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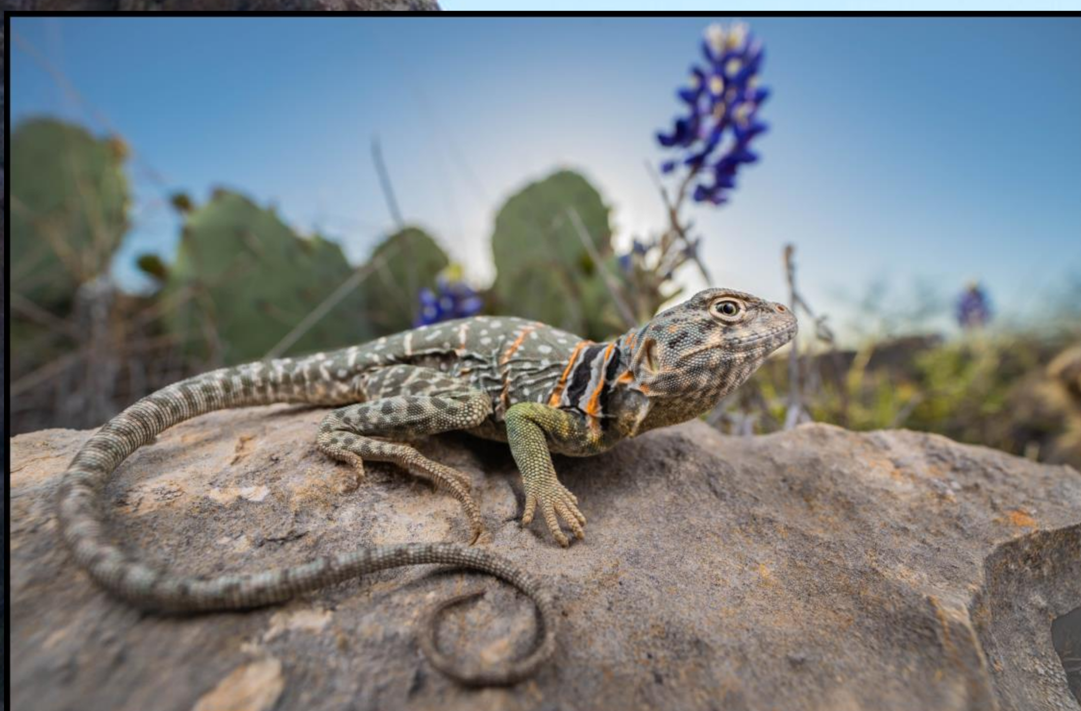

Conservation – Preservation – Education – Public Information
Research – Field Studies – Captive Propagation

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ON THE COVER: Eastern Collared Lizard (*Crotaphytus collaris*), Irion County, Texas (Francisco Portillo). With this photograph, Frank won the SWCHR 2023 *Hans F. Koenig Award for Excellence in Herpetological Photography.*

BACKGROUND IMAGE: Gates' Pass, Tucson Mountains, AZ (Bill White)

ABOUT SWCHR

Originally founded by Gerald Keown in 2007, SWCHR is a 501(c)(3) non-profit association, governed by a board of directors and dedicated to promoting education of the Association's members and the general public relating to the natural history, biology, taxonomy, conservation and preservation needs, field studies, and captive propagation of the herpetofauna indigenous to the American Southwest.

THE SWCHR LOGO

There are several versions of the SWCHR logo, all featuring the Gray-Banded Kingsnake (*Lampropeltis alterna*), a widely-recognized reptile native to the Trans-Pecos region of Texas as well as adjacent Mexico and New MexiCounty

JOINING SWCHR

For information on becoming a member please visit the membership page of the SWCHR web site at
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A CALL FOR PAPERS

Are you a field herpetologist or a herpetoculturist (amateur or professional in either of those capacities) working with species native to the American Southwest? Do you have a paper or an article you have written for which you would like to find a permanent repository? Want to be assured you will always be able to share it with the world? Submit it to the SWCHR *Bulletin* for possible publication. Submitted manuscripts from SWCHR members, as well as non-members, will be considered. There are no page charges to have your articles appear in the SWCHR *Bulletin*, as some other publications now require. To the contrary, **published articles earn the author a free membership in SWCHR for the remainder of the calendar year** (or one-calendar-year extension if they're already a member).

To be accepted for publication, submissions must address herpetological species native to the American Southwest. Such topics as field notes, county checklists, range extensions, taxonomy, reproduction and breeding, diseases, snake bite and venom research, domestic breeding and maintenance, conservation issues, legal issues, etc. are all acceptable. For assistance with formatting manuscripts, contact us at the email address below.

Previously published articles or papers are acceptable, provided you still hold the copyright to the work and have the right to re-publish it. If we accept your paper or article for publication, you will still continue to be the copyright holder. If your submission has been previously published, please provide the name of the publication in which it appeared along with the date of publication. All submissions should be manually proofed in addition to being spell checked and should be submitted by email as either Microsoft Word or text documents.

Send submissions to info@southwesternherp.com.

A Message from the President

I hope everyone is well and finding themselves with a great start to the 2024 field season! We have another great SWCHR *Bulletin*, but also spring weather on our side. I want to keep my rambling to a minimum so that everyone can get to these great papers! But first I do think it is important to remind everyone—both current members and people reading this paper through university access—that papers accepted for publication in our *Bulletin* receive a free membership to SWCHR for the remainder of the calendar year. Even if you are unsure if your observation or experience might be important, please consider submitting it. We are more than happy to hear about your observation; it may be more important than you realize to help fill in our understanding of the natural world and to help build further hypotheses. Now on to my favorite part—being able to have the honor of introducing these amazing papers on behalf of SWCHR.

We begin with the fall quarter update for our SWCHR synopsis of notes and papers from fellow publications. With *Herpetological Review* having just published their September 2023 issue, we wanted to summarize items of interest from our six-state region for that quarter.

Connor Wardle gives us his experiences and perspective on keeping and breeding the Chihuahuan Night Snake (*Hypsiglena jani*). This species is not a widely-kept species in this hobby, so this is a great paper to help fill in those gaps and gain a new perspective on the life of this little snake.

Dusty Rhoads offers us a glimpse into the hard work he has been doing to introduce the First-Ever commercially-available Native Plant Seed Mix to benefit a reptile (and not just any reptile!). For anyone who loves horned lizards, this is the paper you don't want to miss.

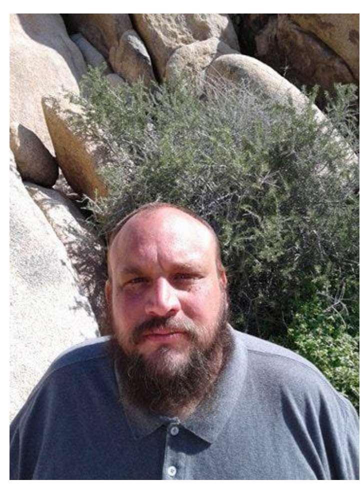
Zoe Hutcherson, our 2023 SWCHR *Laurence M. Klauber Memorial Summer Research Grant* winner, offers us an update on her research into the impact of wildfire on pathogen prevalence in Sacramento Mountains Salamanders (*Aneides hardii*). Again, this is a fine example of study of a lesser-researched species, with implications potentially valuable to similar species and similar habitats.

We close with a paper by Don Becker about his work with Yellow Mud Turtles (*Kinosternon flavescens*) and Ornate Box Turtles (*Terrapene ornata*). Though his studies are outside the southwestern United States, the data logger he created will be of interest and possible use to researchers in any region.

I believe that is enough of my rambling. I am excited to read this and future issues of the SWCHR *Bulletin,* just thinking about what people may discover and build upon based on what they read here. These natural history notes and articles inform our passion for these "lower vertebrates"—it is an exciting thought.

I will see you on the road cuts!

Robert Trembley



Synopsis of SWCHR Region Notes from 2023 Publications—Updated for Fall 2023

compiled by

Robert Twombley

Editor's Note: Since the publication dates of *Herpetological Review* for the 2023 calendar year have been delayed, this update addresses papers and notes published in Volume 54, Issue 3.

SWCHR publishes these abbreviated accounts of Geographical Distribution, Natural History Notes, and other relevant publications, so our readers may be aware of these items pertaining to the herpetofauna of the American Southwest (Arizona, California, Nevada, New Mexico, Texas, and Utah). Accounts are listed by state, then by class/order/suborder as follows: salamanders and newts, frogs and toads, turtles, crocodilians, lizards, and snakes.

Only natural history notes observed in the six-state SWCHR region of interest are included, though other observations may have been recorded from elsewhere in a given species' range. Furthermore, this synopsis should not be considered authoritative—for the full, original accounts, please see the 2023 issues of *Herpetological Review* published by the Society for the Study of Amphibians and Reptiles (2023 was Volume 54 of *Herpetological Review*, the issue number is appended to each listing below). Entries in **bold** signify contributions from SWCHR members.

