintroductions of the Virgin Islands Boa and the Antiguan Racer, the rescue of the Puerto Rican Crested and Kihansi Spray toads, and many others. For this reason, zoo herpetologists may want to look elsewhere for inspiration before they invest the time and money to access the information presented in this book.

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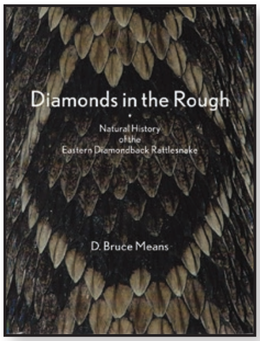
Diamonds in the Rough. Natural History of the Eastern Diamondback Rattlesnake

D. Bruce Means. 2017. Tall Timbers Press, Tallahassee, Florida (www.talltimbers.org). 390 pp. Hardcover. US $100.00.
ISBN: 978-0-9703886-5-0.



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I have always been a fan of larger monographs tackling the autecology of a species, such as the classics by Fitch (1960) and Legler (1960). I also have a great appreciation for synthesis volumes examining specific taxonomic groups such as the box turtles (Dodd 2001) and gartersnakes (Rossman et al. 1996). Such monographic treatises focusing on the fine-scale ecology of a single species are becoming rare and not often undertaken by single authors. Such volumes often represent years to decades of concerted work and in some cases, a lifetime of research. I had expected *Diamonds in the Rough* to follow the path of those classic autecological works using a lifetime of research on *Crotalus adamanteus.* Although the book does contain some excellent information, its presentation and organization are lacking. In addition, it carries a hefty price tag of US $100.00.

The book is arranged into 24 chapters covering a range of topics from the general study sites and methods all the way through human-snake interactions and anthropogenic impacts. I found it odd the author did not adhere to the standard common name, Eastern Diamond-backed Rattlesnake, which has been in use since 2008 (Crother 2008, 2012, 2017) throughout the book. Chapter transitions are faced with images depicting study sites, habitats, the focal species in relevant pictures, and humorous field anecdotal shots (Chapter 4's "Jimmy" conducting radio-telemetry from a motorcycle). The book has numerous illustrations of both color photographs and line drawings peppered among the chapters. In total, there are more than 140 figures that include full-color photographs, x-radiographs, original data graphs, and reprinted data graphs. The book also summarizes a great deal of original information in more than 50 tables and is solidly referenced with more than 500 citations.

There are problems with many of the photographs. Some of the full-color images appear washed out, blurry, or too low in resolution. For example, fig. 7.2 is blurry, and the colors are off, and both images in fig. 5.21 are completely washed out. I think this may be the result of using old film images converted to a digital format. In some cases, it may be difficult to have recent images of behavior on hand, and that one must resort to older images. Some images are also blown up too large for their resolution, as is the case for fig. 5.20. The number of blurred lower resolution images appears to outnumber those that are newer and crisp. Although I do not see this as a major detraction from the book, I feel a better effort could have been made to use more updated photography, get higher resolution scanned images, and work with the color balances and curves of the existing photos.

The book includes many figures presenting original data. As a quantitative person who has dealt with many graph-generating programs, I can tell many of the figures are stock from Microsoft Excel. I have never been a fan of Excel stock graphics without some necessary tweaks. For example, in fig. 10.2 the axis lines, tick marks, and tick labels are by default gray instead of black, making them difficult to read. In addition, all graphs have the default border around the entire graph and upper and right borders around the plot areas. To me, graphs look much cleaner and more professional when these elements are removed. Finally, most of the graphs were reduced too far, which made axes tick labels difficult to read. A perfect example is fig. 10.3. Either the graph sizes or the font sizes should have been increased. Finally, some graphs are even in different fonts (e.g., fig. 11.10 and fig. 11.11 on p. 127). These fonts are not only different from each other but different from the text. Some figures (e.g., 6.3) lack axis labels leaving the reader to wonder whether the y-axis is days between sheds and the x-axis are individual snakes? Overall, I think the author, copy editor, and publisher could have done a better job with the data figures.

Nearly every chapter brims with data tables ranging from the maximum sizes of individuals (table 5.1) to estimates of pre-settlement habitat area occupied (table 20.4). In general, the layout of the tables also reminds me of stock Microsoft Excel tables. Although it is nice to have the alternating rows colored, the extent distracts from the data presented within. Most of the tables are clear, and the reader can quickly grasp the concepts. However, there are a few tables in which the focal columns appear out of order. For example, in table 6.1 the litter summaries are averaged by site by sex. Thus the site is how the data are first partitioned, but the site does not appear until the sixth column. Such an organization causes some confusion with the table. In addition, it is not clear why the author summarizes snakes by sex from sites A–F and G–K separately. Why not just an overall summary? Aside from a few oddities, I found most of the data tables informative and useful for illustrating points made in the text.

Although the 24 chapters of the book cover "major topics," their arrangement and content could have been organized better.

The first four chapters are mainly to set the stage *per se*, detailing the history of the rattlesnake in our culture to site descriptions and methodology used. From there, the remainder of the book focuses on the natural history and ecology of Eastern Diamond-backed Rattlesnakes. An example of the odd organization is that the geographic distribution does not appear until Chapter 13, which I feel should have been placed immediately after morphology. In addition, habitat-related items are a little disorganized and spread across multiple chapters. For example, Chapters 8 and 9 are solid together, but then additional habitat information appears isolated in Chapter 13. The remainder of the book uses a composite of data and anecdotal information to detail the ecology and natural history of the rattlesnake. To some extent, the author achieves a good mixture of this, whereas in other cases the long quotes of field notes are excessive.

Overall, the writing is good. However, I feel the text could have used more of an editorial hand to clear wordy phrases and streamline the writing. Not only would additional editing have benefited the writing, but it probably would have cleared up the issues associated with the plates, figures, and tables, thus making the information more presentable and digestible. Finally, one of the oddest things I found in the book was figure 5.8. I am not a fan of displaying unsafe handling techniques with venomous reptiles. I feel the two images of the author holding rattlesnakes by their head and draping them down to show their length is an improper message to send; add to that the author in one of the pictures is handling a venomous snake without proper footwear and solely in shorts!

Although this book contains some good information on Eastern Diamond-backed Rattlesnakes, it just has too many issues for a positive recommendation. I would not add this book to my collection at US $100.00, a high price considering that *Diamonds in the Rough* received a substantial financial subsidy from Tall Timbers as well as 215 pre-publication subscribers, many of whom are no longer with us in the 25-years since subscriptions were first solicited. I recommend persons interested in rattlesnakes examine the book first before purchasing a copy.

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A Naturalist's Guide to the Reptiles of India, Bangladesh, Bhutan, Nepal, Pakistan and Sri Lanka

Indraneil Das and Abhijit Das. 2017. John Beaufoy Publishing Limited, Oxford, England (http://johnbeaufoy.com/; available through Amazon.com [US] or Natural History Book Service [UK]). 176 pp. Softcover. £11.99, US $17.95. ISBN 978-1-909612-81-5.

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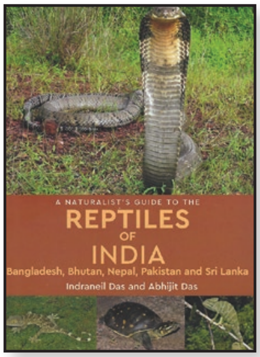

The Indian subcontinent is one of Earth's most recent major geological features (Karanth 2006; Chatterjee et al. 2017). With an area of about 4.4 million km^2, it is home to a nearly endless array of biodiversity. Politically, the Indian subcontinent consists of the Republic of India, the Islamic Republic of Pakistan, the Federal Democratic Republic of Nepal, the Kingdom of Bhutan, the People's Republic of Bangladesh, and the Democratic Socialist Republic of Sri Lanka. These countries not only include pristine habitats within their political boundaries, but also are cradles of an exploding human population. Nearly 50% of world's population resides in South and Southeastern Asia. Such an increasing population demands more space, hence forests face the brunt of habitat loss within this region. At the same time, the Indian subcontinent is home to approximately 700 species of reptiles.

The biogeography of the South Asian fauna and flora is limited by mountains and the sea, with only narrow corridors linking the landscape with adjacent regions. South Asia is isolated by the Himalayas to the north and by the Arabian Sea and the Bay of Bengal surrounding the triangular Indian peninsula, with the island of Sri Lanka to the south. The western limits of South Asia are defined by the arid zones of Baluchistan and Sind (parts of Pakistan) and the Indus River; the eastern limits are delineated by the Chittagong Hill Tracts and the wetlands of Bangladesh.

The herpetofauna of South Asia is not well-studied, especially in the tropical regions (Dubois 1999). Many new species have been described recently (e.g., Agarwal et al. 2016; Agarwal and Ramkrishnan 2017) and there is much more systematic work to be done. At a unique time like this, with decreasing natural habitats and increasing knowledge of new taxa, *A Naturalist's Guide to the Reptiles of India, Bangladesh, Bhutan, Nepal, Pakistan and Sri Lanka* is most welcome. The book offers immediate access to photographs and life history information on 280 reptile species of South Asia. Although this number is less than half of the total reptile fauna, the species covered are important medically as well as taxonomically.

The cover of the book has an imposing and impressive photograph of a King Cobra. Below, the title and authors are presented above images of a Tokay Gecko, Indian Flapshell Turtle, and an Indian Chameleon. The text of the book opens with a clear and precise 'Introduction.' The next chapter, 'Climate and Vegetation', aptly describes the physiographic features of the region and more or less follows Das (1996). 'Conservation of Reptiles' is a general summary of anthropogenic effects. This section is followed by 'Snake-Bite Management' that discusses how to deal with snake bites. The importance of this chapter increases the value of the book, as South Asia is home to many potentially lethal snakes. Moreover, this rapidly developing region lacks access to snake anti-venom in remote areas. *A Naturalist's Guide* appreciably covers more than 30 such potentially lethal snakes. These venomous snakes are widespread within their respective distribution ranges, generally in good numbers. Moreover, these snakes are found in close approximation to humans, be it in agricultural fields, along coastlines, or in rural residences. To round out the introductory material, the authors present accurate and clear color diagrams of snake and lizard scale nomenclature and a short glossary of important terms.

The species accounts begin with tortoises and turtles. As many as 30 species of tortoises and turtles, referable to four families (Testudinidae, Geoemydidae, Trionychidae, and Cheloniidae), are discussed. Generic names and distributional ranges have been updated. Five species of the family Testudinidae are included, of which the endemic *Indotestudo travancorica* is a highlight. Fourteen species of the family Geoemydidae are covered. Information on the rare Arakan Hill Turtle (*Heosemys depressa*) and a pictorial presentation of sexual dimorphism in *Batagur* species are noteworthy. Seven trionychid species are amply discussed, and the Indian Flapshell account includes a fine plastron photograph that will aid in identification. Lastly, four species of marine turtles (family Cheloniidae) are covered along with information on their reproductive biology. Tortoises and turtles of South and Southeast Asia are subject to poaching and illegal trade. Moreover, their extremely secretive life histories make them difficult to study.

The lizard accounts begin with the agamids, a very diverse group. As might be expected, covering all species of this family within South Asia would be impossible. Singular and long-used common names are applied to "species" that are actually composed of multiple species hidden within species complexes. The agamid section begins with information on the identification and natural history of the monotypic taxon *Bufoniceps laungwalaensis* and is well done. This account is followed by accounts of the highly diverse genus *Calotes*. Nine species are presented covering the type species *C. calotes* and the widespread *C. versicolor* complex (Zug et al. 2006). Adequate coverage of endemic species from the Western Ghats, Sri Lanka, and northeast India is included. However, *Calotes rouxii*, a widespread peninsular endemic, and *C. grandisquamis*, a Western Ghats endemic, could have been addressed here. Three of the five described species of the insular endemic Sri Lankan genus *Ceratophora* are included. *Cophotis*, another endemic genus of same region, has only been covered by its type species (*C. ceyalnica*); adding *C. dumbara* would have completely summarized this genus. The Bay Island endemic genus *Coryphophylax* is represented by a single species, *C. subcristatus*. However, *C. brevicaudus* should have been used as a general account. Nevertheless, information on the identification and ecology of this endemic genus has been neatly presented.

Only a single account covers the genus *Draco*, *D. norvilii*, thus missing the widespread peninsular endemic *D. dussumieri*. The genus *Japalpura* is covered by three species (*J. andersoniana*, *J. kumaoensis*, and *J. planidorsata*); much of the diversity within *Japalura* is Tibetan and southern Chinese, which is beyond South Asia. The single species *Laudakia tuberculata* aptly represents this Palearctic genus, as *L. tuberculata* is widespread in the western and central Himalayas. Another Sinhalese endemic, *Lyriocephalus scutatus*, is covered properly, giving due descriptive justice to this flamboyant lizard. Only one species of three known endemic kangaroo lizards, *Otocryptis weigmanni*, is covered. Another Palearctic element, *Paralaudakia caucasia*, is also included. Two peninsular species of *Psammophilus* are known, both of which are covered in this volume. The genera *Ptyctolaemus* and *Saara* are correctly represented by one species each, *P. gularis* and *S. hardwickii*. *Salea horsfieldi* is endemic to the Western Ghats, but another species of this genus, *S. anamallayana*, has been unduly overlooked. The genus *Sitana* is covered by the widespread species *S. ponticeriana* and an insular (Sri Lanka) endemic, *S. bahiri*. *Trapelus* is represented by two species, both Palearctic. The family *Chamaeleonidae*, with a single species (*Chamaeleo zeylanicus*) throughout the region, is naturally included.

The diverse cosmopolitan family Gekkonidae is represented by 35 species. All species are aptly described with respect to their identification characters. Highly diverse genera such as *Cnemaspis*, *Cyrtodactylus*, *Cyrtopodion*, *Gehyra*, *Gekko*, *Hemidactylus*, *Hemiphyllodactylus*, *Lepidodactylus* and *Phelsuma* are discussed, and their respective accounts are accurate. The family Eublepharidae is represented by two broadly distributed species, *E. macularius* and *E. hardwickii*. The family Lacertidae is represented by five species referable to four genera: *Acanthodactylus*, *Eremias*, *Ophisops*, and *Takydromus*. *Acanthodacytlus* and *Eremias* are represented by a single species each, *A. cantoris* and *E. acutirostris*; *Ophisops includes* two species, *O. jerdoni* and *O. leschenaultii*. The diverse genus *Takydromus* is represented by only a single species, *T. khasiensis*. *Takydromus sikkimensis* and *T. sexlineatus* should have been included, as both are found in the eastern Himalayas. The family Scincidae has been nicely done, covering 27 species. The natural history of skinks is little-known and they are one of the least studied groups taxonomically among South Asian terrestrial reptiles. The greatest diversity of scincid species is found in the genera *Eutropis*, *Lygosoma* and *Sphenomorphus* on the mainland and *Lankascincus* endemic to island of Sri Lanka. However, even lesser known skinks such as *Ophiomorus raithmai* have been covered. After the Scincidae, two additional families, Anguidae and Dibamidae, are documented by accounts of a single representative species. The section on the family Varanidae covers the three widespread and common species: *V. bengalensis*, *V. flavescens*, and *V. salvator*.

The section on snakes is the most extensive part of this book, with snake accounts comprising nearly half of the species. Presenting information on the more than 300 species of snakes within the region is not possible in a single field guide. Still, the authors have carefully accounted for all the widespread species. More than 140 species referable to 15 families are included and cover the ophidian fauna in almost all habitat types within this region. There is a justifiable emphasis on the family Colubridae, and the guide adequately covers the common and regionally important species. Accounts of the families Boidae, Pythonidae, and Xenopeltidae include all the known species from South Asia. As mentioned earlier, medically important snakes of families Elapidae and Viperidae are covered, although I think that accounts of *Echis carinatus sochureki* (or *Echis sochureki*) and *Naja oxiana* should have been added. Both taxa are medically important and are not restricted in range. Lastly, the crocodile section is complete with all three species known from this region.

The taxonomy of the species discussed in the book is accurate, due no doubt to the authors' expertise in this field. A green text box that provides a short but specific introduction to each family/group and gives the major characters and global diversity of that taxon is helpful. The very last section before the index provides a comprehensive checklist of the reptiles known from this region. This compilation of all species names with updated generic placements and the latest IUCN status is commendable. It is very intriguing to see that so many taxa in this checklist are assigned a NE (not evaluated) status. This is because of a paucity or unavailability of any data on these taxa. At the same time, India, Pakistan, Nepal, Bhutan, Bangladesh and Sri Lanka are home to diverse types of natural habitats, sadly being lost to deforestation.

The last complete treatise on the reptiles of the Indian subcontinent was by Malcolm A. Smith (1931, 1935, 1943). This series still forms the basis for taxonomic and systematic research on South Asian herpetology. An increasing awareness of species concepts and newly available technologies warrant new surveys and well-planned systematic research. Still, there are many areas within this region about which we know little of the herpetofauna. Biological inventories offer fresh insights to species' distribution and diversity, especially in a biologically rich region such as South Asia. *A Naturalist's Guide to The Reptiles of India, Bangladesh, Bhutan, Nepal, Pakistan and Sri Lanka* will come in handy in developing an inventory and assessment of reptiles because of its clear photographs, ample identification characters, and behavioral and ecological notes. The size of the book is perfect to carry in the field; good color photographs and a well-written text add to the virtue of this field guide. This book delivers what promises in its title and stands as a ready answer to the identification of reptiles for both present and upcoming generations of field biologists. I highly recommend it for all reptile enthusiasts, book collectors, 'in field' scholars, libraries of faunal research institutes, and academic faculty members who teach herpetology.

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Sea Turtle Health and Rehabilitation

Charles A. Manire, Terry M. Norton, Brian A. Stacy, Charles J. Innis, and Craig A. Harms (editors). 2017. J. Ross Publishing, Plantation, Florida (http://www.jrosspub.com/sea-turtle-health-and-rehabilitation.html). 1045 pp. Hardcover. US $295.00. ISBN: 978-1-60427-099-0.

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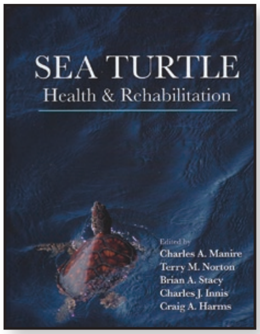

Sea Turtle Health & Rehabilitation is a comprehensive text covering sea turtle husbandry, medicine, and surgery. The book presents information on life history, rescue, medical diagnostics, care, and rehabilitation of sick and injured sea turtles. In-depth information is provided on the physiological systems of sea turtles, causes and sources of illness and injury, diagnostic procedures, and medical and surgical treatments. There are chapters on important subjects related to sea turtle health and conservation, such as mortality investigation, fisheries interactions, oil spills and other environmental toxins, pathogens, cold stunning, and harmful algal blooms.

Prior to the publication of this unique work, the sea turtle community relied on scattered presentations, chapters, and papers to obtain information on sea turtle biology, care, and rehabilitation. Available references focused more on anatomy and biology (Eckert et al. 1999; Wyneken 2001; Wyneken et al. 2013). Of these, none had the in-depth medical information available here. The contents range from the life history of sea turtles to the more detailed and complex issues related to the diagnosis and treatment of conditions that are frequently encountered in sea turtles. Richly illustrated, the material is designed to provide value to the reader facing their first sea turtle or their thousandth. This is an important volume for veterinarians, veterinary students, sea turtle rehabilitators, biologists, and conservationists who work with sea turtles. The book is medically-focused, so topics in life history and conservation, while presented here, are best referenced from other sources.

The five-man editorial team represents leaders in the field of sea turtle medicine, pathology, and management. Each editor also contributes significantly as an author. As each oversees a section that directly relates to their area of specialization in sea turtle medicine, they create a balance that flows through the text providing maximum benefit to the reader. As authors, they clearly demonstrate both the breadth and depth of their experience. The contributor list includes material from 49 additional experts. This team extends the editors' contributions to present a global base of information and depth of material in areas like toxicology, ophthalmology, nutrition, clinical pathology, and the intricacies of the stranding response networks.

The material is divided into six sections. The introduction is edited by Terry Norton and presents identification, taxonomy, natural history, and a big picture perspective on sea turtle rehabilitation. This is a small section and designed to set the stage—not to serve as a comprehensive reference on the taxa or the process of stranding response. Blair Witherington is the best possible author to select for this well-presented overview of the taxa. Norton provides perspective on the expanding efforts of rehabilitation and the introduction rolls seamlessly into the second section, Husbandry, again edited by Norton. Here, the nuts and bolts of a rehabilitation facility, including water quality and biosecurity, are reviewed with gold standards suggested. Norton teams up with two professional nutritionists to address husbandry, including an extensive presentation on nutrition. The nutrition chapter includes information on growth rates and body scoring to assess condition. Body scoring is presented in multiple areas in the book and might better have been focused in the clinical examination section. The in-depth material in this chapter includes tables of dietary evaluations. In spite of this, I searched in vain for a recipe for hand feeding formula or simple diet formulation. This material is available in the appendices and Web Added Value material associated with this chapter. Tables of blood and tissue vitamin and mineral concentrations are awkward because healthy animals are intermixed with values from unhealthy animals and the tables fail to identify the sample sizes used to create the values presented.

Section three, Basic Veterinary Techniques, edited by Charles Manire, thoroughly depicts techniques from clinical examination to necropsy with lovely chapters on imaging and clinical pathology. These chapters present the material in a "how to" fashion laying out unique anatomical features as well as common injuries and descriptions of illnesses. The imaging chapter details positioning techniques for radiographs and ultrasonography and includes examples of results from these modalities as well as CT, MR, and nuclear scintigraphy exams. The clinical pathology chapter by Nicole Stacy and Charles Innis is an extraordinary compilation of hematologic and biochemical data along with excellent plates illustrating blood cells and cytological preparations of disease states. The section wraps up with a discussion of how to and what to expect related to the necropsy exam of sea turtles. As with the nutrition chapter, the "what can I use today" materials such as a necropsy report form are available in the appendices and on line.

Sections four and five edited by Brian Stacy and Charles Innis present the meat of the text. Here, medicine and surgery are organized by system (section 4) and as current therapies (section 5). This structure affords easy presentation of foundational material as well as current best practices. Each systemic chapter describes normal anatomy, revisits the clinical exam, and provides well-illustrated overviews of diseases and injuries. This section will be the most valuable to anatomic pathologists working with material from sea turtles. The Current Therapy section begins with emergency medicine, therapeutics, and analgesia/anesthesia. Appendix 7 tabulates therapeutics in a formulary for ease of reference. Unfortunately, therapeutics related to analgesia and anesthesia are not included in this table and the reader must refer to chapter 22 for this information. A more comprehensive therapeutics table would be appreciated. Surgical techniques such as hook removal and shell repair (the sea turtle doctor's bread and butter) are well illustrated to encourage clinicians facing these common concerns with step-by-step instructions and images.

