CAPTIVE PROPAGATION AND HUSBANDRY OF REPTILES AND AMPHIBIANS

1991

Edited by Richard E. Staub

Northern California Herpetological Society
Special Publication #6

Proceedings
of the
Northern California Herpetological Society's

1991

CONFERENCE ON CAPTIVE PROPAGATION AND HUSBANDRY OF REPTILES AND AMPHIBIANS

Edited by
Richard E. Staub

Preface

The papers in this volume were presented at the Northern California Herpetological Society's (NCHS) Fifth Conference on Captive Propagation and Husbandry of Reptiles and Amphibians, which was held on February 16-18, 1991 at the University of California, Davis. These proceedings, the conference, and the month-to-month activities of NCHS are achieved solely through selfless contributions made by volunteers. An immense number of man-hours are required to plan and coordinate every phase of the conference. The below individuals were instrumental in the success of the conference, and were responsible for coordinating the speakers who presented papers at the conference, reserving the lecture facilities, organizing the registration, planning the banquet, reserving the motel arrangements, designing and producing the conference T-shirt, organizing advertisers for the program, providing transportation to and from the airport for the speakers, and ensuring coffee and donuts were there every morning. I am sure I have failed to remember someone's efforts that were worthy of recognition, and to this end, I apologize.

James Rexroth
Dan Aguilar
Dale DeNardo
John Berger
Sam Bacchini
Gerold Merker
Heino Kemnitz
Ralph Gowen
David Muth
Doug Kubo

The ultimate goal of this conference is to disseminate information on reptile and amphibian husbandry that will assist all herpetoculturists. These proceedings represent the endproduct of this endeavor. The papers were edited, formatted, and integrated into a congruous volume, while preserving the author's style and intention. Ralph Gowen and Dale DeNardo assisted greatly in the process of reviewing the papers for this volume. Tom Greek also contributed his skill as an artist by redrawing a figure in the text.

Again, I would like to thank everyone who contributed to the success of the conference and to all individuals and groups that support NCHS.

Rick Staub
Conference Chairman

On the Cover

Tusked chameleon, *Chamaeleo balteatus*, recently acquired by the Oklahoma City Zoological Park.
Photo by J. Tashjain.

On the Back Cover

Chinese crocodile lizard, *Shinisaurus crocodilurus*. Young lizards would frequently perch atop adults.
Photo by Robert George Sprackland.

Northern California Herpetological Society

The Northern California Herpetological Society is a federal and state non-profit corporation dedicated to conservation, education, and research of reptiles and amphibians. Founded in 1981 by a group of students at the University of California at Davis, NCHS has grown into a society with members across the United States. NCHS held its first conference on the Captive Propagation and Husbandry of Reptiles and Amphibians in 1983 and has held a conference every two years since.

Membership in NCHS is open to anyone with an interest in reptiles and amphibians. Members of NCHS receive a monthly newsletter which contains primarily original articles written by the society's members. NCHS holds a general meeting on the first Friday of every month on the campus of the University of California, Davis.

Proceedings are available from each conference. For current prices and further information about NCHS write to:

Northern California Herpetological Society
P. O. Box 1363
Davis, California 95617-1363

Table of Contents

Two speakers presented papers at the conference, but had previously published their data. Below are the references for their publications.

David J. Morafka, Department of Biological Sciences, California State University, Dominguez Hills.

> Morafka, D.J. and C.J. McCoy. 1988. The Ecogeography of the Mexican Bolson Tortoise (*Gopherus flavomarginatus*): Derivation of its Endangered Status and Recommendations for its Conservation. IN: Annals of Carnegie Museum. Carnegie Museum of Natural History. Pittsburgh, Pennsylvania. pp 5-71
>
> Adest, G.A., G. Aguirre L., D.J. Morafka, and J.V. Jarchow. 1989. Bolson Tortoise (*Gopherus flavomarginatus*) Conservation: I. Life History, II. Husbandry and Reintroduction. Vida Sylvestre Neotropical 2(1):7-20

Dave Blody, Curator of Herpetology, Fort Worth Zoo, Fort Worth, Texas

> Blody, D.A. and D.T. Mehaffey. 1989. Reproductive Biology of the Annulated Boa, *Corallus annulatus*, in Captivity. Int. Zoo Yb. 28:167-172

Hugh McCrystal from the Arizona Sonora Desert Museum in Tuscon, Arizona presented his data on montane rattlesnakes. His research is being funded by the Arizona Game and Fish Department and should be published soon.

EGG RETENTION IN REPTILES

Dale DeNardo, D.V.M.

University of California, Berkeley
Berkeley, CA 94720

INTRODUCTION

Captive propagation of reptiles has increased dramatically in the last ten years, and, consequently, so has the frequency of egg retentions. Reports on dystocia in reptiles are numerous, however they are scattered throughout the literature. This paper reviews the literature on reptile dystocias, and summarizes much of what is known about the causes and treatments of this problem.

PREVALENCE

Unfortunately, dystocias are a common medical problem in reptiles. In a survey of approximately 1600 reptiles at various facilities, Lloyd (1990) reported that dystocias occur in approximately 10% of the captive reptile population annually, with 42% of the dystocias in his survey occurring in snakes, while 39% were in turtles, and 18% were in lizards. The majority of published reports and cases seen by the author involve snakes; however, there is no indication of what fraction of captive reptiles are snakes. Therefore, it is unclear whether the high frequency of dystocias in snakes is a result of snakes being more prone to dystocia or snakes being bred more commonly. Similarly, while most dystocias occur in egg-layers rather than live-bearers, it is probably a reflection of the numbers of egg-laying animals versus live-bearing animals being bred rather than a true indication of prevalence.

While reports of dystocias are numerous for snakes and chelonians (see below), the literature on dystocia is extremely limited for lizards and non-existent for crocodilians. This does not mean that dystocias do not occur in these reptiles. In fact, the most common species of reptile presented to the author for dystocia has been the green iguana (*Iguana iguana*).

ETIOLOGY

Dystocia can be caused by a variety of factors, but in most cases the cause is not determined. Determining the exact cause of a dystocia will allow one to correct the problem and decrease the risk of recurrence. Factors leading to dystocia cover a vast gamut of situations, but they can be usefully divided into two broad groups - obstructive and non-obstructive.

Obstructive Dystocias

An obstructive dystocia is a result of a anatomical inability to pass one or more eggs or fetuses through the oviduct and cloaca. The causative defect may be a fetal or maternal abnormality. Fetal abnormalities include oversized or malformed eggs/fetuses. Such fetal abnormalities will cause the deformed egg and all eggs cranial to it in the oviduct to be retained. Maternal abnormalities include a misshapen pelvis, an oviducal stricture, or a non-reproductive mass such as an abscess or cystic calculi. Additionally, obstructive dystocias may be a result of a complication during oviposition. If two eggs try to pass through the pelvis simultaneously or if an egg fractures within the oviduct, a dystocia may result.

Non-Obstructive Dystocias

While it is easy to understand dystocias caused by obstructions, a large number of dystocias occur where no obstructive etiology can be found. In these cases, the eggs or fetuses removed appear normal, the oviduct is normal, and there are no other masses in the coelomic cavity. These non-obstructive dystocias have been attributed to several causes, although very few have been shown to truly cause dystocia. Many dystocias may be a result of poor husbandry. Improper nesting site, improper temperature, malnutrition, and dehydration may all lead to dystocia. Culturing retained ova or fetuses often reveals one or more bacteria, but whether these organisms caused the dystocia or whether they invaded as a result of the dystocia is difficult to determine.

Another potential cause of dystocia is poor physical condition of the female (Grain and Evans, 1984). Captive reptiles are extremely sedentary when compared to their wild counterparts. This decrease in activity may lead to poor muscle tone. Oviposition requires a substantial muscular effort, and if the female is in poor condition due to inactivity, she may fail to complete the task. Such an etiology could explain many dystocias in egg-laying snakes. These snakes normally pass the majority of their clutch, but retain the last egg or two, which, when removed, appear normal. While poor condition may be a major factor in dystocias, like many implicated causes, its significance has not been tested.

DIAGNOSIS

Diagnosis of dystocia is usually simple in egg-laying snakes. The female usually has just laid some eggs, but there still is one or more large swellings present in the lower half of the body. Remember, however, that there are a multitude of conditions which present simply as a swelling (see Russo, 1987, for review). Funk (1987) reported a speckled kingsnake (*Lampropeltis getula holbrooki*) with a renal tumor which was miss-presented as a dystocia. Similar cases have been presented to the author.

Live-bearing snakes are more difficult to diagnose, since it is difficult to accurately predict a parturition date and it is more common for the entire clutch to be retained. This difficulty in distinguishing a true dystocia from what may just seem to be a long pregnancy, usually results in delayed diagnosis and often a poor prognosis. Prolonged straining or cloacal prolapse may indicate unsuccessful attempts to pass fetuses (Marcus, 1981). The use of ultrasonography has recently improved the ability to diagnose dystocia in viviparous snakes, since it allows non-invasive determination of fetal condition. If the fetuses are dead, timely steps can be taken to remove them.

It can also be quite difficult to determine true dystocia in lizards. Frequently iguanas are presented as "impacted", since they are off-feed and, to the owner, appear ready to explode. The best judge of dystocia in lizards is their attitude. If the lizard is behaving normally other than not eating, there is little likelihood of dystocia. Typical signs of dystocia in lizards include depression, inactivity, and lack of interest by the lizard in its surroundings. If these signs are detected, immediate steps should be taken to relieve the problem. While snakes can tolerate a dystocia for extended periods of time (with variable local damage occurring), dystocia in lizards often leads to rapid death. Many lizards, including iguanas can produce a clutch of eggs without the presence of a male. These clutches are frequently infertile and often retained.

Egg retention in chelonians is nearly impossible to detect without the benefits of radiographs. Even with X-rays, the decision as to whether the eggs are retained or not is a difficult one. If eggs are present and any clinical signs such as depression, straining, or swelling are noted, egg removal efforts should be instigated. Glassford and Brown

(1977) reported the elimination of respiratory distress in a box turtle (*Terrapene carolina*) by removing eggs which were identified by radiographs.

TREATMENT

Treatment of dystocias in reptiles is dependent on the species of reptile, the type of dystocia, and the duration of the dystocia.

Physical Manipulation

One of the more common methods used to attempt removal of retained eggs or fetuses in snakes is manual palpation. Eggs are literally forced out by firmly running a finger down the ventrum of the snake. While some hobbyists claim reasonable success with this method, the risks are substantial. Common complications seem in snakes where manual palpation was used singly or in combination with other treatments to attempt removal of eggs include oviducal rupture (Barten, 1985) and oviducal prolapse (Wagner, 1984; Brown and Martin, 1990). These complications often lead to death (Brown and Martin, 1990).

If the egg can be visualized through the cloaca, attempts can be made to extract the egg using some form of guide (see Wagner,1984, for description). Similar procedures used by others have had deleterious outcomes including egg rupture (Jordon and Kyzar, 1978, in a tortoise) and death (Millichamp et al, 1983, in a snake), so extreme care should be taken.

Hormonal Stimulation

Commonly, the first approach to treating dystocias is to stimulate oviducal contractions by using hormones of the posterior pituitary. Clausen (1940) first noted that extract of the posterior pituitary would cause parturition in viviparous snakes which were at a late stage of pregnancy. Similarly, Yntema (1964) used posterior pituitary extract to stimulate oviposition in snapping turtles (*Chelydra serpentina*). Pituitary extract was first used clinically to remove retained infertile ova from a Jamaican boa (*Epicrates subflavus*) (Huff, 1976). Since then, purified oxytocin, the posterior pituitary hormone which naturally causes uterine contraction in mammals and had been shown to stimulate oviposition in lizards (LaPointe, 1964), has been routinely used to remove retained eggs or fetuses from reptiles.

In his survey, Lloyd (1990) reports oxytocin has been used in doses ranging from 5-30 International Units (IU) per kilogram; however doses as low as 1 IU/kg have been effective in turtles (Glassford and Brown, 1977). The drug may be given intramuscularly or intraperitoneally. The efficacy of oxytocin in removing retained eggs or fetuses is variable depending on the species and the duration of the retention. Oxytocin is much more effective when given within the first 48 hours of dystocia. Even within this period, the author has had less than a 50% effectiveness in snakes. Lizards respond slightly better, while chelonians respond quite well (>90% effectiveness).

A multitude of adjunct treatments have been suggested to increase the efficacy of oxytocin, including simultaneously treating the patient with calcium or reproductive steroids (e.g. estrogen, progesterone) and retreating with a second dose of oxytocin 20-60 minutes after the first dose. The efficacy of these adjunct treatments has not been thoroughly studied; however, estrogen and progesterone both failed to have an effect on isolated lizard oviducts treated with oxytocin (LaPointe, 1969). Temperature does influence the effect of oxytocin on oviducal muscles, with maximum response occurring at the animal's preferred body temperature (LaPointe, 1977).

More recently, arginine vasotocin, the natural oxytocin equivalent for reptiles, has been used in the treatment of dystocias. Reptile oviducts are 10 times more sensitive to arginine vasotocin than to oxytocin (LaPointe, 1977).

Lloyd (1990) reported that arginine vasotocin (at 0.01-1.0 micrograms/kg, IV or IP) was 73% effective, with 18% of the cases having been previously unsuccessfully treated with oxytocin. While there is little doubt that arginine vasotocin is more effective at stimulating oviducal contraction in reptiles, it does have its drawbacks. Currently, arginine vasotocin is only available as a research drug (Sigma #V0130), and is relatively expensive. Additionally, arginine vasotocin is extremely unstable, requiring that it be kept frozen in its powdered form. Once reconstituted in sterile water or saline, it must be used within 6 weeks if frozen or 5 days if refrigerated (Lloyd, 1990).

It must be remembered that these posterior pituitary hormones merely cause oviducal contraction, not "egg removal". Using oxytocin or arginine vasotocin on animals which have an obstructive dystocia can have detrimental consequences. Increasing the pressure on eggs or fetuses which cannot proceed down the oviduct may cause egg fracture, oviduct rupture, hemorrhage, and even death.

Percutaneous Ovocentesis

A second treatment for dystocia in egg-laying snakes is to aspirate the contents of the retained egg by inserting a sterile needle through the ventrum of the snake and into the egg. Caution must be taken not to contaminate the coelomic cavity with egg contents. Removing the yolk from the egg will decrease the size of the egg, therefore making it easier for the snake to pass. As with the use of posterior pituitary hormones, aspiration must be performed shortly after the onset of dystocia (usually within 48 hours). With time, the egg contents solidify and become impossible to extract with a needle.

Once the egg's contents have been removed, the snake can be allowed to pass the egg naturally with oviposition occurring from a few hours to a few days after aspiration. To assure a more rapid oviposition of the aspirated egg, oxytocin can be administered after aspiration (Peters and Coote, 1977; Grain and Evans, 1984).