Geographic Distribution

ARIZONA

(None)

CALIFORNIA

Chalcides ocellatus (Ocellated Skink). San Bernardino County: Apparently established introduced populations of this species native to northern Africa, the Middle East, and parts of the Mediterranean. (3)

Lichanura orcutti (Rosy Boa). Imperial County: Bridges a distribution gap. (3)

NEVADA

Diadophis punctatus regalis (Regal Ring-necked Snake). Nye County: The northernmost vouchered record in Nye County. (3)

Sonora occipitalis (Western Shovel-nosed Snake). Clark County: Range extension. (3)

Trimorphodon lambda (Sonoran Lyresnake). Lincoln County: New county record. (3)

NEW MEXICO

Aspidoscelis exsanguis (Chihuahuan Spotted Whiptail). Colfax County: New county record. (3)

TEXAS

Ambystoma maculatum (Spotted Salamander). Panola County: Fills a distribution gap. (3)

Eleutherodactylus cystignathoides (Rio Grande Chirping Frog). Gregg County: New county record. (3)

Hyla chrysoscelis (Cope's Gray Treefrog). Panola County: Fills a distribution gap. (3)

Pseudacris crucifer (Spring Peeper). Fills a distribution gap. (3)

Pseudacris fouquettei (Cajun Chorus Frog). Fills a distribution gap. (3)

Gastrophryne carolinensis (Eastern Narrow-mouthed Toad). Fills a distribution gap. (3)

Anaxyrus speciosus (Texas Toad). Martin County: New county record. (3)

Lithobates berlandieri (Rio Grande Leopard Frog). Andrews County: New county record. (3)

Lithobates palustris (Pickerel Frog). Fills a distribution gap. (3)

Scaphiohus couchii (Couch's Spadefoot). Martin County: New county record. (3)

Spea bombifrons (Plains Spadefoot). Glasscock County, Martin County: New county records. (3)

Macrochelys teminckii (Alligator Snapping Turtle). Brazoria County: New county record. (3)

Pituophis catenifer (Gophersnake). Martin County: New county record. (3)

UTAH

(None)

Natural History Notes

ARIZONA

Rana chiricahuensis (Chiricahua Leopard Frog). ATTEMPTED PREDATION: Report of an unsuccessful attempt by a *Butorides virescens* (Green Heron) to consume a *R. chiricahuensis*. (3)

Sceloporus magister (Desert Spiny Lizard). SEED SHELL ENTRAPMENT: A young *S. magister* was observed with its head seemingly stuck inside a broken acorn shell (*Quercus* sp.). (3)

CALIFORNIA

Ambystoma macrodactylum (Long-toed Salamander). DIET: An *A. macrodactylum* larva consumed an *Anaxyrus boreas* (Western Toad) larva. (3)

Taricha granulosa (Rough-skinned Newt). DIET: Observation of an adult *Taricha granulosa* consuming a *Monadenia fidelis* (Pacific Sideband Snail). (3)

Pseudacris sierra (Sierran Treefrog). HABITAT USE: Documents the use of commercially operating rice fields in the Sacramento Valley for reproduction. (3)

Rana draytonii (California Red-legged Frog). DIET: Observation of a female *R. draytonii* of moderate size captured and consumed a live *Microtus californicus* (California Vole). (3)

Thamnophis couchii (Sierra Garter Snake), *T. hammondii* (Two-striped Garter Snake), and *T. sirtalis* (Common Garter Snake). TAIL LOSS AND INJURY: A report of three instances of tail loss in western garter snakes, two of which are the first records for their respective species. (3)

NEW MEXICO
(None)

NEVADA
Anaxyrus monfontanus (Hot Creek Toad). REPRODUCTION: Documents late-season breeding event following monsoonal rains in this little-known species. (3)
Tantilla hobartsmithi (Smith's Black-headed Snake). PREDATION: A juvenile female *Latrodectus hesperus* (Western Black Widow) was observed preying upon a *T. hobartsmithi.* (3)

TEXAS
Bufo (*Anaxyrus*) *houstonensis* (Houston Toad). CLIMBING BEHAVIOR: Observation demonstrating that exhaustive searches should not be limited to their spring activity period, nor restricted to searching under sheltering objects during daylight hours. (3)
Kinosternon hirtipes (Rough-footed Mud Turtle). DEMOGRAPHY, SEX RATIO, AND SEXUAL SIZE DIMORPHISM: Study found that the adult sex ratio (female: male) at Plata Wetland Complex was male biased (1:2). (3)
Lepidochelys kempii (Kemp's Ridley Sea Turtle). NESTING MIGRATION: Records of adult *L. kempii* tagged during an in-water study in southeast Florida to be observed nesting in Texas. (3)
Crotalus scutulatus (Mohave Rattlesnake). DEFENSIVE BEHAVIOR: The second published account of neck spreading as a defensive behavior in the northern subspecies, *C. s. scutulatus.* (3)
M*icrurus tener* (Texas Coral Snake). DIURNAL AQUATIC MOVEMENT: Observation of a diurnal aquatic movement. (3)

UTAH
(None)

Book Reviews

Caldwell, Janalee P. *Review of* Schuett, Gordon W.; Charles F. Smith, and William Wells. 2023. *Amphibians of the Sky Islands—Coronado National Forest.* ECO Publishing, Rodeo, New Mexico (https://ecouniverse.com). 144 pp. Softcover. US $14.95. ISBN: 978-1-938850-49-3.

Peer-Reviewed and Other Papers of Southwestern Interest
(all from 2023 unless otherwise noted)

Bowers, Brandon C.; Corey M. Fielder, Danielle K. Walkup, Roel R. Lopez, Wade A. Rydberg, Toby J. Hibbitts, Mickey R. Parker, and Paul S. Crump. "Camera-trap Basking Arrays Detect Western Chicken Turtles (*Deirochelys reticulari)a miaria*) in Dynamic Ephemeral Wetland Mosaics." *Herpetological Review* 54(3): 368-372.

Fielder, Corey M. "New Distributional Records for Amphibians in Panola County, Texas, USA." *Herpetological Review* 54(3): 410.

Moore, John and Brandon C. Bowers. "New County Records for the High Plains Ecoregion of Texas, USA." *Herpetological Review* 54(3): 409-410.

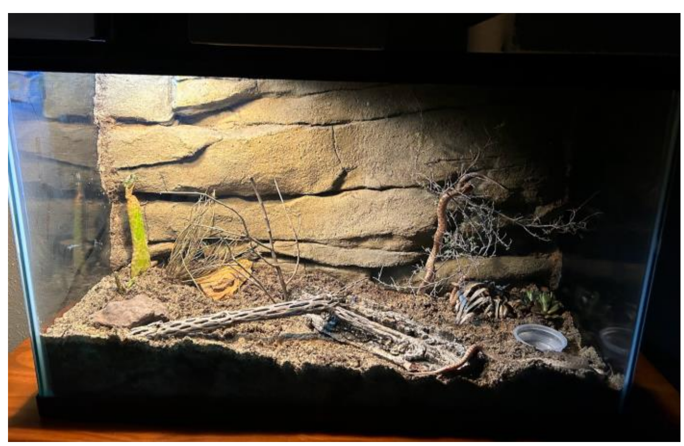
Captive Care and Husbandry of the Chihuahuan Night Snake (*Hypsiglena jani*)

by

Connor Wardle

Let me start off by saying this is by no means the only way to keep this species; this is just what works best for me in my situation. Should you decide to give this species a try I would recommend doing some experimenting to see what works best for your animals and I would encourage you to share your findings with other enthusiasts in the "Hypsiglena Enthusiasts" group on Facebook.

The Chihuahuan Night Snake(*Hypsiglena jani*) or Texas Night Snake is one of, I believe, 10 species of *Hypsiglena* and is found commonly throughout Texas, Oklahoma, New Mexico, Eastern Arizona, Colorado, Kansas, and Mexico. This species typically reaches lengths of 10 to 16 inches; however, I am sure there have been larger specimens recorded. This species primarily feeds on lizards, small snakes, and toads, although it wouldn't surprise me if hatchlings/juveniles consumed soft-bodied invertebrates given their size. This species is often overlooked due to its "difficult" feeding habits, smaller size (especially as hatchlings), and earth-tone coloration. I have greatly enjoyed keeping this species as well as the Desert Night Snake (*Hypsiglena chlorophaea*), and have been maintaining my group for about three years now.



My Chihuahuan Night Snake (*Hypsiglena jani*) display. I still have some work to do on it and some of the plants must grow. Photo by the author.

Housing

I have recently transitioned my 1.1.2 group into a communal 29-gallon Aqueon tank measuring 30" x 12.5" x 18.75" (LxWxH). I have kept them in groups in the past and haven't had conflicts, so I will be attempting it again to hopefully get some captive breeding success. In the past I have kept them in individual Exoterra cube-style enclosures.

One of the unsexed individuals in the new display I built. Photo by the author.