Section 6, Special Topics, is edited by Craig Harms and enhances material introduced in many of the previous chapters. The chapters vary from categories of pathogens such as parasites, toxins, and viruses, to field techniques, fisheries issues, oil spill concerns, and management and mortality investigations. As in prior sections, the appendices provide supporting materials for use in incidents of strandings. The web-based appendices provide links for downloading of forms.

The table of contents is in the expanded form including the chapter headings and subheadings. Normally, I am not a fan of this style as it generally adds pages but not value. In this case, the chapters cover so many topics that having the subheadings is appreciated. In the appendices this is not the case; the format is redundant. Likewise, the Quick Reference Index Citations for Other Relevant Chapters provides an unnecessary internal reference. In a book of this size, trimming the excess is critical.

Overall, the text is a great advance to those working with sea turtles, most specifically those employed in the field of medicine for sea turtle care and conservation. Unfortunately, the price for this text at US $295.00 when purchased direct from the publisher is a bit of a punch in the gut. Comparative texts such as *Reptile Medicine and Surgery* edited by Doug Mader and *Invertebrate Medicine* edited by Greg Lewbart sell at less than half this price. Although the book is excellent and cost is reasonable for the veterinary professional, students and international organizations will be hard pressed to lay down this amount for a single text. The publisher has contacted non-profit organizations committed to wildlife medicine to encourage purchasing and donating books. I am hopeful that they can find a reasonable way to make the material available. Those working with sea turtles have become accustomed to snapping up the latest volume of the *Biology of Sea Turtles* every few years to remain current in this area at a cost around $100. Future editions of *Sea Turtle Health and Rehabilitation* should consider limiting the focus to current therapies and special topics to provide a collection of materials from at a more reasonable cost.

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Freshwater Turtles of Australia

John Cann and Ross Sadlier. 2017. CSIRO Publishing, Clayton South, Victoria, Australia (www.publish.csiro.au) and ECO Publishing and Distribution, Rodeo, New Mexico (www.ecouniverse.com). 448 pp. Hardcover. US $100.00. ISBN: 978-1-938850-19-6. Leather bound edition US $169.95. ISBN: 978-1-938850-24-0.

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Freshwater Turtles of Australia is a substantially updated and expanded version of *Australian Freshwater Turtles* (Cann 1998) and a welcome addition to the literature. The title is somewhat deceiving in that the book also reviews the turtles of New Guinea. A great deal of new research has focused on turtles in Australia and New Guinea since the earlier publication, and it is nicely summarized in this updated book. Cann's first book on Australian turtles was preceded by two other attempts to review what was known about Australian turtles (Goode 1967; Cann 1978), but in much smaller books. Otherwise, publications on Australian turtles lagged behind earlier attempts to produce sizeable compendia of turtles at continental (e.g., Pope 1939; Carr 1952; Ernst and Barbour 1972; Ernst et al. 1994) and global scales (e.g., Pritchard 1979; Ernst and Barbour 1989).

As Cann and Sadlier explain in the Introduction to their new book, the lack of a more complete review of Australian turtles was partly due to the fact that for several decades, research on Australian turtles "…stood under the shadow of priority by [a] US turtle worker…", now deceased, who had amassed a huge collection of chelids from that country. For whatever reason, the expected grand synthesis on Australia's turtles never came out of that university laboratory. Australian naturalists stopped waiting patiently in the 1990s and there was a resurgence of publications on their own turtles. A measure of the increase in knowledge of Australian turtles during that time is provided by a quick scan of my bibliographic database of over 8000 turtle citations as of mid-2018 (see Lovich and Ennen 2013). Prior to 1998, there were 23 citations that had the words "turtle" and "Australia" in the bibliographic entry. From 1998 to the present, there were 63 matching those words. Although these numbers are relatively small, they nevertheless reflect a strong resurgence in interest.

At 32.4 × 24 cm and over 2.6 kg, the publication is essentially a coffee table book that would not be easy to use as a field guide. However, another turtle book by Cann (2008) fills the field guide niche due to its smaller size. *Freshwater Turtles of Australia* is printed on heavy, high gloss paper and is a worthy addition to any herpetologist's library. It is well-written and well-organized, starting with a foreword by Chuck Shaffer that reviews the fascinating history of discovery of Australia's turtle fauna starting in the late 1700s. Highlights of more recent findings since Cann (1998) are nicely summarized by Cann and Sadlier in the Introduction, especially as they relate to conservation, taxonomy, and systematics. The taxonomy of Australian turtles has been especially fluid, and Cann and Sadlier recognize 29 species in Australia and 18 in New Guinea, including one in the former and five in the latter that do not yet have scientific names. Of the 47 species included in the new book, only about 40 are recognized in the most recent checklist of turtles (Turtle Taxonomy Working Group 2017), and not always in the binomial combinations used by Cann and Sadlier. Clearly, more work needs to be focused on the taxonomy of turtles in Australia and New Guinea.

Chapter 1 covers turtles in Aboriginal culture with an excellent collection of photographs showing Aboriginal art and rock paintings depicting turtles. This cultural element adds a human dimension to the book, often missing in scientific treatises. With a traditional cultural knowledge extending back over 60,000–80,000 years, this is an important part of the story of Australia's turtles. An Acknowledgments section follows. The remaining chapters are systematic accounts of the various taxonomic groups: Chapter 2—Long-neck turtles genus *Chelodina*; Chapter 3—Snapping turtles genus *Elseya*; Chapter 4—Saw-shell turtles genus *Wollumbinia*; Chapter 5—Fitzroy River turtle *Rheodytes leukops*; Chapter 6—Mary River turtle *Elusor macrurus*; Chapter 7—Short-neck turtles genus *Emydura*; Chapter 8—Western swamp turtle *Pseudemydura umbrina*; Chapter 9—Pig-nose turtle *Carettochelys insculpta*; Chapter 10—New Guinea freshwater turtles. Species accounts include detailed subsections on description, distribution, and natural history. Following these chapters is a section of References with over 280 citations. By comparison there are over 1000 citations in Cann (1998), presumably because mostly newer research is summarized in the new edition. The concluding chapter contains short bios of the authors.

The authors are highly qualified natural historians with very interesting personal histories. John Cann was born into a family of "reptile-show people" who entertained and educated the public about reptiles for years at their home in La Perouse (near Sydney). Details on their family show, "Snake man," are featured on Wikipedia (https://en.wikipedia.org/wiki/Snake_Man_of_La_Perouse). His father George was the Curator of Reptiles at the Taronga Zoo for 20 years, where John developed his lifelong interest in turtles. A little-known fact is that John's athletic prowess as a young man allowed him to represent Australia and compete in the Olympics (decathlon) in Melbourne in 1956. His physical prowess undoubtedly contributed to his field stamina over many years of catching turtles and other reptiles throughout Australia. Ross Sadlier also grew up in the Sydney area and, like John, his father instilled a passion for animals in his son. Ross worked at the Australian Museum in the Herpetology section for 36 years. Additional details on their careers are given in the book.

The only issue of note that I identified in the book was the authors' use of yet another terminology describing the scutes of the turtle carapace and plastron that are unlike those used in previous publications. Different terms are already used by herpetologists throughout the literature. Dundee (1989) tried to standardize usage but his terminology was not generally accepted (e.g., Ernst and Barbour 1972; Ernst et al. 1994; Ernst and Lovich 2009). What Dundee called costal scutes are pleural scutes in the Ernst publications, and laterals in Cann and Sadlier. Both Dundee and Ernst refer to the large central carapace scutes as vertebrals, but Cann and Sadlier call them centrals. These inconsistencies continue to challenge morphological descriptions of turtle shells and a common terminology remains to be presented and widely accepted.

The book is richly illustrated throughout with high quality photographs of each species, including examples of geographic variation. There are also very good photographs of turtle habitats and a smattering of photos showing people catching turtles in the field allowing the reader to see what is was like for Cann and Sadlier to collect turtles in the bush over the years. This is a great book written by people who know how to both present detailed data on natural history and do so in an engaging and readable fashion. If you are interested in Australian natural history, especially about turtles, you will want to buy and read this book.

Acknowledgments.—I thank Whit Gibbons, Jenna Norris, and Shellie Puffer for reviewing an earlier draft of this review.

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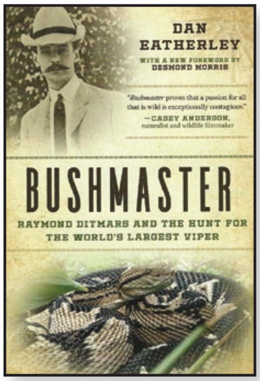
PUBLICATIONS RECEIVED

Bushmaster. Raymond Ditmars and the Hunt for the World's Largest Viper

Dan Eatherley. 2017. Arcade Publishing, New York (arcade@skyhorsepublishing.com). 302 pp. Paperback. US $16.99. ISBN 978-1-62872-766-1, e-book ISBN 978-1-62872-555-1.



For generations of budding herpetologists, the books by Raymond Ditmars (1876–1942), particularly *The Reptile Book* and *Snake-Hunter's Holiday*, were inspirational. Traipsing through exotic regions hunting for snakes filled the daydreams of many of our colleagues growing up from the 1920s through the 1950s. Although often perceived as more of a popularizer than scientist by many of his contemporaries (he had no formal scientific training), Ditmars' contributions to what is now termed public outreach have had a long-lasting impact on our science and the ways we perceive the animals we study and the humans on whose support we depend for their conservation.

Bushmaster is the story of Raymond Ditmars, from his early childhood catching snakes in the vicinity of his New York City home to more adventurous excursions seeking the legendary Bushmaster (*Lachesis muta*; now considered a complex of four species) in Central and South America and Trinidad. The title focuses on the Bushmaster quest, but the book is equally about Ditmars' career as curator, filmmaker, lecturer, and educator through his long association with the New York Zoological Society. The book re-counts the developmental history of the zoo as well as Ditmars' pivotal role in the growth of its reptile, insect, and mammal programs. Much of the narrative is thus about history rather than exciting tales of catching snakes oriented toward amateur snake-catchers. It presents Ditmars' persona as a clear-headed but enthusiastic ophidiophile rather than as a thrill-seeker.

Dan Eatherley provides a readable narrative, alternating between the late 1800s to mid-1900s following Ditmars' career and present-day journeys, as Eatherley retraces Ditmars' steps from New York to the tropics, stopping along the way to see live and preserved Bushmasters and the habitats in which the should be found. Alas, neither actually sees a live Bushmaster in its natural habitat. Captive husbandry has improved substantially since Ditmars' era, even as the snake's habitat succumbs to human onslaught and Bushmasters face unrelenting persecution.

Historians of herpetology, zoo biologists, and naturalists will enjoy this book. Originally published in hardback in 2015, the 2017 paperback edition contains a new Foreword by Desmond Morris. Dan Eatherley is a writer, environmental consultant, and filmmaker based in Exeter, United Kingdom.

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Herpetological History of the Jacksonville Zoo and Gardens

A myriad of zoological parks and roadside animal attractions have dotted the Florida, USA, landscape over the past century and have played key roles in introducing millions of visitors to the state's wildlife, particularly its famed herpetofauna. Although few roadside attractions remain today, Florida is currently home to 17 zoological parks and aquariums accredited by the Association of Zoos and Aquariums (AZA). At least nine of these facilities continue the tradition of displaying and introducing visitors to diverse collections of both native and exotic herpetofauna, and several have made important contributions to the field of herpetology (Card and Murphy 2000; Murphy 2007).

Having recently celebrated its centennial anniversary in 2014, the Jacksonville Zoo and Gardens, located in northeastern Florida, is one of the state's oldest AZA-accredited zoological parks. Over its history, the zoo has displayed a diverse assemblage of herpetofauna and is responsible for several significant captive breeding events, has contributed numerous publications on herpetology and captive management, and has played a crucial role in recovery efforts for several imperiled reptile and amphibian species. Yet, despite its rich herpetological tradition, the Jacksonville Zoo and Gardens has received very little attention in published works highlighting the herpetological history and contributions of zoos (e.g., Card and Murphy 2000; Murphy 2007). In light of this general absence from the historical literature, here we provide an extensive overview of the history of herpetology at the Jacksonville Zoo and Gardens from its early 20th Century inception to the present day. We discuss trends in its animal collection, exhibits, and staffing over time, as well as the many important achievements and contributions made by the zoo and its staff to the fields of herpetology and herpetological husbandry.

Zoo Origins and Overview

The Springfield Park Zoo was originally established in 1914 by the city of Jacksonville in an 18-acre (7.2-ha) park in the neighborhood of Springfield, located just north of downtown. After persisting at this site for more than a decade but experiencing various problems including severe flooding from an adjacent creek and complaints from local residents of foul odors emanating from the zoo (David et al. 2014), the decision was made in 1925 to relocate the zoo to a 37.5-acre (15-ha) site along the Trout River on the north side of the city. There, it was renamed the Jacksonville Municipal Zoo and was initially accompanied by a natural history museum, which displayed a large collection of taxidermied wildlife owned by museum director B. O. Crichlow (David et al. 2014). One of the most celebrated exhibits of the museum was the mount of a large 544-kg Devil Ray (*Mobula mobular*) that had been collected off of St. Augustine, Florida with a dead 22-kg sea turtle lodged in its jaws (David et al. 2014). Although an official date or record of its eventual closing could not be ascertained, the museum and its collection had apparently relocated to Miami, Florida sometime before 1933 (Anonymous 1933).

As a municipality-run facility, many of the zoo's animal keeper staff during its first six decades lacked significant training, experience, or familiarity with animal biology or husbandry and were often workers transferred in from other city departments (Anonymous 1979), sometimes as demotions or retribution for poor work performance elsewhere (P. Sachs, pers. comm.). Like many other municipal zoos of the era, the Jacksonville Zoo (renamed the Jacksonville Zoological Park in 1966) fell on hard financial times by the 1960s. In 1971, the decision was made to transfer management and oversight of the zoo from the city's Department of Recreation and Public Affairs to a newly formed not-for-profit organization, the Jacksonville Zoological Society. This privatization and restructuring, which also took place at many other zoos in the United States around the same period (Kisling 2001), enabled the zoo to become more effectively managed and eventually rebound from its financial woes. As part of this transition, there was an unfortunate loss of many institutional records and paperwork, particularly those documenting the zoo's animal collection and staffing prior to 1971.

Since the early 1990s, the zoo has undergone extensive capital improvements, including the modernization and

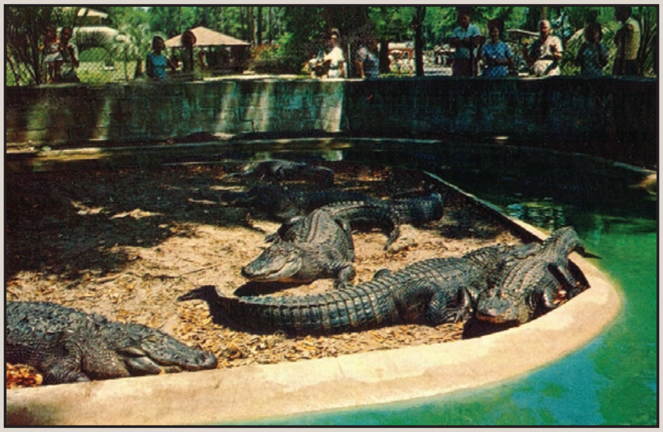

Fig. 1. An early *Alligator mississippiensis* exhibit at the Jacksonville Zoo and Gardens. Undated postcard, ca. 1975.

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PHOTO COURTESY OF PETER SACHS

Fig. 2. The Jacksonville Zoo and Gardens' first building dedicated to exhibiting herpetofauna, ca. 1971–1972.

PHOTO COURTESY OF PETER SACHS

Fig. 3. Reptile curator Peter Sachs receives an "adopt an animal" check in front of the zoo's original reptile building in the early 1970s. An Amazon Basin Emerald Tree Boa (*Corallus batesi*) can be seen on display in background.

PHOTO COURTESY OF PETER SACHS

Fig. 4. Interior service corridor of the zoo's first building dedicated to displaying reptiles. Early 1970s.

construction of many new exhibits, buildings, and gardens. The zoo changed its moniker to the Jacksonville Zoological Gardens in 1993 and then once again in 2003 to the Jacksonville Zoo and Gardens (hereafter JZG), and was officially recognized by the city as a botanical garden the following year. The zoo's current footprint is ca. 117 acres (47.3 ha), of which 29 acres (11.7 ha) remain undeveloped, and as of 1 November 2017, the animal collection comprised a total of 1705 specimens representing 257 species. Over the last decade, JZG has become the most popular and heavily visited attraction in northern Florida, now seeing an annual attendance of around one million visitors.

Herpetology at the Jacksonville Zoo and Gardens

Due to the paucity of available records, it is unclear when an official herpetology department was established at JZG; however, an earlier incarnation of the department around 1970 was known as the "reptile project" (P. Sachs, pers. comm.). There appears to have been six official herpetology curators to date: Peter S. Sachs (1971–1976), Ralph Williams (1980–1981, 1985–1986), David Collins (1982–1985), Jack Meyer (1986–1991), Greg Lepera (1994–2007), and Dino Ferri (2007–2013). Early on, reptile department staff sometimes consisted of just a single person, who performed both keeper and curatorial duties (Anonymous 1979; D. Collins, pers. comm.). In 2014, the herpetology curator position was merged with that of the bird and ambassador animal collections, and is currently held by Mike Taylor. In addition to managing the reptile and amphibian collection, the herpetology department also oversees the zoo's marine and freshwater fishes and invertebrates. Seven full-time keepers (including three aquarists) and a supervisor currently comprise the herpetology department.

Exhibits.—Although an outdoor alligator exhibit has existed in one form or another since the zoo's inception (e.g., Fig. 1.), the first official building dedicated specifically to the keeping of herpetofauna was a free-standing structure that had been constructed on a slab under a covered picnic area on the southwestern side of the zoo in 1957 following the donation of a large private collection of snakes (Figs. 2, 3). This building featured around 30 exhibits with viewing windows encircling its perimeter, a central service corridor (Fig. 4), and retractable roofing panels (P. Sachs, pers. comm.). It was here that most of the zoo's reptile breeding took place during the 1970s to early 90s (see below), until it was finally decommissioned, razed, and replaced by more modern reptile and amphibian buildings throughout the zoo (see below). During the early 1980s, there were also several reptile enclosures scattered throughout the zoo, including an off-exhibit building, a small barn with multiple tortoise pens, a crocodile pool, and a lizard pit (D. Collins, pers. comm.).

Today, JZG is unique among most zoos in that instead of a single, centralized reptile house, its herpetological collection is distributed throughout the zoo in four reptile and amphibian-specific buildings and one mixed-collection building displaying reptiles, amphibians, freshwater fishes, and small mammals. Most of these buildings have a particular biogeographic focus.

The first of the zoo's current herpetology buildings to open was the African Reptile House in 1998, which houses a variety of African taxa in 13 exhibits ranging in size from smaller cubicle-style terraria to a large room-sized exhibit. Although no longer a fixture in the building, Naked Mole Rats (*Heterocephalus glaber*) were previously displayed here alongside African Mole Snakes (*Pseudaspis cana*).

The Wild Florida Reptile House opened three years later in 2001, and displays native Floridian species along with some introduced exotic species in a series of 17 indoor exhibits; the two largest exhibits also feature outdoor components. Adjacent to this building is a large outdoor exhibit for American Alligators (*Alligator mississippiensis*) as well as an Alligator Snapping Turtle (*Macroclemys temminckii*) exhibit.

Range of the Jaguar, a large exhibit complex featuring both indoor and outdoor displays of Central and South American taxa (and a few Caribbean species) opened in 2004, and includes a large walk-through aviary with a 150,000-liter pool housing several neotropical turtle and fish species, several large outdoor mammal exhibits, and a Mayan temple-themed building with 11 indoor reptile and amphibian exhibits (Fig. 5) and a few small mammal exhibits.

Repurposed from a building that formerly housed koalas, and was used as a nursery for newborn animals before that, the zoo's Amphibian Conservation Center (originally named "Save the Frogs") opened in 2007 to coincide with and celebrate the Amphibian Ark's "Year of the Frog" conservation campaign (Grow and Allard 2008; Pavajeau et al. 2008). This building features two independent laboratories dedicated to the ex-situ conservation breeding of threatened amphibian taxa (see below), as well as a central display area showcasing several additional amphibian species. All three rooms are located behind large viewing windows, offering visitors a glimpse into the daily husbandry and conservation work carried out by the zoo's herpetology staff (Fig. 6).

The most recent reptile building to be constructed at JZG is the Komodo Dragon (*Varanus komodoensis*) facility, which opened in 2009 as part of the initial phase of the zoo's Asian exhibits expansion. Architecturally themed as an Indonesian fishing village within Komodo National Park, this facility features both indoor and outdoor exhibits as well as a series of off-exhibit indoor enclosures and an outdoor holding yard.

A temporary exhibition highlighting the prehistoric megasnake *Titanoboa* was developed by the zoo and held from March to September 2015 in the zoo's Discovery Center. It featured a larger-than-life-sized model of *Titanoboa*, osteological mounts of extant snake taxa, and several exhibits displaying living representatives of some of the largest native Floridian (*Crotalus adamanteus*, *Drymarchon couperi*) and exotic (*Python bivittatus*, *Eunectes murinus*) snake species alive today.

Living Collection and Captive Breeding.—Significant efforts have been made by one of the authors (AFR) to historically reconstruct records documenting JZG's living collection since the zoo's inception. Although these efforts are ongoing, from records that could be retrieved, at least 321 species of reptiles and 78 species of amphibians have been maintained by the zoo over its history. The first reptile species to be kept by the zoo was *Alligator mississippiensis*, in 1914. The Gopher Tortoise (*Gopherus polyphemus*) was also an early captive displayed by the zoo, with records dating back to at least 1938; however, given the species' ubiquitous occurrence in the area, including on zoo grounds, it is likely the species was kept even earlier. Additional reptiles documented in an inventory record from 1938 included 24 *A. mississippiensis*, a Common Snapping Turtle (*Cheyldra serpentina*), and several unspecified snake species, including one rattlesnake (*Crotalus* sp.). While most of the herpetofauna maintained during the zoo's early history appears to have been native Floridian species, the collection began to grow and diversify to include non-native taxa in the late 1950s, coinciding with the opening of the zoo's first official reptile building, the emergence of several prominent reptile dealers in Florida such as Ross Allen's Reptile Institute that periodically supplied JZG with specimens, and the 1957 donation of a large private collection of snakes to the zoo.

PHOTO BY ROBERT W. MENDYK

Fig. 5. Interior view of reptile exhibits in the Range of the Jaguar Temple building, 2017.

PHOTO BY ROBERT W. MENDYK

Fig. 6. Public view of one of the two bio-secure conservation breeding laboratories in the zoo's Amphibian Conservation Center, 2017.