A similar procedure has been used successfully in turtles when the egg can be visualized through the cloaca. The egg is aspirated through the cloaca, and the collapsed egg is then removed via a pair of forceps (Rosskopf and Woerpel, 1982).

Surgery in Snakes

If retained eggs or fetuses cannot be removed with the use of hormones and or aspiration, then surgery is indicated. Surgical removal of fetuses was first performed in snakes for embryological studies (Clark, 1937; Zehr, 1962). Since then, salpingotomies (i.e. caesarian sections) have been performed routinely to correct dystocias in snakes (Hime, 1976; McNiell, 1977; Peters and Coote, 1977; Vandeventer and Schmidt, 1977; Mulder et al, 1979; Patterson and Smith, 1979; Millichamp et al, 1983; Grain and Evans, 1984; Barten, 1985; Loomis and Smith, 1987; Brown and Martin, 1990; Lichtenhan, 1991).

Once anesthetized, but prior to surgery, gentle manual palpation may be used to attempt to remove the retained eggs or fetuses. The relaxation of the oviducal sphincter due to the anesthesia decreases the risk of injury from manual palpation and increases the likelihood of success. Never the less, care must be taken to avoid prolapse or rupture. Most surgeries are simple, using a ventro-lateral incision between the first and second scale row of the skin overlying the retained egg or fetus. The oviduct is then incised and the egg/fetus is removed. Additional eggs/fetuses may be removed through the same incision or additional incisions may be required. The oviduct, muscle, and skin are then closed using appropriate suture patterns. Recovery from surgery is relatively quick when gas anesthetics such as isoflorane are used.

While surgery has successfully corrected dystocias of up to 27 months (Patterson and Smith, 1979), it is advised that surgery be performed as soon as possible. Even if the female behaves normally, local changes around the retained eggs or fetuses may be causing damage which can effect future reproduction and even survival of the female (Hime, 1976). Mulder et al (1979) report eggs adhering to the oviduct in a snake with a 6 month retention, while the author has seen adhesions form in as little as 2 weeks. If severe, adhesions may require partial or complete removal of one or both oviducts. Obviously, this additional procedure will reduce or eliminate any future breeding potential.

Surgery in Lizards

Surgery to remove eggs in lizards is very similar to snakes. The procedure often requires two surgical incisions, one on each ventro-lateral aspect. Midline incisions are discouraged to avoid cutting the major central vein that runs just under the skin. Egg retentions involving 20-30 eggs are common in green iguanas (*Iguana iguana*), thus making a caesarian section an extended procedure. If there is no intention of purposely breeding the lizard in the future, it is highly recommended to perform an ovariohysterectomy (i.e. spay) on the lizard rather than a caesarian section. This will greatly shorten the surgery time and avoid recurrence of the problem in the future. Simply preventing a female iguana access to a male will not guarantee that she will not produce eggs again.

Surgery in Chelonians

Surgery can also be employed to remove retained eggs from chelonians. The procedure is less common since oxytocin is so effective in these animals. However, if oxytocin fails or if the eggs are oversized, malformed, or fractured, surgery is required. The shell of chelonians makes surgical removal of eggs much more complicated than the equivalent procedure in snakes. Usually, a bone saw is used to remove a flap from the plastron to gain access to the coelomic cavity. The eggs are then removed in a fashion similar to snakes, and the flap is re-attached using fiberglass and resin (see Frye and Schuchman, 1974, for description). Healing from these surgeries is relatively slow, requiring several days before the chelonian exhibits normal behavior.

An alternative to removing a section of the plastron is to perform the surgery through the inguinal region, just cranial to the hind limb (see Brannian, 1984, and Page, 1985 for descriptions). Access into and out of the coelomic cavity is limited in this procedure, sometimes making it difficult to locate and remove the eggs. However, this soft-tissue approach greatly shortens the healing time.

VIABILITY OF RETAINED EGGS/FETUSES

While countless numbers of retained, and often fertile eggs have been removed from reptiles, there have been only two known incidents of successful incubation of these eggs. Frye (1981) reported the hatching of a red-footed tortoise (*Geochelone carbonaria*) egg which was surgically removed, and the author successfully incubated 1 of 6 eggs removed during necropsy of an eastern box turtle (*Terrapene carolina*) which died acutely. It is uncertain whether either of these eggs were truly retained. Eggs removed with oxytocin from gravid, but not retaining, female lizards and turtles are commonly incubated successfully. The author knows of no reports of snake eggs successfully hatching after hormonal or surgical removal.

There has been success removing live fetuses via caesarian section. Chiodini and Sundberg (1980) report an 80% survival of fetuses removed from the water snake (*Natrix sipedon*) during experimental procedures. The only report of successful surgical recovery of live fetuses from a true dystocia was that of Vandeventer and Schmidt (1977) who removed 9 live and 12 dead neonatal gaboon vipers (*Bitis gabonica*) from a female which approximately a week earlier produced 1 live and 2 dead neonates.

FUTURE REPRODUCTION AND RECURRENCE

The prognosis for future reproduction is good provided no complications were noted during the dystocia and the reproductive tract was left intact. Successful reproduction the year following a surgical correction of a dystocia has been reported in both egg-laying (Brown and Martin, 1990; Lichtenhan, 1991) and live-bearing snakes (Loomis and Smith, 1987). The author has also noted viable second clutches produced within months of correcting dystocias with either oxytocin or egg content aspiration.

While the potential for future reproduction is good, it seems that snakes which have retained eggs in the past are more likely to retain eggs in the future than snakes that have normal oviposition or parturition. The author knows of a snake that has retained a single egg from a clutch four years in a row. Each year the egg has been successfully removed with either oxytocin or aspiration, with one year the snake producing a second clutch within 2 months without a dystocia. The increase incidence of repeat dystocias could simply be related to failure to correct the causative problem, but further work is needed before any conclusions can be made.

Females which have non-correctable anatomical problems which lead to dystocia should not be allowed to breed again. The female should be spayed or denied access to a fertile male. As stated earlier, denying a female lizard access to a male does not insure that the female will not produce another clutch of eggs in the future. Also, male snakes which have been castrated or had a vasectomy have been shown to have mature sperm in their ductus deferens for at least three months post-operatively (Zwart et al, 1979).

CONCLUSIONS

Dystocia is a common problem seen in reptile reproductive programs. Fortunately, there exist several options to safely correct the problem. The sooner a dystocia is treated the less likely secondary complications will develop and the easier the treatment will be. Further work is necessary to better understand the causes of dystocia in order to hopefully reduce the relatively high incidence of dystocias seen in captive reptiles today.

LITERATURE CITED

Barten, S.L., 1985. Oviductal rupture in a Burmese python (*Python molurus bivittatus*) treated with oxytocin for egg retention. Journal of Zoo Animal Medicine 16:141-143.

Brannian, R.E.. 1984. A soft tissue laparotomy technique in turtles. JAVMA 185(11):1416-1417.

Brown, C.W. and Martin, R.A., 1990. Dystocias in snakes. The Compendium - Small Animal 12(3):361-368.

Chiodini, R.J. and Sundberg, J.P., 1980. Caesarian delivery in the snake. Veterinary Medicine / Small Animal Clinician October, 1980:1605-1608 .

Clark, H., 1937. Embryonic series in snakes. Science 85:569-570.

Clausen, H.J., 1940. Studies on the effect of ovariotomy and hypophysectomy on gestation in snakes. Endocrinology 27:700-704.

Cooper, J.E. and Jackson, O.F., 1981. Disease of the Reptilia, Vol. 2; Academic Press, New York. pp.54 5,577.

Frye, F.L., 1981. Biomedical and Surgical Aspects of Captive Reptile Husbandry; Vet Med Publishing Co, Edwardsville, Kansas. pp.297-307.

------------. and Schuchman, S. M., 1974. Salpingostomy and caesarian delivery of impacted ova in a tortoise. Veterinary Medicine / Small Animal Clinician April, 1974:454-457.

Funk, R.S., 1987. Heminephrectomy and nuclear scanning in a speckled kingsnake (*Lampropeltis getulus holbrooki*) with a kidney tumor. IN Rosenberg, R.J. (Ed). Proceedings of the 11th International Herpetological Symposium. pp 137-138.

Glassford, J.F. and Brown, K., 1977. Treatment of egg retention in a turtle. Veterinary Medicine / Small Animal Clinician October, 1977:1641-1645.

Grain, E. Jr. and Evans, J.E. Jr., 1984. Egg retention in four snakes. JAVMA 185(6):679-681.

Hime, J.M., 1976. A report of the scientific sessions of the Zoological Society of London. Journal of Zoology 178:453.

Huff, T., 1976. The use of oxytocin to induce labour in a Jamaican boa (*Epicrates subflavus*). International Zoo Yearbook 16:82.

Jordon, R.D. and Kyzar, C.T., 1978. Intra-abdominal removal of eggs from a gopher tortoise. Veterinary Medicine / Small Animal Clinician August, 1978:1051-1054.

LaPointe, J., 1964. Induction of oviposition in lizards with the hormone oxytocin. Copeia 1964:451-452.

---------------. 1969. Effects of ovarian steroids and neurohypophyseal hormones on the oviduct of the viviparous lizard, *Klauberina riversiana*. J. Endocr. 43:197-205.

---------------. 1977. Comparative physiology of neurohypophysial hormone action on the vertebrate oviduct-uterus. American Zoologist 17:763-773.

Lichtenhan, J.B., 1991. Caesarian section (CBAC) in an amelanistic corn snake. Reptile and Amphibian Magazine Nov/Dec 1 91:16-19.

Lloyd, M.L., 1990. Reptilian dystocias review - causes, prevention, management and comments on the synthetic hormone vasotocin. 1990 Proceedings American Association of Zoo Veterinarians 290-296.

Loomis, M. and Smith, R., 1987. Fertility following caesarian section in an Aruba Island rattlesnake (*Crotalus unicolor*). International Zoo Yearbook 26:187-188.

Marcus, L.C., 1981. Veterinary Biology and Medicine of Captive Amphibians and Reptiles, Lea and Febiger, Philadelphia. pp.195-196.

McNeill, R., 1977. Dystocia in a snake. The Veterinary Record June 18, 1977:536

Millichamp, N. J., Lawrence, K., Jacobson, E.R., Jackson, O., and Bell, D., 1983. Egg retention in snakes. JAVMA 183(11):1213-1218.

Mulder, J.B., Hauser, J.J., and Perry, J.J., 1979. Surgical removal of retained eggs from a kingsnake (*Lampropeltis getulus*). J. Zoo Animal Medicine 10:21-22.

Page, C.D., 1985. Soft-tissue celiotomy in an Aldabra tortoise. J. Zoo Animal Medicine 16:113-114.

Patterson, R.W. and Smith, A., 1979. Surgical intervention to relieve dystocia in a python. The Veterinary Record June 16, 1979:551-552.

Peters, A.R. and Coote, J., 1977. Dystocia in a snake. The Veterinary Record May 14, 1977:423.

Rosskopf, W.J. Jr. and Woerpel, R., 1982. Treatment of an egg-bound turtle. Modern Veterinary Practice August, 1982:644-645.

Russo, E.A., 1987. Diagnosis and treatment of lumps and bumps in snakes. Compendium Small Animal 9(8):795-806.

Vandeventer, T.L. and Schmidt, M., 1977. Caesarian section on a western Gaboon viper (*Bitis gabonica rhinoceros*). International Zoo Yearbook. 17:140-142.

Wagner, E. and Foster, J., 1984. Removing retained eggs from snakes. IN McKeown, S. and Caporaso, F. (Eds). Proceedings from the 8th Annual International Herpetological Symposium. pp 112-113.

Yntema, C.L., 1964. Procurement and use of turtle embryos for experimental procedures. Anatomical Record 149:577-586.

Zehr, D.R., 1962. Stages in the normal development of the common garter snake, *Thamnophis sirtalis sirtalis*. Copeia 2:322-329.

Zwart, P., Dorrestein, G.M., Stades, F.C., and Broer, B.H., 1979. Vasectomy in the garter snake (*Thamnophis sirtalis radix*). J. Zoo Animal Medicine 10:17-21.

MEDICAL CARE OF TURTLES AND TORTOISES

Douglas R. Mader, M.S., D.V.M.

Long Beach Animal Hospital
3816 E. Anaheim St.
Long Beach, CA 90804

INTRODUCTION

An important aspect of the medical care in any species is a fundamental knowledge of the "normal animal". If a caretaker does not know the normal for a given species, then there is no way that they will be able to recognize the abnormal. A thorough understanding of an animal's biology and life history are basic to diagnosing problems and establishing treatments.

In exotic animal medicine, the vast majority of my diagnoses are management related, as opposed to disease related. In the same thought, the bulk of my *prescriptions* are not drugs, but rather *instructions* on how to correct deficiencies in husbandry. That does not mean that drugs and diagnostic aids are not used. They are, but they are usually secondary to correcting fundamental problems in husbandry.

With that preface, it should be noted that the bulk of this discussion will focus on understanding what to look for in the normal animal, thus greatly enhancing the recognition of early warning signs of disease.

BASICS

Turtles, or chelonians, as the group is properly called, range in size from as small as only a few grams in weight and several centimeters in diameter, to as large as 150 kg and 2 m around. They are found in the oceans, lakes, rivers, ponds, swamps, mountains, and deserts. Some of the species have a life expectancy of up to 200 years.

In general, turtles and tortoises are more active when compared to snakes, and, according to their owners, have very individualistic and distinct personalities. Their activity is usually confined to the daylight hours since all chelonians are diurnal, unlike many of the snakes, which, if nocturnal, may restrict the majority of their activity to the nighttime hours.

These animals are creatures of habit. Their typical day usually follows a series of repetitive patterns. These patterns are also repeated regularly on a day to day basis. The day often begins with the animal seeking out some sort of warmth. This could be a basking place if the animal is outdoors, or a heat lamp if confined indoors. Chelonians, like snakes, are ectothermic, or cold-blooded (the old term used to be poikilothermic). The ambient environmental temperature influences the animal's internal body temperature. The internal body temperature has a direct influence on the animal's metabolism, activity levels, and its ability to eat and digest food. Temperature also has a direct influence on the chelonian's immune system and its ability to ward off disease. The animals are more efficient in resisting disease at the higher temperatures.

After the animal's internal body temperature reaches the proper degree of warmth during the morning basking, the turtle or tortoise will then focus its attention on obtaining food. This task is simplified if the animal is kept in captivity and food is offered to it, but in the wild, this may mean foraging to collect the proper quantity and quality of food stuffs. After the animal gathers and eats its morning meal it may then return to basking or find yet another resting area. These behaviors are often repeated numerous times throughout the day.

The habitats provided for chelonians in captivity should be designed and constructed with a knowledge of their natural behaviors in mind. Separate areas for refuge, basking, soaking, swimming and food collection should all be provided.