Lighting/Heating

Given that this species is primarily crepuscular/nocturnal, lighting is not required. That being said, with my intent to breed this species and produce true captive-bred *H. jani*, I feel like a natural light cycle to show seasonal changes won't hurt. I provide a ZooMed Reptisun 10.0 UVB bulb as well as a 50-watt heat bulb. The basking spot is around 83 degrees Fahrenheit with an ambient temperature of 75 degrees Fahrenheit. During the spring I provide a 12:12 day-to-night cycle, summer increases to 14:10, and in the fall they get a 10:14 cycle. During the winter months they brumate in a wine cooler.

Substrate

When setting up displays like this particular enclosure, I really enjoy making it look as close to the dirt I find out in the field when observing these guys. I came up with my own mix of play sand and top soil which resembled an area I have found these guys in Alpine, Texas. This species tends to burrow quite a bit, so I provide a nice and deep three-inch substrate layer, which is beneficial for the desert scrub plants as well. In the past I have used aspen shavings and haven't had issues with it; I just wanted a more visually pleasing enclosure for my group.

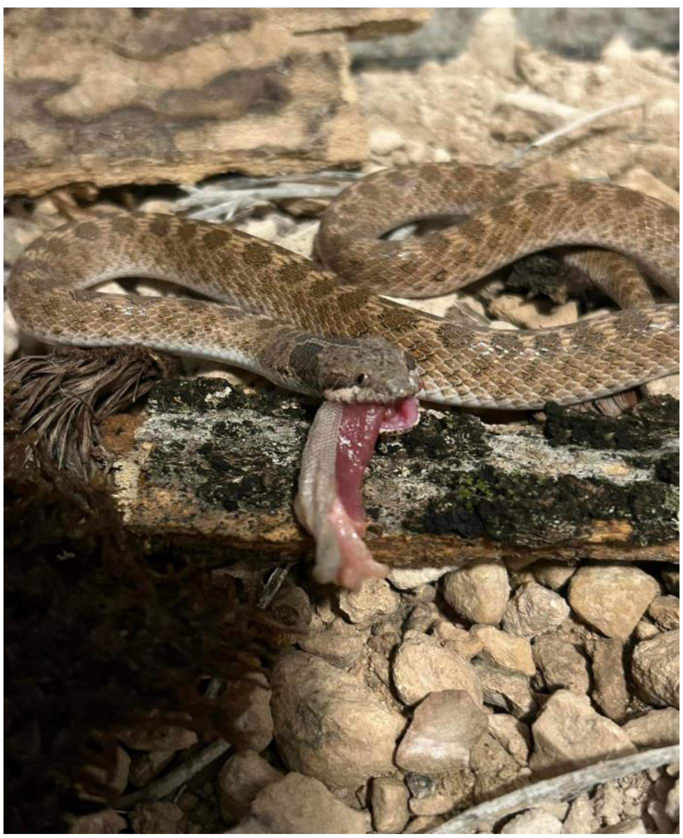
My big girl exploring the new set up. Photo by the author.

Humidity

I do not measure the humidity within any of my enclosures, but I do provide humid hides to provide a gradient within the enclosure. These humid hides are typically made from old food containers and are filled with damp sphagnum moss. These hides are very cost-effective to produce and the animals make great use of them. Through the use of these humid hides my animals have always had perfect sheds, so I have not needed to gauge the humidity level. I have noticed that my animals tend to not drink from water bowls. I do still provide them, but they tend to prefer a light misting of the enclosure where they can drink off the rocks and glass. I lightly mist their enclosure twice a week to maintain the animals' hydration.

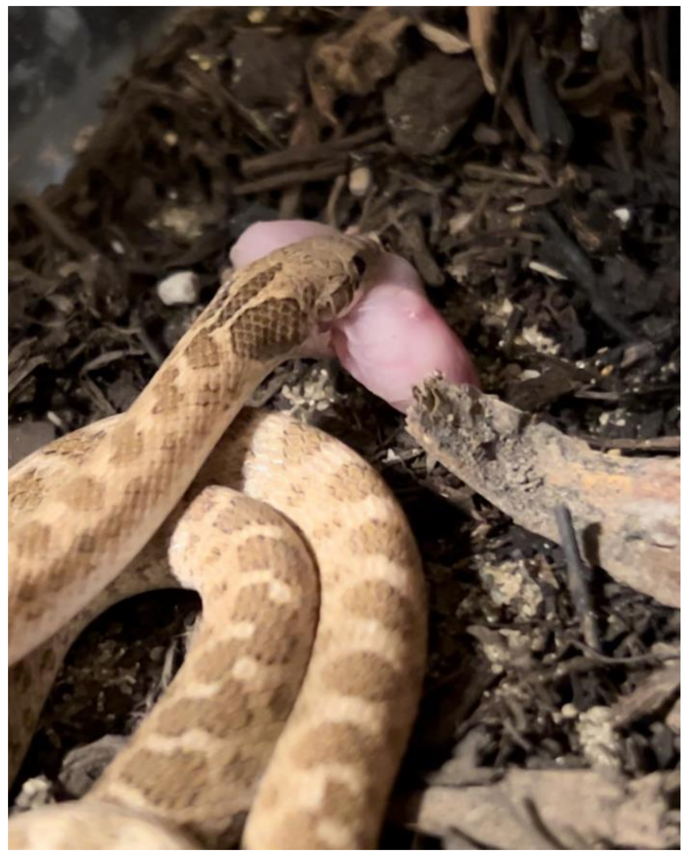
Diet

As mentioned at the beginning of this article, in the wild *H. jani* tend to prefer lizards, toads, and invertebrate prey. That being said, from my captive care I believe their diet may be a little more varied than we may think. In captivity I have had my animals take anoles (*Anolis* spp.), spiny lizards (*Sceloporus* spp.), Side-blotched Lizards (*Uta stansburiana*), house geckos (*Hemidactylus* spp.), chicken heart, rodents (hairless pinky mice), and even day-old button quail. As hatchlings, I assist-fed them mouse tails and chicken-flavored baby food. I would feed them every three to four days, alternating between each food source. Eventually it got to the point where I could just set the tail in the mouth of the snake and they would consume it themselves; however, I still needed to tube-feed the baby food. If I do get lucky and end up having success with my group and get hatchlings again, I would like to try the slurry used at MTOXINS Venom Lab for their hatchling Eastern Coral Snakes (*Mucrurus fulvius*), which is a Carnivore Care type mix providing a more complete diet.



My big female eating a piece of chicken heart scented with shed from a Rubber Boa (*Charina bottae*). I couldn't get this animal feeding originally, until it finally took a frozen/thawed Variable Sand Snake (*Chilomeniscus stramineus*). This animal hasn't given me issues since. Photo by the author.

Right now, all of my adult night snakes will take rodent prey; some more readily than others. I was having trouble getting one of my animals started and nearly lost it, until a friend reached out with some Side-blotched Lizards to use for scenting. These Side-blotched Lizards understandably were the "golden ticket" to switching these guys over. With *Hypsiglena* primarily feeding on less-dense prey in the wild, I am careful with my rodent usage in their diet. I tend to feed them more like a Rock Rattlesnake (*Crotalus lepidus*), maybe two to three meals a month; if an animal ends up getting a little chunky they go a little longer between meals. I did mention I use hairless rodents with these guys, and that again is due to them being primarily lizard feeders in the wild. I haven't seen any signs that they are not able to digest rodent hair, but I would rather not take a chance and have something bad happen. There is one animal in my collection which is a bit more heavyset in comparison to my other *Hypsiglena*, and for that reason she is given a supplemented chicken heart which has less fat in comparison to a rodent.



My male feeding on an unscented pinky mouse. This animal now has a feeding response similar to any Common Kingsnake (*Lampropeltis getulus* complex). It is really interesting watching them use their rear fangs when feeding. Photo by the author.

Brumation

There is nothing notably different with how I brumate my night snakes compared to my other colubrids. I stop feeding after Halloween (October 31st) and start slowly bringing their temperature down after two weeks of fasting. Just before Thanksgiving (fourth Thursday in November) they are at 50 degrees Fahrenheit until Valentine's Day (February 14th). Just after Valentine's Day, I begin a two-week process of warming them up, and I start feeding in March. I do brumate these animals together and have not noticed any issues with that.

Eggs upon laying, with my finger for reference. Photo by the author.

Incubation

One of the night snakes I had in the past came to me after somewhat recently mating in the wild. This female laid two eggs on October 3rd, 2022. After incubating at 80 degrees Fahrenheit for 63, days both eggs hatched out on December 5th. Should I get eggs again down the line, I will make sure to take proper weight and length records—something I am regretting skipping over now.

Hatchlings in comparison to my finger. Photo by the author.