Successful reproduction has occurred in at least 53 reptile and 13 amphibian species at JZG (Tables 1, 2). However, it is difficult to ascertain, particularly in older records, whether some of these records represented true captive breeding events or offspring produced by wild-caught females that arrived at the zoo gravid. The earliest documented reptile births at the zoo were of the Sidewinder (*Crotalus cerastes*) and West African Gaboon Viper (*Bitis rhinoceros*) in 1970; the earliest record of amphibian reproduction occurred in the African Clawed Frog (*Xenopus laevis*) in 1979. Many reptiles and amphibians have reached significant ages at JZG (e.g., Mendyk and Smith 2016); noteworthy records of longevity are presented in Table 3.

At least 83 lizard species have been kept by the zoo. Noteworthy taxa have included the Sungazer (*Smaug giganteus*), Fiji Banded Iguana (*Brachylophus fasciatus*), Ricord's Ground Iguana (*Cyclura ricordi*), and the Bahamian Rock Iguana (*C. rileyi*). A locally collected amelanistic Eastern Glass Lizard (*Ophisaurus ventralis*) was displayed in the early 1970s (P. Sachs,

Table 1. Breakdown of reptile taxa successfully reproduced at the Jacksonville Zoo and Gardens 1970–2017. *Asterisked entries represent captive birth (from a recently acquired gravid female) rather than true captive breeding.

Order	Family	Species	Common name	Earliest known reproductive event
Testudines	Emydidae	*Pseudemys concinna peninsularis*	Peninsular Cooter	1989
		Terrapene carolina bauri	Florida Box Turtle	2013
	Geomydidae	*Heosemys annandalii*	Yellow-headed Temple Turtle	2013
		Rhinoclemmys pulcherrima incisa	Painted Wood Turtle	1985
	Kinosternidae	*Kinosternon odoratum*	Common Musk Turtle	unknown
	Testudinidae	*Aldabrachelys gigantea*	Aldabra Tortoise	1984
		Chelonoidis carbonaria	Red-foot Tortoise	1986
		Geochelone elegans	Indian Star Tortoise	1987
		Geochelone sulcata	African Spur-thighed Tortoise	1990
		Gopherus polyphemus	Gopher Tortoise	1984
		Kinixys belliana nogueyi	Bell's Hinged-back Tortoise	2000
		Malacochersus tornieri	Pancake Tortoise	2013
		Stigmochelys pardalis babcocki	Leopard Tortoise	1985
	Chelidae	*Chelodina novaeguineae*	New Guinea Snakeneck Turtle	1983
	Pelomedusidae	*Pelusios subniger subniger*	East African Black Mud Turtle	2000
Squamata – Sauria	Agamidae	*Pogona vitticeps*	Bearded Dragon	1999
		Xenagama taylori	Taylor's Shield-tailed Agama	2017
	Anguidae	*Ophisaurus ventralis*	Eastern Glass Lizard	2013
	Cordylidae	*Smaug warreni*	Warren's Girdled Lizard	2015
	Eublepharidae	*Eublepharis macularius*	Leopard Gecko	1979
	Gekkonidae	*Hemidactylus turcicus*	Mediterranean House Gecko	1978
		Thecadactylus rapicaudus	Turnip-tailed Gecko	1990
	Iguanidae	*Iguana iguana*	Common Iguana	unknown
Squamata – Serpentes	Boidae	*Boa constrictor constrictor*	Red-tailed Boa Constrictor	1972
		Chilabothrus angulifer	Cuban Boa	1980
		Chilabothrus chrysogaster	Turks Island Boa	1974*
		Chilabothrus fordi	Haitian Ground Boa	1980
		Chilabothrus inornatus	Puerto Rican Boa	1975
		Chilabothrus strigilatus fosteri	Bimini Boa	1972
		Chilabothrus s. strigulatus	Bahamian Boa	1976
		Chilabothrus subflavus	Jamaican Boa	1976
		Corallus hortulanus hortulanus	Amazon Tree Boa	1982
		Epicrates alvarezi	Argentinian Rainbow Boa	1988
	Pythonidae	*Python regius*	Ball Python	1997
		Python sebae	Northern African Rock Python	2012
	Colubridae	*Drymarchon couperi*	Eastern Indigo Snake	1984
		Hydrodynastes gigas	False Water Cobra	1972
		Lampropeltis triangulum hondurensis	Honduran Milksnake	1984
		Nerodia clarkii compressicauda	Mangrove Salt Marsh Snake	2002
		Orthriophis taeniurus	Taiwan Beauty Snake	1971
		Pantherophis alleghaniensis	Eastern RatSnake	1977
		Pantherophis guttatus	Red Ratsnake	1976
		Pituophis melanoleucus mugitus	Florida Pinesnake	1985
		Pituophis ruthveni	Louisiana Pinesnake	2015
	Viperidae	*Atheris chloroechis*	Western Bush Viper	2000
		Bitis rhinoceros	West African Gaboon Viper	1970
		Bothriechis schlegelii	Eyelash Viper	1986
		Crotalus atrox	Western Diamond-backed Rattlesnake	1984
		Crotalus cerastes laterorepens	Sidewinder	1970
		Lachesis muta muta	South American Bushmaster	2007
		Atropoides occiduus	Jumping Pitviper	1992
		Sistrurus miliarius barbouri	Dusky Pygmy Rattlesnake	1987
Crocodylia	Crocodylidae	*Crocodylus niloticus*	Nile Crocodile	1983
		Osteolaemus tetraspis tetraspis	West African Dwarf Crocodile	1980
	Alligatoridae	*Alligator mississippiensis*	American Alligator	unknown

TABLE 2. Breakdown of amphibian taxa successfully reproduced at the Jacksonville Zoo and gardens 1979–2017.

Order	Family	Species	Common name	Earliest known reproductive event
Caudata	Ambystomatidae	*Ambystoma mexicanum*	Axolotl	1997
	Proteidae	*Necturus beyeri*	Gulf Coast Waterdog	2016
	Salamandridae	*Neurergus kaiseri*	Kaiser's Newt	2010
		Notophthalmus perstriatus	Striped Newt	2008
		Notophthalmus viridescens dorsalis	Broken-striped Newt	1995
		Notophthalmus viridiscens piaropicola	Peninsular Newt	2001
Anura	Bufonidae	*Atelopus zeteki*	Panamanian Golden Frog	2008
		Peltophryne lemur	Puerto Rican Crested Toad	2011
	Dendrobatidae	*Dendrobates auratus*	Green and Black Poison Dart Frog	1989
		Dendrobates tinctorius	Dying Poison Dart Frog	2008
	Bombinatoridae	*Bombina orientalis*	Oriental Fire-bellied Toad	1982
	Hylidae	*Agalychnis callidryas*	Red-eyed Treefrog	2009
	Pipidae	*Xenopus laevis*	African Clawed Frog	1979
		Pipa parva	Sabana Suriname Toad	2016

TABLE 3. Selected longevity records for reptiles and amphibians kept by the Jacksonville Zoo and Gardens as of 1 November 2017. Longevity is represented in years. Note that many wild-caught reptiles and amphibians were acquired as adults; therefore, their true ages may be far greater than the values reported here which represent the total number of years lived in captivity. Abbreviations: CB = captive-bred; CH = captive-hatched; WC = wild-caught; UNK = unknown. Sexes are represented as Male.Female.Unknown. *Asterisked values represent specimens that are still living in the collection.

Class	Order	Species	Common name	Origin	Sex	Longevity
Reptilia	Squamata – Sauria	*Dracaena guianensis*	Caiman Lizard	WC	0.1	18.1
		Eublepharis macularius	Leopard Gecko	WC	1.0	24.3
		Heloderma horridum horridum	Mexican Beaded Lizard	CB	1.0	31.6*
		Tupinambis rufescens	Red Tegu	UNK	1.0	23.7
	Squamata – Serpentes	*Acrantophis dumerilii*	Dumeril's Boa	CB	1.0	35.1*
		Agkistrodon contortrix contortrix	Southern Copperhead	CB	0.1	17.6
		Atheris chlorechis	West African Bush Viper	WC	0.1	17.9
		Bothreichis schlegelii	Eyelash Viper	CB	0.1	19.8
		Crotalus adamanteus	Eastern Diamondback Rattlesnake	WC	1.0	20.8
		Dendroaspis viridis	West African Green Mamba	CB	0.2	19.9*
		Epicrates cenchria cenchria	Brazilian Rainbow Boa	CB	1.0	22.3
		Naja haje haje	Egyptian Cobra	WC	1.0	20.4*
		Pituophis melanoleucus mugitus	Florida Pinesnake	CB	1.0	22.2*
		Proatheris superciliaris	Lowland Swamp Viper	WC	1.0	15.8
		Pseudaspis cana	Mole Viper	WC	0.1	13.6
		Sistrurus miliarius barbouri	Dusky Pygmy Rattlesnake	WC	1.0	20.9
	Testudines	*Chelus fimbriatus*	Mata Mata	WC	1.0	35.5*
		Chelus fimbriatus	Mata Mata	WC	0.1	32.6*
		Dermatemys mawii	Central American River Turtle	WC	1.0	37.2*
		Dermatemys mawii	Central American River Turtle	WC	0.1	24.3*
		Dermatemys mawii	Central American River Turtle	CH	0.1	23.8*
		Terrapene coahuila	Coahuilan Box Turtle	CB	1.0	28.2*
Amphibia	Anura	*Rhaebo guttatus*	Smooth-sided Toad	WC	0.0.1	19.5
	Caudata	*Notophthalmus viridescens dorsalis*	Broken-striped Newt	WC	0.1	20
		Notophthalmus viridescens piaropicola	Peninsular Newt	WC	0.1	13.5
		Siren intermedia intermedia	Eastern Lesser Siren	WC	0.0.1	15.4
		Siren lacertina	Greater Siren	WC	1.0	13.6
	Gymnophiona	*Typhlonectes natans*	Rio Cauca Caecelian	WC	1.0	14.0*

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Fig. 7. An amelanistic gopher snake (*Pituophis* sp.) at JZG, early 1970s.

PHOTO COURTESY OF PETER SACHS

Fig. 8. An amelanistic *Crotalus atrox* displayed at JZG, early 1970s.

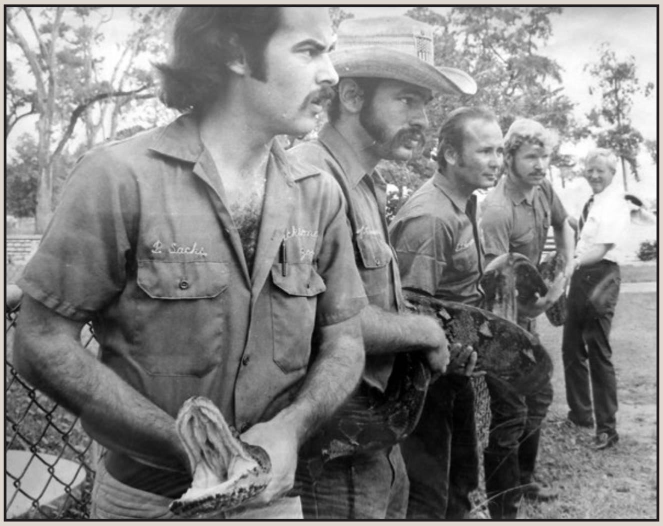

PHOTO COURTESY OF PETER SACHS

Fig. 9. Peter Sachs (left) and colleagues (from left to right: Jeffery Crocker, Leonardo Runyan, and Tim Krause) restrain a ~7-m long *Malayopython reticulaus* acquired in 1971.

pers. comm.). Florida's only native amphisbaenid, the Florida Worm Lizard (*Rhineura floridana*), has also been kept. *Varanus komodoensis* was added to the collection in 2007. Besides commonly bred species such as the Inland Bearded Dragon (*Pogona vitticeps*) and Leopard Gecko (*Eublepharus macularius*), lizard reproduction at JZG has been limited. Warren's Girdled Lizard (*Smaug warreni*) was successfully reproduced for the first time at the zoo in 2015, although the offspring did not survive past a few months. Taylor's Shield-tailed Agama (*Xenagama taylori*) was successfully reproduced in 2017.

About 165 species of snakes have been maintained by JZG, with venomous taxa making up an important component of the zoo's snake collection over the past several decades. From the mid-1990s to mid-2000s, there was a strong focus on African bush vipers of the genus *Atheris*, with five species kept: *A. ceratophorus, A. chloroechis, A. desaixi, A. squamiger*, and *A. superciliaris*. Some of the more obscure venomous taxa kept have included the Cape Coral Snake (*Aspidelaps lubricus*), Peringuey's Adder (*Bitis peringueyi*), Leaf-nosed Viper (*Eristicophis macmahoni*), and the Desert Black Snake (*Walterinnesia aegyptia*). As many as ten species belonging to the neotropical boid genera *Chilabothrus* and *Epicrates* were kept throughout the 1970s and 1980s; additional noteworthy snake taxa included the Indian Python (*Python molurus*), Bahamian Pygmy Boa (*Tropidophis canus*), and the Ratonel (*Pseudoboa neuwiedii*). Now a common fixture in the reptile hobby, JZG may have been the first zoo to display an amelanistic Red Rat Snake (*Pantherophis guttatus*) in 1971, acquired from noted breeder and originator of this color mutation, H. Bernard Bechtel (P. Sachs, pers. comm.). Around this time, the zoo displayed a few other amelanistic snakes including a gopher snake (*Pituophis* sp.) (Fig. 7) and Western Diamondback Rattlesnake (*Crotalus atrox*) (Fig. 8), as well as a 7-m-long Reticulated Python (*Malayopython reticulatus*) (Fig. 9). During the 1970s and 80s, the zoo successfully birthed at least six species of *Chilabothrus* (formerly assigned to *Epicrates*): *C. angulifer, C. chrysogaster, C. inornatus, C. striatus fosteri* (Fig. 10), *C. s. strigulatus, C. subflavus*, and *C. fordi*. Some of these may have represented world's first breeding events; however, at least one birth reported for *C. chrysogaster* (Anonymous 1975) appears to have been a case of a gravid female giving birth shortly after arriving at the zoo (see Huff 1978; Smith 2011). JZG may have been the first zoo to successfully reproduce the False Water Cobra (*Hydrodynastes gigas*) in 1972 (Fig. 11). The South American Bushmaster (*Lachesis muta*) was reproduced on several occasions in the late 2000s (Eisele 2009).

Fifty-nine chelonian species are known to have been kept, including such notable taxa as the Central American River Turtle (*Dermatemys mawii*), Flat-backed Spider Tortoise (*Pyxis planicauda*), North American Wood Turtle (*Glyptemys insculpta*), Blanding's Turtle (*Emydoidea blandingii*), and Aldabra Tortoise (*Aldabrachelys gigantea*). Throughout the 1980s, JZG hatched at least seven species of tortoise, which included the first captive breeding of *A. gigantea* in the western hemisphere (Collins 1984; Fig. 12). Additional noteworthy chelonians bred at the zoo include the Indian Star Tortoise (*Geochelone elegans*), Pancake Tortoise (*Malacochersus tournieri*), and Yellow-headed Temple Turtle (*Heosemys annandalii*).

At least 14 species of crocodilian have been kept, including the Sunda Gharial (*Tomistoma schlegelii*), Orinoco Crocodile (*Crocodylus intermedius*), Cuban Crocodile (*C. rhombifer*), West African Crocodile (*C. suchus*), Dwarf Crocodile (*Osteolaemus tetraspis*), and Slender-snouted Crocodile (*Mecistops cataphractus*). Despite the temperate climate of northern Florida (where nighttime temperatures in winter may drop as low as -9°C) and the zoo's lack of indoor holding space for colder winter months, two species of crocodile, the Nile Crocodile (*C. niloticus*) and *O. tetraspis* were successfully reproduced in the 1980s.

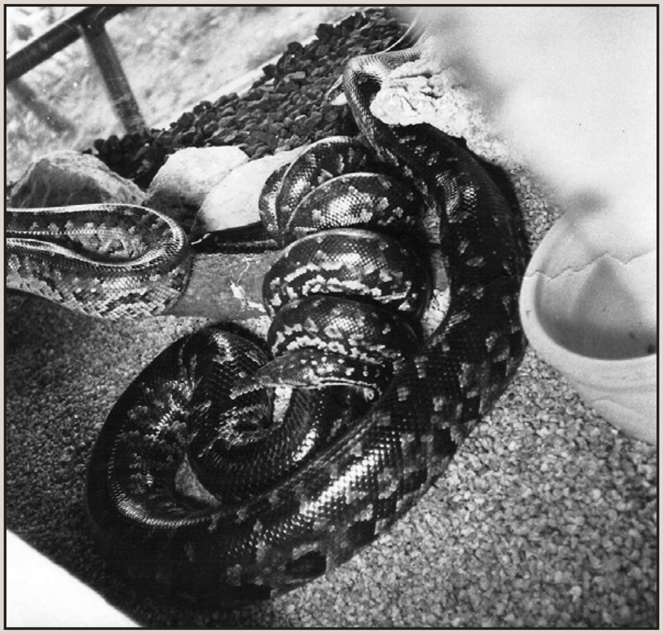

PHOTO COURTESY OF PETER SACHS

Fig. 10. Copulation in Bimini Island Boas (*Chilabothrus striatus fosteri*) at JZG, around 1973–1974.

PHOTO COURTESY OF PETER SACHS

Fig. 11. Copulation in *Hydronastes gigas* ca. 1973–1974.

PHOTOS COURTESY OF DAVID COLLINS

Fig. 12. Captive breeding of *Aldabrachelys gigantea* at JZG, 1980s.

Since 1946, at least 77 species of amphibians have been kept, including 55 anurans, 21 caudates, and one gymnophionan. Several poison dart frogs (*Dendrobates* sp.) were imported from Costa Rica and kept by the zoo in the early 1970s, but were never officially displayed for the public (P. Sachs, pers. comm.). Following the establishment of the zoo's Amphibian Conservation Center in 2008, several noteworthy reproductive events have taken place at JZG. Caudates have been a strong focus. Most notably, the Striped Newt (*Notophthalmus perstriatus*) has been produced since 2008 over multiple generations (see below); additional newt species bred include the Eastern Newt (*N. viridescens dorsalis* and *N. v. piaropicola*) and Kaiser's Newt (*Neurergus kaiseri*). Although captive breeding success with the Gulf Coast Waterdog (*Necturus beyeri*) was first achieved by the Cincinnati Zoo in 2013 (Stoops et al. 2014), JZG appears to have been the first facility to successfully reproduce this species in captivity without the use of assisted reproductive technologies in 2016, and then building upon this success again in 2017.

Several noteworthy importations of reptiles were spearheaded by the zoo. A group of *Pseudaspis cana*, the first to be displayed at JZG, was acquired from the Pretoria Zoo in South Africa in 1999. In 2011, a group of 17 Jamaican Boas (*Chilabothrus subflavus*) was imported from Europe to diversify and bolster the AZA-managed North American captive population. In 2010, the zoo assisted with processing a large confiscated group of *Heosemys annandalii* that had been seized in Hong Kong and imported by the Turtle Survival Alliance (TSA) (Hagen 2011). A few dozen adult individuals arrived at JZG, where they were processed and then shipped out to other facilities in the United States; six of these individuals were retained by JZG for breeding purposes. At least two *Alligator mississippiensis* were exported from JZG to the Riyadh Zoo in Saudi Arabia in 1988.

As of November 2017, the herpetology department's living collection was comprised of 58 species of reptiles and 20 species of amphibians. On account of the biogeographic scope of most of the zoo's reptile and amphibian buildings, the collection is heavily focused on African, Central and South American, and Floridian taxa, although some Asian species such as *Varanus komodoensis* and *Heosemys annandalii* are kept as well. The snake collection continues to have a strong emphasis on venomous taxa (15 species) including all native Floridian species (6 species), with a venomous to non-venomous taxon ratio of 1.07 (N = 31).The zoo currently maintains 12 species of reptiles and one amphibian whose captive populations are managed by AZA Species Survival Plans (SSP), representing 20.7% and 5.0% of each collection, respectively. Of the 15 SSP-managed species that have been kept by JZG over its history, six have successfully been

Table 4. Reptile and amphibian species found on grounds at the Jacksonville Zoo and Gardens. *Single asterisks denote species previously reported, but have not been recorded in 10+ years; **double asterisks denote a species involved in the zoo's long-term snake mark-recapture study. "General abundance" is a qualitative assessment using a scale of Abundant–>Common–>Uncommon–>Rare.

Class	Order	Family	Taxon	Common Name	Status	General abundance
Amphibia	Anura	Bufonidae	*Anaxyrus terrestris*	Southern Toad	Native	Abundant
		Eleutherodactylidae	*Eleutherodactylus planirostris*	Greenhouse Frog	Exotic	Abundant
		Hylidae	*Hyla cinerea*	Green Treefrog	Native	Abundant
			Hyla squirella	Squirrel Treefrog	Native	Abundant
			Osteopilus septentrionalis	Cuban Treefrog	Exotic	Abundant
		Microhylidae	*Gastrophryne carolinensis*	Narrow-mouth Toad	Native	Uncommon
		Ranidae	*Rana catesbeiana*	American Bullfrog	Native	Abundant
			Rana sphenocephala	Southern Leopard Frog	Native	Common
		Scaphiopodidae	*Scaphiopus holbrookii*	Eastern Spadefoot Toad	Native	Uncommon
	Caudata	Sirenidae	*Siren lacertina*	Greater Siren	Native	Unknown
		Amphiumidae	*Amphiuma means*	Two-toed Amphiuma	Native	Unknown
Reptilia	Crocodylia	Alligatoridae	*Alligator mississippiensis*	American Alligator	Native	Common
	Squamata – Sauria	Dactyloidae	*Anolis carolinensis*	Green Anole	Native	Common
			Norops sagrei	Brown Anole	Exotic	Abundant
		Scincidae	*Plestiodon fasciatus*	Five-lined Skink	Native	Common
			Plestiodon laticeps	Broad-headed Skink	Native	Common
			Scincella lateralis	Ground Skink	Native	Common
		Anguidae	*Ophisaurus ventralis*	Eastern Glass Lizard	Native	Uncommon
		Gekkonidae	*Hemidactylus turcicus*	Mediterranean Gecko	Exotic	Abundant
	Squamata – Serpentes	Colubridae	*Coluber constrictor***	Black Racer	Native	Abundant
			*Farancia abacura***	Mud Snake	Native	Rare
			*Opheodrys aestivus***	Rough Green snake	Native	Common
			*Nerodia fasciata***	Banded Water snake	Native	Abundant
			*Pantherophis alleghaniensis***	Yellow Ratsnake	Native	Abundant
			*Pantherophis guttatus***	Red Ratsnake	Native	Common
			*Regina alleni**	Striped Crayfish Snake	Native	Unknown
			Rhadinaea flavilata	Pinewoods Snake	Native	Uncommon
			*Thamnophis sirtalis***	Eastern Garter Snake	Native	Abundant
			*Thamnophis sauritus***	Eastern Ribbon Snake	Native	Common
		Elapidae	*Micrurus fulvius**	Eastern Coral Snake	Native	Unknown
		Typhlopidae	*Ramphotyphlops braminus*	Brahminy Blind Snake	Exotic	Rare
		Viperidae	*Crotalus adamanteus***	Eastern Diamond-backed Rattlesnake	Native	Uncommon
			*Sistrurus miliarius**	Pygmy Rattlesnake	Native	Unknown
	Testudines	Chelydridae	*Cheyldra serpentina osceola*	Florida Snapping Turtle	Native	Abundant
		Emydidae	*Malaclemys terrapin*	Diamondback Terrapin	Native	Rare
			Pseudemys rubriventris	Red-bellied Cooter	Native	Common
			Terrapene carolina bauri	Florida Box Turtle	Native	Common
			Trachemys scripta elegans	Red-eared Slider	Exotic	Common
			Trachemys scripta scripta	Yellow-bellied Slider	Native	Common
		Kinosternidae	*Kinosternon baurii*	Striped Mud Turtle	Native	Common
			Sternotherus odoratus	Common Musk Turtle	Native	Abundant
		Testudinae	*Gopherus polyphemus*	Gopher Tortoise	Native	Common
		Trionychidae	*Apalone ferox*	Florida Softshell Turtle	Native	Abundant

reproduced: the Puerto Rican Crested Toad (*Peltophryne lemur*), Panamanian Golden Frog (*Atelopus zeteki*), Louisiana Pine Snake (*Pituophis ruthveni*), *Lachesis muta*, *Heosemys annandalii*, and *Malacochersus tournieri*. Several clutches of eggs have also been received from *Dermatemy*s *mawii* over successive years, but have not yet proven to be viable.