DIETS

Proper nutrition is critical to maintaining healthy turtles and tortoises in captivity. Unlike snakes which all have a very similar diet (all snakes require some type of animal protein as the mainstay of their diets), chelonians vary from being herbivorous to omnivorous to carnivorous.

In general, the tortoises are vegetarians. On occasion they may eat some meat, but it is not a major part of their natural diet. In captivity, tortoises are often fed canned dog or cat food, although this practice is not advised due to the excessively high protein levels. A better diet would include fresh vegetables (romaine lettuce, string beans, broccoli, cauliflower, sweet potatoes, carrots, etc.), small amounts of fruit (less than five percent of the diet), fresh flowers (hibiscus, rose petals, dandelion flowers and stems) and a supplementation of vitamins and minerals.

Box turtles have a primarily omnivorous diet. Fresh vegetables, flowers and fruit should be supplemented with wax worms, night crawlers, sow bugs, crickets, slugs or snails and an occasional fish or baby mouse.

Terrapins and other water turtles are mostly piscivorous, however, many will eat items found near or in the water such as algae, moss, and aquatic plants. Captive terrapins will fair well on feeder fish, trout chow, and commercially prepared turtle feeds. Water turtles will only eat while in the water.

SEXING

Sex determination is fairly simple for the experienced keeper. There are a few gender characteristics which can be utilized in identifying males and females. In some species of the box turtle, the male has red irises and the female has orange/brown eyes. In tortoises, the male usually has a concaved plastron (the bottom half of the shell). This is an evolutionary adaptation which allows the male to mount the female without sliding off the side of her domed shell. In some of the water turtles, the male has much longer front claws.

The most reliable characteristic across the species is the tail length and the placement of the cloaca relative to the shell. The male's sexual organ, in its non-reproductive position, is tucked away within the base of the tail, just in front of the cloaca. Because of this the tail is usually broader and longer than the female's tail, and the cloaca is usually located out from under the edge of the carapace (the upper part of the shell). The female has a shorter, narrower tail with the cloaca positioned well underneath the border of the carapace. After seeing a male and female side-by-side, one can easily visualize the difference.

PHYSICAL EXAMINATIONS

Every animal in your collection should be examined and weighed at least monthly, preferably bi-weekly. Before a hands-on examination is performed, stand back and watch the animal interact in its environment, and with its cagemates. A healthy chelonian will be active and alert. When approached it should make some sort of gesture of notice. This may be a simple head turn from the advancing person, or it may be as overt as lifting its head with a subsequent gazing toward the activity. This does not mean that a tortoise or turtle is not healthy if it does not exhibit these behaviors, but it is an important part of the overall examination and should be factored into its health assessment.

Watch the animal walk around its enclosure. A normal chelonian will be able to walk around with its shell lifted well off the ground. Inability to lift its body is a sign of weakness or other internal disease.

Not all chelonians will be shiny. Many of the species have a glossy layer of keratin over their carapace. If kept free of dirt and animal feces, the keratin layer makes the animal very attractive. Often times this shine will be lost due to the soiled conditions of housing. The glossy appearance will be enhanced if the dirt layer is cleaned off with mild soap and warm water, followed by buffing with a dry cloth.

Chelonians do not shed in pieces like snakes. Small amounts of flaky skin is normal, but if large pieces of skin or scales are sloughing off, it is a sign of serious disease. Abnormal shedding of the skin can be caused from improper husbandry, poor nutrition, or a number of internal diseases.

After a general examination is performed from afar, a hands-on examination should be performed on each individual. Start out with a thorough evaluation of the outside of the animal. This will give the keeper a chance to check for the presence of ticks, wounds, shell damage, or other signs of disease. The carapace and the plastron should be free of deformities, and most importantly should not be discolored or bruised. Purple-black bruising and small blood spots can be signs of internal disease.

Many animals are shy and will retreat into their shell. Some of the larger chelonians are so strong that it is almost impossible to get them out for a more thorough physical examination without tranquilization. If the head, neck, and limbs can be grasped, gentle tension will often suffice to extend the body part for examination.

All four limbs should be extended from beyond the margins of the shell. Each limb should be palpated for deformities, fractures, or other injuries. The toes and toenails should be intact and free of lesions. The nostrils should be dry and free of crusts or discharge. There should be no saliva, mucus, or bubbles around the lips. The eyes should be clear and without secretions. Although chelonians do not have external ears, they do have an external eardrum called the tympanic membrane. This membrane should be intact and not swollen. The animal's tail should be examined for the presence of maggots, parasites, or other abnormalities. Pasted feces or a foul smell can be an indication of disease.

COMMON DISEASES

Ectoparasites (Ticks)

Ticks are commonly found behind the ears and in the area of soft skin under the shell just in front of the back legs. The ticks, which vary in size from 4-10 mm, and in color from jet black to brown with yellow spots, are very obvious. Their size and color depends on the species of the tick, its age, and whether or not it has recently taken a blood meal.

All ticks are potential disease threats. The ticks attach to the animal in the wild and then stow-away under the shells of the chelonian during transport. Many species of ticks are known vectors for malaria and other serious diseases.

Animals with ticks should be avoided. Caution should be taken to ensure that the tortoise did not arrive with ticks, only to have them removed prior to being offered for sale. A close-up examination of the skin in the neck region and in front of the back leg will reveal scabs, dry skin and possibly remaining pieces of disembodied ticks.

Ticks can be removed, but treating the tick related diseases may be difficult. The problem can take on added significance when an infested animal is brought into a healthy, tick-free collection. A single gravid (pregnant) female tick is capable of producing over a thousand baby ticks.

The tick can be removed by grasping it by the mouth-parts (not the head!) with a pair of very fine tweezers and gently pulling. Always check to be sure that the piercing mouth-piece has been extracted. If left in the skin it may become infected. If it is necessary to purchase animals infested with ticks, then the ticks should be removed by an experienced person and the animal should be thoroughly checked for signs of disease. The evaluation should include a blood screening to check for malaria.

Hypovitaminosis A

A common disease which affects terrapins is called hypovitaminosis A, or simply, vitamin A deficiency caused from poor nutrition. A terrapin with hypovitaminosis A typically has puffy eyelids which are swollen shut, a thick whitish nasal discharge, and a copious production of saliva. The condition can be fatal if not treated. If proper medical care is given, the animals usually respond in two to three weeks.

Treatment usually consists of correcting dietary deficiencies and supplementing with vitamin A as needed. When an animal has a deficiency in vitamin A, there is often a concomitant respiratory infection. Vegetarian animals, like tortoises, are unlikely to develop hypovitaminosis A since a vegetable diet is usually high in vitamin A. Since vitamin A is a fat soluble vitamin and can be stored in the liver for as long as six months, even an animal that is not eating well is not at a great risk of a deficiency.

Caution should be taken when supplementing vitamin A. Unfortunately, it is easy to overdose an animal with injectable vitamin A. I do not recommend administering injectable vitamin A to tortoises. It is also possible to overdose an animal using oral vitamin A with the same catastrophic results.

Hypervitaminosis A (excessive vitamin A) causes a sloughing of the entire thickness of the skin. The skin takes on a par-boiled or third degree burn appearance. The skin falls off in sheets, leaving connective tissue and muscle beneath. This can be a rapidly lethal condition, and regardless of how quickly attempts at medical therapy are initiated, the prognosis is usually grave.

Respiratory conditions

Many tortoises will develop a nasal discharge when stressed. If the discharge is clear, there are no bubbles forming during respiration, and the animal is eating and otherwise active, veterinary care is probably not immediately warranted. If the tortoise is acclimated and the stress alleviated, the nasal discharge will often resolve spontaneously.

If the nasal discharge progresses from a clear exudate to a milky secretion, and the tortoise begins to blow bubbles when it breathes, stops feeding or otherwise acts lethargic, the condition has become serious. Pneumonia may ensue and veterinary attention becomes imperative.

The bulk of the respiratory problems in tortoises that we see in our hospital are husbandry related. Simply correcting the husbandry practices corrects the majority of nasal discharges without medical intervention. The key is "hot and dry". Most tortoises are desert animals, and low ambient temperatures and high environmental humidity can lead to the frequent nasal discharges. These chronic discharges can ultimately lead to more serious disease conditions. Recognizing these problems early on and making the necessary corrections before the animal's health becomes jeopardized is imperative.

Severe respiratory problems need medical intervention. Bacterial cultures and sensitivities are the key to successful treatment of many of these conditions. Once the disease agent has been identified, then proper antibacterial therapy can be initiated.

Beak deformities

Tortoises and turtles do not have teeth. Prehension and mastication are accomplished with a keratinized ridge of skin which lines the lips, much like the beak of a bird. On occasion this beak will become overgrown. When this happens the animal has difficulty eating. This condition can ultimately lead to starvation and death.

Correction of this deformity is easily accomplished by grinding down the overgrown edge with a hand held burr or grinding wheel. It should be noted, however, that this is a chronic condition and in time the beak will once again need trimming.

Aural (ear) abscesses

Both tortoises and turtles are prone to bacterial abscesses behind the tympanic membranes. They usually occur as a large, firm swelling just behind the eye. The cause of the abscess is not always apparent. If left untreated, they can rupture internally (toward the brain) and kill the afflicted animal. Treatment is simple and effective if caught early.

The tympanic membrane is cleaned with an iodine solution. Then an incision is made around the bottom portion. Small forceps or an ear curette is inserted into the ear canal itself and the caseated pus behind the membrane is carefully removed. This is then flushed out with a solution of betadine. This flushing is repeated daily until the membrane closes, which may take a couple of weeks. Systemic antibiotics are usually not necessary.

Shell diseases

Both tortoises and turtles are susceptible to diseases of the shell. Septicemic Cutaneous Ulcerative Dermatitis (SCUD) is a common disease of aquatic terrapins. It is characterized by ulcers forming either on the carapace or the plastron. The lesions are crater-like in appearance and tend to increase in diameter as the disease progresses. If caught early, daily baths in an iodine based solution will effectively control the disease. However, if left untreated, injectable antibiotic therapy will be necessary.

Shell trauma is also a frequent occurrence in chelonians. Lawn mowers, dog bites, drops, etc. are all common causes of shell damage. This can range from simple non-penetrating cracks to severe shell loss. Minor cracks can often be treated with gentle cleansing and fiberglass polymer patching. However, large deficits in the shell may require orthopedic surgery and bone grafting, along with fiberglass repair. Any shell trauma, regardless of how minor it may appear on the surface, should be evaluated by an experienced veterinarian.

Dehydration

Dehydration is common in animals that are not housed properly, are sick, or are flipped over. Dehydration can be recognized by evidence of sunken eyes, excessively wrinkled skin (more than usual for a given animal), and dry mucus membranes. Dehydration can predispose a reptile to a condition called visceral gout, which is usually fatal.

Since dehydration can be life-threatening in a reptile, any animal demonstrating any signs of dehydration needs fluid supplementation. Mild cases of dehydration can be handled effectively with the administration of oral liquids. Pedialyte® and Gatorade® are readily accessible fluid replacement solutions. Both are available in retail stores.

Oral fluids can be administered via a stomach tube and a large syringe. A good rule of thumb is that the average stomach can hold approximately 3% of the animals body weight. For example, a tortoise weighing 2 kg (4 lbs, 6 oz), can be orally administered 0.06 kg (60 ml) of fluids. For replacement fluids, you would want to orally administer approximately 60 ml (2 oz.) of liquid.

The next step is to calculate the extent of the animal's dehydration. The actual calculation is fairly easy, but it requires a sound record keeping system. The amount of dehydration can be calculated by weighing the animal, and subtracting this weight from its last known healthy weight (hence the importance of frequently weighing all of the animals in your collection). For example, let's say the tortoise just mentioned previously had a weight of 2.1 kg (4 lbs, 10 oz). A quick mathematic calculation tells us that it has lost approximately 5% of its body weight.

Since we already know that the stomach can only hold approximately 3% of the animal's body weight, then it becomes apparent that to correct this fluid deficit the fluids will need to be supplemented over a period of time. Do not attempt to rapidly overhydrate a dehydrated animal, instead replace the fluid loss gradually. A good plan is to replace about one half of the deficit (in this case 2.5% of 4 lbs, 10 oz, or 1.9 oz) initially, and the remaining half (an additional 1.9 oz) in 24 hours.

This is an oversimplified approach, however, it is a start and can easily be applied to cases of mild dehydration (mild dehydration can be considered as a 5% loss of the body weight or less). Moderate or severe cases should be referred to qualified veterinarians for replacement fluid therapy. Intracoelomic or intravenous fluid administration may be necessary.

Salmonellosis

Salmonellosis is perhaps the single-most infamous zoonosis associated with reptiles. A zoonosis is any disease which can be transmitted from animals to man. There have been over 200 different serotypes of *Salmonella* isolated from reptiles, including aquatic turtles, land tortoises, lizards, snakes, and crocodilians. The red-eared slider was the terrapin that received the majority of the negative publicity. At the time, that salmonellosis was a major disease concern, the slider was the most common type of turtle kept as pets in the United States.

In the early 1970's, it was estimated that about 280,000 cases of human Salmonellosis were contracted from pet turtles. In 1975 the Food and Drug Administration passed a law making it illegal to sell viable turtle eggs or live turtles with a carapace length of less than four inches in length in the United States. It was felt that animals larger than four inches in length did not pose the same threat and were still legal for trade.