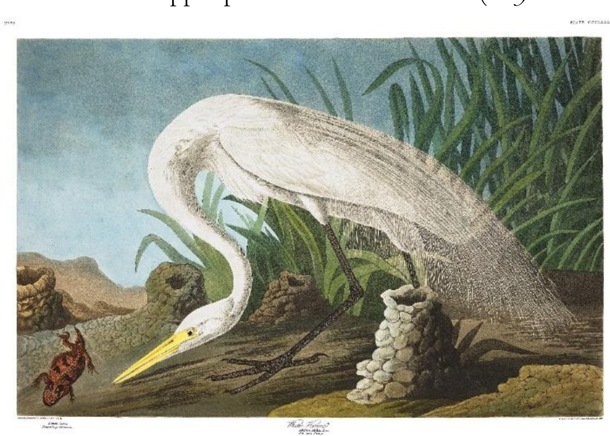
The First-Ever Commercially Available Native Plant Seed Mix for a Reptile

by

Dustin Rhoads

It's a comfortably cool morning in October 2022, and here I am along with ecological restorationists Emily Neiman, Leslie Boorhem-Stephenson, and Orion Weldon, planting 100 pounds of a brand-new native plant seed mix for Texas Horned Lizards (*Phrynosoma cornutum*) at country music singer Willie Nelson's ranch in Spicewood, Texas. I could say this journey started early 2010 when I read a book by entomologist Douglas Tallamy about how you can invite and sustain native wildlife and their food webs in your yard with native plants; but in reality, this journey started long before that—at least as far back as my birth. Then again, I could even make the case that this story can trace its roots to the spring of 1837.

Six months after Charles Darwin completed his *HMS Beagle* expedition, John James Audubon started his own from New Orleans, traveling aboard a US revenue schooner along the Gulf Coast. From at least late April thru mid-May, Audubon visited Galveston in the then-newly founded Republic of Texas to complete his legendary *Birds of America* illustrated work. Audubon encountered many birds in Galveston, and among them was the species that became the symbol for the society that would later bear his name—the Great Egret (*Ardea alba*). On plate 386 of his book is an ecological diorama that is vaguely (but not totally) familiar to my many wanderings around my youthful stomping grounds: an egret is there among "crawdad" (Cambaridae) chimneys—both familiar sights along the irrigation ditches around my neighborhood. Somewhat less familiar, however, is the tall bunchgrass—perhaps Eastern Gamagrass (*Tripsacum dactyloides*)—in the background. Even less familiar is another animal that Audubon had instructed engraver Robert Havell, Jr. to etch onto the copper plate—a Horned Lizard (*Phrynosoma* sp.).



Audubon's "White Heron" (aka Great Egret) from *Birds of America* (1838). JJ Audubon painted the egret from scenes on location in Galveston and Charleston. Image in public domain, courtesy of the National Audubon Society.

Fast-forward to 1980. I was born in the place where Audubon stepped off the USCGC *Campbell*, was greeted by the secretary of the Texas Navy, Samuel Rhoads Fisher, and encountered the ecological setting in which he painted the Great Egret and saw other natural wonders like Ivory-billed Woodpeckers (*Campephilus principalis*). By this time the area was a quite-changed, blue-collar Texas oil refinery town a few minutes' walk from Galveston Bay, in a typical working-class neighborhood meticulously turfed and routinely manicured with a monoculture of exotic St. Augustine carpet grass (*Stenotaphrum secundatum*) lawns and lined with mature and equally exotic Chinese Tallow trees (*Triadica sebifera*). But only two-and-a-half decades before my birth, my childhood neighborhood was a nearly treeless coastal prairie filled with a diversity of native bunchgrasses like cordgrass (*Spartina* spp.), bluestems (*Andropogon* spp.), paspalums (*Paspalum* spp.), Switchgrass (*Panicum virgatum*), muhlies (*Muhlenbergia capillaris*), and gamagrass (*Tripsacum* spp.)—species that had been there for tens of thousands of years, up until only a quarter century before I graced the scene. This was the setting that welcomed Audubon to my place of birth nearly 200 years ago but had ironically eluded me by just one score and five.

By the time I was born, the rich historical community of Texas native wild grasses had been replaced with a depauperate roster of foreign plants. The limited animal species I could find in my yard as a child matched the limited parameters of such trimmed, controlled landscaping. But even though they were few in diversity (consisting of only three natives encountered with any consistency), herps were my window into the exquisite beauty of wildness because, for a toddler, they were the most accessible—I could *touch* them. We had lime-colored Green Anole lizards (*Anolis carolinensis*) that could climb way up the trees, and down below the canopy shaded in the leaflitter, Gulf Coast Toads (*Incilius valliceps*) and Rough Earth Snakes (*Haldea striatula*) kept their small bodies cool and damp, buffered from the Texas heat. These were the few reptile and amphibian species that could be found on a regular basis because they didn't need to cross busy roads as often to find mates, shelter, or food. They could also subsist because they were the few herp species that could live on the paltry few invertebrates that could eke out a living in a conventional suburban yard landscaped with exotic plants and doused regularly with pesticides, herbicides, and fertilizers. We also had one single non-native herp species—Mediterranean Geckos (*Hemidactylus turcicus*)—which hunted moths on the outside walls at night. Encounters with these few taxa cemented my interest in nature and herpetology from a young age. I was grateful for their presence. But by the time I was four years old, I had become well-aware that this assemblage of reptiles and amphibians was not only different compared to what it was just a quarter century prior, it was also missing many key players.

Top photo: When I was a teenager in 1993, my family moved into a subdivision called "Swallows Meadow" on the north side of Texas City, a couple minutes' walk from Galveston Bay. A 40-acre remnant of coastal prairie crisscrossed with dirt bike trails was nestled between our neighborhood and an adjacent subdivision, North Village, connected by a single unpaved road. My younger brother, Austin, found Texas Horned Lizards (*Phrynosoma cornutum*) in that field in the late 1990s (and didn't tell me about it) while I was too busy being a teenager riding my bicycle to my girlfriend's house in North Village.
Bottom photo: Today, that 40-acre prairie remnant is now houses—a subdivision called Bayside Landing. There are no more Horned Lizards in Texas City, and they are all but gone in the entire Coastal Prairie ecoregion. I never saw a wild Texas Horned Lizard until I was 27. (*Google Earth* images, 1995 and 2023, respectively.)

The adults in my life—my parents, aunts, uncles, grandmothers, older neighbors, and friends' parents who grew up or lived in the same area and noticed my peculiar affinity for reptiles—told me of the animals they had encountered there in the 1950s and 1960s. Frequent visitors to their yards were such delightful guests as Box Turtles (*Terrapene carolina triunguis*), Rough Green Snakes (*Opheodrys aestivus*), Leopard Frogs (*Lithobates sphenocephalus*), Speckled Kingsnakes (*Lampropeltis holbrooki*), Glass Lizards (*Ophisaurus attenuatus*), Garter Snakes (*Thamnophis* spp.), and the coveted Texas Horned Lizards. The ecstasy conjured up in my mind of what it could've been like finding such "exotic" fare in such a pedestrian place as my backyard induced in me such a pleasing reverie that I would nearly salivate at the thought of it, and yet simultaneously seethe in envy at the squandered luck that my elders possessed but did not seem to appreciate as much as I did.

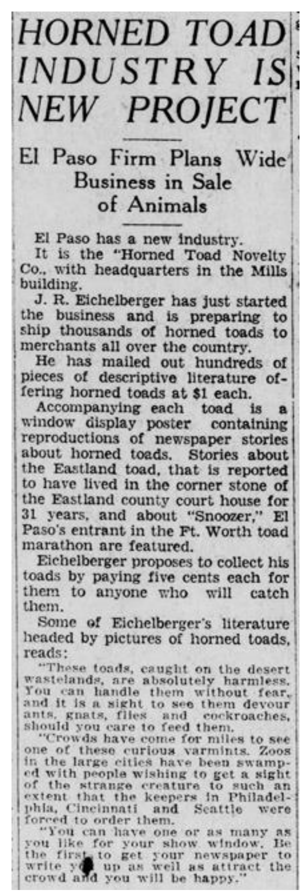
What was the cause of it? Where did these animals go? Could we ever get them back? Would Galveston County kids ever have them in their yards again? As I grew and eventually pursued degrees in ecology with a personal emphasis on studying Texas herps, these questions yet remained with me. It was shortly after finishing my undergraduate studies when I came across the aforementioned book, *Bringing Nature Home: How You Can Sustain Wildlife with Native Plants*, by Douglas Tallamy, that seemed to answer those questions. Tallamy's main argument was one of community: that in terrestrial environments all energy from the sun is converted to food energy by plants; that those plants don't *want* to get eaten and defend themselves with chemical and structural adaptive defenses; that the main reason that energy locked up in plants eventually travels throughout the food web *anyway* is via specially adapted insects that have evolved past the plants' defenses; that more than 90 percent of those herbivorous insects specialize in one or a few types of native plants that they have co-evolved with (and thus diversity of plants supports diversity of wildlife higher up the food chain); and that most wildlife species—including nearly all songbirds, practically all reptiles, absolutely all amphibians, and many fishes and mammals—depend on eating those native insects that dined on those native plants. If you could trace any animal species' food web back to the native plants that sustain its food web, you could figure out which species of native plants to sow to create that animal's habitat. Simple.

As I followed this train of thought, I found other books, web sites, and even commercially-available native plant seed mixes that were made to support birds, butterflies, and bees—in other words, species that could reasonably traverse a road or highway to get to the native plant in your yard. But there were none of these resources for reptiles, and it bothered me because even though non-volant terrestrial vertebrates face more challenges spreading across our highway-ridden landscapes, Tallamy's principle still applies the same for those organisms.