In addition to the herpetology department's collection, 11 reptile and six amphibian species are presently maintained by the zoo's education department for educational outreach.

Wild Herpetofauna on Zoo Grounds.—Many zoological parks in the United States and Europe are located within urban centers and subsequently lack an abundance of free-ranging wildlife on their grounds, particularly reptiles and amphibians. In contrast, given its geographical location along the Trout River and the extensive wetlands, wooded areas, and planted gardens that occur throughout its grounds, JZG is presently home to at least 28 species of reptiles (four of which are non-native) including one venomous species, the Eastern Diamond-backed Rattlesnake (*Crotalus adamanteus*), and 11 species of amphibians (two of which are non-native) (Table 4; Fig. 13). Three additional snake species, the Eastern Coral Snake (*Micrurus fulvius*), Pygmy Rattlesnake (*Sistrurus miliarius*), and Striped Crayfish Snake

PHOTO COURTESY OF STEVE GOTT

FIG. 13. Herpetology keeper Sean Wallace (left) and senior herpetology keeper Emily Fyfe (right) safely restrain a large *Crotalus adamanteus* captured on zoo grounds in 2013.

PHOTO BY ROBERT W. MENDYK

FIG. 14. A wild *Pantherophis alleghaniensis* retrieved after ingesting several plaster-filled "dummy eggs" in one of the zoo's aviaries. Surgical excision of the eggs was required after attempts to manually palpate and eject the eggs were unsuccessful.

(*Regina alleni*) have previously been recorded on grounds (D. Collins and G. Lepera, pers. comm.), but have not been observed in over a decade. Snakes that are opportunistically captured on zoo grounds by herpetology staff and deemed large enough for PIT tag implantation are monitored as part of a long-term mark-recapture study on their ecology (see below).

Some species of wild herpetofauna present noteworthy challenges in terms of safeguarding JZG collection animals. Wild alligators are a persistent threat to collection animals, particularly smaller wading birds and waterfowl maintained in the zoo's open, outdoor exhibits. Hatchling and juvenile alligators periodically enter wetland areas on zoo grounds from the adjacent Trout River or through drainage culverts when small, and then quickly grow to formidable sizes feeding on an abundant supply of wild amphibians, insects and fishes. Nuisance alligators retrieved from exhibit areas are usually relocated to the Trout River.

Florida Snapping Turtles (*Chelydra serpentina osceola*) are also abundant on zoo grounds, and on several occasions have bitten and seriously injured the feet of wading birds and waterfowl in the collection. On one occasion, a Southern Pudu (*Pudu puda*) fawn was grabbed and pulled underwater by a snapping turtle, but was able to escape without injury. Problematic individuals are relocated to other wetland sites on the property or the Trout River. Interestingly, two wild snapping turtles living in a natural wetland area inside a mixed bird and hoofstock exhibit had learned to leave the water to opportunistically feed on pelleted crane food from food bowls up on land.

Ratsnakes of the genus *Pantherophis* are notorious raiders of bird nests and chicken coups (e.g., Medsger 1920) and are common throughout JZG. Yellow Ratsnakes (*Pantherophis alleghaniensis*) and Red Ratsnakes (*P. guttatus*) are frequently encountered attempting to enter the zoo's aviaries to feed on collection birds' eggs and their young. On several occasions, these snakes have inadvertently consumed "dummy" or "nest eggs"—sand or plaster-filled eggshells placed in nests as substitutes for the actual eggs. Unable to digest or pass these replica eggs (Fig. 14), surgical intervention by the zoo's veterinary staff has been required for multiple individuals after manual palpation and ejection of the eggs proved unsuccessful. Once captured and removed from aviaries, these snakes are PIT-tagged and released away from the aviary in another part of the zoo. Some of these snakes end up returning to their original capture sites, and "repeat offenders" that continue to target aviaries are relocated off-grounds or accessioned into the zoo's living collection.

The introduced Cuban Treefrog (*Osteopilus septentrionalis*) has been established in southern Florida since the 1920s (Barbour 1931), but has only reached northeastern Florida in recent years. It is substantially larger than all of Florida's native treefrog species, and is known to occupy the same refuge sites and predate indigenous species such as the Green Treefrog (*Hyla cinerea*) (Wyatt and Forys 2004; Glorioso et al. 2012). Although not necessarily a threat to JZG's collection animals, its presence does jeopardize indigenous treefrog populations on grounds. After several years of casual sightings on zoo grounds, which most likely represented individual specimens transported up from southern Florida with tropical plant shipments, the number of *O. septentrionalis* opportunistically collected in 2016 exploded to more than 100 individuals, with breeding adults, tadpoles, and juveniles now appearing throughout the zoo. Efforts to remove opportunistically collected individuals over the past several years have failed to prevent the establishment of this species on zoo grounds.

The Greenhouse Frog (*Eleutherodactylus planirostris*) is another introduced anuran that is now established throughout the state of Florida. On zoo grounds, they are frequently encountered beneath rocks, logs, and other objects, and occasionally enter animal buildings and exhibits. Several individuals, which were probably inadvertently transported with live plants or mulch, were found living inside the zoo's bushmaster and poison dart frog exhibit. In addition to successfully reproducing in the exhibit undetected (this species deposits its eggs terrestrially and has direct development with no tadpole stage), male *E. planirostris* have been observed attempting to mate with Green and Black (*Dendrobates auratus*) and Golfodulcean (*Phyllobates vittatus*) Poison Dart Frogs in the exhibit (B. Eisele and M. Beshel, pers. comm.).

CONSERVATION

Ex-situ Conservation Breeding and Repatriation.—JZG is currently engaged in three ex-situ conservation breeding and repatriation programs where captive-bred offspring produced at the zoo are released into the wild in an effort to help restore extirpated populations or bolster declining wild populations. In 2008, the zoo joined the *Peltophryne lemur* SSP and recovery program, a conservation initiative led by the United States Fish and Wildlife Service in partnership with Puerto Rico's Department of Natural and Environmental Resources and several AZA-accredited zoos. The program carefully oversees the captive management and breeding of *P. lemur* in zoos and the subsequent release of captive-bred tadpoles into selected wetland sites in Puerto Rico (see Johnson 1990). As of November 2017, more than 8000 *P. lemur* tadpoles have been produced by JZG at its Amphibian Conservation Center and delivered to Puerto Rico for release.

In response to rapidly declining wild populations of *Notophthalmus perstriatus* throughout its range in Florida and Georgia, a multifaceted recovery project was initiated in 2010 by the Coastal Plains Institute, in collaboration with the Florida Fish and Wildlife Conservation Commission and the United States Forestry Service, to protect, conserve, and study the decline of the species. Recovery work for this species focuses on the Apalachicola National Forest (ANF) in the Florida panhandle, the species' last reported stronghold in the state (Means et al. 2013). In 2011, JZG joined the project to help establish ex-situ assurance colonies of *N. perstriatus* together with the Memphis Zoo, and to produce captive-bred offspring for repatriation into recipient wetlands within the ANF (Means et al. 2012). As of November 2017, JZG has released more than 1000 *N. perstriatus* into the wild, and together with the Memphis Zoo and most recently the Central Florida Zoo's Orianne Center for Indigo Conservation, more than 1300 individuals have been repatriated since the project's inception (Means et al. 2016; Fig. 15). Offspring produced at JZG have been sent to other AZA-accredited facilities in Florida to establish additional ex situ breeding colonies. Conservation support grants received from the American Association of Zoo Keepers (2015) and the Foundation for the Conservation of Salamanders (2017) have enabled JZG to expand on this project by increasing the zoo's holding capacity for rearing larval newts.

PHOTO BY ROBERT W. MENDYK

FIG. 15. Captive-bred *Notophthalmus perstriatus* produced at JZG in preparation for release in the Apalachicola National Forest, Florida.

Since 2011, JZG has participated in conservation and recovery efforts for *Pituophis ruthveni*, an imperiled colubrid native to western Louisiana and eastern Texas (Rudolph et al. 2006). Captive-bred offspring produced by zoos participating in the *P. ruthveni* SSP, including JZG, are released into the wild in an attempt to bolster current populations (e.g., Himes and Hardy 2006). Although some setbacks were experienced prior to 2014, JZG produced its first three captive-bred *P. ruthveni* offspring in 2015, followed by additional offspring in 2016 and 2017.

Although currently listed as a species of least concern by the IUCN, JZG is working to reproduce *Necturus beyeri*, an aquatic salamander that naturally occurs from western Florida to eastern Texas. By maintaining *N. beyeri* as an analog species, JZG seeks to develop husbandry and breeding techniques for this species that can then be applied to the closely related and endangered Alabama Waterdog (*N. alabamensis*) for future conservation breeding and repatriation efforts. Reproductive success with *N. beyeri* was first achieved at JZG in 2016 (three live offspring) and repeated again in 2017 (35 offspring), and we hope that these initial successes can be refined to produce consistent and repeatable techniques that can soon be applied to *N. alabamensis.*

Capacity Building.—In addition to directly participating in several herpetological conservation and recovery projects, JZG is also committed to supporting a wide range of conservation projects and initiatives led by other organizations and institutions throughout the globe. Through its Conservation Awards, Research and Events (CARE) committee, JZG has provided financial and logistical support to numerous projects worldwide, with more than US $73,000 in funding support allocated specifically to herpetology-related projects since 2013. This has also included providing support to graduate students studying local herpetological conservation issues in Florida and the southeastern United States, and equipment support for radiotelemetry field studies on *Chilabothrus subflavus.*

In 2015, JZG, in collaboration with the TSA and the Belize Foundation for Research and Environmental Education (BFREE), sponsored the 2nd Hicatee Conservation Workshop and Forum in southern Belize, which brought together a consortium

PHOTO COURTESY OF STEVE GOTT

FIG. 16. JZG herpetology staff (senior keepers Brian Eisele and Emily Fyfe) measure a wild *Coluber constrictor priapus* with zoo visitors as part of the department's long-term mark-recapture snake study, in 2011.

of stakeholders involved in the conservation of *Dermatemys mawii* across its range to discuss current conservation efforts and future actions for the species (Barrett et al. 2016). Additional funding support from JZG has been earmarked for supporting the Hicatee Conservation and Research Center (HCRC) at BFREE as well as the design and construction of a new educational exhibit in Belize which will highlight *D. mawii* and its conservation challenges. JZG herpetology and hospital staff members routinely travel to Belize to assist with reproductive health assessments of the HCRC's captive assurance population.

RESEARCH

In recent years, JZG's herpetology department has initiated several in-situ, ex-situ, and database-driven research projects focusing on various subjects including ecology, physiology, behavior, captive management, and life expectancy. Ethological investigations are currently focused on the behavioral repertoire and courtship of the poorly-known *Dermatemys mawii* (e.g., Mendyk and Eisele 2016), with hopes that this research can help inform husbandry and reproductive management practices for this critically endangered species. Herpetology staff members are also studying the life expectancy and longevity of various reptile groups in captivity through analysis of life history data obtained from published studbooks and animal record keeping databases such as the Zoological Information Management System (ZIMS) (e.g., Mendyk 2015), with a study on the life expectancies and longevities of rattlesnakes (*Crotalus* and *Sistrurus*) in captivity currently underway. Building upon earlier studies on thermal husbandry in captive reptiles (Mendyk et al. 2014, 2016), JZG seeks to study the thermoregulatory behaviors and thermal preferences of species within its collection, which can have strong implications for improving captive management practices in zoos.

In addition to husbandry-based studies, JZG is engaged in field research on reptile and amphibian ecology. Similar to studies carried out by other zoos with wild reptile and amphibian populations on grounds (e.g., Roberts and Mitchell 1998; Hartdegen 2004; D. Smith, pers. comm.; C. Baker, pers. comm.), JZG has been conducting a long-term mark-recapture study of free-ranging snakes since 2008. Over the course of this study, herpetology staff have collected important ecological data that will shed light on the diversity and relative abundances, sex ratios, seasonal activity patterns, habitat associations, home range sizes, growth rates, injuries and other health issues of the native snake species found on zoo grounds. Specimens opportunistically encountered are captured, measured and weighed, probed for sex determination, and PIT-tagged before being released. With the exception of venomous snakes (*Crotalus adamanteus*) which are relocated to undeveloped areas of the zoo due to public safety concerns, non-venomous species are usually released at or near the site of their capture. To maximize this project's educational impact, herpetology staff will usually process captured snakes out in public areas of the zoo to encourage questions from visitors and provide them with opportunities to see local snake species up close and in person (Fig. 16). To date, more than 400 individual snakes of nine species (Table 4) have been PIT-tagged and continue to be monitored on zoo grounds.

CITIZEN SCIENCE AND EDUCATIONAL OUTREACH

In 2015, JZG joined FrogWatch USA, a citizen science-based initiative led by the AZA to collect important field data and monitor changes in anuran populations and communities throughout North America (Inkley 2006). Through its Northeast Florida FrogWatch Chapter, JZG hosts annual training sessions and workshops for local citizen scientists interested in participating in the program and leads occasional nighttime field outings with FrogWatch participants and zoo volunteers.

Like most zoos, JZG features a separate education department that focuses exclusively on educational outreach; however, visitor engagement and education are also significant activities of the zoo's herpetology department. Each year, herpetology staff engages thousands of visitors on herpetological matters through a combination of scheduled keeper chats, public feedings and other demonstrations (e.g., processing wild snakes in the mark-recapture study), and behind the scenes tours, with the goal of educating and fostering appreciation and respect for reptiles and amphibians. Further educational outreach is carried out off-grounds through public lectures and presentations given by JZG herpetology staff and through participation at various conservation events (e.g., Claxton Rattlesnake Festival; see Morell 2017).

STAFF PUBLICATIONS

Since the 1970s, JZG staff has contributed numerous publications on various aspects of herpetology and captive management. Many of these works have focused on the husbandry and reproduction of species at the zoo. For instance, Collins (1984) described the husbandry and first successful breeding of *Aldabrachelys gigantea* in the Western Hemisphere. Williams (1986a,b), Meyer (1987), and Meyer et al. (1989) described in detail the husbandry and breeding of several tortoise species in the collection. Lepera (2004b) outlined some of JZG's safety protocols for working with venomous taxa which are still in use today. Eisele (2009) documented the zoo's repeated reproductive success with *Lachesis muta*. Beshel (2014a,b) discussed current ex-situ amphibian conservation initiatives at the zoo, whereas Means et al. (2014, 2015, 2016), and Mendyk and Beshel (2017) described husbandry and breeding parameters for *Notophthalmus perstriatus* at JZG. Scent enrichment for several reptiles in the collection was described by Burr (1997), and Mikus

(2014) outlined an operant conditioning program for behaviorally managing a group of adult *Alligator mississippiensis*. As part of a training manual on crocodilian captive management, Carter and Lepera (2001) discussed techniques used for identifying individual animals, Pastika and Lepera (2001) provided an overview of transportation methods, and Rost (2001a) discussed record keeping. Ferri (2012a) prepared and published the AZA North American regional studbook for *Chilabothrus subflavus*.

Additional works have focused on broader management topics, such as identifying problematic areas of husbandry in zoos and offering potential solutions for their remedy. Mendyk et al. (2014) described the thermal husbandry of monitor lizards; as a follow-up to this study, Mendyk et al. (2016b) quantified and characterized differences in thermal husbandry practices between zoos and private herpetoculturists, and highlighted the importance of communication between these two groups. Mendyk (2015a) assessed the life expectancy and longevity of monitor lizards in North American zoos and discussed factors that may be affecting long-term keeping and breeding success. Annotated bibliographies on the captive management and reproduction of monitor lizards were provided by Mendyk (2015c, 2016c, 2017). Mendyk (2018) discussed folklore reptile husbandry in zoos, highlighting outdated or inappropriate keeping practices that may be affecting the success of captive management programs.

Several published works have focused on historical aspects of zoo herpetology. For example, Mendyk (2015d) reported on the history of a troubled reptile zoo in New York, whereas Mendyk et al. (2015) described the history, husbandry, and behavior of a Bornean Earless Monitor (*Lanthanotus borneensis*) maintained by the Bronx Zoo during the 1960s and 70s. Mendyk and Smith (2016) briefly discussed the history of *Dermatemys mawii* in zoos, and presented new captive longevity records for the species.

Some publications have focused on the natural history, ecology, and behavior of reptiles. Groves and Sachs (1973) provided descriptions of the eggs and offspring of the Scarlet Kingsnake (*Lampropeltis elapsoides*) in Florida. Wilson and Meyer's (1985) book, *The Snakes of Honduras*, provided a comprehensive overview of the natural history and distribution of Honduran snakes. Mendyk and Adragna (2014) reported on the discovery, status, and distribution of two new introduced populations of the Italian Wall Lizard (*Podarcis siculus*) in New York. Mendyk et al. (2016a) studied the reproductive biology of invasive female Northern African Rock Pythons (*Python sebae*) collected from southern Florida. Mendyk and Eisele (2016) described caudal prehensility in *Dermatemys mawii*. Ferri (2012b) briefly discussed collecting water samples for amphibian eDNA testing. Taylor and Mendyk (2017) described a case of kyphosis in a Florida Softshell Turtle *Apalone ferox*, and Wood et al. (2017) studied foraging behavior in Hawksbill Sea Turtles (*Eretmochelys imbricata*) in southern Florida. Mendyk et al. (2018) described the resiliency of a wild clutch of Southern Black Racer (*Coluber constrictor priapus*) eggs that hatched after experiencing extensive flooding and total submersion after a direct hurricane strike.

Jacksonville Zoo and Gardens staff have also contributed various works on the veterinary management and surgery of reptiles in captivity. Page (1985) described soft tissue celiotomy in an *Aldabrachelys gigantea*, and reviewed anesthesia techniques in reptiles (Page 1993). Page et al. (1988, 1991) described pharmacokinetics in *Gopherus polyphemus*, and Mautino and Page (1993) discussed various aspects of the biology and medical management of captive chelonians. Page et al. (1986) described the medical management of a debilitated Leopard Tortoise (*Stigmochelys pardalis*). Mendyk (2015b) described the use of predatory soil mites for treating parasitic mite infestations in lizards and discussed the potential role of biological control agents as an alternative to chemical therapeutics.

Finally, a number of brief, miscellaneous herpetological notes were published in the Jacksonville Zoo and Gardens' newsletter *Kitabu* (Anonymous 1979, 1997b; Williams 1986b; Meyer 1987; Moore 1988, 1989, 1991, 1992, 1999; Lepera 1997, 2001, 2004a,b,c; Rost 1997, 2001b; Tomlinson 1999; Pastika 2000; Kapustin 2004) and *Wild* (Mendyk 2015e, 2016a,b), which it later changed its name to in 2005.

ADDITIONAL ACTIVITIES AND MISCELLANY

Several staff members of JZG's herpetology department have served in various capacities for national and international conservation organizations and committees and have served as instructors for several courses and professional workshops. Dino Ferri served as chair of the AZA Snake Taxon Advisory Group, vice chair of the AZA Wildlife Conservation Management Committee, and as a steering committee member of the AZA Chelonian Taxon Advisory Group. Former herpetology supervisor Steve Gott served as a trustee on the board of the World Chelonian Trust. Former herpetology supervisor Robert Mendyk was a founding member of the IUCN Species Survival Commission's (SSC) Monitor Lizard Specialist Group, and served on steering committees of the AZA Lizard Taxon Advisory and Komodo Dragon Taxon Advisory Groups. Greg Lepera served as an instructor for the AZA Crocodilian Biology and Captive Management course over several years and senior herpetology keeper Mark Beshel has been an instructor for the Amphibian Ark's Biology, Management and Conservation of North American Salamanders Training Course since its inception in 2016.

While at JZG, Robert Mendyk served as herpetoculture section editor for the journal *Herpetological Review* (published by the Society for the Study of Amphibians and Reptiles) and editor-in-chief of *Biawak*: *Journal of Varanid Biology and Husbandry* (published by the International Varanid Interest Group).

In recognition of their contributions to the field of herpetology, Greg Lepera and Robert Mendyk were presented with the International Herpetological Symposium's Joseph Laszlo Memorial Award in 2000 and 2017, respectively.

OUTLOOK

The herpetological history outlined here documents one aspect of the Jacksonville Zoo and Gardens' maturation into a modern zoological park—a facility that serves both the public interest and the scientific community as an important center for conservation, education, and research. Reptiles and amphibians have played a crucial role in shaping the zoo's growth and transformation over the past several decades, and will likely continue to be a driving force behind its continued evolution. Much is planned for the future of the herpetology department in terms of its collection and conservation and research activities.

Working within the biogeographic framework of its herpetology collection, JZG seeks to increase its role and participation in AZA-managed SSP programs. Additionally, since maintaining sustainable captive populations continues to be a major challenge for zoo herpetology departments (e.g., Ziegler et al. 2016), JZG also plans to focus on reproducing rare and threatened species that are not managed by SSPs but are still in much need of captive breeding (e.g., Ziegler et al. 2016, 2017). Along similar lines of sustainability, it will also be important for the zoo to focus on breeding common species that are of educational or display value, particularly native taxa. Currently in Florida zoos, many indigenous reptile and amphibian species are rarely reproduced because of their perceived commonness and widespread availability as wild-caught specimens. As stewards of wildlife conservation, zoos should be distancing themselves from the practice of field-collecting (or purchasing wild-caught) reptiles and amphibians if more responsible options are available. To achieve this, a concerted effort will be needed among Florida zoos to produce sustainable captive-bred populations of important native species. JZG has recognized this need as it seeks to establish captive-bred lineages of several indigenous species, and welcomes collaboration, participation, and feedback from other institutions on these efforts.