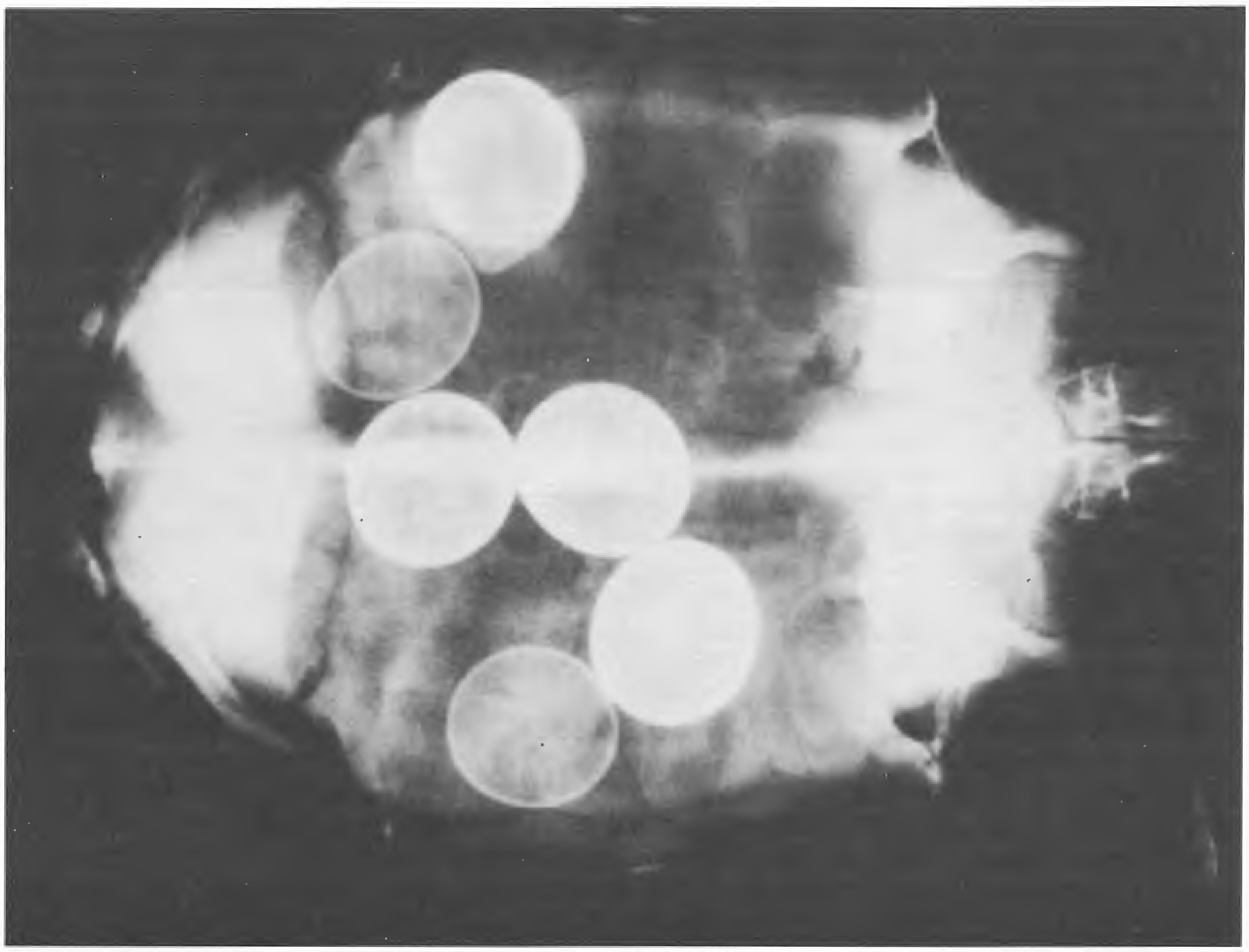
Salmonellosis in reptiles is usually asymptomatic (does not cause overt disease). On occasion an animal may develop a loss of appetite, become lethargic, or have diarrhea. More commonly, the animal appears healthy and acts as a carrier, infecting other animals and people for up to twelve months.

Salmonellosis is best diagnosed by a veterinarian. Microbiological cultures of the aninal's feces, cloaca, or blood may identify the organism. A positive test result is diagnostic for the disease, but a negative *Salmonella* culture can be misleading since the organism may not always be identified, even from a known positive animal. It is wise to recheck all negative animals two or three times to be certain that the *Salmonella* is not present. Concerned chelonian owners are encouraged to set up a screening program with their veterinarian for Salmonellosis and other important reptilian diseases.

CONCLUSION

Perhaps the single most important point to this discussion is that you have to know the normal animal before you will know what is abnormal. This is a concept which we stress in our hospital to all of our technical staff and veterinary student preceptors. It is fundamental to providing quality medical care for any animal, and becomes intensified when dealing with exotic species where the scope of knowledge is limited.

Early recognition of disease is the key to rapid diagnosis and successful treatment. Reptiles are stoic by nature. Prognosis for full recovery and return to function is augmented when the basic husbandry practices are optimal from the start.



Some medical problems, such as these retained eggs in this desert tortoise (*Gopherus agassizii*), require veterinary intervention for proper care. Photo by Douglas R. Mader.

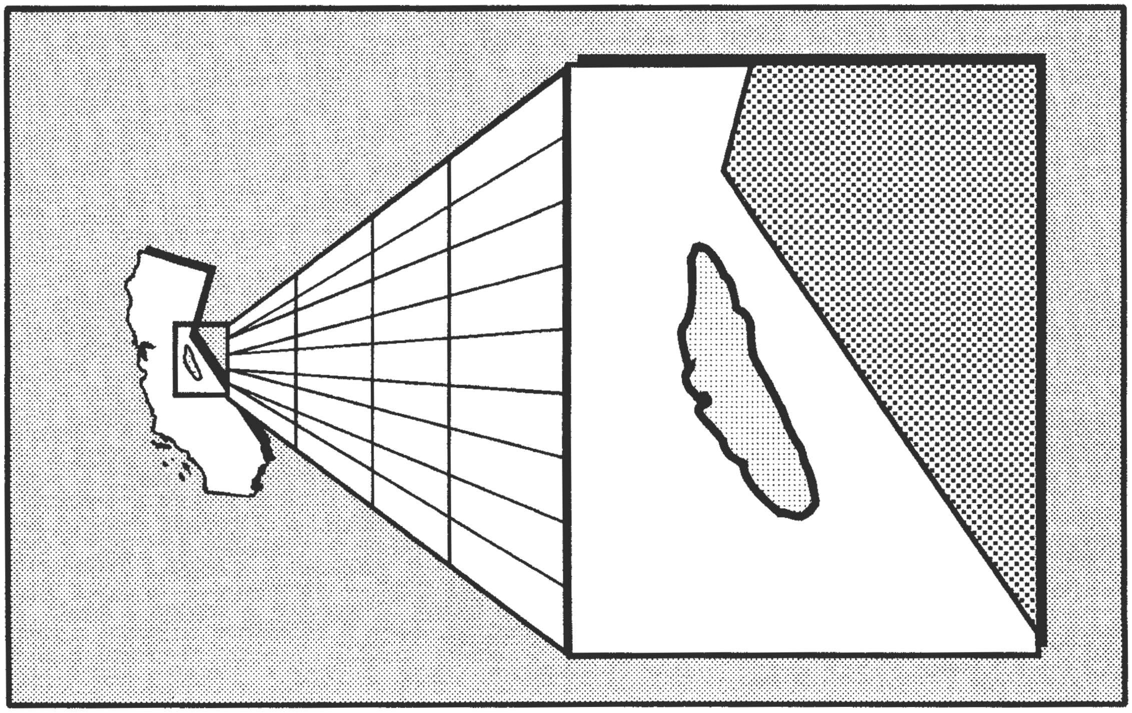


Figure 1. Historical range of the Yosemite toad.

CAPTIVE HUSBANDRY AS A TECHNIQUE TO CONSERVE A SPECIES OF SPECIAL CONCERN, THE YOSEMITE TOAD

David Lamar Martin

Assistant Curator
Museum of Vertebrate Zoology
San Jose State University

INTRODUCTION

The Yosemite toad (*Bufo canorus*) is a montane toad endemic to the high elevations of the Sierra Nevada Mountains of California. This toad, like other high elevation anurans throughout the world, has been declining in numbers for unknown reasons. An investigation of this decline requires the use of test individuals, which can no longer be taken from the wild without detriment to these populations. Few attempts have been made at long term rearing and captive breeding of this species. Included is a review of the known natural history information and new observations made by the author, including tadpole cannibalism and the inclusion of red spider mites in the diets of juveniles. The success of captive propagation of this and other species could serve not only for experimentation, but could also be used for restocking wild populations should the decline continue.

Dramatic declines in amphibian species throughout the world have been reported in recent years (Bury and Luckenbach, 1976; Bury et al., 1980; Hammerson, 1982; Hayes and Jennings, 1986). Many of these declines can be explained by local effects such as deforestation, industrial pollution, and land development (Wake, 1990, 1991; Wake and Morowitz, 1990; Baringa, 1990), but anuran declines in apparently pristine environments, such as National Parks and the Wilderness Areas in National Forests, are largely unexplained at this time (Corn and Fogleman, 1984; Sherman and Morton, 1984; Stolzenburg, 1989; Corn et al., 1989; Bradford, 1989, 1991; Hart and Hoffman, 1989; Wake, 1990; Wake and Morowitz, 1990; Baringa, 1990; Martin, 1990a, 1991a, 1991b; Milstein, 1990, Bradford et al., in prep).

The Yosemite toad is a montane toad which is endemic to three National Parks and three National Forests within the Sierra Nevada Mountains of California (Figure 1). This amphibian has experienced dramatic declines in both range and density in recent years (Sherman & Morton, 1984; Wake, 1990; Martin, 1990a, 1991a, 1991b, in prep.; Milstein, 1990; Jennings & Hayes, in prep.), but the population decline of this species and many other high elevation anurans occurring in pristine environments remains a mystery. An investigation into the causes of this decline will undoubtedly require the use of test animals to explore the possible causes and solutions to the decline of this and other anuran species. Unfortunately, natural populations of these animals are difficult to find, and when found they are usually just large enough to support a viable population. Therefore, these populations could not maintain themselves under the added pressure of collection for laboratory testing of decline hypothesis. Surplus animals from captive breeding programs conducted at zoos, for example, would be an excellent source for critically threatened or endangered anurans to be used in laboratory testing, but at this time there no captive breeding program for any of the threatened or endangered anurans of California, including the Yosemite toad. For this reason test animals must be carefully selected from wild populations so as not to reduce extant populations below the level of viability, which ultimately would result in the extinction of the populations being studied.

Due to the recent drought in California, the vernal pools inhabited by the tadpoles of the Yosemite toad are drying up before metamorphosis can be completed. The pools are drying up because of insufficient snow pack in the Sierra Nevada Mountains needed for snowmelt run off to maintain these pools to the middle of July (Martin, in prep.). Yosemite toad tadpole metamorphosis normally occurs in late July or early August. The tadpoles, which would desiccate and die after the pools have dried, can be collected and used for experimentation without detriment to wild populations, as these individuals would have died had they not been rescued. At this point the key to success; however, is being able to rear and maintain these individuals in captivity for long periods of time.

Unfortunately, few attempts at captive rearing of the tadpoles of the Yosemite toad have been successful, and only limited attempts have been made at maintaining adult toads in captivity for extended periods. The lack of information on the basic husbandry of this animal makes the likelihood for the success of a captive rearing program all the less probable. However, if all of the known information on the natural history of this animal is compiled and utilized to establish a captive management program, the possibility for successful rearing and eventual captive propagation of this animal is much greater.

NATURAL HISTORY

The Yosemite Natural History Survey of 1914-1915 discovered a small toad inhabiting the boggy marshes in the higher elevation areas surveyed within Yosemite National Park. This small toad was later described by Dr. Charles Lewis Camp (1916) as the Yosemite toad or *Bufo canorus*. The specific name *canorus*, which is Latin for tuneful, was given to this small toad because of the high pitched tremolo uttered by the male toads during the breeding season. Camp (1916) also noted that *B. canorus* is extraordinary in its pronounced sexual dimorphism, and Storer (1925) stated that the dimorphism and dichromatism of *B. canorus* is more pronounced than that of any other North American anuran.

The adult male Yosemite toad (50-65 mm snout-vent length) is a smooth animal with a uniform olive-green to tan-yellow coloration. The female Yosemite toad is slightly larger (55-80 mm snout-vent length) than the male and has a ground color of light tan which becomes brownish tan on the back and parotoids. The dorsal and lateral surfaces of the female are marbled with irregular black patches that are encircled by a thin white line. There are large tubercles in the center of each patch which are tipped with a brownish-rust color (Stebbins, 1951; Wright and Wright, 1933; Camp, 1916; Karlstrom, 1962).

Karlstrom (1962) suggests that the color differences between the sexes may be for concealment in the different habitats frequented by these individuals. In the males, the uniform olive to tan coloration blends well with the tan-colored silt on which the males tend to remain during the breeding period. The large black patches of the female Yosemite toad break up the outline and allow the blending of the animal into a varied background, such as grassy vegetation (Sherman, 1980).

Juvenile Yosemite toads have the same basic coloration as that of the adult female toad, but they also have a distinct vertebral stripe. Female toads begin to increase the number of melanophores at about two and a half to three years of age, which results in larger black patches joining to form a marbling effect that breaks up the vertebral stripe. Male toads, on the other hand, begin to lose the warts and their associated melanophores at about one and a half to two years of age, resulting in a lightening of the dorsal surface from an olive-green to a lime-yellow coloration. The color dimorphism of Yosemite toads occurs at approximately the same time as does sexual maturity (Karlstrom, 1962).

Yosemite toad tadpoles (30 mm maximum length) are densely pigmented animals with a rounded or truncated rostrum and a short deep-bodied tail (Stebbins, 1951: Karlstrom, 1962). The tadpoles live in snowmelt-fed vernal pools and small ponds with an average depth of 5-10 cm. These shallow pools cool at night to 0°C and often are encrusted with ice, but they quickly warm to 27-33°C by mid-morning due to the intense solar irradiation in these high elevation areas. The high pond temperatures serve to speed up the development of the tadpoles, which allows them to metamorphose before the vernal pools dry up.

The eggs of the Yosemite toad contain almost black ova (2.1 mm diam.), which are encircled by two distinct jelly envelopes giving an over all average diameter of 4.1 mm. The eggs are laid in elongated single or double strands, or in a cluster several eggs deep. The eggs are laid in the margins of the breeding pools where the water depth is 4-6 cm. It has been estimated that a single female may be capable of producing 1,500 to 2,000 eggs in a single breeding season (Karlstrom and Livezey, 1955).

The Yosemite toad is found in the vicinity of vernal pools, kettle sinks, and lake shores contained within subalpine and alpine-arctic meadow type vegetation. This vegetation is surrounded by dense clusters of willows, which in turn are bordered by lodgepole and white-bark pine forests. These meadows and the Yosemite toads inhabiting them can be found from 2,150 to over 3,400 m in elevation, however most Yosemite toads are found more precisely between 2,400 and 3,000 m in elevation in the Sierra Nevada (Mullally, 1953; Stebbins, 1951, 1966; Camp, 1916; Karlstrom, 1962). These highly productive meadows provide excellent areas for breeding and growth during the short four month active season. Additionally, the rodent populations and their associated burrows found on the margins of the meadows provide excellent shelters for the adult Yosemite toads to escape the night and winter subfreezing temperatures (Morton, 1981; Sherman, 1980).

The Yosemite toad is found in an environment where night-time temperatures during the active season commonly drop to 0°C and rise to 25-30°C during the day. This high flux in temperature has resulted in a toad with a wide temperature tolerance ranging from -2-30°C (Karlstrom, 1962). But the preferred body temperature of these animals is between 19-25°C (Cunningham, 1963). These animals thermoregulate by behavioral adaptation to their environment. The Yosemite toad escapes the over-night freezing temperatures and the high mid-day temperatures by retreating to their subterranean burrows that provide insulation from the extreme environmental temperatures (Karlstrum, 1957, 1962). Additionally, these animals use solar insolation to raise or maintain their body temperatures within the preferred temperature range (Cunningham, 1963). This method of thermoregulation is achieved by basking or remaining motionless with the back oriented in the direction of the sun just outside the burrow. Cunningham (1963) showed that the body temperatures of Yosemite toads are commonly above either the air or soil temperatures, but their body temperatures are usually closest to the substrate temperature. This similarity suggests that solar insolation or basking is of great importance to the maintenance of optimal thermal levels in these ectothermic animals.