After graduating with a Masters of Science degree in Biology, studying camouflage in Texas Horned Lizards, from Texas Christian University in Fort Worth in December 2019, I wanted to work for a company or organization that would allow me to work on that problem. To learn my native plants, I volunteered at the Fort Worth Nature Center greenhouse and participated in Native Prairies Association of Texas workshops. Finally, in spring of 2022, I was hired as an ecological restorationist for Native American Seed (NAS)—a company whose mission is "helping people restore the earth" via native Texas wildflowers and prairie grasses. With guidance and help from NAS veteran employees, meetings with San Antonio Zoo staff, Texas Parks and Wildlife botanists, and others, I was able start working on putting together a native plant seed mix for a reptile that all Texans love—the Texas Horned Lizard. It's fitting, isn't it? Few reptiles epitomize better the symbology of the American West.

When we are trying to restore Horned Lizards, what we are actually trying to restore is the community that supported them. Horned Lizards are part of a grassland community. And in some ways, they are latecomers to the grassland community and specialize in filling a niche role. The wildlife communities that welcomed Texas Horned Lizards to the central grassland of North America are paradoxically analogous to the communities embodied by the Euroamerican settler land ethic regime that replaced them. As Ernest Callenbach said in his book *Bring Back the Buffalo! A Sustainable Future for America's Great Plains*:

> The first few trappers, explorers, and fur traders seemed to pose little threat. But after them came buffalo hunters, preying on the bison. Then came traders, making their living off the hunters and trappers and Indians. Then settlers took possession of the land to extract the accumulated richness of its soil, and gold miners invaded the Black Hills. Bandits, gunfighters, lawyers, and storekeepers arrived to live off the townsfolk and settlers. Finally, the military and its civilian helpers killed or rounded up the remaining Indians. …The water and land were first exploited by open-range cattle barons and their hired guns, but then came 'the plow that broke the Plains.' Wave after wave of farmers built sod houses, plowed, planted, watched their crops shrivel or blow away, and went bust. So in time a new variety of predator appeared on the Plains: not carnivore, not even human, but nonetheless voracious. Banks gobbled up the farms. Giant grain-trading corporations learned to manipulate commodity prices, producing waves of bankruptcies. Seed companies, fertilizer companies, and equipment companies racked up sales to failing farmers. These new predators were mostly legal fictions called corporations: self-replicating organisms driven by an ineluctable need to maximize profits, protected by law from personal liability claims. They steadily sucked money from the farmers, driving them to try ever harder to squeeze money from the land. For a time, the farmers fought back through populist political organizations. They even formed a new party and sent a few representatives to statehouses and to Washington, but their uprisings were soon beaten down.


HORNED TOAD INDUSTRY IS NEW PROJECT

El Paso Firm Plans Wide Business in Sale of Animals

El Paso has a new industry.

It is the "Horned Toad Novelty Co., with headquarters in the Mills building.

J. R. Eichelberger has just started the business and is preparing to ship thousands of horned toads to merchants all over the country.

He has mailed out hundreds of pieces of descriptive literature offering horned toads at $1 each.

Accompanying each toad is a window display poster containing reproductions of newspaper stories about horned toads. Stories about the Eastland toad, that is reported to have lived in the corner stone of the Eastland county court house for 31 years, and about "Snoozer," El Paso's entrant in the Ft. Worth toad marathon are featured.

Eichelberger proposes to collect his toads by paying five cents each for them to anyone who will catch them.

Some of Eichelberger's literature headed by pictures of horned toads, reads:

"These toads, caught on the desert wastelands, are absolutely harmless. You can handle them without fear, and it is a sight to see them devour ants, gnats, flies and cockroaches, should you care to feed them.

"Crowds have come for miles to see one of these curious varmints. Zoos in the large cities have been swamped with people wishing to get a sight of the strange creature to such an extent that the keepers in Philadelphia, Cincinnati and Seattle were forced to order them.

"You can have one or as many as you like for your show window. Be the first to get your newspaper to write you up as well as attract the crowd and you will be happy."

A news clipping from 1928. Even if you discount the horned lizard curio and short-term pet resale industry that permeated throughout the first sixty-plus years of the twentieth century, the fact that the tallgrass prairie is perhaps the most endangered ecosystem on the planet with less than 1% of it currently remaining is enough to understand 'what happened to the horny toad' in the historic tallgrass prairie range, i.e. all grasslands along and east of today's I-35 corridor. (From the *El Paso Evening Post*, May 26th 1928.)

The historian in me perceives that the disappearance of Texas Horned Lizards from the entire eastern half of their historic range—and many minor pinpoint extinction events throughout the whole range—is, when boiled down, merely a later-stage continuation of the dismantling and commodifying of the Great Plains that began 200 years ago. Horned Lizards are simply one of the last pieces of the prairie that held on. The land itself and its wild resources were seen as an endless commodity in "the era of the myth of inexhaustibility, the belief that the West is so vast, that the resources are so vast, that they can never be exhausted," as historical novelist Michael Punke has stated. Texas Horned Lizards were not immune. As the prairie of the "American Serengeti" was first de-buffaloed, de-prairie dogged, de-wolfed, and eventually de-grassed of its natives, only the smaller species that could hang on to the scantiest patches of degraded prairie remained: "Horny Toads" and the Prairie Seed Harvester Ants (*Pogonomyrmex* spp.) they ate were among these. However, they too eventually succumbed; first in the tallgrass prairie of the Southern Great Plains, but eventually too in the midgrass prairie as their populations have continuously receded westward. *You tell me*, dear reader—after the annihilation of the American Serengeti's big animals, what other Southern Great Plains megafauna could persist free-range at least a few more years on one-acre vacant sandlots, alleyways, and turnrows in smalltown America up-and-down the country's central breadbasket?

Horned Lizards are, in a way, johnnies-come-lately to the prairie. About 57 million years ago, before the Great Plains was a grassland, it was a humid forest dominated by palm trees and soon inhabited by primitive horses, rhinos, and camels with puny teeth adequate for eating soft leaves. Then about 25 million years ago the tooth anatomy of their descendants changed—they all developed taller, higher-crowned teeth more suited for eating a tough, wiry newcomer called *grass*. A few more million years pass and social insects that specialize on grasses (like Seed Harvester Ants and Grass Harvester Termites, *Tenuirostritermes cinereus*) arrived on the scene, and a few million years after that, a toad-bodied lizard that specialized on eating these insects arrived too.

Horned Lizards, as you know, are insectivores and there are, of course, other insects that "fill in the corners" of the Horned Lizard diet. In creating a native plant seed mix for Horned Lizards, the priority was to include as many native plant species as possible to attract and support as many insects as possible, not only staples of Horned Lizard diet like Harvester Ants/Harvester Termites but also insects that seem important outside the usual diet like other species of native ants (Formicidae) and sweat bees (Halictidae), for instance. In particular, there was a focus on including native plants with extrafloral nectaries, elaiosomes, and species that support aphids (Aphididae)—since all those are directly important in the food web of native ants.

We also included a wide variety of bunchgrasses (more than 30 species) of varying heights to dually provide not only food for Horned Lizard food but also to provide shade structures and cover under the grasses' skirts, while allowing bare ground micro-trails to exist between the root crowns of plants for Horned Lizards to bask, forage, and camouflage against the soil.

The Horned Lizard Habitat Mix™ from Native American Seed (*Junction, Texas*). Photo by Lindsey Ebert (Rehorning Texas).

When Audubon incorporated the Horned Lizard in his Great Heron watercolor painting, he borrowed a specimen from Richard Harlan (who had only a decade before described in a paper Texas Horned Lizards brought to Thomas Jefferson from the Lewis and Clark expedition). This was not a Texas Horned Lizard but a lizard that Audubon called a "*Phrynosoma orbiculare*" (*sic*; Holbrook in his 1838 book corrected Audubon's taxonomy—it was a Blainville's or Coast Horned Lizard, *i.e.*, *Phrynosoma coronatum*) from the west coast of the continent, collected in 1834 by Harvard lecturer Thomas Nuttall. I have not come across any notes that explicitly stated Audubon saw Horned Lizards when he visited Galveston in the spring of 1837, but I like to think he did. They would have been common. As a teenager in the 1990s living where Audubon visited, I became aware of a couple tiny remnant populations of Texas Horned Lizards in Galveston County coastal prairie still hanging on—one in a field between my neighborhood and an adjacent neighborhood (and that field is now houses), and the other in a field in Dickinson, Texas. Those populations have likely winked out, as have most of them east of the Interstate Highway 35 corridor. What can we do to restore them?