The Jacksonville Zoo and Gardens' ideal location in northeastern Florida affords many opportunities for the zoo to support and contribute to local reptile and amphibian conservation efforts and field research. Moreover, with several nearby universities and AZA-accredited zoological facilities, there are many opportunities to forge collaborative partnerships with these institutions as well as local, state and federal wildlife agencies. Expanding upon its current conservation projects, JZG seeks to add additional species to its ex situ captive breeding and repatriation efforts. The zoo also plans to continue providing financial and logistical support to external conservation and research projects, with a particular emphasis on biodiversity hotspots such Guyana, Democratic Republic of Congo, Indonesia, and Florida.

As highlighted by several authors discussing the importance of zoo-academic herpetological collaborations, zoos have unique opportunities to work together with outside researchers on a variety of research topics (Murphy and Chiszar 1989; Chiszar et al. 1993; Pough 1993; Chiszar and Smith 2005) yet may not be reaching their potential in this regard (Pough 1993; Card et al. 1998). To maximize its scientific contributions to the fields of herpetology and herpetological husbandry, JZG is taking several steps to increase its research activities. In addition to increasing the size of its reptile and amphibian collection by including more individuals of species that would facilitate larger sample sizes for biological investigations, JZG is soliciting partnerships with academic institutions and promoting the zoo as a living research laboratory and source for biological materials to outside researchers. In addition to the archetypal studies conducted on reptile and amphibian behavior, physiology, and veterinary management in zoos (Chiszar et al. 1993; Chiszar and Smith 2005), a new animal wellness initiative at JZG (see Maple 2016) offers opportunities to study the behavior of captive reptiles and amphibians and evaluate the appropriateness and success of captive husbandry regimes.

Finally, the various conservation, research, and publishing activities of JZG's herpetology staff, coupled with the many contributions made by other zoos over the past century (e.g., Murphy 2007), exemplify the value and importance of herpetology departments in zoos. Today, as zoos face increasing scrutiny from animal rights groups and other critics, it is imperative now more than ever, that zoos demonstrate their value and importance to society through their educational and scientific endeavors. The vast body of herpetological research and conservation programs carried out by zoos offers many opportunities for zoos to boast and publicize these activities and contributions. Retrospective historical accounts such as the present article can also be valuable for illustrating the significance of zoos. Additionally, in an era where some zoos are moving away from taxonomic specialization among their staffing by restructuring traditional taxonomically defined departments (e.g., herpetology, ornithology, mammalogy) to those defined by biogeography (e.g., Asia, Neotropics, Australia, etc.) or zones within the zoo, this is likely to have a negative impact on the scientific output and contributions of zoological parks, particularly those herpetological in nature. It is doubtful that JZG could have achieved all that it has over the past 40 years in terms of herpetological husbandry and breeding, conservation, and research if it did not have specialized herpetology staff. Therefore, it is our hope that in addition to contributing a further chapter to the annals of zoo history, this article illustrates the importance and necessity of maintaining taxonomic specialization in zoological parks, particularly formal herpetology departments.

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Why Are There Not More Agamid Lizards in Zoo Collections?

> Of the two groups of the Squamata, the lizards are of course the older. They have the conventional body plan of a typical land vertebrate: four legs, five toes to a foot, and the sprawling gait of the earliest reptiles. Most of the adaptations that have allowed them to spread and prosper are relatively unspectacular changes in the old four-legged look – exceptions being the various groups in which the legs have been lost completely. As vertebrates, lizards are a fairly representative group and it has been suggested that the lizards would be more suitable as a type with which to introduce freshman biology students to vertebrate anatomy than the universally used frog. Perhaps it sounds cynical to say so, but I think the answer there is that the frog being tailless, fits dissection pans more gracefully.
>
> —Archie Carr, *Life Nature Library: The Reptiles,* 1963

Some of the most beautiful and specialized lizards on the planet comprise the family Agamidae (see Scott Moody's phylogenetic and historical biogeographical relationships of the family, 1980). A sizeable number are strikingly ornate, with arresting colors and patterns. Many have an array of ornaments—crests, spines, frills, fins, dewlaps, nasal appendages, spinose "beards" and other accoutrements. A few display remarkable anti-predator behaviors that deploy these unusual structures to intimidate enemies. Agamids are found from the western Pacific through Australia and Asia to Africa and Europe. The family is large, comprising 57 genera and 483 species (Uetz et al. 2017). As a group, they are generally diurnal; most use well-developed hind limbs with five clawed digits to escape predators. They feed on a variety of insects, reptiles, nestling birds, small mammals, and plant materials.

Agamids are mostly terrestrial and have keen vision. Some use remarkably effective camouflage to avoid detection. They wave arms, do pushups, bob heads, extend dewlaps and crests, bite, compress or inflate bodies, vocalize, run bipedally, glide, lash tails, and engage in other fascinating behaviors, some of which are directed toward conspecifics.

The female Rainbow Lizard (*Agama agama*) lays eggs in a hole dug with snout and claws. The hole is constructed in sandy, wet, damp soil that is exposed to sunlight most of the day and covered by vegetation. Eggs are usually laid in clutches ranging from five to seven ellipsoidal eggs. This taxon is a thermoregulated embryo species resulting in all males at 29°C and all females at 26°C (Crews et al. 1983). The eggs hatch within eight to ten weeks. A complete treatment of life and anatomy of this lizard has been published by Vernon Harris (1963, 1964).

What other group of reptiles has such colorful standard names? Jacky Lizard, Frilled Lizard, Bicycle Dragon, Thorny Devil, Sailfin Lizard, Lashtail, Forest Dragon, Great Anglehead Lizard, Lyrehead Lizard, Secret Toadhead Agama, Lionhead Agama, Crested Flying Dragon, Eyebrow Lizard, Picklenape, Bloodsucker, and a host of others equally intriguing.

Hence, it is surprising that so few living agamids are displayed in zoo collections, especially since they do not hide constantly like many snakes, and many species adapt well to captivity. Many of the above behaviors would be visible to the zoo visitor. Here is an account of a complex array of unique behaviors observed by one of us (JBM) at the Dallas Zoo: "An elaborate combination of specialized motor patterns were observed in the agamid lizard, *Goniocephalus dilophus,* when confronted by an intruder: 1) when most threatened, bipedal bounding or hopping flight with forelimbs rotated in a circumductory plane [movement of a limb in a circular direction], mouth closed; 2) when less threatened, lateral presentation with open mouth, tongue rolled forward, gular sac expanded, side hopping [sideways hopping movement in which this lizard hops first to one side and then the other]; and 3) when least threatened, frontal orientation, short rushes toward the intruder with mouth open, biting attempted. Expansion of the hyoid apparatus allowed for greater visibility of the yellowish dentary region. The soft gum tissue and tissue exterior to the masseter muscle deepened to a bright reddish hue, due probably to vascularization. A guttural hissing sound produced by lung evacuation through the glottis accompanied the display" (Murphy et al. 1978, Fig. 12). With relatively little effort, interesting graphics and displays could be designed to show off these remarkable creatures. To whet a zoo worker's appetite, some of the most spectacular ones are depicted here.

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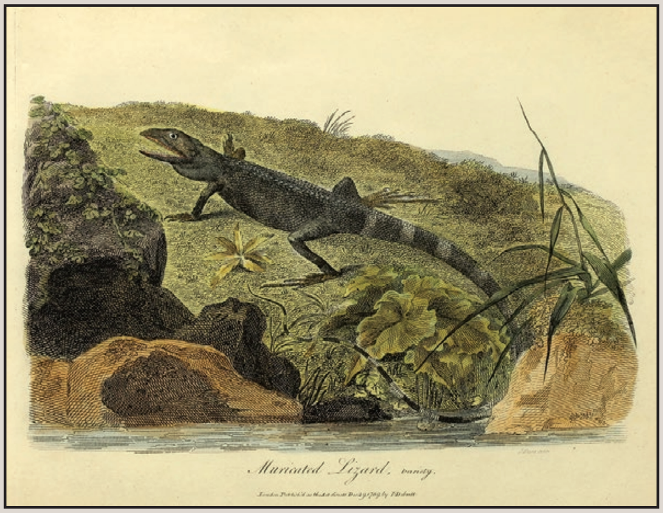


Fig. 1. Muricated Lizard, sometimes called Jacky Lizard (*Amphibolurus muricatus,* described as *Lacerta muricata*). Reproduced from *Journal of a voyage to new South Wales, with sixty-five plates of non descript animals, birds, lizards, serpents, curious cones of trees and other natural productions* by John White, 1790.

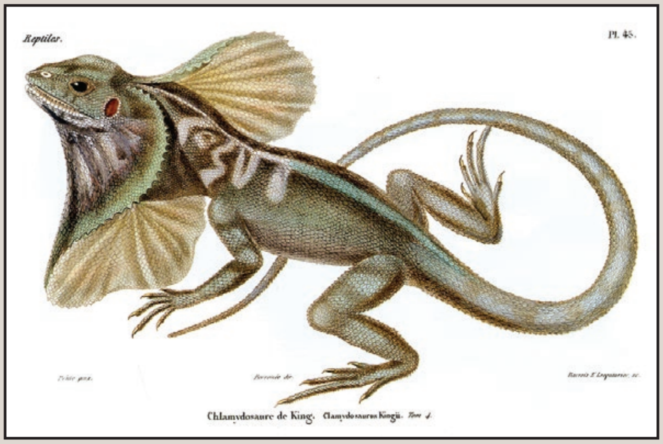


Fig. 2. Frilled Lizard (*Chlamydosaurus kingii*). Reproduced from *Erpétologie Générale ou Histoire Naturelle Complète des Reptiles* by A. M. C. Duméril, G. Bibron and A. H. A. Duméril, 1839. This species is found in Australia and New Guinea. A specimen at the Dallas Zoo flared its frill, hopped bipedally and vocalized with grunting sounds. The French naturalists, Constant Duméril, Gabriel Bibron, and the senior author's son, Auguste Duméril, produced this classic herpetological work, based on the collections of the Muséum d'Histoire Naturelle in Paris (9 volumes in 10 plus an atlas of colored plates, 1834–1854). All of the saurian plates were illustrated by artist Jean Gabriel Prêtre (1780-1845), produced between 1836 and 1839. These books have been reprinted by SSAR of nearly 7,000 printed pages and 250 plates in six volumes with an introduction by Roger Bour, including extensive new biographies of all three authors.

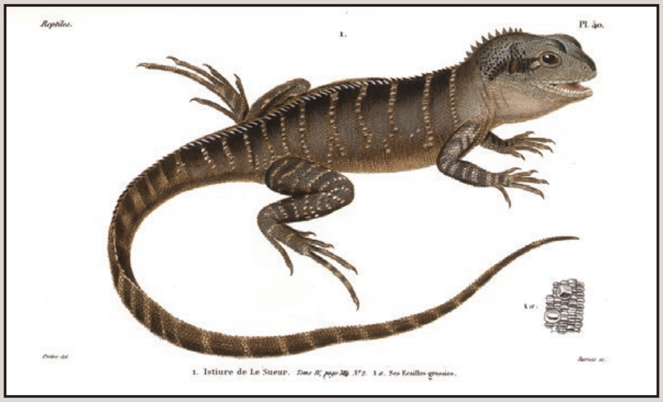


Fig. 3. Eastern Water Dragon (*Physignathus lesueurii*). Reproduced from *Erpétologie Générale ou Histoire Naturelle Complète des Reptiles* by A. M. C. Duméril, G. Bibron, and A. H. A. Duméril, 1839. This species is found in Australia. A large captive population lives in a spacious semi-aquatic display at the U.S. National Aquarium in Baltimore, Maryland.

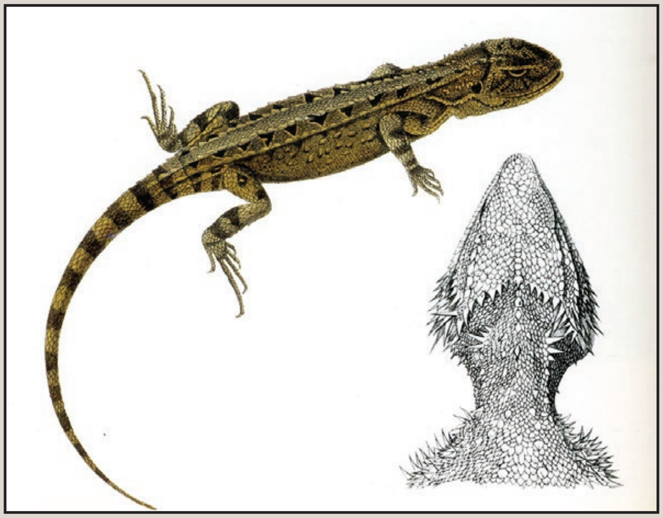


Fig. 4. Bearded Dragon (*Pogona barbata*). Reproduced from *Le règne animal…* by Baron Georges Cuvier, 1836-1839. An interactive display at the Smithsonian's National Zoo in Washington, D.C., used this species to demonstrate shuttling thermoregulation. See Avery (1994) for a discussion of effects of temperature on captive reptiles and amphibians.

Fig. 5. Jerdon's Forest Lizard (*Calotes jerdoni*; top) from India, Burma, and Bhutan, and Khasi Hills Forest Lizard (*Calotes maria*) from India. Reproduced from Albert Günther (1870) *Descriptions of a new Indian lizard of the genus* Calotes.

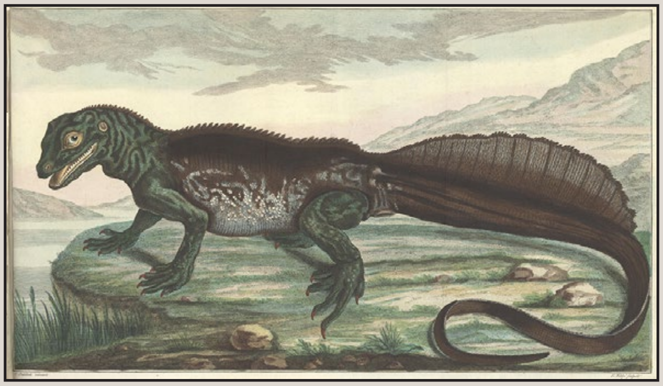


FIG. 6. Sailfin Lizard (*Hydrosaurus amboinensis*). Reproduced from *Johannis Alberti Schlosser medicinae doctoris ... Epistola ad virum expertissimum, peritissimumque Ferdinandum Dejean ... De lacerta Amboinensi...* J. A. Schlosser, 1768. This impressive large and semi-aquatic species is found in Indonesia and New Guinea. A captive group of Philippine Sailfin Dragons (*Hydrosaurus pustulatus*) maintained at the Dallas Zoo would often swim in a pool of water. This species is hardy and breeds readily in captivity. Recently-hatched lizards were used as a stimulus to study tail-luring in Death Adders (*Acanthophis antarcticus*). These lizards were separated from the snakes with a glass panel and were susceptible to the lure, lunging at snake tails. See Carpenter et al. (1978), Chiszar et al. (1990).

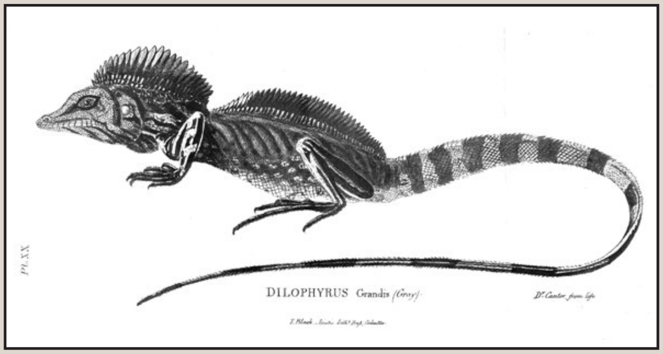


FIG. 7. Great Anglehead Lizard (*Dilophyrus Grandis*, now *Gonocephalus grandis*). Reproduced from *Catalogue of reptiles inhabiting the Malayan Peninsula* by T. E. Cantor, 1847. This species is found in Indonesia, Malaysia, Thailand and Viet Nam. We have never seen one alive in a zoo.

FIG. 8. Okinawa Tree Lizard (*Japalura polygonata*). Reproduced from George Boulenger (1887) *On a collection of reptiles and amphibians made by Mr. H. Pryor in Loo Choo Islands.* This species is native to Taiwan and Japan. "In the five centuries of published works in the field of herpetology, only two can be said to cover comprehensively and scientifically *all* of the species of amphibians and reptiles of the world. One, in English, was by George A. Boulenger and based on the collections of the British Museum in London (9 volumes, 1882–1896). These books were reprinted in 1961–1966, clothbound, and despite their age, remain among the most frequently consulted references in herpetology today." (Murphy 2017).

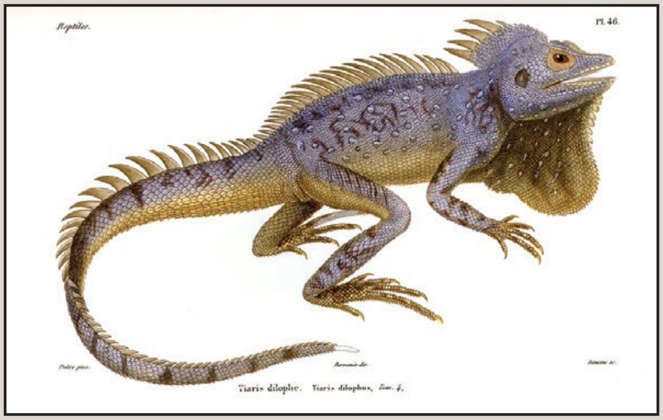


FIG. 9. Indonesian Forest Dragon (*Hypsilurus dilophus* now *Goniocephalus dilophus*). Reproduced from *Erpétologie Générale ou Histoire Naturelle Complète des Reptiles* by A. M. C. Duméril, G. Bibron and A. H. A. Duméril, 1839. This species occurs in Indonesia. See text for a description of behaviors.

FIG. 10. Boulenger's Tree Lizard (*Dendragama boulengeri*) and Sumatra Nose-horned Lizard (*Harpesaurus beccarii*). Reproduced from *Note Erpetologiche - Alcuni nuovi Sauri raccolti in Sumatra dal Dr. O. Beccari. Ann. Mus. civ. stor. nat. Genova 2* by Giacomo Doria, 1888. Both species are endemic to Indonesia.

FIG. 11. Green Water Dragon (*Physignathus mentager*, now *Physignathus cocincinus*). Reproduced from *The Reptiles of British India* by Albert C. L. G. Günther, 1864. Many zoos display this lizard species, which has been reproduced in captivity.

FIG. 12. Ornate Mastigure (*Uromastix ornatus*, now *Uromastyx ornata*). Reproduced from *Zoology of Egypt. Vol. l. Reptilia and Batrachia* by John Anderson, 1898. This species is native to Israel, Egypt, Yemen, and Saudi Arabia. Most species of this genus have been maintained alive in zoo collections. They thrive as long as temperatures are high enough.

FIG. 13. Harduns (*Trapelus hispidus, Stellio vulgaris*) from Africa, Egypt and Syria. Reproduced from *Bilder-Atlas zur wissenschaftlich-populären Naturgeschichte des Wirbelthiere* by L. J. F. J. Fitzinger, 1867.

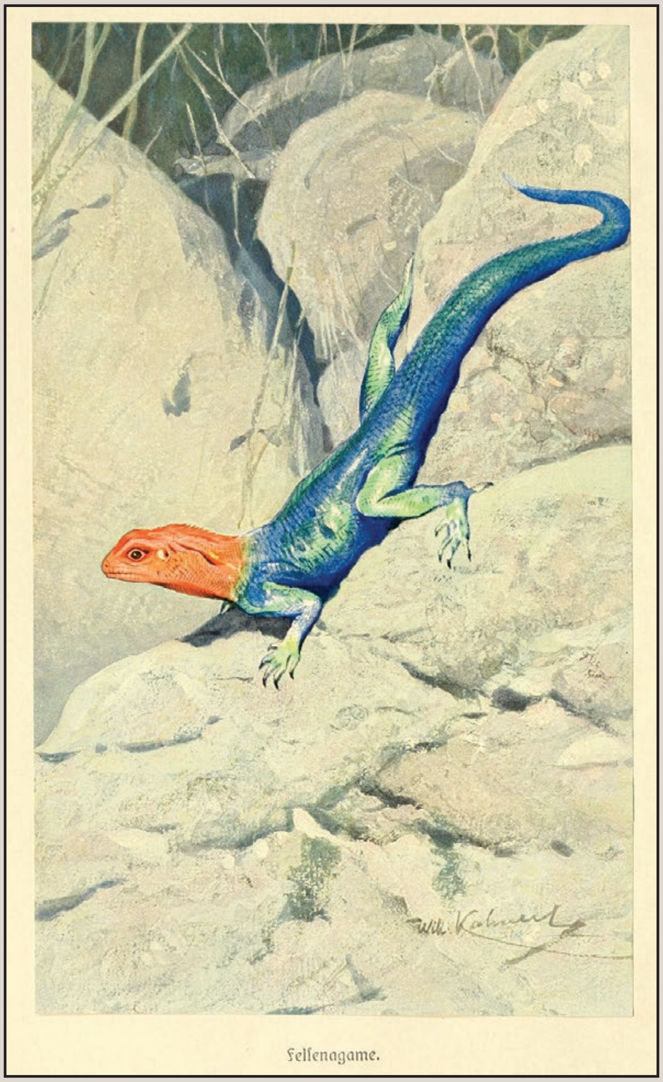
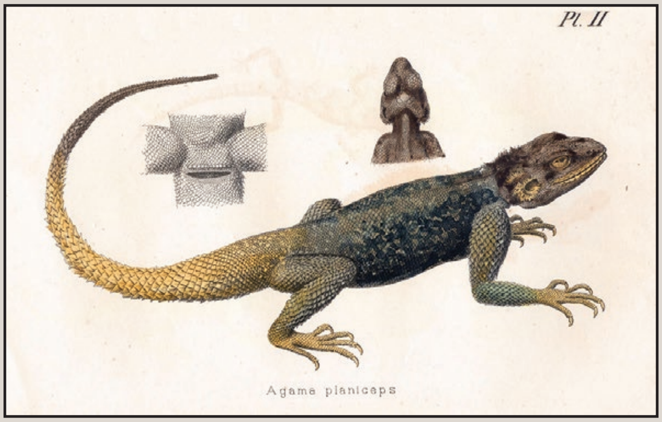


Fig. 14. Common or Red-head Agama (*Agama agama*). Reproduced from *Brehms Tierleben. Allgemeine kunde des Tierreichs. Volume 2* by Alfred E. Brehm, 1911. A captive population of this species, along with *Cordylus, Platysaurus, Chamaesaura* and *Pseudocordylus,* lived in a large display of mixed African lizards at the Dallas Zoo. Brehm coined the term "Vivarium," directed the Hamburg zoo from 1863 onward, and founded the Aquarium Unter den Linden (Berlin's main street) in 1869. His ten-volume comprehensive treatment of animals set the standard for such compilations. The reptile section (vol. 4) was revised by Franz Werner in 1912–1913. This invasive species and Indochinese Tree Agama (*Calotes mystaceus*) have been established in Florida, USA.