Studies on the food habits of the Yosemite toad have been extremely limited. Grinnel and Storer (1924) conducted the first stomach content survey on a toad collected from Porcupine Flat on June 29, 1915. The stomach of this animal contained two tenebrionid beetles, several weevils of various species, numerous large ants, one centipede and some fir needles, probably taken incidentally. The next investigator to examine the stomach contents of the Yosemite toad was Mullally (1953) who examined an unspecified number of toads from Gaylor Lakes. The stomachs were reported to contain spiders and a random assortment of insects including ladybird beetles, dragonfly nymphs, mosquitoes and lepidoptera larvae.

The most recent survey of food habits was a pilot study conducted by the author (Martin 1990b) during the summer of 1988. A total of ten toads were collected, including the following: three newly metamorphosed toadlets; three individuals that were approximately two months post metamorphosis or yearlings; two one-year-old individuals and an adult male and female. A total of ten families from six insect orders were identified as food taken by Yosemite toads, as well as spiders and previously unreported spider mites, crane flies, spring tails, owl flies, and damsel flies. Newly metamorphosed Yosemite toadlet stomachs (Figure 2) were found to contain 70% spider mites and 10% owl flies, but they did not contain spiders, chalcid wasps, or ants. Yearling stomachs (Figure 3) also contained spider mites and owl flies, but spiders and chalcid wasps made up 45% and 20% of the food items found in the stomachs, respectively.

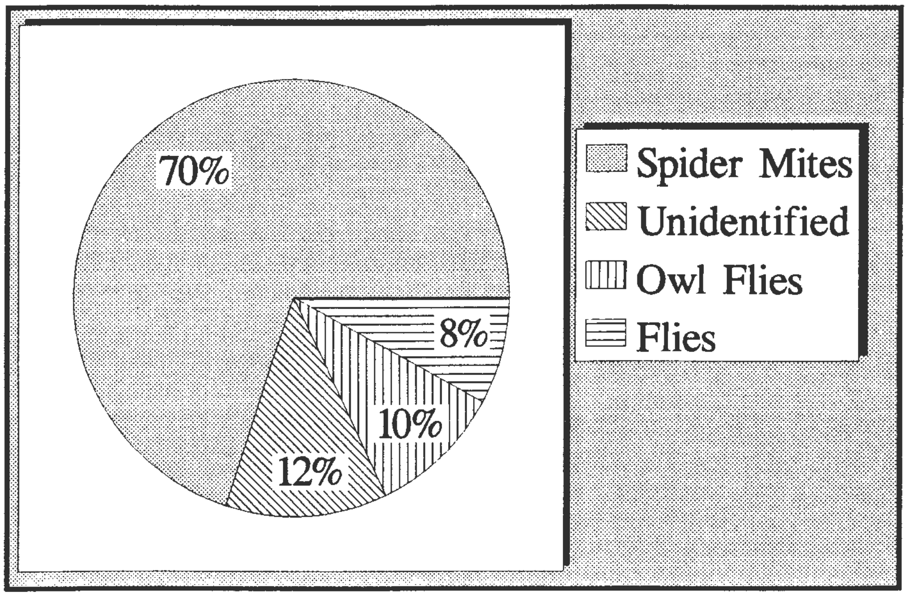


Figure 2. Yosemite toad toadlet stomach contents.

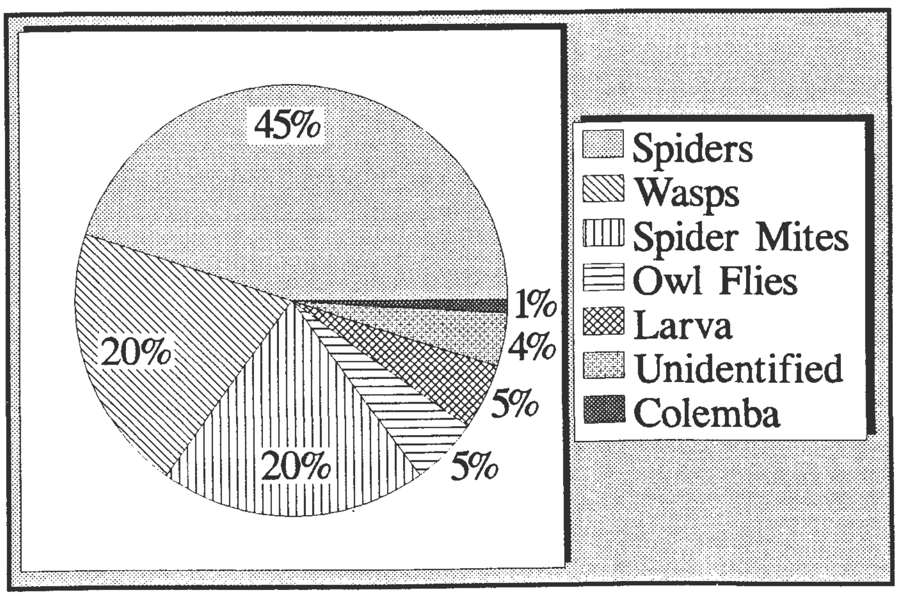


Figure 3. Yearling Yosemite toad stomach contents.

One-year-old toad (Figure 4) stomachs did not contain any mites or owl flies, but they did contain 15% spiders, 5% chalcid wasps, and 75% ants. If these five principle food items are arranged in order of size and compared with toad age class a partitioning of the food resources can be seen with the toadlets taking the smaller mites, the yearling toads taking the medium-sized spiders predominantly, and the one-year-old toads taking the larger ants (Figure 5).

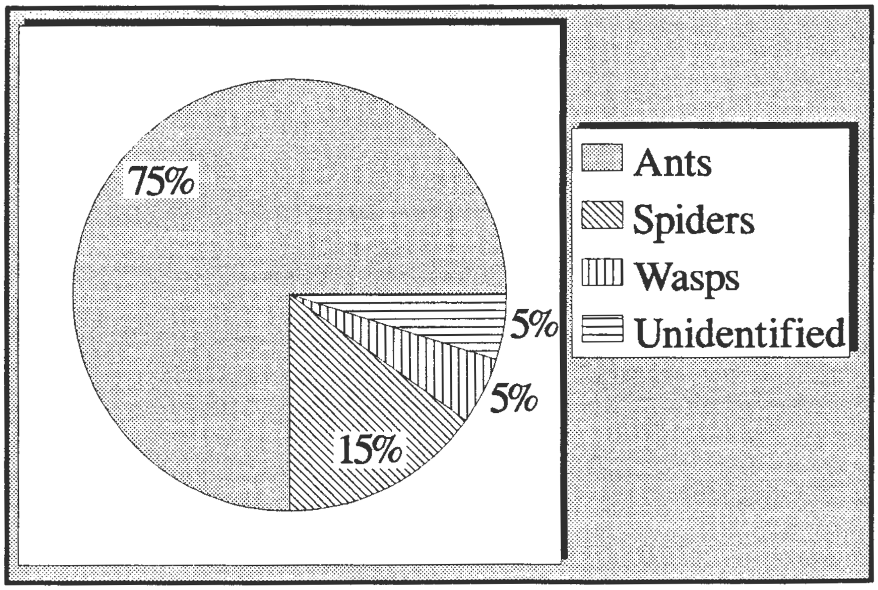


Figure 4. One year old Yosemite toad stomach contents.

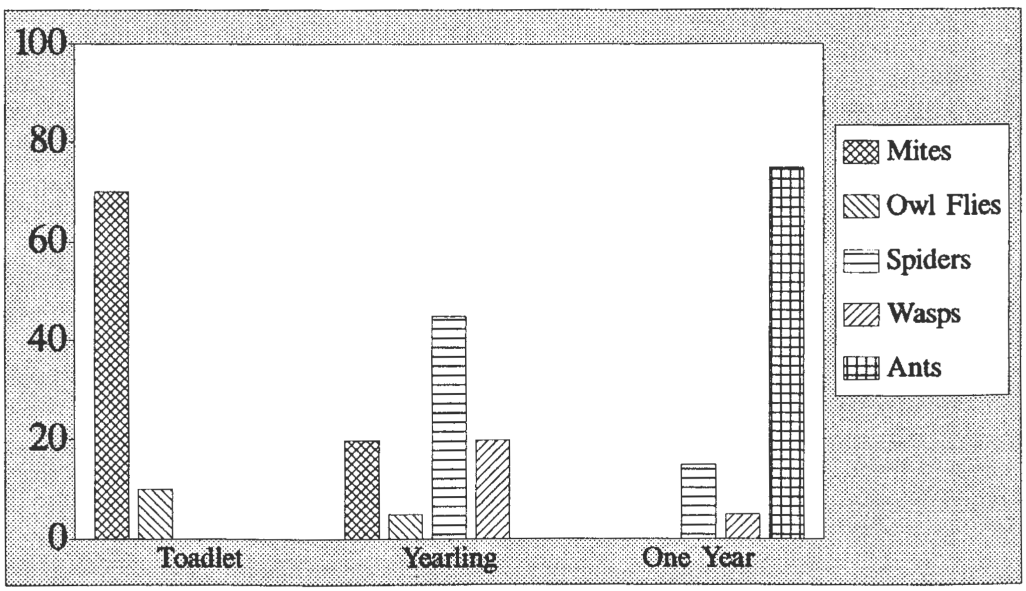


Figure 5. Food resource partitioning between Yosemite toad juveniles.

Adult Yosemite toads consume a wide variety of much larger prey items, as has been shown in previous studies. The adult female Yosemite toad examined by the author (Martin, 1990b) showed a large number of soft-bodied crane flies (75%) in her stomach (Figure 6). She also contained 10% spiders, but this percentage was comprised of two large spiders. The adult male toad (Figure 7), on the other hand, consumed about 40% beetles and only 35% crane flies. The remainder of the stomach contents were comprised of a variety of insects and spiders. The above data provides much insight into the feeding habits of the Yosemite toad, but the small number of animals examined makes it difficult to draw any concrete conclusions.

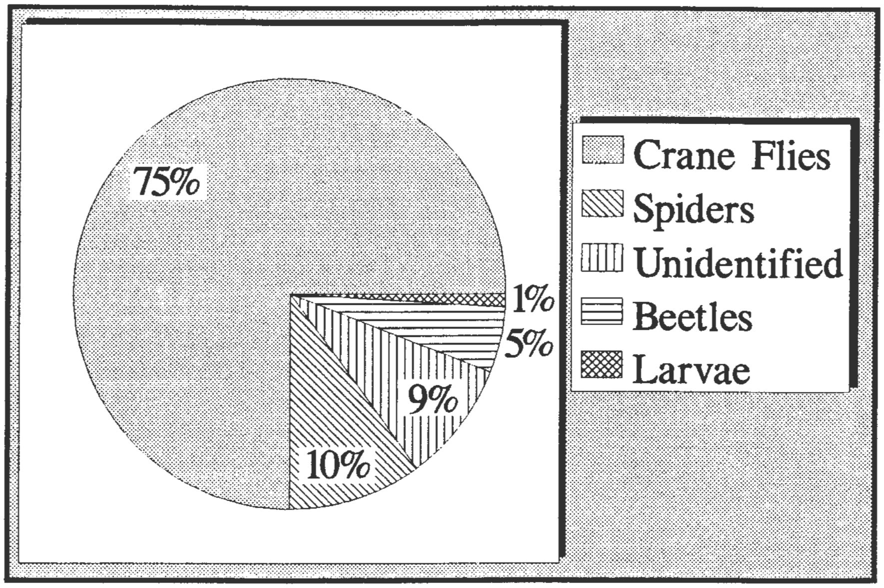


Figure 6. Three year old female Yosemite toad stomach contents.

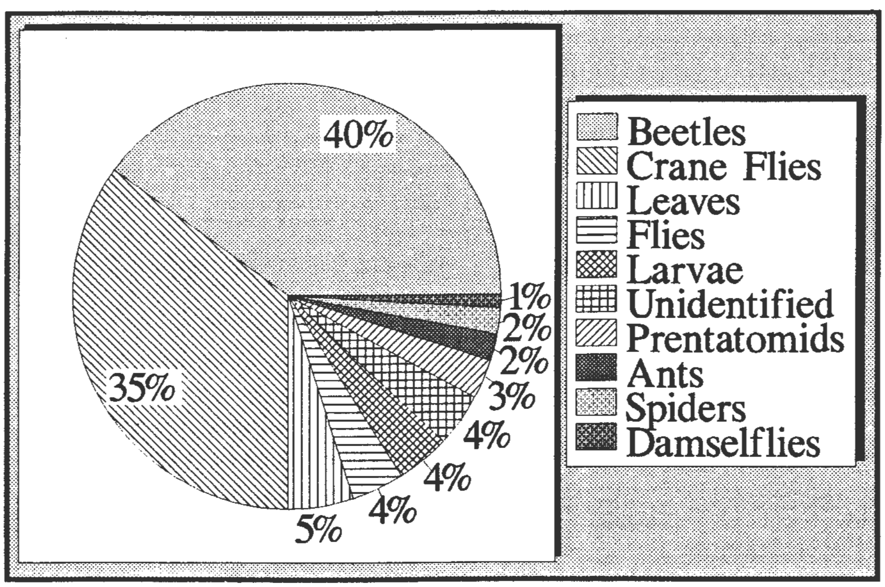


Figure 7. Four year old male Yosemite toad stomach contents.

The only information available on the food habits of Yosemite toad tadpoles is observational data collected by the author (Martin, in prep.) which reports the feeding of tadpoles on detritus, plant materials such as algae, and two unusual food sources. The first unusual food source is pollen grains from the surrounding lodge-pole pine forests. The pollen grains fall from the trees and become suspended in the surface layer of ponds where Yosemite toad tadpoles are found. The tadpoles roll to expose their ventral surface and then feed on the pollen grains from under the surface of the meniscus. The high energy content of the pollen most likely makes it an extremely advantageous food source. The second unusual food source reported is the consumption of Yosemite toad tadpoles by others of the same species. It is unknown if the behavior represents true cannibalism or simply opportunistic feeding on a dead neonate, but this feeding behavior stimulates frenzies which promote attacks on other living tadpoles of the same species. The result of these attacks can be seen as notches in the tails of tadpoles and other wounds to the body. It is reasonable to assume that individuals repeatedly attacked during these frenzies may succumb to their wounds and become the next source of protein in the pond. This carnivorous behavior is not limited to others of the same species, but includes observations of feeding on Pacific tree frog (*Hyla regila)* larva, predaceous diving beetle larva, and a dead ground squirrel, as well as bites taken out of the hand of the observer during investigation of this behavior. This behavior occurs late in the development of these tadpoles and increases in frequency as the animals get closer to metamorphosis. It would appear that the increased amount of protein in the diet of these tadpoles assists with the successful completion of metamorphosis in these animals. More information on the food habits of the Yosemite toad needs to be collected, such as periods and methods of foraging, food preferences, and the relative nutritional value of food items taken.