There are, of course, other inputs besides native plants that generate and maintain habitat suitable for Texas Horned Lizards. Keystone members of their community as well as natural wild processes build and maintain infrastructures so integral that if you remove them, it tripwires the entire community. History of the last 200 years in America has taught us that. Some of these members are species like Bison (*Bison bison*), Prairie Dogs (*Cynomys* spp.), Pronghorn (*Antilocapra americana*), Elk (*Cervus canadensis*), Wolves (*Canis lupus*), Dung Beetles (Scarabaeidae), and all other native insects in general. Some of these natural wild processes are things like lightning-ignited prairie fires, drought, and wind, but also raw materials like deep perennial roots and fertile soil. However, you first need the native plants if you hope to channel all of the sun's energy into that specific prairie community that supports Horned Lizards.

What we have created at NAS is a seed mix that will hopefully be a step in that direction of restoring this community, the current iteration comprising more than 83 species and varieties of native bunchgrasses and wildflowers of the Southern Great Plains. It's not a *perfect* solution in every situation or county, but it's a *good* solution, all Texas-native, and a damn sight better than Bermudagrass (*Cynodon dactylon*), Bahiagrass (*Paspalum notatum*), Buffelgrass (*Cenchrus ciliaris*), St. Augustine, or any of the other invasive, introduced, exotic carpet grasses choking the ecological landscape to death in a sea of monocultures. It's not only the first commercially available native plant "ecosystem in a bag" seed mix for Texas Horned Lizards, but also, to my knowledge, the first-ever commercially available native plant seed mix for a reptile (but I'd be happy to be wrong). My hope is that this will trend, so that we continue to learn the "native plant food web formula" for every species on earth. Onwards. And who knows? Maybe Willie Nelson will pen a song for the "Horny Toads" to one day return to his ranch.

Author's Note: An earlier version of this story appeared in the May 2023 issue of Phrynosomatics*, the newsletter of the Horned Lizard Conservation Society.*

SWCHR Laurence M. Klauber Memorial Summer Research Grant Update: Impact of Wildfire on Pathogen Prevalence in Sacramento Mountains Salamanders (*Aneides hardii*)

by

Zoe A. Hutcherson
Department of Biology, Eastern New Mexico University, Portales, New Mexico

Figure 1. Sacramento Mountains Salamander (*Aneides hardii*) from Otero County, New Mexico, USA, 2023. Photo by Drew R. Davis.

Study Background

The Sacramento Mountains Salamander (*Aneides hardii*; Figure 1, 2) is a unique lungless plethodontid salamander endemic to the Sacramento Mountain range within south-central New Mexico (Degenhardt *et al.* 1996). Across this range, it can be found under logs and bark in spruce-fir forests at elevations >2400 m (Figure 3; Scott and Ramotnik 1992; Scarpetta 2019). Though it is currently not afforded any Federal designation or protection, it is listed as state-threatened in New Mexico, and it remains of significant conservation interest due to its restricted range and population isolation (Osborne *et al.* 2019). Globally, amphibians face many threats, with major attention given to the role of pathogens in population and species declines (Collins and Storfer 2003; Wake and Vredenburg 2008). Major pathogens of concern include *Batrachochytrium dendrobatidis* (*Bd*), *B. salamandrivorans* (*Bsal*), and various strains of ranaviruses, which have all been contributed to observed declines in amphibians (Gray *et al.* 2009, 2023; Olson *et al.* 2021), though the occurrence of these pathogens is poorly understood (*Bd*, ranavirus) or presumed absent (*Bsal*) in New Mexico (Duffus *et al.* 2015; Johnson and Fritzler 2019; Grear *et al.* 2021). Given their potential threat, it is important to understand the prevalence and distribution of these pathogens in order to better understand the risk they pose to the Sacramento Mountains Salamander.

In addition to pathogens, amphibians face many other threats that may contribute to population declines. Environmental hazards, notably wildfires, can also pose formidable risks to amphibians (Hossack and Pilliod 2011). Wildfires, in particular, have been documented to exert significant adverse impacts on plethodontid salamanders, resulting in direct mortality and habitat degradation (Hossack and Pilliod 2011). However, studies have also highlighted instances where wildfires appear to mitigate the prevalence of *Bd* in other amphibian species (Hossack *et al.* 2013). These findings suggest complex interactions between wildfires and amphibian health, and a deeper understanding of these dynamics is needed to better understand how they affect Sacramento Mountains Salamanders.

Figure 2. Sacramento Mountains Salamander (*Aneides hardii*), from Otero County, New Mexico, USA, 2023. Photo by Drew R. Davis.

With evidence suggesting negative interactions between wildfires and pathogen prevalence, there is potential that vulnerable populations of Sacramento Mountains Salamanders may find relief from the diseases that have devastated amphibian populations globally due to regional wildfires. The core objective of this study is to examine the relationship between wildfire history and the prevalence of pathogens in the Sacramento Mountains Salamander. Furthermore, given the paucity of information on pathogens in Sacramento Mountains Salamanders, this study aims to help provide baseline levels of pathogen prevalence in this species.

Study Aims

The overarching aim of this study is to examine the intricate interplay between wildfire history and the prevalence and intensity of pathogens in corresponding populations of Sacramento Mountains Salamanders. To achieve this overarching objective, the study will pursue the following specific aims:

1. *Investigate correlations between wildfire incidents and pathogen prevalence.*—Through comprehensive field surveys and laboratory analyses, the study aims to elucidate the correlations between the occurrence of wildfire and the prevalence of three amphibian pathogens, including *Bd* and ranavirus, within populations of Sacramento Mountains Salamanders.

2. *Evaluate the impact of wildfire on salamander health and habitat quality.*—By conducting visual encounter surveys and employing non-destructive sampling techniques, the study seeks to assess the direct and indirect impacts of wildfires on salamander health and habitat quality, thereby shedding light on the mechanisms underlying the observed correlations between wildfires and pathogen dynamics.

3. *Establish baseline data on pathogen prevalence.*—In the absence of prior studies elucidating pathogen prevalence within Sacramento Mountains Salamander populations, this research endeavors to fill this knowledge gap by generating baseline data on pathogen prevalence and infection intensity.

Figure 3. Characteristic spruce-fir forest habitat within the Sacramento Mountains, New Mexico, USA, 2023. Photo by Drew R. Davis.

Methods

In summer 2023 I began a field study to assess the impact of wildfire on pathogen prevalence in Sacramento Mountains Salamanders within their native habitat in Otero County in south-central New Mexico. Sampling sites were chosen based on documented wildfire occurrences and visited to survey for salamanders. The study design encompassed visits to sites affected by wildfires approximately 5, 15, and 30 years ago, as well as sites with no recorded fire activity for over 50 years. Visual surveys were conducted by searching under fallen logs and rocks throughout the area (Figure 4). When salamanders were encountered, I collected microhabitat data, including air and soil temperatures, soil moisture, humidity, wind speed, canopy cover, and elevation. Salamanders were handled using clean nitrile gloves, weighed, measured, and photographed, and any outward signs of disease infection were recorded (Figure 5). After this, I swabbed individuals to collect a *Bd* swab sample and collected a small tail tissue clip (ca. 3 mm) for ranavirus assays. Gloves were changed and all tools were cleaned with a 10% bleach solution between each subject to prevent cross-contamination and the transmission of pathogens among sampled individuals. All samples were transported to the laboratory of Dr. Drew Davis at Eastern New Mexico University (ENMU), stored at -20° Celsius, and are awaiting analysis.

Figure 4. Photo of the author with a Sacramento Mountains Salamander (*Aneides hardii*). This individual was the first specimen to be captured during sampling.

Figure 5. Photo of the author processing a Sacramento Mountains Salamander (*Aneides hardii*).

Progress To Date

A total of 41 Sacramento Mountains Salamanders were collected in July and August 2023 (Figure 6). Microhabitat data was collected at every site where salamanders were detected and are summarized in Table 1. Swab and tissue samples were collected from each individual and are awaiting laboratory analysis. Renovations to the Roosevelt Science Center at ENMU have resulted in delays in conducting the laboratory analyses to screen for these three pathogens of interest. The renovations to the building are set to be completed in May 2024, with an anticipated move into the new laboratory over the summer. Therefore, I aim to run these samples and screen for these pathogens over Fall 2024.

Swabs and tissues will be extracted using a QIAGEN DNeasy Blood and Tissue Kit following the manufacturer's protocol. All samples will be run in triplicate and pathogen (*Bd*, ranavirus) detection will be determined via qPCR (StepOnePlus, Applied Biosystems). The *Bd* assay will target the internal transcribed spacer (ITS-1) ribosomal RNA gene as described by Boyle *et al.* (2004), while those used for ranavirus will target the major capsid protein (MCP) gene as described by Forson and Storfer (2006). Each plate will contain a negative control (DI water) and a standardized dilution series of gBlocks (IDT) that contains the target sequence of DNA to use in a standard curve. This method provides an estimate of the number of gene copies present in a sample of a particular pathogen. Samples will only be considered positive if two of three replicates are positive, and gene copies will be determined by averaging values from each amplified sample.

Figure 6. Juvenile Sacramento Mountains Salamander (*Aneides hardii*) specimen captured during fieldwork sitting on the author's pinky finger, conveying the size of juveniles.