CREDIT: PROVIDED BY KRAIG ADLER.

Fig. 15. Namib Rock Agama (*Agama planiceps*). Reproduced from the title page of *Herpétologie d'Angola et du Congo* by J. V. Barbosa du Bocage, 1895; these drawings were made by Enrique Casanova.

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Fig. 16. "The external appearance of this Lizard is the most ferocious of any that I know, the horn of the head and the numerous spines on the body giving it a most formidable aspect" John Edward Gray (I841). Four agamids are pictured in this chromolithograph from *Kunstformen der Natur* [*Art Forms in Nature*] by Ernst Haeckel in 1904. This Thorny Devil (*Moloch horridus*) in bottom right corner is difficult to keep in captivity as it is an ant specialist. This plate (79) is called "Lacertilia/Eidechsen." In the 1960s, curator Ed Almandarz at Lincoln Park Zoo in Chicago tried all ant species in the region with no success—all were totally ignored. In Australia, Pianka and Pianka (1970) wrote that a captive only accepted one ant taxon of eight offered (*Tridomyrex rufoniger*) and discussed the "hydroscopic nature" of the skin.

Acknowledgments.—This contribution is dedicated to the memory of Charles Lewis Camp (1893–1975), who published *Classification of the Lizards* in 1923. This important work was reprinted by SSAR in 1971, with a new preface by the author and an introduction by Garth Underwood. Smithsonian librarians Polly Lasker assisted with citations and Leslie Overstreet provided a guide to reading plates.

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Addendum

How to Read Signed Plates

Prepared by Leslie Overstreet
Smithsonian Libraries

Typically, two people were involved in producing printed images: the original artist who created the image and the craftsman who transferred it to the wood block, metal (copper or steel) plate, or lithographic stone for printing. Their names commonly appear along the bottom edge of the illustration, with the artist in the lower left corner and the craftsman (wood-cutter, engraver, or lithographer) in the lower right. Sometimes, the name of the printer or "publisher" also appears in the lower right corner. Their roles are specified by the following abbreviations:

a.f., aq., aquaf., aquaforti = etched by
cael., caelavit = engraved by
d., del., delin., delineavit = drawn by
des., desig. = drawn by
dessiné = drawn by
direx., direxit = directed by (head of workshop)
eng., engd. = engraved by
ex., exc., excu., excud., excudit, excudebat = printed by; published by
f. ,fe., fec., fecit, fa., fac., faciebat = made by
gez., gezeichnet = drawn by
grav., gravé = engraved by
imp., impr., impressit = printed by (*imp. lith.* = printed on a lithographic press)
inc., inci., incid., incidit, incidebat = engraved by
inv., invent., inventor = designed by (the original artist)
lith., litho., lithog. = lithographed by
p., pictor, pin., pingebat, pinsit, pinx., pinxit = painted by (original work)
ph.sc., photosculpsit = engraved by light
s., sc., scul., sculp., sculpsit, sculpebat, sculptor = engraved by

ABOUT OUR COVER: *Feihyla hansenae*

Feihyla hansenae (Hansen's Asian Treefrog) is an arboreal-breeding member of the Rhacophoridae, a speciose family of Old World frogs (ca. 421 species; AmphibiaWeb 2018. <https://amphibiaweb.org> University of California, Berkeley. Accessed 3 Aug 2018). It can be found in ponds or seasonal pools during the rainy season in northern, east central, and southeastern Thailand (Yodthong et al. 2014. Asian Herpetological Research 5:179–196), although its range likely extends into neighboring countries of Cambodia, Laos, and Myanmar. The species was described (as *Philautus hansensae*) in 1927 by Doris M. Cochran (1898–1968), who for many years was in charge of the herpetological collection at the United States National Museum. Recent molecular surveys of samples from across the range revealed the existence of two deeply differentiated lineages (Yodthong et al. 2014, *op. cit.*), hinting that more than one species might be contained within what is presently considered *F. hansenae* (Aowphol et al. 2013. Zootaxa 3702:101–123). Unique among congeners, *F. hansenae* exhibits parental care. Females stay with their offspring throughout the egg stage, hydrating the eggs when needed and warding off potential predators (Poo and Bickford 2013. Ethology 119:671–679; Poo et al. 2016. Biological Journal of the Linnean Society 118:901–910).

Our cover image was recorded by **Sinlan Poo**, a behavioral ecologist who obtained her Ph.D. from National University of Singapore. Originally from Taiwan, she has worked on research projects in the Neotropics (Panama and Ecuador), East and Southeast Asia (Taiwan, Thailand, Sri Lanka, and Singapore), and the United States. Her dissertation work on the reproductive ecology of *Feihyla hansenae* is the first experimental study quantifying the costs and benefits of parental care in a Southeast Asian amphibian. Currently she is a postdoctoral fellow at the Memphis Zoo in Tennessee, USA, where her research is focused on assisted reproduction and conservation of endangered frogs and toads in North and Central America. She encountered the group of four male *F. hansenae* attempting to mate with one female at the Sakaerat Environmental Research Station, Nakhon Ratchasima Province, Thailand, during September 2015. The photo was taken using a Pentax K10D with a Tamron 90mm f/2.8 SP AF Di macro lens, stopped down to f/13, at an ISO 100 and shutter speed at 1/125 sec. The camera's built-in flash provided illumination. Additional details about this encounter are reported elsewhere in this issue (Poo and Low, pp. 516–517).


PHOTO BY CINDY POO

SSAR BUSINESS

Roger Conant Grants-in-Herpetology 2018 Award Winners

Successful Grant Applicants: An award in the amount of $500 was made to each of the following individuals.

Conservation:

- **Matthew Atkinson** (University of Central Florida); "The impacts of the emergent protistan pathogen Perkinsea on Florida's amphibian communities." Advisor: Anna Savage
- **Joseph Kennedy** (The University of Mississippi); "The role of reproductive interference and endocrine stress in the decline of green treefrogs following Cuban treefrog invasions." Advisor: Christopher Leary.

Education:

- **Kristen Hecht** (Florida Natural History Museum): "Public engagement of herpetologists: attitudes and scope." Advisor: Max Nickerson.

Field Research in Herpetology

- **Ying Chen** (Queen's University): "Understanding the genetic basis of advertisement call attributes in spring peepers (*Pseudacris crucifer*)." Advisor: Stephen Lougheed
- **Olivia Feagles** (University of Wisconsin-Milwaukee): "Testosterone: the power behind multimodal signaling in gray treefrogs (*Hyla versicolor*)?" Advisor: Gerlinde Höbel

International Research in Herpetology

- **Laura Braunholtz** (Newcastle University, UK): "Roads through rainforests: measuring and mitigating biodiversity impacts in Brunei." Advisor: Marion Pfeifer
- **Milan van der Velden** (Free University of Brussels): "Comparative osteology of the tepui-associated toad *Oreophrynella* with insights on the origin and function of opposable fingers and toes in the genus." Advisor: Philippe Kok

Laboratory Research in Herpetology

- **Matthew Dickson** (California State University-Northridge): "Evolution on your porch: testing the mechanisms underlying rapid adaptive change among introduced house gecko populations in response to climatic variation." Advisor: Robert Espinoza
- **Aaron Griffing** (Marquette University) "Differential regenerative ability in New Caledonian geckos (*Correlophus*): an untapped evolutionary model to study tail regeneration." Advisor: Tony Gamble

Travel

- **Kinsey Brock** (University of California-Merced): "Causes and consequences of color polymorphism." Advisor: Danielle Edwards
- **Jessica Reimche** (University of Nevada-Reno): "Molecular evolution of an adaptive trait: the genetic mechanisms of TTX resistance in the Sierra garter snake (*Thamnophis couchii*)." Advisor: Chris Feldman

Undergraduate Research in Herpetology

- **Courtney Check** (College of William and Mary): "Movement ecology and nonbreeding habitat usage of two *Anaxyrus* species." Advisor: Matthias Leu.
- **Kenen Goodwin** (Utah State University): "Characterizing temporal variation in the skin-associated microbiome of the salamander *Ambystoma mavortium*." Advisor: Zachariah Gompert

SSAR congratulates each of this year's winners and extends its appreciation to the following reviewers: Roberto Brenes (Carroll College, Wisconsin), Priya Nanjappa (U.S. Fish & Wildlife Service), Sean Sterrett (University of Massachusetts-Amherst; Massachusetts Cooperative Fish and Wildlife Research Unit), Emily Moriarty Lemmon (Florida State University), Sarah Milton (Florida Atlantic University), Matthew Chatfield (Unity College, Maine), and Joshua Kapfer (Committee Chair).

……

Roger Conant Grants-in-Herpetology Program: Guidelines for Proposal Submission

Proposals will be accepted for the 2019 SSAR Roger Conant Grants-in-Herpetology Program starting on 15 September 2018. This program is intended to provide financial support for deserving individuals (with a primary emphasis on student researchers) or organizations involved in herpetological research, education, or conservation.

Who Can Apply?

All applicants must be students AND members of SSAR with the following exceptions:

- Those applying for the International category must be students but DO NOT have to be SSAR members.

- Those applying for the Herpetological Education category DO NOT have to be a student or SSAR member.

- Those applying for support of regional herpetological society projects DO NOT have to be SSAR members.

In keeping with the Society's goal of encouraging participation by the broadest possible community of applicants, preference may be given to individuals who might not have access to other funding sources. Applications must be submitted by individuals only (but see special considerations for education categories below).

Grant Amounts and Deadlines

Successful applicants in each of the categories listed below will receive an award in the amount of US $500. A total of 12 proposals will be selected each year through peer review, to receive funding. The deadline for submission is 15 December 2018 (see special note on membership dues below). The awards will be announced by May or early June 2019.

Grant Proposal Categories

- *Conservation of Amphibians and Reptiles.*—Proposals should outline a conservation-oriented research project. This project may focus on species endangered or threatened at the state, national, or international level, or address research on potentially threatened habitats or species, or on introduced injurious species.

- *Field Research in Herpetology.*—Proposals may address needs for field station fees or equipment and materials in field oriented projects, or the field work portions of broader studies. This might include in-situ behavioral studies, ecological, life history, or sexual selection studies. Survey work by individuals or regional societies may be submitted here or in TRAVEL below depending on how the funds are to be used.

- *Laboratory Research.*—Proposals may address needs for equipment or materials in laboratory projects or laboratory portions of broader projects. This might include studies in behavior, biochemistry, molecular biology, biomechanics, or physiology.

- *Travel.*—Proposals may address support for travel to field study sites near or far, or to utilize distant collections or facilities. If funding is sought to get from one place to another, proposals should be submitted in the TRAVEL category. Proposals normally submitted in the CONSERVATION or FIELD RESEARCH categories should be submitted here if travel funding is being sought.

- *International.*—Proposals may address needs in any of the above five categories. The applicant must be a student, but not necessarily a SSAR member. Preference will be given to students with limited access to research funds and in countries where herpetological research has historically been under-funded. Note Regarding Eligibility in the International Category: In 2011, the SSAR Board of Directors decided that proposals in this category would only be accepted if they were submitted from institutions OUTSIDE of the United States. Proposals from students who are primarily affiliated with United States institutions are not eligible for this category. **Also, SSAR cannot send funds to international applicants hailing from countries to which monetary transfers are prohibited by U.S. laws. If you are a researcher in one of these countries, we cannot consider your application.**

- *Herpetological Education.*—Proposals may address an educational project or start up support for an educational program in a zoo, museum, park, nature center, regional herpetological society, etc. The project must focus on a herpetological topic. Note: Although proposals for institutional projects are accepted, education proposals must be submitted by an individual (either sole applicant or principal contact person for the project). Applicants need not be students or SSAR members.

- *Undergraduate Research in Herpetology.*—Proposals in this category can outline any research project focused on questions related to amphibian or reptile taxonomy, biology, ecology or conservation. Funding can be used to purchase equipment, materials, or as re-imbursement for travel to conduct research (i.e., offset the cost of gas for trips to field sites, museums to investigate specimens, etc.). Applicants must be currently seeking a Bachelor's Degree at an institution of higher learning. Applicants need not be affiliated with US institutions, and international applicants need not be SSAR members. Students that apply for this award must be working directly with a research advisor at an academic or professional institution (i.e., University, Regulatory Agency, Zoological Garden, Museum, etc.). This advisor must submit the letter of support for the student's work. **This letter of support must explicitly confirm that the applicant is currently a student and has the time, ability and guidance to successfully complete the proposed project**. Strong preference will be given for applicants that have limited funding from other sources.

Additional information and requirements are posted on the SSAR website at: ssarherps.org/pages/GIH.php. All proposals must be submitted electronically to kapferj@uww.edu as a single PDF file named "lastname-category.pdf" no later than 15 December 2018 to be considered (letter of support may be included with the proposal or sent separately). Exceptions to electronic submission and file format may be allowed for special cases with prior approval by SSAR-GIH Chair. Failure to meet these guidelines may result in elimination of a proposal from consideration.

Successful applicants are encouraged to submit the results of their research for publication in the *Journal of Herpetology* or *Herpetological Review*, and/or to present their findings at the annual meeting of SSAR. Submit proposals or questions regarding application procedures to: Joshua M. Kapfer, Chair, SSAR Grants in Herpetology, Department of Biological Sciences, University of Wisconsin-Whitewater, Whitewater, Wisconsin 53190, USA (e-mail: kapferj@uww.edu).

Grants-in-Herpetology Donor Information

Financial contributions by SSAR members, institutions, and other benefactors support this program significantly, and can increase the number and/or size of awards. Your tax-deductible (for US taxpayers) contribution to this program will directly benefit meritorious research and education in herpetology. Contact the SSAR Treasurer for additional information about contributing to the Grants-in-Herpetology Program. If you are employed by an organization that will match donations made to nonprofit organizations, please notify your employer that you have made a donation to the Grants-in-Herpetology Program.

SSAR would like to thank the many grant reviewers and donors who have been instrumental in the success of this committee for the last several decades.

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Dean E. Metter Memorial Award for 2018

The Dean E. Metter Memorial Award Committee was unanimous in recommending **Arianne Messerman** for this year's award. Arianne is a doctoral student with Manuel Leal in the Division of Biological Sciences at the University of Missouri—Columbia. Her bachelor's degree is in Biology from Kenyon College and she completed a master's degree in Environmental Management (Concentration: Ecosystem Science and Conservation) from Duke University.

She requested funds from SSAR to be used to continue field work in Missouri on an assemblage of Flatwoods Salamanders. Professor Leal writes that "she developed a project that integrates field-based ecological measurements, laboratory-based physiological measurements, and experimental manipulations to evaluate the potential contributions of physiological and life-history traits to the current and future distribution of salamanders."

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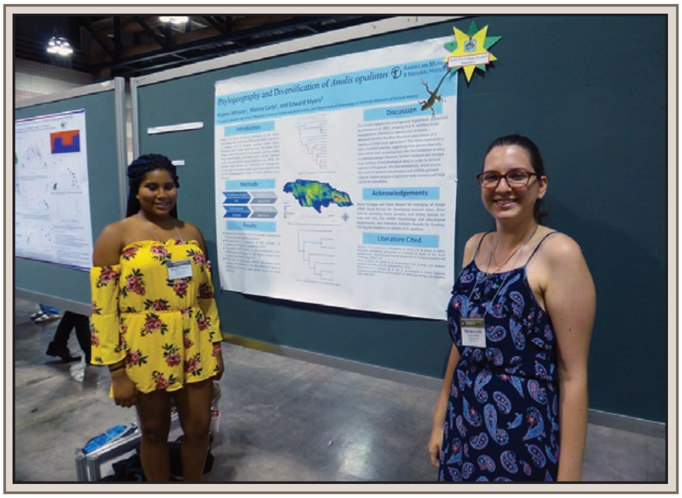
PHOTO BY C. THAWLEY

Support for Pre-College Students to Attend SSAR Annual Meeting

SSAR is pleased to announce that it will provide funds for selected junior and senior high school students who are budding herpetologists and wish to interact with advanced students and professionals with similar interests. Some pre-baccalaureate students already have knowledge about amphibians and reptiles, but wish to learn more about the field of herpetology. Attending our annual meeting, which will be held at the Snowbird Resort, near Sandy, Utah, from July 24–28, 2019, would provide such an opportunity. See the following website for more information: conferences.k-state.edu/JMIH2019. The meeting is the Joint Meeting of Ichthyologists and Herpetologists (JMIH).

An anonymous donor has made funds available to support several students in 2019. These awards will cover the student's registration fees and may possibly help to defray some other expenses.

Students who wish to apply must provide the following information by email:

A one-page, double-spaced statement about why you wish to attend and what you expect to gain from the experience. Please include what grade in school you will be entering in the fall of 2019. Please send this statement as an attachment.

A letter of support from your teacher or guidance counselor endorsing your application and certifying that you are a current student at his/her school. Please send this letter as an attachment.

One of your parents must agree to attend the meeting with you to act as chaperone. The parent must send an email agreeing to attend the meeting with you. Applications without this email of agreement will not be considered.

Budget. If you need to have more than the registration fee covered, please explain what amount you would need and what it would be used for. Note that we cannot cover a parent's expenses, and only under unusual circumstances can we allot much more than the cost of registration fees to any one student. The budget information should be included in the email message sent with your statement.

All application materials must be sent before 25 February 2019. Send to: Dr. John Maerz, Chair of the SSAR Pre-College Awards Committee, at jcmaerz@uga.edu. Only complete applications that include the four items above will be considered.

Awards will be announced by 5 March 2019. All funds will be made available at the meeting in Snowbird.

A requirement of these meeting awards is that students must provide a few paragraphs about their experiences at the meeting. (For example: Was the experience valuable, and if so, in what ways? How can the experience be improved?). This report should be sent to Dr. Maerz no later than one month after the meeting ends.

For detailed descriptions of previous SSAR meetings, with photographs, see the December issues of the society's bulletin, *Herpetological Review* (https://ssarherps.org/publications/herpetological-review/)

The photo on the preceding page depicts Kyomi Johnson (L) and Marina Carbi (R), who were pre-college recipients of funds to support their participation in JMIH 2018. They are pictured with their poster on "Speciation and Phylogeography of *Anolis opalinus* on Jamaica."

NEWSNOTES

Desert Tortoise Council Awards Available for 2019

The Desert Tortoise Council offers three award programs in support of travel and research for students. The **David J. Morafka Memorial Research Award** was established in memory of the late Dr. David J. Morafka, distinguished herpetologist and authority on North American tortoises. The Morafka Award supports research that contributes to the understanding, management, and conservation of tortoises of the genus *Gopherus* in the southwestern United States and Mexico: *G. agassizii, G. morafkai, G. evgoodei, G. berlandieri*, and *G. flavomarginatus*. The award amount is US $2,000.

The **Lockheed Martin Diversity Grant** is intended to fund research by minority and international students. The Council favors grant applications for research that contributes to the understanding, management, and conservation of tortoises of the genus *Gopherus* in the southwestern United States and Mexico but will consider proposals for similar research on other imperiled chelonian species across the globe. The grant amount is US $1,000.

The **Glenn R. Stewart Travel Fund** honors the DTC's cofounder. This Fund supports students working with North American *Gopherus* tortoises by assisting with travel costs to attend and participate in the 2019 Desert Tortoise Council Symposia. The Fund will support up to US $500 (each) in travel costs for up to two students annually.

The application deadlines for the Morafka Award and Diversity Grant are **1 December 2018**; the deadline for the Student Travel Fund is **24 November 2018**. Detailed information about these award programs, including eligibility, evaluation criteria, and application procedures can be found at: http://www.deserttortoise.org.

Reptile Database Update

The latest release of the Reptile Database was released on 2 July 2018.

As usual, herpetologists weren't exactly idle, so we made literally thousands of edits to this release. Here are some highlights:

Species database. Over the past 4 months, the number of species increased from 10,711 to 10,793, an increase of 82 species. 66 new species have been described since March 1. Nine species have been revalidated from synonymy and 16 subspecies were elevated to full species. In addition, 8 species moved to another genus and 4 changed their gender. Finally, 10 species were synonymized, resulting in a total list of 113 new or changed species names.

New man-made species. Note that we decided not to accept the two recently described "synthetic" species, *Aspidoscelis neavesi* and *A. priscillae*. Both species were created by crossing sexual species, so that the resulting hybrids turned out to be asexual, parthenogenetic offspring. Such hybrids occur naturally and thus have been recognized as valid species for decades. In fact, more than 70 parthenogenetic reptile species listed in the Reptile Database are of natural origin and have been established in nature for a long time (note that quite a few of them have been shown to be only facultatively parthenogenetic). While the experiments by Cole et al. (2014, 2017 provide interesting insights into the origin of parthenogenetic species, their new lab-made clones do not occur in nature, which is the main reason why our Scientific Advisory Board voted not to recognize them at this point, at least not as regular species. Nevertheless, you can find their names in the database as synonyms under their parental species, *Aspidoscelis inornatus, A. uniparens*, and *A. exsanguis*. The IUCN also released a similar statement on synthetic species.

Literature update. This release of the database contains **46,318 references**, compared to 45,535 in the February 2018 release, an increase of 783 publications.

Literature curation help still needed. To keep up with the flood of publications, we would like to renew our call for help with curation. If you enjoy reading reptile papers, especially related to taxonomy, phylogenetics, and biogeography, please let us know. Actually, we are happy to cover all kinds of other topics, given that we are adding natural history data to the database too, but we cannot cover all these other topics ourselves, so they are even more dependent on your help. We are experimenting with a new model now where we put papers on cloud storage, so our curation team (you?) can access them and make annotations (e.g., using Acrobat Reader or Preview on Macs). Email us for more details.

Social media updates. About 6 months ago we started to post new species on social media, including Facebook, Twitter, and Instagram, managed by our social media editors **Amy McLeod** and **Mark Herr**. If you want to know whether there are new reptile species please follow those posts. We have also started a new web page with database updates before they go online in the public web database. We are planning to expand those posts to other taxonomic and possibly other herpetological updates.

New checklists and geographic datasets. Since our last release almost 1,000 entries have had their distribution data edited. Among countless small edits and updates, we have used the latest *Field Guide to East African Reptiles* (see below) to update the data on Eastern Africa, and the checklist for **Guerrero** (Mexico, 182 species!), based on Palacios-Aguilar & Flores-Villela 2018.