HUSBANDRY

Captive management of the Yosemite toad has been conducted on a limited basis (Hansen, Karlstrom, McCready and McKeown, pers. comm.: Blair, 1959, 1964; Bradford et. al., in prep.). Generally, only a few individuals were maintained for demonstration or research purposes with little effort put toward long term captive management or reproduction. Those individuals that were maintained as tadpoles were used for experiments on growth, interrelationships between similar species, and pH tolerances, but these animals either died or were sacrificed shortly after metamorphosis. Adult individuals were maintained for experimentation or demonstration for periods up to four years. In one case, adults were maintained in an environmentally controlled chamber at five degrees centigrade for several years, but this temperature is well below the preferred body temperature of these animals, and most likely represents an extended period of estivation rather than true captive management. The only captive breeding of Yosemite toads was conducted by Bradford et. al. (in prep.) who collected gravid females and calling males from breeding ponds and later allowed contact between those individuals in the lab after injection of 0.1 mg/kg synthetic luteinizing hormone-releasing hormone into both males and females. Once again only a few offspring survived to metamorphosis, but none survived past metamorphosis, and the adults were not maintained for periods in excess of a few months. One notable exception is herpetoculturist and biologist Alan McCready, who maintained wild-caught Yosemite toad tadpoles in outdoor enclosures where he reared a limited number of these tadpoles through metamorphosis and maintained those individuals for several years.

The main problem with the past captive management of these animals has been the lack of natural history information employed to maintain these animals. Simply put, a Yosemite toad can not survive in the typical frog aquaria set up, but if an individual examines the natural history information available on the animal being maintained and attempts to duplicate the natural conditions where the animal is found, greater success with captive management can be achieved.

TADPOLE REARING TANK

Yosemite toad tadpoles are maintained in 95-115 l glass or plexi-glass aquaria which are fitted with a fine mesh screen top to prevent the escape of the newly metamorphosed toadlets. Each tank is capable of housing 20-25 tadpoles in a dense aggregation, as is typically found under natural conditions, without an excessive amount of stress or feeding pressure. Tanks with larger numbers of individuals had greater numbers of deaths and cannibalism. Tanks with larger numbers also required greater amounts of food and fouled much more quickly than tanks with fewer numbers of individuals.

The aquaria contained gravel which was excavated out to form a central pool with banks of gravel on all sides. The pool is constructed in such a way that it is eight to ten centimeters at the deepest point, similar to the pools in their natural habitat. The sides slope to allow a minimum of a two centimeter dry gravel border all the way around the pool, providing an easy escape route for newly metamorphosed toadlets in any direction (Figure 8). Previous tadpole rearing setups utilized the sloping tank method where half the tank is filled with water and the tank is sloped so that the opposite end of the tank is dry. It was found that newly metamorphosing toads would swim against the glass on the water side of the aquarium, perhaps looking for land to crawl onto, become exhausted, and drown. The gravel should be sealed in a nonreactive plastic or epoxy (typical of high quality aquarium gravel), and be a neutral or contrasting color like tan, to facilitate the observation of tadpoles and new metamorphs without handling stress. The gravel should be approximately 0.5 cm in diameter. Smaller sized gravel should be avoided because of possible ingestion by newly metamorphosed toadlets resulting in blockages of the intestinal tract. Consequently, larger aquarium gravel has proven to be difficult for the young toadlets to maneuver on. The gravel should be placed on top of an under-gravel filter plate with an air stone and circulation tube affixed in the center. The top of the circulation tube should be covered with a fine mesh screen to avoid tadpoles from entering the tube and becoming trapped (Figure 8).

Water used in the tadpole tank must be high quality water containing some trace minerals, but excessively "hard" water has caused stress and even death in some of the tadpoles collected in 1989. Measurable chlorine levels in water have also been linked to high mortality rates and should be avoided at all costs. Additional losses occurred after using distilled water. The cause of death is unknown, but it may have occurred due to the lack of certain trace elements or to an osmoregulatory imbalance created by the lack of solutes in the water. The author uses bottled "Mountain Spring Water" for tadpoles and all other amphibian applications because the water contains some trace minerals, but it does not contain chlorine. The water is maintained at a temperature of approximately 20°C with a four degree temperature fluctuation range. The water is not kept at the high temperatures found under natural conditions because the low night temperature drop proved to be problematic in the much warmer central California climate. Also, the high temperatures accelerate the process of development, resulting in smaller individuals at transformation which did not have sufficient food stores to survive metamorphosis. By reducing the water temperature to a constant 20°C, development is slowed down and stress is reduced resulting in tadpoles with greater body size at transformation and greater survivorship through metamorphosis.

Lighting over the tadpoles enclosure is maintained on a 12:12 cycle, which means there is 12 hours of light followed by 12 hours of darkness in each 24 hour period. The lighting used consists of a 20 watt broad-spectrum florescent tube (Vita-lite®) connected to a programmable light timer.

The tadpole tanks are cleaned approximately once weekly or when the water is fouled. In order to clean the cage the tadpoles are captured using a small nylon fish net and transferred to a plastic tub containing fresh water of the same temperature. The rearing tank is then filled with a solution of 1% household bleach in tap water and allowed to stand for five minutes. The tank is then flushed three to four times with tap water. Next the tank is filled and allowed to overflow for approximately 10 minutes during which time the gravel is mixed to allow all organic materials to float to the surface. The tank is then drained and the gravel and tank are placed out in the sun and allowed to completely dry, which also evaporates any residual bleach. The cleaning process can be greatly sped up if a spare batch of gravel, which has already been disinfected and dried, is used to replace the drying, newly disinfected gravel. Once the tank is dry the gravel is rinsed three to four times with mountain spring water before it is finally filled and the pond is reconstructed in the gravel. The tadpoles are then returned to the tank and not fed until the next day.

FEEDING

The larvae are fed boiled spinach and a small amount of high quality frozen (thawed) fish food once daily . The excess food should be removed from the aquaria every day to keep the surplus from fouling the water. Once weekly, raw diced beef heart is added to the larval rearing tank a few hours before cleaning. The inclusion of this high protein source helps to reduce the incidence of cannibalism, but care should be taken not to add too much beef heart or the tank will foul, possibly resulting in the death of the tadpoles.

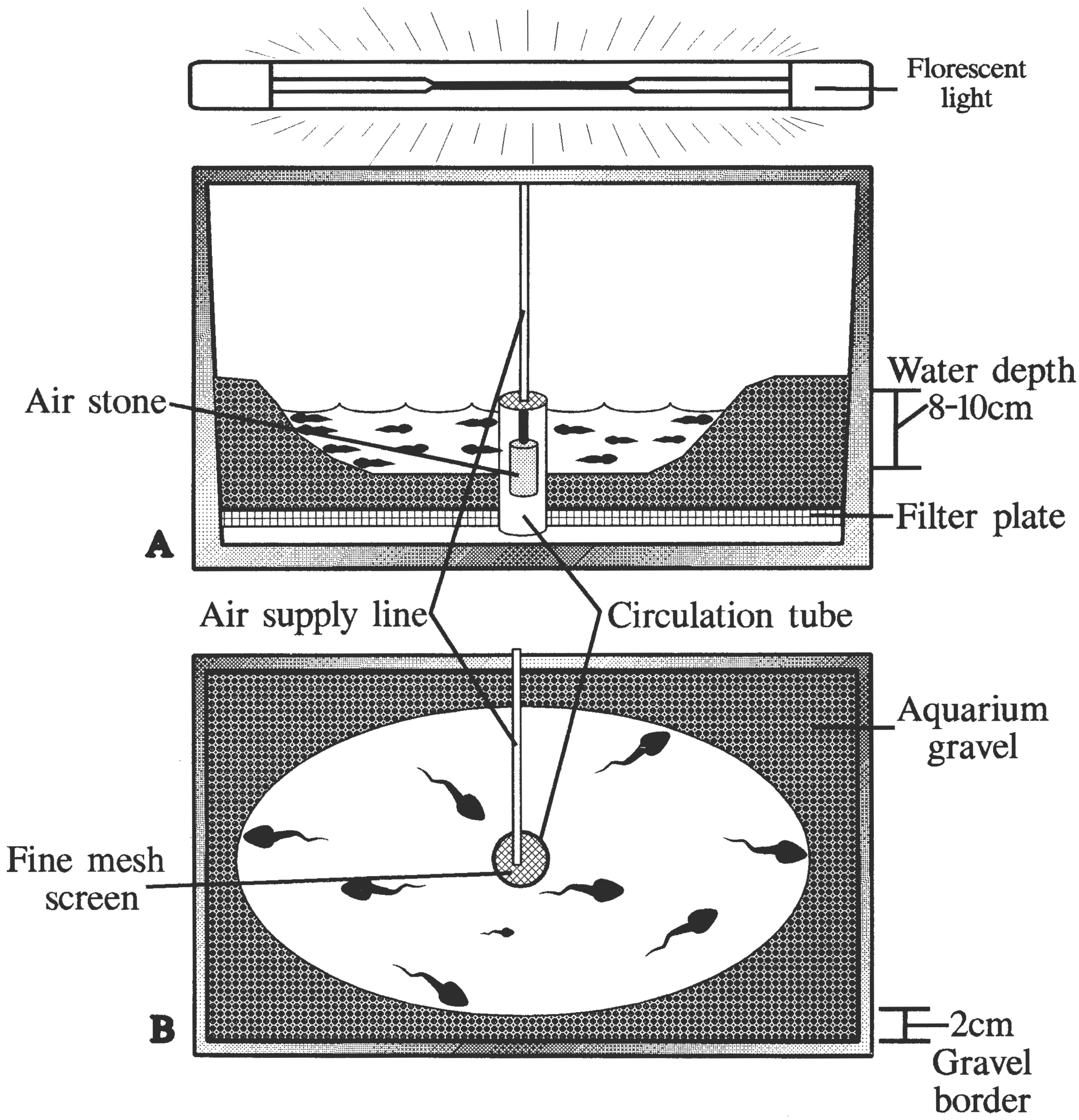


Figure 8. Tadpole rearing tank. A. Side view. B. Top view.

The newly metamorphosed toadlets are extremely active and will grow rapidly even when food is not readily abundant. A plentiful supply of small food items, however, is paramount to the continued survival of these small toadlets. Because of the small size of the toadlets (3-4 mm snout-vent length), they must be offered extremely small food items. Traditional items such as fruit flies and pinhead crickets are too large for the toadlets, but plant mites, which are similar to the spider mites isolated from the stomachs of wild individuals, prove to be an ideal food source. Cultures of plant mites were obtained from Biotactics (Los Angeles, Ca.) and maintained on green beans. Mites are collected from the plants and piled in one corner of the rearing tank early in the day where the newly metamorphosed toadlets readily consume them. The toadlets are transferred to the adult enclosure one week after metamorphosis.

JUVENILE AND ADULT ENCLOSURES

The adults are housed in 95-115 l enclosures like those used for the tadpoles. Unlike the tadpoles, the adults are housed with only four juveniles or two adults per tank to reduce feeding pressures and density stress. Like the larval tank, the adult tank is fitted with a tight fitting fine mesh screen top. This top must be anchored securely to the tank to avoid the escape of the toads, which have proven to be rather adept climbers.

The tank is divided into three different micro-climates. The first, a dry-land component, comprises a third to a half of the cage and is sectioned off from the rest of the cage with a 10-15 cm high solid divider (Figure 9). This divider can be constructed of glass or preferably plastic with the top edge polished smooth and held in place with a bead of silicone. The dry land area is then filled with four to five centimeters of gravel, followed by potting soil or peat dirt that does not contain either perlite or vermiculite. The soil should be kept moist but not wet. Rocks or pieces of wood and bark can be added to the dry-land area to provide objects to burrow under. Plastic plants can also be added to the enclosure to give it a more natural appearance and add a sense of security to the toads, but silk and live plants should be avoided. The coloration of silk plants runs when it gets wet and may be toxic to amphibians. Live plants are usually destroyed in a short period of time by these active and some what clumsy creatures. Additionally, live plants can act as a bacterial storage area since they are usually not disinfected or thrown away when the rest of the enclosure is cleaned. The remaining half to two thirds of the cage is constructed much like that of the larval enclosure (Figure 9), providing a damp micro climate for basking and a pool or water micro climate for soaking. The gravel used in the adult enclosure must be 1-2 cm in diameter for juveniles and 2-3 cm for adults. The size change is to avoid accidental ingestion of gravel by aggressively feeding toads, which has resulted in intestinal blockages and death to several toads. The under gravel filter plate is still used, but it does not have a circulation tube or an air stone. Instead there is a constant level siphon attached to the bottom of the cage to set the water level in the pool (Figure 9), and allow the excess water to be drained to the filtration unit.

Water that overflows from the adult tank is delivered to a second tank containing a reverse flow sand/gravel filter and a storage area that comprises approximately a fourth of the tank (Figure 9). The water drained from the adult enclosure flows through a pipe to the bottom of the filter tank where it flows up through successively smaller layers of gravel until it passes through the top layer of sand (this filter is for physical filtration only, not bacterial filtration). At that point there is 3-4 cm of free water above the sand before it flows over the divider into the storage tank. From the storage tank the water is pumped by a small submersible fountain pump (some aquaria power heads can be used) back to the adult enclosure two to three times a day. The pump is controlled using a programmable light timer which turns the pump on for 10-15 minutes in the morning, afternoon and evening. The water is pumped through 0.64 cm PVC tubing to the top of the adult enclosure where 8 drip system heads are attached to the screen top of the adult enclosure. Two of the drip system emitters are spray heads which are placed to wash down the glass on the wet side of the cage. It may be necessary for one or both of the spray heads to be of the half round variety to avoid soaking the dry-land portion of the aquaria. The remaining six emitters are drip type and are used to wash down the gravel in the adult enclosure.