Acknowledgements

I am deeply grateful to SWCHR for granting me this opportunity, which has been pivotal in advancing my research endeavors. This opportunity has not only provided crucial financial support but has also instilled in me a profound sense of gratitude and excitement for the future of my research. The support from SWCHR is a testament to their commitment to fostering diversity and inclusion within the scientific community, and I am deeply thankful. Additionally, I would like to express my heartfelt appreciation to the New Mexico Alliance for Minority Participation (NM AMP) for their unwavering support. Finally, I extend heartfelt gratitude to my advisor, Dr. Drew Davis, for his unwavering support and enthusiasm for this project. His guidance and dedication have been invaluable, and I am truly fortunate to have him as a mentor. I am inspired by his passion for research, and I am committed to upholding the same level of dedication throughout my academic journey. All sampling was conducted under a NM Department of Game and Fish Scientific Collecting Permit (#3872) and followed an approved ENMU IACUC protocol (2023-DAV-005).

TABLE 1. Mean, standard deviation (SD), minimum, and maximum values of microhabitat variables recorded from locations where Sacramento Mountain Salamanders (*Aneides hardii*) were sampled across Otero County, New Mexico, USA.

Variable	Mean	SD	Minimum	Maximum
Air temperature (°C)	25.13	2.09	21.78	27.11
Top soil temperature (°C)	16.99	3.81	13.67	25.06
Soil temperature (°C)	14.79	1.57	12.39	18.89
Soil moisture (%)	11.90	5.79	0.10	19.60
Humidity (%)	33.98	7.99	23.90	47.30
Wind speed (kmph)	1.76	1.13	0	3.06
Canopy cover (%)	95.27	2.14	92.72	98.96
Elevation (m)	2650.60	59.23	2493.87	2790.75

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The BUM Logger: an Automated Device for Remote Sensing of Soil Temperatures

by

Don Becker

On a small nature preserved in Eastern Iowa, there is a population of state-listed Yellow Mud Turtles (*Kinosternon flavescens*) and Ornate Box Turtles (*Terrapene ornata*). A small team of three people—Jim Scharosch, Josh Otten, and I—have been monitoring these turtles to better understand their behavior and habitat needs, which will allow us to better support the protection and preservation of these species in the future. One important part of this research is to make annual predictions of when the turtles will emerge from the ground to start their active season. Predictions were previously based on soil temperatures reported by a nearby weather station maintained by Iowa State University, but the data has some limitations. With a little creativity, a few purchases, and a couple spare parts, I was able to create a temperature logger so our team can more accurately monitor soil temperatures and develop predictions for turtle movement.

The first station deployed into the sand prairie. It was too short; future stations were taller. Photo by the author.

One of the data limitations that led to this project is that the data collected by Iowa State University is recorded on a location a few miles from our study site, and may have a different soil composition and sun exposure. To address this issue, we buried Thermochron iButton temperature loggers on our site at the same depths used by the weather station. The temperatures we recorded were similar enough to the weather station to be used for our predictions, but there was still a drawback to the data. Only four depths are used for monitoring: 4, 12, 24, and 50 inches (and the 24-inch depth had malfunctioned). With data only from those depths, we were not able to accurately interpolate the data to estimate the temperatures at depths in between the sensors.

Our first solution was just to bury more iButton loggers, but even though iButtons are relatively inexpensive, the costs can add up when you want to put them every 6 inches at multiple locations. They could easily eat up a decent portion of our equipment budget, and with a limited life span, they would have to be replaced every few seasons, adding more cost and labor. That could not be our only option.

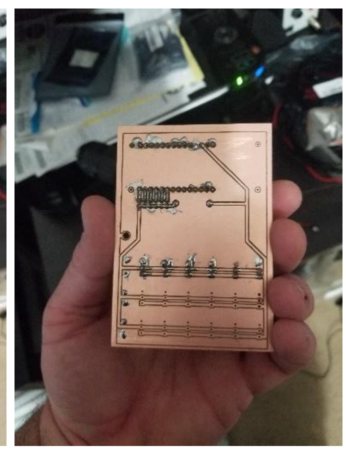
I had been working with Arduino boards along with some other similar development boards for another project, and had read posts on forums discussing using Arduinos to monitor soil temperatures in green houses. The DS18B20 sensors from Analog Devices appeared to be popular, had a lot of community support, were available in a water proof package with long cables, and because they use the 1-Wire protocol, many of them could be easily connected to an Arduino. If that wasn't enough, they are also cheap! I had a spare Arduino UNO sitting on a shelf, so I ordered some sensors and got to it.

The sensors I ordered came with JST-XH female connectors on the cables, which made them much easier to use. I soldered some male connectors to a protoboard along with a 4.7 KΩ resistor and a pin header, and started to write some code. I thought for sure it would take me a little while to figure out how to read data from the sensors, but the DallasTemperature Library made things very simple. When I ran the provided examples, everything just worked.

After reading the sensors, the next challenge was to determine how to store the data. The Arduino UNO does not have any form of storage built into it. It also doesn't have a real-time clock, so there is no way to tell what time it was when you read data from the sensors. Adafruit sells a data logging shield for Arduino UNO compatible devices that includes an SD card slot and a real-time clock. I was again surprised that the example code provided by the SD and RTCLib libraries just worked.

This all seemed too good to be true. I was able to take a bunch of off-the-shelf components, slap them together, and start logging data from the temperature sensors. So far, the Arduino had been powered by the USB cable connected to my PC. How would I power it when it was out at the turtle site—Batteries? Solar? I had to find out how much power it was using before determining how I was going to power it. While plenty of people have tested the power consumption of the Arduino UNO, I measured it myself with a multimeter, and found it was drawing 50 milliamps (mA) from the USB port, which was the same amount others had measured.

A standard AA battery has a capacity of 2,850 milliamp hours (mAh), meaning if a device was drawing 1mA of current from the batteries, it could do so for 2,850 hours. The Arduino UNO requires 50mA, so an AA battery would only last for 57 hours. You would need 4 AA batteries in series to provide the required voltage to the Arduino, but wiring them in series does not provide additional battery life. A standard D battery has a capacity of 10,000mAh, but that would still require 4 batteries, and would only last a bit over 8 days. I live close enough to the site that I could replace batteries every few days, but the cost of batteries would really add up over time. I considered using rechargeable 18650 batteries, but fortunately a new option was presented.



Front (L) and back (R) views of the first prototype board. Photos by the author.

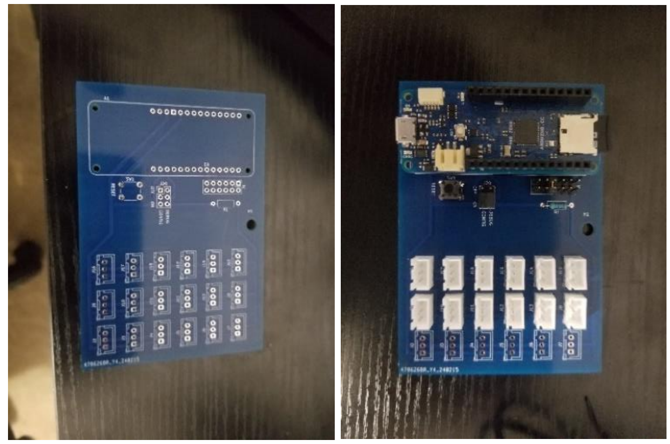
While investigating ways to reduce the power consumption of Arduino boards, I learned about the Arduino Low Power Library. This library allows you to put your Arduino board into a deep sleep with very minimal power consumption. This is the point in the development timeline that things stopped "just working." It turns out the library is not supported by AVR-based Arduino boards such as the Arduino UNO. I was a little discouraged, but not for long.

One board supported by the Low Power Library is the Arduino MKR Zero, which has a built-in SD card slot, real-time clock, and is designed to be powered by a Lithium Polymer (LiPo) battery. Additionally, it had a measured a power draw of only 15mA! That meant I could now use a 10,000mAh LiPo battery and power the board for 27 days. That alone was a big improvement, but I bought this board so I could use the Arduino Low Power Library, and put the board into a deep sleep. When the board initially went into deep sleep, it was only drawing 500 microamps (µA). It was another improvement, but not the final number. With a few recommended software tweaks I found on forums, I was able to get that down to 150 µA. With such a low power consumption while in a deep sleep state, a 10,000mAh LiPo battery could in theory power the board for over 1,000 days when logging the temperature every 5 minutes. In the real world, cold weather can impact battery performance, but 1,000 days is a pretty good start.

First test assembly in the weather proof box. Photo by the author.