How up-to-date is the database? In case you wondered ... there is no simple way to measure this. However, one approximation is the most recent paper cited in each species entry. By that measure, only 49 species don't have any citations before 2000, but 10,418 species have citations after 2010, and 5874 have citations published after 2015. Notably, 1,777 species have been updated with citations published in 2018 (although there are more that were updated this year without 2018 citations!). That is, overall we are quite up to date. But let us know if anything is missing.

New photos. During the past 4 months, 64 photographers have submitted 692 new photos of 342 species. We have now 11,954 photos of 4,309 species (= 40% of all species) from about 760 photographers. The **new photos in this release were contributed** by A. A. Nadolnyi, Adriana Dado, Alfred Schleicher, Alvaro Camina, André Koch, Antoine Fouquet, Antonio Cadiz Diaz, Brian Bush, César J. Pollo, Christopher C Austin, Colin Tilbury, Cristian Hernán Fulvio Pérez, Daniel Velho, Dave Showler, David Hodkinson, Diego Paucar, Elí García-Padilla, Frank McCann, Frank Tillack, Gary Brown, Hector M. Diaz Perdomo, Henrik Bringsøe, Igor Doronin, Marina Doronina, Igor Joventino, Ishan Agarwal, Israel Solano Zavaleta, Ivan Ineich, Jake Wilson Binaday, Jannico Kelk, Jingsong Shi, John Lyakurwa, Jorge Alberto Zuñiga Baos, Laurent Chirio, Luciano Javier Avila, Luke Bloch, Luke Verburgt, M. M. Beskaravayniy, Manuel Iturriaga Monsisbay, Marc Faucher, Marcelo Ribeiro Duarte, Marcos Di Bernardo, Maren Gaulke, Marina Doronina, Maxim A. Koshkin, O. V. Kukushkin, Olga Alishevskaya, Patrick Campbell, Paul Freed, Peter Soltys, Pongpol Adireksam, Roberto Sindaco, Roman Zuev, Ryan Ellis, S. V. Leonov, Sagar Khunte, Siria Ribeiro, Stephen Mahony, Stephen Zozaya, Steve Wilson, V. Deepak, V. Giragosov, V. S. Marchenko, O. V. Kukushkin, and Werner Conradie. The most generous photographers this time were **Laurent Chirio with 180 photos**, followed by **Gary Brown with 116**, our own **Paul Freed with 70**, and **Marcelo Ribeiro Duarte with 54**. As always, our sincerest thanks to all contributors! Please keep sending in those photos — we still have ~6000 species to cover!

Bioinformatics summer projects. If you are interested in helping out at the Reptile Database with your programming or other computer skills, you are welcome to join us for a number of smaller projects, e.g. aiming at text mining, data analysis, or **reformatting** project (e.g. converting **bibliographies** to a format that we can import). Most of these are not very demanding, but some experience with a programming language (Perl, Python, R etc.) or database application (SQL, Access, Filemaker) would greatly help. We also have some projects aiming at the automated **extraction of images from pdfs** or from other sources to set up a database of (copyrighted) images for further image analysis or **character extraction**. Finally, we are eager to expand data analysis to geographic or phylogenetic data. Please contact us for further details.

As usual, please send us corrections, additions, papers, or comments, or post them to our Facebook page.

Contributed by the Reptile Database Team
Peter Uetz, Paul Freed, Jiri Hošek, Amy McLeod, Mark Herr
http://www.reptile-database.org

MEETINGS

International Herpetological Symposium

2018 marked the 41st anniversary of the International Herpetological Symposium. The primary purpose of the IHS is to provide a forum for the dissemination of information and results of research pertaining to the natural history, conservation biology, and captive management and propagation of amphibians and reptiles. Each year the IHS is held at a different location, often locally hosted by a zoological, herpetological, or herpetocultural institution. In June (2018), IHS was generously hosted by the Houston Zoo in Houston, Texas, and facilitated in partnership with the East Texas Herpetological Society. The keynote speaker was Twan Leenders, with the Roger Tory Peterson Institute of Natural History, whose presentation "Conservation Starts at Home—a Herpetologist's Journey Through the World's Backyards" was the perfect kick-off to the conference (Fig. 1). The banquet speaker, Carl Franklin, with the Amphibian and Reptile Diversity Research Center at the University of Texas Arlington, thoroughly entertained and educated the audience with his presentation "New Knowledge and Dance Moves from Old Animals." The meeting hosted over 40 presenters from all over the world, as well as "An Evening with Harry Greene," which included an inspiring presentation, book signing, and lively discussion over evening cocktails.

Fig. 1. Keynote speaker Twan Leenders of the Roger Tory Peterson Institute of Natural History presenting "Conservation Starts at Home-a Herpetologist's Journey Through the World's Backyards" at IHS Houston, Texas.

Presenters included a group of inspired young speakers referred to as "Junior Herpetologists." The International Herpetological Society's JUNIOR HERPETOLOGIST and NEXTGEN HERPETOLOGIST program was initiated in 2015 by the IHS board supported by several sponsors (especially Timberline) to promote the younger generation's interest and involvement in herpetology, conservation, education, and herpetoculture (Fig. 2). Full travel grants are awarded annually for winners and their chaperones to attend the meetings and present papers. This program is growing, and during the 2018 conference, Junior/NextGen Herpetologists had the opportunity to participate not only in the presentations and meeting events, but evening field-herping excursions and an immersive "hands in the gloves" reptile necropsy wet-lab conducted by veterinarian Dr. Cord Offerman.

During the 2018 IHS, the Grants Program committee selected three projects to receive a total of US $3,000 in one of four categories: Herpetological Natural History, Herpetological Conservation Biology, Captive Propagation, and Herpetological Education. IHS strongly encourages researchers and educators to apply. Proposals are due by May 31 of each year with recipients notified by September. Applicants may be anyone from the herpetological community and recipients are expected to present their findings at a future symposium. Grants are made possible by the generous contributions of attendees and donors during our live auction. Special thanks to Terry Vandeventer, Master Bladesmith, who generously donated one of his custom creations to the IHS auction, and to Todd Goodman of Timberline for his outstanding winning bid.

To promote new research at the undergraduate level, IHS implemented the Collegiate Speed Session during the 2018 meeting. This session was a hit, with positive feedback from participants and attendees alike. If you are interested in participating in this session, more information can be found on our website under Speaker Information.

Each year, IHS honors those who have demonstrated significant contributions in the field of herpetology. The Joe Laszlo Memorial Award for 2018 went to Micha Petty of the Louisiana Exotic Animal Resource Network for his presentation "Innovations in outreach: interpreting herps to the public." The IHS Award in Conservation was presented to Maria Elena Barragan-Paladines of the Fundación Herpetológica Gustavo Orces for "New approaches toward the conservation of venomous snakes in Ecuador." Last but not least, the IHS presented Paul Moler, Florida Fish and Wildlife Conservation Commission, with the Lifetime Achievement Award for his longstanding and ongoing dedication to the protection of reptiles and amphibians and their habitats.

Fig. 2. (From left to right) Junior Herpetologist program coordinator, Russ Gurley; Junior and NextGen Herpetologist award winners Paul Coyne "The Evolutionary Differences within the Chelidae Family," Michael Skibsted "*Macrochelys temminckii*: the Living Dinosaur," and Tyler Tobias-Jones "Dragons, Dinosaurs, and Detective Work: the Potential Impact of Paleobiology in Herpetological Conservation;" special guest Harry Greene; Todd Goodman, Owner and CEO of Timberline and sponsor of the Junior Herpetologist program.

Unlike most herpetological societies or associations, IHS does not have a voting membership, but an electoral body. That body consists of the members of the Board of Directors, the Advisory Council, publication editors, and chairs of various committees. Dedicated zoologists, herpetologists, industry professionals, and private herpetoculturists volunteer their time in planning and organizing the annual symposia and other IHS related programs.

Over the years, an increasing number of people with varied interests in herpetology have attended IHS meetings, and this curious mix has allowed IHS to develop its unique flavor. Although IHS was historically visualized as an organization with an interesting blend of academia and herpetoculture, the current attendees far exceed those bounds. In short, IHS has something for everyone, and all are welcome. Starting in 2019, IHS will offer special registration rates for youth attendees (17 and under) as well as student discounts. We encourage you to join us next year at our first international meeting in over a decade hosted in Belize (19–22 June 2019). The call for abstracts is now open, and speaker slots are filling up fast. All local Belizean residents receive half off of registration costs with valid ID. IHS Officers are hard at work organizing a meeting to remember, this is one you will not want to miss. We hope to see you all in Belize!

More information on speaker instructions, grants, proceedings, hotel and registration can be found on the IHS website:

<http://www.internationalherpetologicalsymposium.com>

Jennifer L. Stabile
President – IHS
jens@fieldprojects.org

Meetings Calendar

Meeting announcement information should be sent directly to the Editor (herpreview@gmail.com) well in advance of the event. We also welcome brief reports of meetings; please consult the Editor for details.

19–20 October 2018—Rattlesnakes of Arizona: Book Signing and Mini-Symposium, Rodeo, New Mexico, USA. Information: https://www.geronimoevent.com/home

5–8 November 2018—XV Reunión Nacional de Herpetología and 30th Anniversary Meeting of Sociedad Herpetológica Mexicana, Xalapa, Veracruz, Mexico. Information: http://sociedadherpetologicamexicana.org.mx/

10–11 January 2019—California-Nevada Amphibian Populations Task Force Meeting, Arcata, California, USA

21–23 February 2019—Desert Tortoise Council Symposium, Tucson, Arizona, USA. Information: www.deserttortoise.org

20–24 May 2019—2º Congreso Nacional de Vipéridos Mexicanos y Ofidismo, Aguascalientes, Mexico. Information: congresoviperidos@gmail.com

19–22 June 2019—42nd International Herpetological Symposium, Belize. Information: http://www.internationalherpetologicalsymposium.com

11–14 July 2019—Biology of Pitvipers 3, Rodeo, New Mexico, USA. Information: https://www.biologyofthepitvipers.com/home

24–28 July 2019—Joint Meeting of Ichthyologists and Herpetologists, Snowbird, Utah, USA.

2–6 September 2019—14th Herpetological Association of Africa Conference, Hogsback, Eastern Cape, South Africa. Information: s.edwards@ru.ac.za

September 2019—SEH 2019: 20th European Congress of Herpetology, Milan, Italy. Information: http://seh-congress-2019.unipv.it/seh2019-at-milan/

CURRENT RESEARCH

The purpose of Current Research is to present brief summaries and citations for selected papers from journals other than those published by the American Society of Ichthyologists and Herpetologists, The Herpetologists' League, and the Society for the Study of Amphibians and Reptiles. Limited space prohibits comprehensive coverage of the literature, but an effort will be made to cover a variety of taxa and topics. To ensure that the coverage is as broad and current as possible, authors are invited to send reprints to the Current Research section editors, Ben Lowe or Chava Weitzman; e-mail addresses may be found on the inside front cover.

Morphology and Antimicrobial Properties of Aquatic Salamander Skin Glands

Amphibians possess two types of exocrine glands in their skin: mucous and granular. Mucous glands generally serve to keep the skin moist, however, specialized mucous glands of some lineages have been co-opted for the purpose of pheromone production. Typical granular glands have toxic and anti-microbial properties, though in some lineages, they produce pheromones and store nutrients. A growing body of work has investigated the potential for granular secretions to help amphibians fight *Bd* infection, with some positive results. These investigations into amphibian skin glands have largely been restricted to terrestrial or amphibious species. The authors of this paper aimed to shed light on the morphology and properties of the skin glands of an aquatic amphibian, the Three-toed Amphiuma (*Amphiuma tridactylum*). They were specifically interested in determining whether the exocrine products of *A. tridactylum* skin glands inhibit *Bd* and *Bsal* infections, which they considered likely, given the absence of cases of infected amphiumas. They also looked for evidence of skin gland specialization akin to that seen in other amphibians. The authors collected skin secretions from two live *A. tridactylum* (one of each sex) and enriched them for antimicrobial peptides. These products were analyzed using reversed-phase high-performance liquid chromatography and were also applied to *Bd* and *Bsal* cultures to determine the minimal inhibitory concentration (MIC), the smallest amount of secretion that completely inhibits fungal growth. Skin samples were collected from two salamanders of each sex (including one reproductively active male and one inactive male). These samples were studied histologically, and variation in mucous and granular gland density was determined for various body sections and planes. The liquid chromatography analysis showed that skin secretions did not differ between sexes and were mostly composed of a single compound type. The *Bd*- and *Bsal*-inhibition experiments revealed that the skin secretions of *A. tridactylum* do indeed inhibit the growth of these fungi. This marks the first study to find *Bsal*-inhibition properties in amphibian skin secretions (*Bsal* MIC = 187 $\mu g\ ml^{-1}$). The estimated *Bd* MIC, 75 $\mu g\ ml^{-1}$, is very low relative to estimates for other evaluated species indicating a powerful anti-*Bd* property of amphiuma skin secretions. Across the body, glands of each type are not distributed evenly, and in the case of granular glands, not uniform in size: mucous glands occur at a higher concentration on the head, and granular glands reach their highest concentration and largest size posterior to the head. The largest granular glands were observed on the base of the tail of the reproductively active male. These findings reveal aquatic salamanders possess skin glands that are both antimicrobially active and show regional specialization, warranting further investigation.

Pereira, K. E., B. I. Crother, D. M. Sever, C. L. Fontenot, Jr., J. A. Pojman, Sr., D. B. Wilburn, and S. K. Woodley. 2018. Skin glands of an aquatic salamander vary in size and distribution and release antimicrobial secretions effective against chytrid fungal pathogens. Journal of Experimental Biology (*in press*) DOI: 10.1242/jeb.183707

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Toxin-Resistance Alleles Confer Different Locomotor Consequences for Gartersnakes

Adaptations do not arise in a vacuum: changes to an existing character to further one function often have pleiotropic effects and can potentially be maladaptive relative to other functions. One well-studied system of evolutionary tradeoffs with respect to adaptation involves gartersnakes (Natricidae: *Thamnophis* spp.) and western newts (*Taricha* spp.). *Taricha* possess tetrodotoxin (TTX) in their skin secretions, a chemical that acts on the nervous system and is one of the most toxic non-protein substances known to science. Previous work has established that these gartersnakes and newts have entered into an "evolutionary arms race," with several *Thamnophis* lineages having independently acquired mutations that alter the voltage-gated sodium channels (Na_v 1.4) associated with their skeletal muscle allowing them to consume *Taricha* despite their TTX-laden skin secretions. Previous work has also suggested that this TTX resistance has a negative, pleiotropic effect on locomotor performance. However, a direct genotype–phenotype connection has not been made, and the underlying mechanism of reduced motor performance associated with the TTX-resistant allele is not completely understood. To this end, the authors of this paper genotyped snakes from two lineages of *Thamnophis sirtalis* (California and Pacific Northwest; 7 and 11 populations, respectively) for which TTX resistance and crawl speed has previously been evaluated. Specifically, a gene that encodes an important region of the Na_v 1.4 protein was sequenced. Tests were performed to determine whether a correlation between genotype and motor performance exists. To evaluate the functionality of various Na_v 1.4 proteins, alleles for non-TTX-resistant and TTX-resistant proteins from each of the two lineages were cloned, converted to mRNA transcripts, and injected into *Xenopus laevis* oocytes (resulting in the incorporation of these proteins). Two to seven days after injection, oocytes were evaluated for ionic current and TTX-binding affinity. These experiments resulted in dramatically different consequences for the two lineages. Pacific Northwest snakes showed no correlation between Na_v 1.4 genotype and locomotor phenotype, while the two were strongly correlated in California snakes. The authors suggest the TTX-resistant allele present in California snakes represents an escape from the arms race, with homozygous resistant snakes suffering no locomotor consequences upon consuming *Taricha*. This TTX-resistant allele is more derived relative to that found in Pacific Northwest populations, with more amino acid substitutions. Alternatively, Pacific Northwest snakes with TTX-resistant alleles suffer no locomotor consequences due to their genotype, but their less-derived TTX-resistant alleles confer less resistance relative to those of California populations. These differences were also reflected in the protein-performance experiments. Although both alleles showed reduced TTX affinity and shifts in voltage dependence relative to the non-resistant allele, the magnitudes of these trends were much greater for the California allele than the Pacific Northwest allele. These results further clarify the dynamics underlying a foundational system for the study of natural selection.

Hague, M. T. J., G. Toledo, S. L. Geffeney, C. T. Hanifin, E. D. Brodie Jr., and E. D. Brodie III. 2018. Large-effect mutations generate trade-off between predatory and locomotor ability during arms race coevolution with deadly prey. Evolution Letters (*in press*) DOI: 10.1002/evl3.76

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Bidirectional Dispersal of *Goniurosaurus* between Island and Mainland

The lizard genus *Goniurosaurus* (Eublepharidae) consists of 18 described species found in southeastern China, northeastern Vietnam, the Ryukyu Archipelago, and Hainan Island. A previous morphological analysis of the genus found a pattern consistent with two dispersal events from the mainland to Hainan Island, followed by speciation. Specifically, Hainanese *G. bawanglingensis* was found to be related to one group of *Goniurosaurus* species in Vietnam and adjacent China, while another Hainanese species (*G. hainanensis*) fell out within another Vietnamese clade that includes *G. lichtenfelderi*. However, previous molecular studies failed to include *G. bawanglingensis*, and no phylogenetic study has included the recently described Hainanese *G. zhoui*. To further clarify the evolutionary relationship of Hainanese *Goniurosaurus*, the authors of this study acquired genetic samples from the three Hainanese *Goniurosaurus* species, from which they collected molecular sequence data for mitochondrial and two nuclear genes. Along with published *Goniurosaurus* sequence data, these new sequence data were subjected to phylogenetic analyses, including the construction of a time-calibrated tree. Further, ancestral state analyses were performed to identify the ancestral geographic ranges of the recovered lineages and to reveal how the number of body bands has evolved. These analyses showed that *G. zhoui* is sister to a *G. lichtenfelderi* + *G. hainanensis* clade. In turn, this clade is sister to *G. bawanglingensis*. Therefore, instead of multiple colonizations of Hainan, the most parsimonious explanation is a single colonization of Hainan, followed by intra-island diversification, then colonization of the mainland by Hainanese *Goniurosaurus* (the ancestor of *G. lichtenfelderi*). The divergence between mainland and Hainanese *Goniurosaurus* (the latter including *G. lichtenfelderi*) dates to between 22 and 49 million years ago, the lower bound nearing the proposed timing of Hainan's separation from the mainland. The split between *G. lichtenfelderi* and the rest of the Hainanese clade was identified as occurring ~3.5 million years ago, coinciding with glacial periods. Character state reconstruction suggests that a decrease in body band number has occurred independently two or three times. This study shows that Hainan Island has served as a biodiversity source as well as a sink and highlights the importance of considering large continental islands as potential sources of diversity.

Liang, B., R-B. Zhou, Y-L. Liu, B. Chen, L. L. Grismer, and N. Wang. 2018. Renewed classification within *Goniurosaurus* (Squamata: Eublepharidae) uncovers the dual roles of a continental island (Hainan) in species evolution. Molecular Phylogenetics and Evolution (*in press*) DOI: 10.1016/j.ympev.2018.06.011

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Advanced Spring Phenology in Turtles Varies by Geography and Species

Species are limited by different abiotic and biotic factors at different points in their distribution, and yet predictions of how species will respond to warming climates are often extrapolated from responses in one or few local populations. At distributional edges, species are more likely to be limited by abiotic variables, and these sites often cannot represent how whole species will respond to changing environments. The authors of this study analyzed long-term nesting behavior data from multiple sites alongside climate data to identify variation and changes across geography and temperature cues. Analyses focused on the date of first nesting in six freshwater turtle genera at six sites in North America, with data on 10–36 sampling years per site. Though the authors found some significant shifts in first nesting occurring sooner across sampling years, there were no clear latitudinal trends across species associated with changes in the onset of nesting. For most populations, however, warmer temperatures in April corresponded with earlier nesting onset. Additional data on spring emergence and the onset of basking mirrored the nesting trends observed. Overall, this in-depth sampling indicates intraspecific variation in spring behaviors among populations in the plastic phenotypes targeted by this study. Despite inconsistent responses to latitude among species, these results suggest that no one population of a species should be deemed representative in predictions of how that species will react to climate change. The authors additionally note that changes in thermal environments will also affect turtle development and sex ratios, and how populations respond to these pressures will also vary.

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Dollo's Law and Digit-Number Evolution in *Brachymeles* Skinks

Digit reduction and loss within amniote lineages has been a frequent subject of scientific study. These studies have revealed a pattern where digit #1 is generally the first to be lost, followed by digit #5 (Morse's Law). Hovering over these investigations is Dollo's Law: complex structures, once lost, cannot be re-evolved to their original state. However, the authors of the present paper point out that Dollo's Law as originally postulated does allow for the re-evolution of characters subsequent to being lost; they just wouldn't be identical to their original state. The authors of this paper aimed to evaluate the scincid genus *Brachymeles* with respect to both Dollo's and Morse's laws. *Brachymeles* is represented by 41 described species and is restricted to the Philippines (except for two species occurring in Borneo and Thailand). *Brachymeles* species span the limb/digit spectrum, with limbless, limbed but lacking five digits, and pentadactyl species. In apparent violation of Dollo's Law, previous studies have suggested the pentadactyl condition has re-evolved within *Brachymeles*. The authors collected limb and digit number data for 39 *Brachymeles* species and 30 outgroup taxa. Measurements of bones, and phalangeal formulae (patterns of numbers of bones per digit), were acquired for 11 *Brachymeles* species and 3 outgroup taxa. The limb musculature of a large pentadactyl species (*B. kadwa*) was analyzed through dissection. The authors also assembled a molecular dataset (1 mtDNA locus/3 nuDNA loci) for the lizards in the more inclusive morphological dataset. The concatenated dataset was subjected to a time-calibrated phylogenetic analysis. The resulting tree was used to infer the ancestral morphological states, and various models of limb/digit evolution were evaluated. These analyses revealed very strong evidence for the pentadactyl state re-evolving in an ancestor of the clade that includes *B. kadwa*. A model where limbs do not re-evolve did not perform significantly worse than models allowing for limb re-evolution. Alternatively, a model allowing for digit-increase evolution with different rates for gain and loss significantly outperformed other models (digit loss was 22.5 times more likely than digit gain). However, in keeping with a more nuanced Dollo's Law, the pentadactyl limb of *B. kadwa* is morphologically different than that of other pentadactyl skinks. Specifically, the limb of *B. kadwa* is less complex with fewer phalanges and muscles. The pattern of digit loss in *Brachymeles* runs counter to Morse's Law: digit #5 is the most frequently lost digit. In contrast, the pattern of digit loss seen in other skink lineages follows Morse's Law (e.g., *Lerista*, *Hemiergis*). This study highlights the importance of testing these "biological laws" in each instance they are suspected to apply.