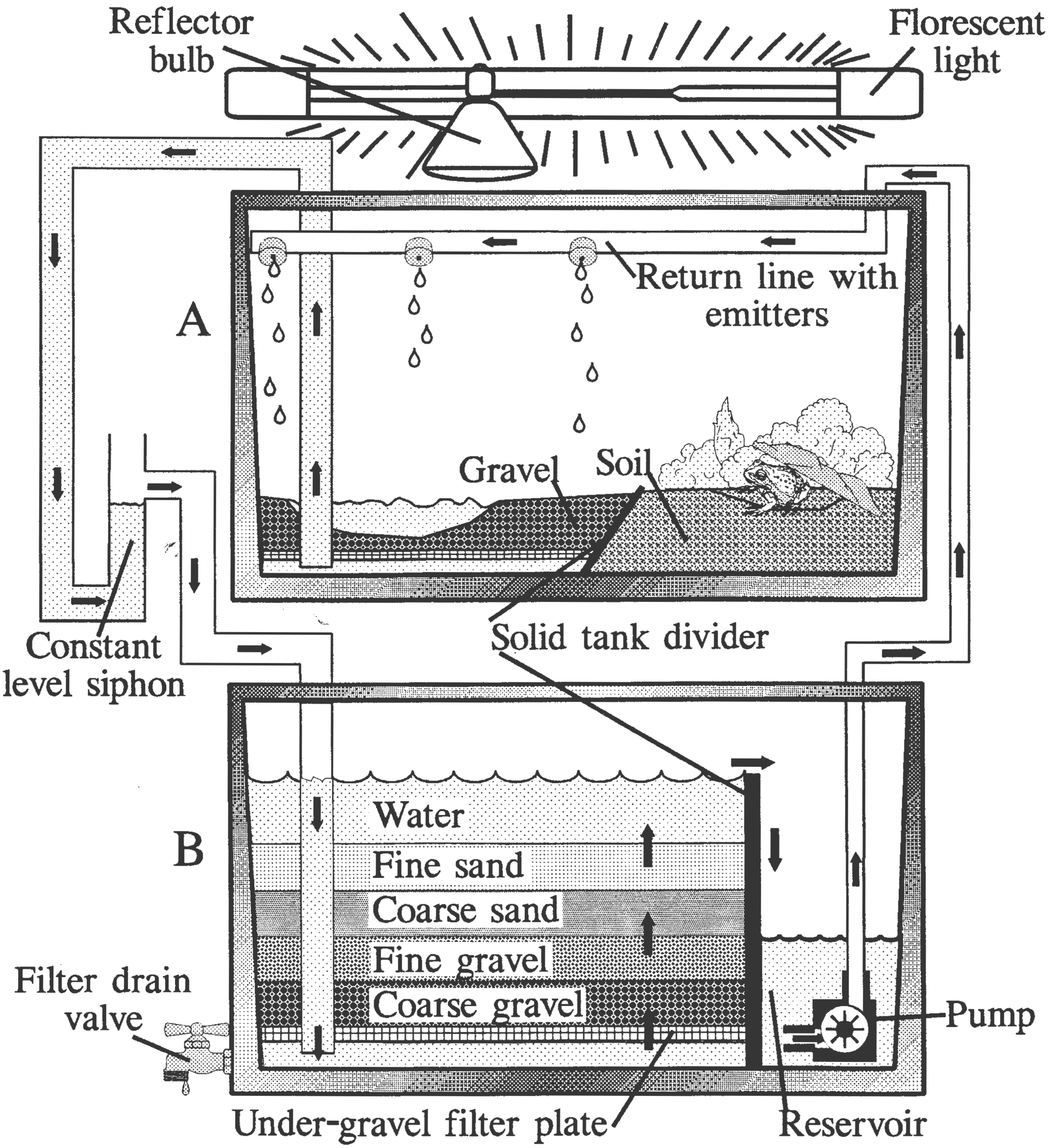


Figure 9. Adult circulating cage system. Arrows indicate direction of water flow. A. Adult enclosure. B. Filter tank.

The ambient temperature where the Yosemite toads are kept should be maintained between 10-20°C. Higher temperatures should only be achieved under basking lamps to allow for the animal to thermoregulate. The water flowing through the system should be maintained at room temperature. The preferred body temperature of the Yosemite toad is in the range of 19-25°C which, as previously stated, is achieved through basking (Cunningham, 1956). To provide a basking site, a 15 watt GE reflector type incandescent plant bulb is used. The focus of the reflector bulb is placed half way between the water and wet gravel area of the adult enclosure so the animals can uptake water from the ventral surface to make up for the evaporative loss on the dorsal surface which occurs during basking. The temperature of the basking spot is adjusted by the distance between the basking site and the reflector bulb, but the temperature at the basking site should never climb above 27°C. The basking lights are turned off at night to provide a temperature differential between day and night.

Background lighting is provided by two florescent fixtures. The first is a full spectrum florescent tube (Vita-lite®), and the second is a UV A tube (GE BLB). The possible benefits of the UVA tube are unknown at this time, but the toads seem to take on a more natural coloration and feed more vigorously under this type of light. However, caution must be exercised until more research is conducted on the potential negative impacts to amphibians caused by long term exposure to this type of light. The two florescent tubes and the basking light are connected to a programmable light timer which should be programmed on a 12:12 cycle for toads up to two years of age. Adults (toads > 2 years) should be maintained on a natural light cycle.

This type of filtered cage system has the advantage of reducing the amount of cleaning required to maintain healthy animals, and thereby decreasing the stress that can be caused by handling during cleaning. The water in the filtration system should be changed approximately once weekly utilizing the filter drain valve (Figure 9). Water should then be back flushed through the filter media and the pump should be run to empty the storage tank. Once the storage tank is drained it should be filled with bottled mountain spring water and the pump run again to flush out the adult enclosure. It should be noted that the weekly cleaning of the enclosure is performed without removing or disturbing the adults contained within. This system can keep the water clean for longer periods of time; however, caution should be exercised because ammonia levels can build up in this type of system without any visual clues to the decreasing water quality. Chemical test kits for fish aquaria can be used to help monitor water quality. Once the system is flushed it should be filled with mountain spring water and returned to automatic control.

The entire cage system should be taken apart about every 6-8 weeks and thoroughly cleaned. The gravel and enclosure should be cleaned in the same manner as the larval rearing tank. The soil and wood from the dry-land component should be completely replaced, but the gravel under the soil and any decorative rocks can be cleaned along with the rest of the tank (unsealed rocks must dry for several days to a week to ensure complete removal of residual bleach). The filter should be completely disassembled and cleaned using a 1% bleach solution once or twice yearly depending on the size of the toads in the enclosure and the amount of bacterial growth.

FEEDING

The newly transferred toadlets are offered a mixture of plant mites, pinhead crickets, and fruit flies every day. Once the toadlets reach 2 cm in length, the mites are discontinued and the toads are fed crickets and fruit flies every two to three days depending on the robustness of the toads. Yosemite toads, like most amphibians, will quickly become obese if allowed to do so. The quantity and frequency of feeding must be closely monitored. The size of the insects offered to the toadlets are gradually increased as the toadlets grow. Diced worms and meal worms are added to the diet as are silk worms, wax worms, and the occasional beetle, but the main diet of these animals is crickets that are maintained on laying mash and oranges (de Vosjoli, 1990). The crickets offered to the toadlets less than one year of age are dusted with vitamin powder (Super-preen®) and calcium carbonate once a week. The diet supplementation is gradually reduced until the toads reach three years of age, at which point supplementation is only given approximately once a month.

The most important factor in feeding Yosemite toads is the time of day the toads are fed. Toads in all age classes react best when fed in the morning after approximately one hour of basking. Food which is placed in the cage earlier in the morning, before the body temperature of the toads can be raised to the preferred level, elicits little response, but food that is placed in the cage after one hour or more of basking results in a vigorous feeding response. Yosemite toads that were fed in the mid to late afternoon have been found bloated and inactive the next day. Some mortality has occurred, particularly when the toads were fed late in the day with a lower than normal night time temperature drop. This suggests that food should not be offered after the mid-point of the light period, when the toads will not have sufficient basking time to completely digest the food ingested.

CONCLUSION

The Yosemite toad captive rearing project is still in the beginning stages of its development, but the early success of this program is directly attributable to the incorporation of the known natural history information into the captive management plan of this species. Information such a food preferences and basking behavior gave clues as to the nutritional and thermal requirements of this animal, which intuitively would seem not to be required or to be of little importance to the survival of this animal. The inclusion of protein in the diets of tadpoles has proven critical for surviving metamorphosis. The high thermal and UV light requirements of these animals is also unique, especially when one observes the apparently delicate amphibian skin. Once again the natural history information provided clues as to the husbandry requirements of these animals.

The early success of this project has produced 68 nine-month-old toads and the successful maintenance of four adult toads. The captive-reared toads have now been divided up into three colonies to avoid the potential dangers of an epizootic incident wiping out the entire captive population, and to allow the honing of the captive management plan to maximize the survivorship of these increasingly rare animals. The first satellite colony was established at the Sacramento Zoo, Sacramento, California, under the care of keeper Mark Olin. Mr. Olin is preparing to receive large numbers of tadpoles next summer and is working on larger more productive tadpole rearing facilities. The second satellite colony was established some time later at the Chaffee Zoological Gardens, Fresno, California, under Curator Sean McKeown. Mr. McKeown and his staff are working on fine tuning the husbandry of the young toadlets. Finally, the mother colony is being kept by the author, who is working on pathogen control and treatment (especially under dense conditions). The author is also working toward uncovering the requirements and facilities needed to captively breed the Yosemite toad without using hormone injections.

There are many other amphibian species which are declining throughout the world, but determining the cause for the decline of these animals is difficult due to their complex life cycles and the multiple ecological interactions within their environment. If potential causes of the decline of these animals are discovered, then laboratory test specimens which are captively produced will be of paramount importance to the successful verification of these hypothesis. Finally, if the decline should continue to the point of extinction of wild populations, the captively reared individuals and the captive management plan, which hopefully will have already been successfully defined, can be used to restock wild populations.

LITERATURE CITED

Baringa, M. 1990. Where have all the froggies gone? Science 247:1033-1034.

Blair, W.F. 1959. Genetic compatibility and species groups in U.S. toads (*Bufo*). Texas Journal of Science, 11(4):427-453.

--------------. 1963. Evolutionary relationships of North American toads of the genus *Bufo*: A Progress report. International Journal of Organic Evolution, 17(1):1-16.

Bradford, D. F. 1989. Allotropic distribution of native frogs and introduced fishes in high Sierra Nevada lakes of California: Implication of the negative effect of fish introductions. Copeia 1989(3):775-778.

----------------. 1991. Mass mortality and extinction in a high-elevation population of *Rana muscosa*. J. Herpetol. 25(2):174-177.

----------------., C. Swanson and M. S. Gordon In prep. Acidic deposition in the Sierra Nevada: Effects of low pH and inorganic aluminum on two declining species of amphibians. Journal of Herpetology 27 (In press).

Bury, R. B. and R. A. Luckenbach. 1976. Introduced amphibians and reptiles in California. Biological Conservation (10):1-14.

--------------, C. K. Dod Jr. and G. M. Fellers. 1980. Conservation of the Amphibia of the United States: A review. United States Department of the Interior, Fish and Wildlife Service, Resource Publication (134):1-34.

Camp, C.L. 1916. Description of *Bufo canorus*, A new toad from the Yosemite National Park. University of California Publications in Zoology, 17(6):59-62.

Corn, P. S. and J. C. Fogleman. 1984. Extinction of montane populations of the northern leopard frog (*Rana pipiens*) in Colorado. Journal of Herpetology 18(2):147-152.

-------------, W. Stolzenburg and R. B. Bury. 1989. Acid precipitation studies in Colorado and Wyoming: Interim report of surveys of montane amphibians and water chemistry. U.S. Fish and Wildlife Service Biological Report 80(40.26):1-55.

Cunningham, J.D. 1963. Additional observations on the ecology of the Yosemite toad, *Bufo canorus*. Herpetologica, 19(1):56-61.

de Vosjoli, P. 1990. The Right Way to Feed Insect-eating Lizards. Advanced Vivarium Systems, Lakeside, California.

Grinnel, J. and T.I. Storer. 1924. Animal Life in the Yosemite. University of California, Berkeley.

Hammerson, J. A. 1982. Bullfrog eliminating leopard frogs in Colorado? Herpetological Review 13(4):115-116.

Hart, J. and E. Hoffman. 1989. Possible effects of acidic deposition on a Rocky Mountain population of the tiger salamander *Ambystoma tigrinum*. Conservation Biology 3(2):149-158.

Hayes, M. P. and M. R. Jennings. 1986. Decline of ranid frog species in Western North America: Are bullfrogs (*Rana catesbeiana*) responsible? Journal of Herpetology 20(4):490-509

Jennings, M. R. and M. P. Hayes. In prep. Species of special concern status in California. Report to the California Department of Fish and Game.

Karlstrom, E. L. 1957. The use of CO-60 as a tag for recovering amphibians in the field. Ecology 38(2):187-195.

-------------------. 1962. The toad genus *Bufo* in the Sierra Nevada of California: Ecological and systematic relationships. University of California Publications in Zoology 62(1):1-104.

-------------------. and R.L. Livezey. 1955. The eggs and larvae of the Yosemite toad *Bufo canorus* Camp. Herpetologica, 11:221-227.

Martin, D. L. 1990a. Population status of the Yosemite Toad (*Bufo canorus*): An interim report (Abstract). Program and Abstracts of the Joint Annual Meeting of the Herpetologists' League and the Society for the Study of Amphibians and Reptiles, Tulane University, New Orleans, Louisiana.

--------------. 1990b. Food habits of the Yosemite toad (*Bufo canorus*). Program and Abstracts of the Joint Annual Meeting of the Herpetologists' League and the Society for the Study of Amphibians and Reptiles, Tulane University, New Orleans, Louisiana.

--------------. 1991a. The dramatic decline of a species of special concern: The Yosemite Toad (*Bufo canorus*) (Abstract). Program and Abstracts of the Joint Annual Meeting of the Herpetologists' League and the Society for the Study of Amphibians and Reptiles, Penn State University, University Park, Pennsylvania.

--------------. 1991b. Population census of a species of special concern: The Yosemite Toad (*Bufo canorus*) (Abstract). Program and Abstracts of the Fourth Biennial Conference of Research in California's National Parks, University of California, Davis, California.

--------------. In prep. Population status of the Yosemite toad (*Bufo canorus*): A California endemic species of special concern.

Milstein, M. 1990. Unlikely harbingers. National Parks 64(7-8):18-24.

Morton, M.L. 1981. Seasonal changes in total body lipid and liver weight in the Yosemite toad. Copeia, 1:234-238.

Mullally, D.P. 1953. Observations on the ecology of the toad *Bufo canorus*. Copeia, 3:182-183.

Sherman, C. K. 1980. A Comparison of the Natural History and Mating System of Two Anurans: Yosemite Toads (*Bufo canorus*) and Black Toads (*Bufo exsul*). University Microfilms International No. 8106225:i-394.

--------------- and M. L. Morton. 1984. The toad that stays on its toes. Natural History 93(3):72-78.

Stebbins, R.C. 1951. Amphibians of Western North America. University of California Press, Berkeley. 539 pp.

----------------. 1966. A Field Guide to Western Reptiles and Amphibians. Houghton Mufflin Company, Boston. xiv+279 pp.

Stolzenburg, W. 1989. What are the toads telling us? Colorado Outdoors 1989(3/4):24-27.

Storer, T.I. 1925. A synopsis of the amphibia of California. University of California Publications in Zoology, 27:1-307.

Wake, D. B. 1990. Excerpts from the frog log. Unpublished Manuscript 14 pp.