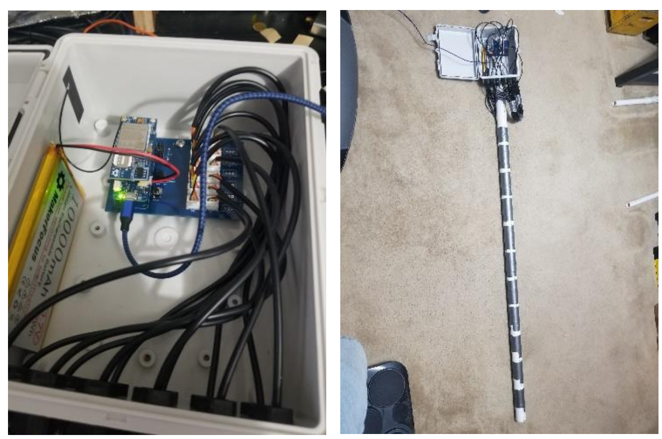
All I had to do now was connect sensors to the Arduino. Using KiCad I designed a small board that had 18 connectors for sensors, a place to solder in a pull up resistor, and a small jumper block so I could change which pin the Arduino used to communicate with the sensors, just in case a conflict came up in the future. To set the time on the board, I had to open a separate Arduino sketch, manually modify some time values, and then upload it to the board. After the time was set, I would have to upload the logging firmware again. There was a lot of code that was the same in the two sketches, but not an easy way to share code between them. After a bit of head scratching, I decided to have the code check one of the analog pins, and if it detected a high value, it would start a configuration shell instead of logging data. Now all I had to do was use a jumper wire to connect the 3.3V pin of the Arduino to A0 and hit the reset button, and I was presented with a configuration shell where I could set the time and manage the connected sensors.

While, I could have deployed the logger at this point, I still had a few more tweaks. I didn't want to worry about finding a jumper wire any time I need to configure the board. I also didn't like how the printed circuit board (PCB) that I cut out on the computer numerical control (CNC) machine looked. I went back to KiCad and added another jumper block. Now all I had to do was move a small jumper to "ON" and the board would enter the configuration shell. I also added a jumper to enable debugging that prevented the board from going into a deep sleep, which helped to debug issues and flash new code. Rather than cut the new board out on the CNC, I decided to have it manufactured by JLCPCB.



First manufactured PCB (L); assembled with components and Arduino MKR Zero (R). Photos by the author.

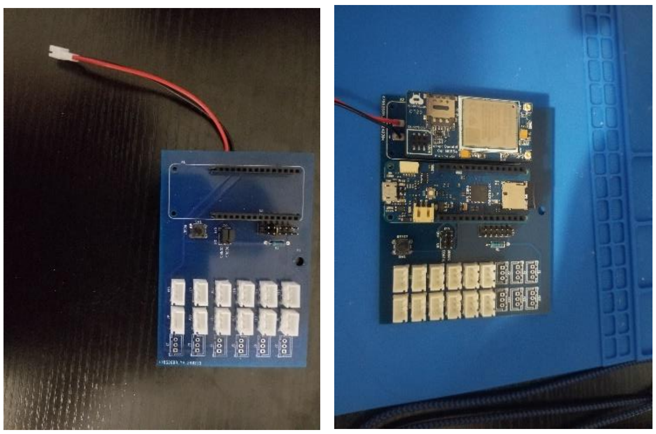
At this point, the logger was ready to be placed out in the sand prairie. I purchased a weatherproof box to protect the electronics, some cable glands to seal the gaps around the sensor cables, and a length of PVC pipe on which to mount everything. I drilled holes for the cable glands, fastened all the sensors at the right places on the PVC pipe, and… decided I wanted to make another change. The idea of putting a cellular modem on the logger had already crossed my mind. I started getting ads for cellular plans that were meant for "Internet of things" (IoT) devices, and realized they were much cheaper than I had thought. Now I just needed a modem.



First PCB assembled with modem in weatherproof box (L); station wired and attached to the PVC pole with tape (R). Photos by the author.

A quick search turned up a modem made by iot-bots.com that was designed to work with Arduino MKR boards. Surprisingly, I ordered it, plugged it in, ran sample code, and it worked. I could now remotely monitor our soils! I was elated, until I unplugged the Arduino from the USB port, and the modem stopped working. It turns out that the MKR Zero does not provide the required 5V power that the modem requires when they are powered by a LiPo battery. The LiPo battery has a maximum voltage of 4.2V when fully charged, and the Arduino does not have any hardware to boost the voltage up to 5V.

I was a bit discouraged again. In order to power the modem, I would need to power the Arduino via the USB plug or an external 5V power source. I still wanted to use the LiPo batteries, so I ordered some low-cost boost converters that would step up the voltage to 5V. When they showed up, I soldered some wires on to connect the batteries, and watched as the modem flashed its pretty blue lights, and a new entry had been added to the database on the web server where it was sending data. I was back in action…or so I thought.



First manufactured PCB with battery connector soldered on (L); second manufactured PCB assembled (R) including modem on the side. Photos by the author.

Every time I hit the reset button on the Arduino, the code would reset, the modem would send data, the board would go into deep sleep, and then the modem would not work again until I hit reset. Everything else was working fine. Every five minutes a new log entry was being added to the SD card, but not being sent to the web server. I spent a few days emailing back and forth with Max from iot-bots.com, who was very helpful and dedicated. While he thought I was not providing enough power to the modem, I didn't think that was the issue (and ended up being right), but the discussion was helpful in another way.

While going back and forth about the issue, Max told me that the modem had its own boost converter, and could be powered with as little as 3.3V. The modem had to use the 5V pin on the Arduino, because the 3.3V pin was not able to provide enough power. He suggested connecting the battery directly to the VIN pin on the Arduino to power everything. That would allow the modem to draw whatever power it needed from the battery directly, and the voltage regulator on the Arduino would still be able to power everything else. I soldered a battery connector to the underside of the PCB, and everything powered up, but the modem still wasn't working after deep sleep.

In the end the issue was not with the modem at all. Initially the board was using 500 µA of power when in deep sleep, and I was able to find some suggestions on forums to reduce the power consumption even further. One of those suggestions was to put all the digital pins on the Arduino into pull-up mode. I had done that so early in the process that I completely forgot about it. When I changed the code to not change the mode of the pins used by the modem, everything started working. Now it was actually time to put it out in the sand prairie.

The last thing to test was whether the modem could make a connection from the turtle site. On the day of installation, I met a few people on site and helped them with a few things while I waited to see if the modem could make a connection. In the office, it usually made a connection once every three or four tries. I watched my server as we worked. Twenty minutes, nothing. Thirty minutes, still no log entry. At forty minutes, I really started to worry. Then, around fifty minutes into waiting, it finally connected. Success!

Unfortunately, a few days after I put it out, I woke up and saw that six of the sensors were not providing temperatures. Why six? When I ordered the sensors, they came in packs of six. Did I get a bad batch? On the PCB, the connectors are in rows of six. Was there something wrong with my design or my soldering? Nope, none of the above. Mice decided to chew through the cables, and just happened to take out six. Funnily enough, Josh and I had discussed whether or not the mice would eat the cables and thought they wouldn't, out in an ecosystem with plenty of food. We were wrong.

Wire damage caused by mice (L); aluminum window screen used to protect wires from mice (R). Photos by the author.

We decided to leave the logger out with what was left of the sensors while I waited for replacement parts. Once we saw it was working, we decided we wanted a couple more. I wasn't happy with the power setup, so I made a couple more changes to the PCB. The modem no longer plugs into the Arduino directly. Instead, it sits next to it, and has an independent power source. Adding the modem reduces the battery life substantially. By having an independent power source, the logger can continue to log data to the SD card, even if the modem battery goes dead. The modem power source is also connected to one of the analog pins on the Arduino so I can monitor the battery level remotely. I also added a way to connect external power to the Arduino itself, and added a 5V regulator so we have other options for providing power. If the LiPo batteries don't like the cold weather this winter, we now have the option to use alkaline batteries or even a solar panel. When all the new parts arrived, a new station was wired up, but this time we protected the wire with some metal window screen. So far everything is working great, and the mice have not chewed through the cables. Hopefully it stays that way and we are able to successfully track soil temperatures and link the data to turtle activity in the coming years.

Source code and KiCad files for PCBs are available at: https://github.com/donfbecker/BUM-Logger

Rebuilt station with cables running inside the PVC pipe, deployed at second location. Note the taller height compared to the first station. Photo by the author.

SWCHR CODE OF ETHICS

As a member of the Southwestern Center for Herpetological Research, I subscribe to the Association's Code of Ethics.

Field activities should limit the impact on natural habitats, replacing all cover objects, not tearing apart rocks or logs and refraining from the use of gasoline or other toxic materials.

Catch and release coupled with photography and the limited take of non-protected species for personal study or breeding use is permitted. The commercial take and sale of wild-caught animals is not acceptable.

Collecting practices should respect landowner rights, including but not limited to securing permission for land entry and the packing out of all personal trash.

Captive-breeding efforts are recognized as a valid means of potentially reducing collection pressures on wild populations and are encouraged.

The release of captive animals including captive-bred animals into the wild is discouraged except under the supervision of trained professionals and in accordance with an accepted species preservation or restocking plan.

The disclosure of exact locality information on public internet forums is discouraged in most circumstances. Locality information posted on public internet forums usually should be restricted to providing the name of the county where the animal was found. When specific locality data is provided to one in confidence, it should be kept in confidence and should not be abused or shared with others without explicit permission.

Other members of the Association are always to be treated cordially and in a respectful manner.

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SWCHR
PO BOX 131262
SPRING TX 77393