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Exotic Competitor Impacts Foraging of Native Predator

Convergent evolution is a phenomenon that has fascinated biologists ever since Darwin. A classic herpetological example of convergent evolution is between the Sidewinder Rattlesnake (*Crotalus cerastes*; hereafter, "Sidewinder") of North America's Mojave and Sonoran deserts and the Horned Desert Viper (*Cerastes cerastes*; hereafter, "Horned Viper") distributed from North Africa to the Arabian Peninsula. However, the authors of the present paper suggest that the Sidewinder's pit organ represents a "constraint-breaking adaptation," allowing it to hunt efficiently in complete darkness. The authors aimed to test this hypothesis by determining whether the behavior of a natural predator and its prey (Horned Viper and Anderson's Gerbil, respectively) would be altered by the presence of Sidewinders. They predicted that either Sidewinders would cause Horned Vipers to shift their activity to moonlit nights or Sidewinders would inhibit Horned Viper activity regardless of moon cycle. The experiment was conducted in a large outdoor vivarium on the Sede Boker Campus of Ben-Gurion University in Israel's Negev Desert. The vivarium was divided into four sectors, each maintaining six gerbils with 18 gerbil feeding stations. One sector had no snakes (negative control), one had two Horned Vipers, one had two Sidewinders, and one had one of each species. Preliminary tests revealed that Sidewinders readily hunted and consumed gerbils. Snakes were allowed to forage in the sectors over four periods (two each new moon and full moon; all three consecutive nights except one four consecutive nights new-moon period). Horned Viper activity was assessed by observing characteristic tracks left in the sand when in ambush position. Gerbil activity was assessed by the amount of food removed from the feeding stations. As expected, Horned Viper activity was higher on full moon nights. When Sidewinders were present, Horned Viper full moon activity increased and new moon activity decreased relative to when Sidewinders were absent. Gerbil activity was similar for the two single snake species treatments, but was reduced in the two-snake species treatment. The authors suggest that the gerbil and the Horned Viper recognize the Sidewinder as a predator and a competitor, respectively, as indicated by their behavioral changes. Because the gerbils showed greatest activity reduction when in the presence of two snake predator species, the authors argue that the Sidewinder facilitates the Horned Viper on moonlit nights, while interfering on moonless nights. The nuanced relationships between native predator and prey and exotic predator found in this study highlight the complex interactions that must be considered when evaluating the effect of invasive species.

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Convergent Dwarfism in Island Reptiles Supported by Resource Reduction

Island dwarfism and gigantism are common in species with both mainland and island populations. The evolution of these size differences is thought to be associated with resource restrictions in the case of dwarfism and predator release in the case of gigantism. Importantly, declines in resource availability could result in tradeoffs affecting life-history traits other than size. In this study, the authors report evidence for island dwarfism in three reptile species of California and test the hypothesis of smaller size corresponding with limited prey items by analyzing head morphology and blood glucose levels. Individuals from one to two populations of Gopher Snakes (*Pituophis catenifer*), Western Yellow-Bellied Racers (*Coluber constrictor*), and Southern Alligator Lizards (*Elgaria multicarinata*) were measured in mainland California and nearby California Channel Islands. On average, all three species had significantly smaller snout–vent length on islands relative to mainland populations, ranging from 10–31% smaller depending on the species. A principal component analysis reduced the dimensionality of three head measurements relative to body size. Island populations of both snake species had smaller relative head sizes than mainland populations, though in the lizards, this comparison was only marginally significant. Female Gopher Snakes also tended to have smaller relative head sizes than males on islands, indicating sexual dimorphism in the dwarfs in characteristics not dimorphic in mainland populations, which could be due to differing foraging strategies between the sexes. Blood glucose also tended to be lower in island individuals, with patterns differing by sampling year and sex depending on the species. The authors posit that inconsistencies in blood glucose patterns were likely affected by a drought that occurred during the study. Small head size and lower blood glucose both support the hypothesis that island populations encounter fewer resources, and prey resources are likely smaller. In vertebrates, glucose levels can increase with stressors, but the presence of dwarfism and not gigantism in these species does not support a hypothesis that low glucose levels could be due to predator release. The California Channel Islands, which were never connected to the mainland, were colonized by these species many thousands of years ago. The authors hypothesize that such a long period of isolation would allow for their divergence in body size to be an evolutionary change, and is less likely to be caused by plasticity in these traits. A continuation of this study will include additional species in these reptile communities.

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Community Richness, Host Species, and Spatial Factors Affect Ranavirus Presence

The ability to predict disease dynamics is a fundamental aspect of management and conservation successes, but patterns of disease at different scales are affected by different drivers and variables. In the case of ranaviruses, some host species are susceptible to disease while others may be reservoirs for the pathogens, and abiotic factors such as pond depth or water quality can affect disease prevalence and infection intensity. Using data from 76 ponds in California's East Bay region, this study aimed to detect the influence of 21 biotic, abiotic, and landscape predictor variables on ranavirus presence. Prevalence data came from nearly 1400 juvenile amphibian samples from 2013, representing five non-threatened species (3 frog and 2 salamander species). One-third of the samples tested positive for ranavirus, and two-thirds of the ponds had at least one infected individual. Rough-skinned Newts (*Taricha granulosa*) had the highest ranavirus prevalence of 60%, while American Bullfrogs (*Rana catesbeiana*) had the lowest at 16%. Using multimodel inference, the authors detected that shorter distance to an infected pond and an increase in richness of amphibians, fishes, and macroinvertebrates were both associated with an increased likelihood of ranavirus presence. These results suggest that the movement of infected individuals or water among nearby ponds is important in the spread of disease. Increased taxonomic richness, on the other hand, could allow for more reservoir species or more opportunities for ranavirus to spread to new wetlands. Individual-level analyses, which included only data from the ponds with ranavirus, determined that smaller individuals (shorter snout–vent length) and individuals in ponds with lower vertebrate richness relative to host density were more likely to host ranavirus, and different species had different likelihoods of hosting the pathogen. The opposite effect of richness on individual-level infections versus site-level presence could indicate a dilution effect, but more information on transmission of ranaviruses within and between host species is needed to tease apart the role of species richness in disease prevalence. Overall, abiotic factors were found to be less important to ranavirus presence than biotic and landscape factors. The lack of an association between environmental stressors or the presence of proposed reservoir species such as bullfrogs provides important new insights into our still-limited understanding about this disease.

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Identifying Causes of *Anolis* Radiation

The genus *Anolis* is widely recognized as having undergone an adaptive radiation, with hundreds of species described between mainland and island habitats. Thorough examination of this radiation, however, had not been conducted to identify likely drivers of this diversification. In this study, the authors used large datasets on *Anolis* and its possible sister group, Corytophanidae, to test for the presence of rapid diversification and two possible drivers: 1) the dewlap as a key innovation, and 2) expansion to island habitats. Phylogenetic estimates included data on all 379 *Anolis* species, while phenotypic analyses incorporated data on 336 of these species for which head scale size, relative hind limb length, and body size data were available. From phylogenetic analyses, the authors did not detect a significant difference in speciation rate of dewlapped *Anolis* when compared with non-dewlapped corytophanids and *Anolis* (males of two anole species lack a dewlap); they suggest caution in accepting this conclusion, considering, among other things, the obvious richness disparity that exists between these groups. The authors also did not find a significantly increased speciation rate in island versus mainland species, but argue that the invasion of anoles onto islands and other geographic uplifts that also allow for local endemics could maintain a comparable and elevated speciation rate in the genus as a whole. Within *Anolis*, adaptive radiation on islands was supported by an increased rate of body length evolution, which could have allowed for these lizards to quickly fill open ecological niches on islands uninhabited by their competitors. Interestingly, this support for divergence of phenotypic evolutionary rates disappeared when corytophanids were included in the analysis. Though the authors did not find dewlaps to be the key innovation allowing for *Anolis* adaptive radiation, they urge that another trait may still support the key innovation hypothesis and discuss multiple reasons why the adaptive radiation in this genus should continue to be investigated.

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Panzootic Chytrid Fungus Diversified in Last 120 Years by Human Movement

Chytridiomycosis is a panzootic disease in frogs, affecting populations around the world. Years of research on the origin of the lineage of *Batrachochytrium dendrobatidis* (*Bd*) causing this disease has given inconclusive results, with studies estimating a most recent common ancestor in the virulent lineage ranging two orders of magnitude and geographic origin theories pinpointing multiple regions around the globe. In this study, the authors analyzed genomic data from over 200 *Bd* isolates representing every continent where the pathogen has been found and all three orders of Amphibia to reveal phylogenetic relationships among the lineages and detect the likely origin of this pathogen. In addition to detecting lineages previously described, these analyses found a new lineage from the Korean peninsula with higher diversity than the other groups. From interpretations of Tajima's *D* values, most lineages seem to have experienced selection or instability in population size, though values for the new Korean lineage show mutation-drift equilibrium, indicative of the pathogen being endemic to the region. The authors further estimated that the lineage responsible for most of the worldwide chytridiomycosis likely originated 50–120 years ago, with some variation stemming from mitochondrial versus nuclear estimates. The spread and diversification of this lineage (named *Bd*GPL) thus coincides with an increase in the amphibian trade in the 20th century and an increase in worldwide human movement beginning in the 19th century. In addition to the rapid diversification of *Bd* by selection, the authors detected at least three instances of hybridization between lineages, allowing for further diversity. Alongside genomic studies, this paper investigated the relative virulence of the different lineages and found *Bd*GPL and an additional lineage found in Europe and Africa, *Bd*CAPE, to have similar virulence greater than the other lineages, though virulence was species- and life-stage-specific. The authors urge for more thorough sampling for *Bd* in Asia, where they have found high genetic diversity in relatively few isolates, and where *Bd* and its close relative, *B. salamandrivorans*, seem to have originated. The rapid worldwide spread of this pathogen in the recent past reveals that a focus should be placed on biosecurity for amphibian species protection.

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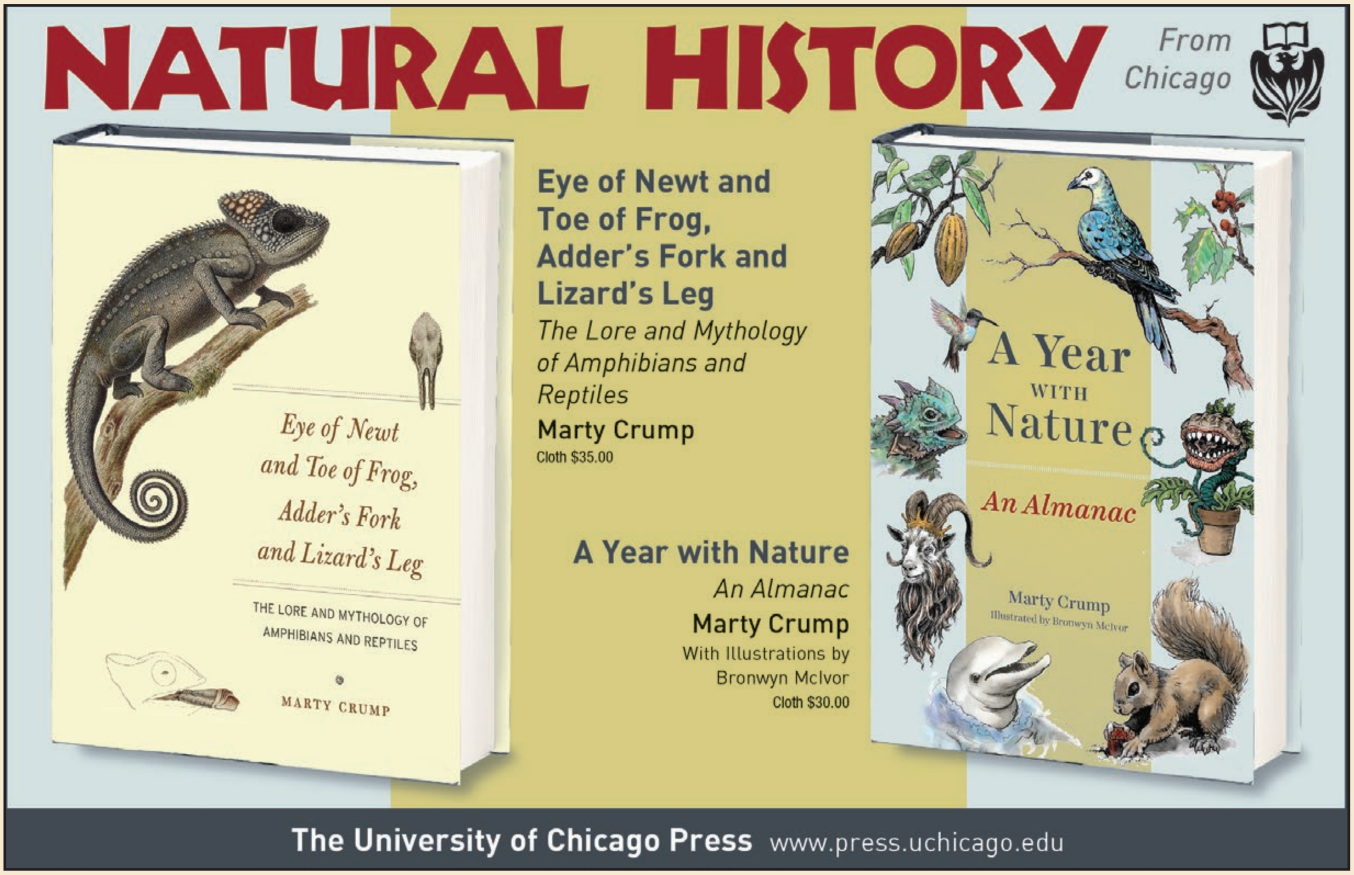

Eye of Newt and Toe of Frog, Adder's Fork and Lizard's Leg
THE LORE AND MYTHOLOGY OF AMPHIBIANS AND REPTILES
MARTY CRUMP
A Year with Nature
An Almanac
Marty Crump
Illustrated by Bronwyn McIvor

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The **International Herpetological Symposium** (**IHS**) has established a grant program to provide financial assistance to individuals or organizations conducting herpetological research, conservation, and education. Proposals are due by **April 30** of each year, and recipients are announced by September. Grants are available annually in amounts up to US $1000 and will be awarded to applicants whose projects represent a significant contribution to herpetology in one of the areas listed below:

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- ***Herpetological Conservation Biology***.—Proposals in this category should address new research on threatened, imperiled, or a surrogate for such amphibian or reptile species, or the phenomena that affect the maintenance, decline, and restoration of their natural habitat.
- ***Captive Propagation***.— Proposals in this category should address research in captive behavioral studies or new techniques in captive maintenance and breeding of amphibians or reptiles.
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- Recipients will be required to present their findings at a future symposium, either in person or via poster at their own expense.
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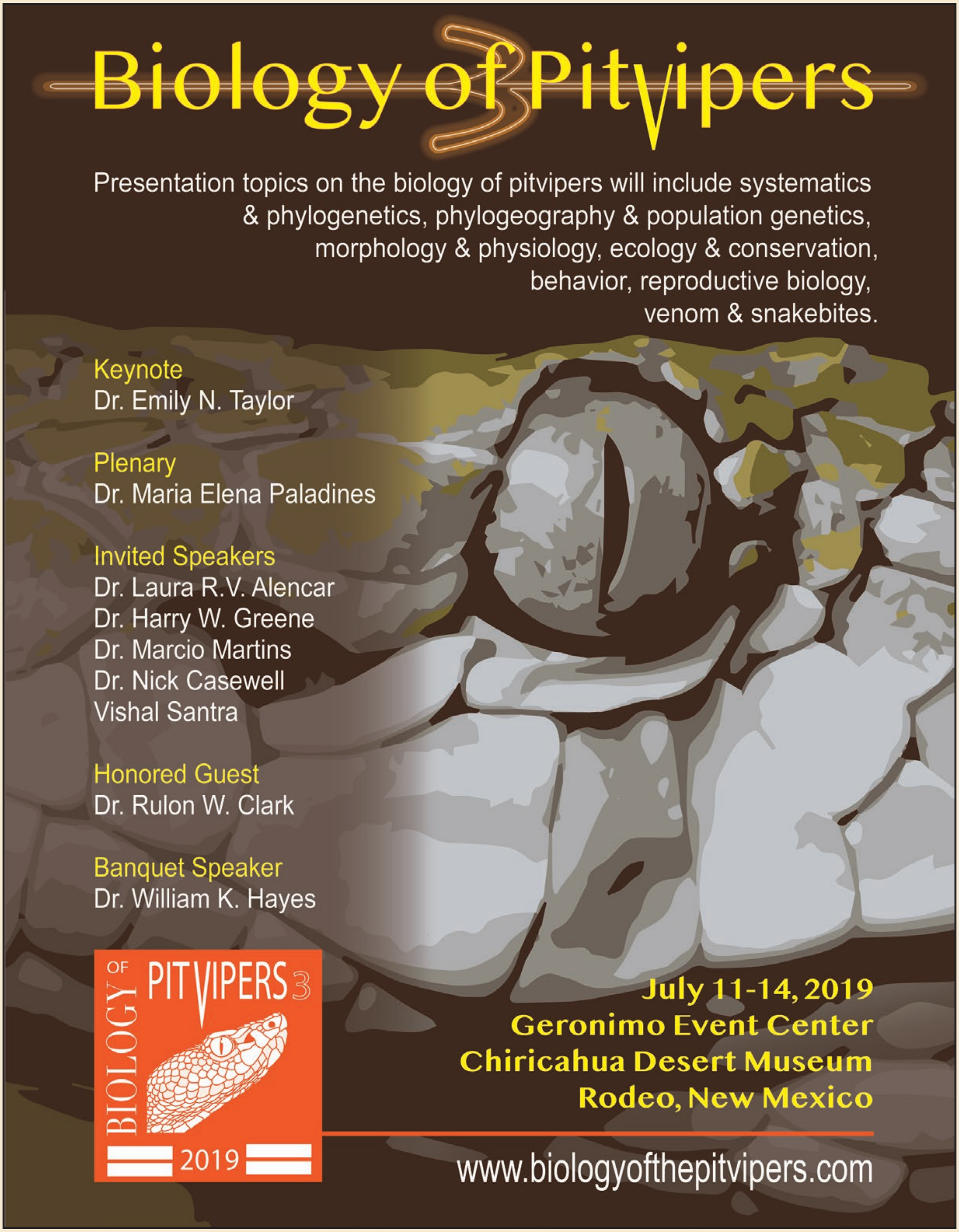

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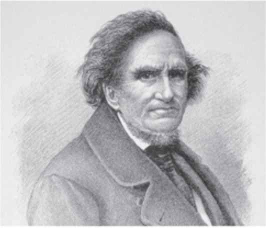


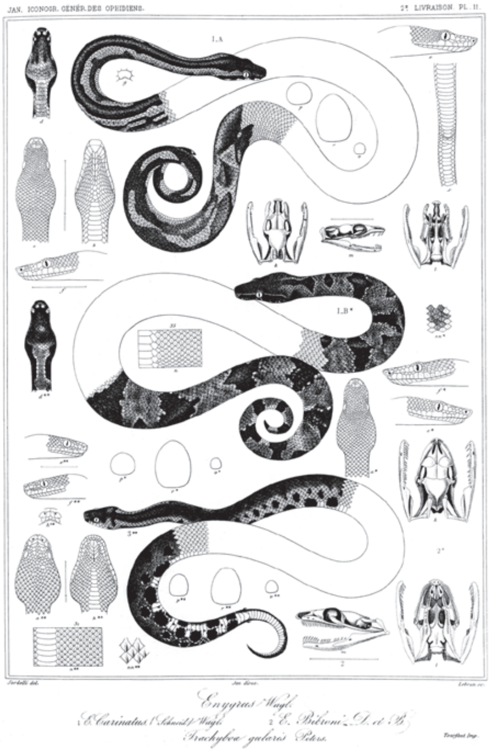
The Herpetological Contributions of Giorgio Jan (1791–1866)

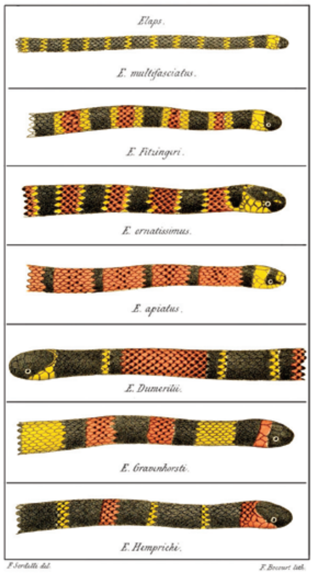
With an Introduction, Annotated Bibliography, Synopsis of Herpetological Taxa, and a Comprehensive Guide to the ***Iconographie Générale des Ophidiens***

by JAY M. SAVAGE and ROY W. McDIARMID

Giorgio Jan (1791–1866) undertook one of the most ambitious herpetological projects of all time—to monograph and illustrate the world's snake fauna. The result was the *Iconographie Générale des Ophidiens* (1860–1881), the most comprehensive, and still one of the most useful of all illustrated works on snakes. This facsimile reprint of the *Iconographie* includes Ferdinando Sordelli's 300 plates with more than 8500 individual figures depicting 953 species of snakes, as well as all of the original wrappers as issued with the 50 livraisons comprising the work as a whole. Also included are the facsimiles of 22 additional publications by Jan constituting his entire herpetological output. These include Jan's plan and prodromus for the *Iconographie*, several family level monographs meant to accompany the *Iconographie*, and his most extensive text, *Elenco sistematico degli Ofidi descritti e disegnati per l'Iconografia generale* (1863), as well as 23 additional plates.

This publication also features 168 pages of **new material** by renowned herpetologists and bibliophiles **Jay M. Savage** and **Roy W. McDiarmid**. This includes historical background and an annotated bibliography of Giorgio Jan, as well as synopses of the taxa recognized and described by Jan, the authorship of names in the *Iconographie*, and much more. The global scope of Jan's work and the invaluable Comprehensive Guide by Savage and McDiarmid make this a book that belongs on every herpetologist's shelf.





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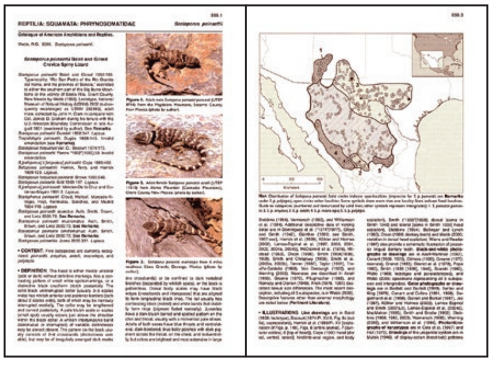
CATALOGUE OF AMERICAN AMPHIBIANS AND REPTILES

The *Catalogue* consists of individual accounts of species and genera prepared by specialists, including synonymy, definition, distribution map, and comprehensive list of literature for each taxon. The accounts cover amphibians and reptiles of the entire Western Hemisphere. As of 2014, SSAR is making all CAAR accounts OPEN ACCESS as a service to the herpetological community. To access individual accounts, visit https://repositories.lib.utexas.edu/handle/2152/44116.



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