-------------. 1991. Declining amphibian populations. Science 253:860.

-------------. and H. Morowitz. 1990. Declining amphibian populations: A global phenomenon. Report of Workshop findings to the National Research Council Board on Biology.

Wright, A.A. and A.H. Wright. 1933. Hand Book of Frogs and Toads of the United States and Canada. The Comstock Publishing Co., Inc., Ithaca, New York.

Yosemite toad (*Bufo canorus*) tadpoles feeding on a dead ground squirrel. Photo by David Lamar Martin.

CONSERVATION OF THREATENED AMPHIBIANS: THE INTEGRATION OF CAPTIVE BREEDING AND FIELD RESEARCH

Bob R. Johnson

Curator of Amphibians and Reptiles
Metro Toronto Zoo
P.O.Box 280
West Hill, Ontario, Canada M1E 4RS

INTRODUCTION

Management strategies for threatened amphibians increasingly involve the integration of research data from the wild and captive populations. The application of field data to captive breeding programs and the return of captive bred animals to the wild are essential components in the conservation of the Puerto Rican crested toad, *Peltophryne lemur*. Along with 13 other zoos participating in the American Association of Zoological Parks and Aquariums' (AAZPA) Species Survival Plan (SSP), Metro Toronto Zoo has maintained this species in captivity for 10 years. We cannot yet assure the survival of this species, and there are very real concerns that, in the interval, amphibians on a global scale have declined in number and distribution.

Captive managers have a role to play in supporting wild amphibian populations. One such role would involve short or long term maintenance of amphibian species identified as being in danger of extirpation or with species below minimum viable population size. For example, if research has identified 10 species of Bolitoglossid salamanders in decline because the patch of forest in which they lived was on the verge of disappearing, we could bring the whole group into captivity. Let's suppose that five of these species used to inhabit the darkest reaches of the primary forest or were adapted to living in bromeliads high above the forest floor and none could be bred more than once in captivity. These would be doomed to extirpation unless another patch of primary forest could be found for the immediate release of these five species.

From the five remaining species, three lived on the forest edge and two exploited open patches in the forest. For this reason, all five did well in captivity and bred for several generations. Because we obtained thirty individuals from each of the five species, we can predict that all of the genetic diversity once represented in the wild population was still present in the five captive populations.

Now let's suppose that after our forest patch had been destroyed for the charcoal that each tree represented, there began to grow a secondary tangle of scrub forest which was now given some degree of protection on the realization of what it once represented. We could immediately return the two captive species which prefer open habitat. This would represent a short term conservation and captive breeding program.

The remaining three species could be re-introduced to our patch of forest once the larger trees had time to establish themselves above the scrub. And fortunately this process was enhanced with the planting of primary forest species as part of a local conservation and education initiative for the local schoolchildren. This hypothetical scenario is not too far from the reality of possibilities for short and long term captive breeding programs. The Puerto Rican crested toad provides a practical example of captive propagation as a component of conservation programs.

The Puerto Rican crested toad is the only native Bufonid of the Puerto Rican bank. It is known to breed in one pond on the south west coast and four man-made cattle troughs in the northwest. The northern population is estimated to consist of 3000 toads while no more than 25 toads have been sighted at any one breeding episode in the north. In fact, it is not known whether the northern population has bred in the last two years and seems doomed to extirpation.

The largest and best studied population of toads breed in the Tamarindo section of the Guanica State forest. The breeding pond is located in a precarious location within 100 meters of the sea and could be breached or inundated by heavy seas during severe storms or hurricanes. The site serves as a parking lot for users of a nearby beach during the dry season. The parking area is now closed during breeding episodes. In fact, until 1984, the flooded parking area was regularly drained by well meaning employees of the nearby town of Guanica. As a result there was not enough water remaining for the tadpoles to complete their 18 day larval period before metamorphosing. Recruitment to the Guanica population during this period was not possible and the only other breeding pond was destroyed by a hurricane in 1985.

CAPTIVE BREEDING PROTOCOLS

Captive breeding protocols were first outlined by Paine (1984). Aestivation and husbandry protocols were further refined by Devison (1991). Breeding details are available in these papers.

Amphibians have colonized dry habitats because they are adapted to conserve body fluids by forming a protest cocoon of shed skin, by increasing blood urea levels, or drought avoidance by aestivating in humid underground chambers, usually in rock.

However, the extreme demands placed on the physiology of drought adapted amphibians often means that these conditions are actually required to stimulate egg maturation or to maintain a full complement of eggs in readiness for a brief period of reproduction. The micro environments utilized by aestivating amphibians are not well studied and their re-creation in captivity remains a challenge.

An increase in parasite burdens might be expected after any period of breeding stress and toads should be routinely monitored during and after breeding attempts, particularly if exposed to cold or drought. Toads quickly learn which area provides protection and which provide micro-environments necessary for temperature and water regulation. If these are routinely moved or replaced their internalized world map is disturbed, resulting in stress which may be responsible for future infective diseases or parasite loads.

Environmental stimuli such as photoperiod, temperature, and rainfall are expressed physiologically by the secretion of reproductive hormones into the blood stream. Now the amphibian becomes sensitized to secondary, and then tertiary, behavioral stimuli which did not previously stimulate a response from the toad. For example, male calls only stimulate the females after they have experienced a rise in temperature and an increase in rainfall. Our challenge is to recognize those factors responsible for stimulating or facilitating reproduction and then re-create these in captivity in a sequence which approximates the thresholds encountered in the wild. Therein lies the importance of field data to captive husbandry.

The importance of water quality is often overlooked. Odum (1985) has summarized the conditions and techniques favorable for amphibian reproduction. We still have a lot to learn from a cross over from the aquarist in this regard. We should not expect amphibian larvae from the amazon basin to thrive in the same aquatic environment re-created for tadpoles from the calcareous Caribbean Islands. National Aquarium in Baltimore and Aquarium of the Americas in New Orleans have experimented with various "tea" recipes which are used to condition water. Again the field literature must be consulted to re-construct appropriate aquatic environments.

Amphibian diets are all too often taken for granted. Once the environmental triggers responsible for reproduction are isolated, diet may be the most important aspect of a sustainable amphibian reproduction program. The quality and frequency of feeding are important considerations. We all understand the importance of supplementing the usual invertebrate foodstuffs with an appropriate calcium and vitamin D_3 source.

Bone fractures have been observed in the long bones of post metamorphic toads, perhaps indicative of calcium and vitamin D_3 deficiencies induced in the larval stage, which were difficult to compensate for in the early stages of growth. Both black lights within one foot of the tadpoles and toadlets or high intensity ultraviolet lights placed over one end of the holding tanks and no closer than three feet would be appropriate trial sources of ultraviolet lighting. Some species, such as Bufonids, will actually bask under high intensity lights, mirroring behaviors observed in the wild.

We prefer to feed toads on a growth diet for actively growing toadlets or after a period of inanition associated with drought or cold induced conditioning prior to breeding. Many toads will not feed after a period of rest as the metabolic slowdown associated with this physiological change utilizes very little of the lipid reserves, (10% to 15% on average).

Once growth curves begin to level, we reduce the quantity and frequency of feeding. Of course, it is necessary to have weekly or monthly weights to confirm growth rates. With practice you will recognize the growth phases and frequent weighing is not necessary. After a season of normal activity, we further reduce the frequency and quantity of feeding to only maintain lipid stores responsible for weight gains.

Continuous feeding throughout the year risks the stressing of kidneys during cycles of metabolic slowdown. Kidney damage and death or gout are symptomatic of this regime of continuous feeding. During field work in Puerto Rico, toads were never observed to feed and there is a possibility that toads require several years to build up lipid reserves before breeding. The only invertebrate species active at the same time as the toads were grasshoppers, cicada, small beetles, and an abundance of snails. Grasshoppers and cicada foraged along the beach areas and were not sympatric with the fossorial toads which moved away from the beach immediately after breeding. Some tropical species, such as Dendrobatids with relatively high metabolic rates, are an exception to this feeding schedule.

Failure to recover after the enforced period of inactivity, or failure to ovulate may be related to insufficient lipid stores. Toads are well fed prior to any breeding attempt and, if lipid stores appear depleted after aestivation, no attempt is made to stress the toad further by attempting a breeding.

Initially, toads were set up in a deep bed of moistened peat moss and sphagnum. Currently, limestone rocks are piled on the bottom of the Aestivation chamber to create a maze of underground tunnels before adding the peat substrate. Toads bury deeper as the substrate gradually dries. After about thirty days the substrate has dried completely and the toads are inactive. Water dishes are added as an emergency moisture source and these are occasionally utilized. Toads may be left undisturbed for a period of about thirty days after the substrate has dried out. At any sign if distress or dehydration and loss of body condition, the toads should slowly be rehydrated. A light misting may be adequate to initiate the rehydration process.

Rehydration should simulate the environmental conditions that might be experienced by the aestivating amphibian in the field. The immersion of toads in water should be avoided as the kidneys may not be functioning efficiently during the period of metabolic shutdown and the toad may swell with water. In our experience, rehydration is best completed over a three day period before the males are introduced to the breeding tank.

While the toads are rehydrating, tape recorded calls are played. These stimulate males and indicate to females that conditions are favorable for breeding. Calls often stimulate activity in both sexes and males in particular will orient themselves to the direction of the call playback and call in sequence with the taped "competitor".

Males are placed in a breeding tank which has a well developed algal culture growing in it. This mimics the sequence observed at breeding ponds in the field. Two to three days later, females, who have been exposed to the calls of males for three days, are introduced to the breeding tanks. Amplexus is usually immediate.

Eggs are wrapped around an abundant supply of plastic plants and flowerpot shards provided for this purpose. It has been our experience that attached and undisturbed eggs have a significantly higher hatch rate. Amplexant toads are removed from the breeding tank and the eggs are left to hatch.

Hatching occurs within 24 hours and neonate tadpoles remain suspended from egg strings. Hatchling tadpoles retain the external gills and are best left undisturbed until these are absorbed, usually within a day. Only enough toads are raised to meet immediate needs and the remainder are cropped. Our success rate increases if we attempt to raise only the number needed at metamorphosis, which varies from 100 for distribution to other breeders to 1500 if the toadlets are destined for release.

Just prior to metamorphosis, water temperatures are raised to 28°C to simulate the rise in water temperature measured in ponds in the field. These beach areas are used extensively by toadlets to bask and to breath without having to swim to the surface.

Toadlets will move en masse out of the water and seek refuge under objects despite their exposure to dehydration. This behavior has obvious advantages to toadlets in the wild who must seek refuge from the hot sun and the deteriorating water conditions of the drying pond. Clumping behavior is common in many bufonids as it was for the Puerto Rican crested toad. As soon as toadlets leave the water, they seek refuges. This has adaptive advantages in the wild, as ground temperatures exceed 45°C around the drying ponds in Puerto Rico. A single mass of toadlets in one clump reduces the surface area to body mass ratio with a corresponding reduction in evaporative water loss. Clumping under small rocks was observed around the breeding pond and this may represent a critical factor in juvenile mortality.

Several species of amphibians adopt leaf-litter postures. Once again, this behavior was observed in captivity prior to our understanding of its adaptive significance in the wild. In the leaf-litter of the dry forests in which this species is found, this posture is an effective camouflage, making the toadlets virtually invisible amongst the leaf litter.

RADIO TRACKING AND STRATEGIES FOR AMPHIBIAN RELEASE PROGRAMS

Release and translocation programs for amphibians are not, as yet, well developed. Of the 3,000 toadlets released, the only evidence of survivorship has come from the discovery in 1989 of two animals released as part of a group of 640 in 1988. The small size of the released toadlets makes follow-up difficult but nonetheless important. To date, the only measure of success has been the presence or absence of toadlets at release sites.

A harness designed to carry a radio transmitter in knapsack fashion enabled us to outfit twelve toads with radio transmitters. Initial research focussed on the habitat use and survival of two year old, captive raised toads. The results of this aspect of ongoing field research into habitat use and home range are the subject of future publications (Johnson, in press). Mongoose predation was a significant factor on the survival of captive raised toads.

After waiting two years before there was enough rainfall to stimulate emergence and reproduction, a research team from Metro Toronto Zoo tracked post reproductive movements for 20 days. Much as expected, the toads utilize crevices and solution holes in the limestone as daytime retreats from the dehydrating effects of ground temperatures which soar to 50°C during the day. Toads migrated up to 2 km during our study period. After a three day period of activity in which the distance traveled averaged 150 m per 24 hours period, toads settled into an area and movement was reduced to an average of 6 m per night.

Data on pH, salinity, calcium, and temperature was collected at Guanica by the Metro Toronto Zoo research team in 1990. It is essential that the chemistry and ecological characteristics of the breeding ponds be characterized. First, a pond profile would allow managers to maintain the pond's essential character given sudden or long term changes which may adversely affect the present population. Second, the need to identify existing ponds suitable for crested toad reproduction is imperative. Failing this, it will be necessary to construct breeding ponds which would include the essential components of the present pond.

If northern lines are extirpated, southern animals may be translocated to the historic range. Studies of mitochondrial DNA are underway to determine the extent to which northern and southern populations have diverged. If the northern and southern populations can not be differentiated, then it is possible to translocate toads from the south to enhance the population size of the northern populations. It is anticipated that new ponds, whether naturally formed during rains or man made to fill during rain, would receive tadpoles from two sources. Some tadpoles would be translocated from the present breeding pond to adjacent ponds outside the migratory range of the crested toad, and a second pond would receive captive bred tadpoles returned to Puerto Rico for release.

Given the prolific reproductive output of amphibians, there is a very real possibility of genetically swamping wild populations if releases represent the contribution of a small number of founders. To reduce this possibility, it may be necessary to utilize as many founder animals as possible or to limit the representation from each breeding pair, and to release tadpoles, the lifestage with the highest mortality.

Animals intended for release should be acclimated to the conditions of the release environment. Post metamorphic migration away from the breeding pond requires further study to determine the importance in relocating breeding ponds. Post metamorphic movements may orient the toadlet to its new environment and provide a "map" so that the toad can later return to its natal pond for breeding.

The importance of the discovery of a breeding population of toads in Guanica Forest by Miguel Canals cannot be underestimated, nor can the series of actions he initiated to secure this site for subsequent breedings. The ditch which each year drained the flooded parking area in which the toads bred was dammed; a series of posts prevented cars from driving into the breeding area and from destroying vegetation used by the toads during egg laying; and during the three week period the road to the parking lot is closed to prevent contamination of the water. A public information campaign through local schools and newspapers has resulted in local acceptance of the changes which have occurred in this high use area to ensure the survival of the toad itself. The survival of the crested toad thus far has been dependant to a large extent on the efforts of DNR Forest Manager, Miguel Canals.

More recently, Metro Toronto Zoo has funded the production of 2,000 posters which have been distributed throughout Puerto Rico. The posters have illustrations which aid in distinguishing between the endemic crested toad and the introduced marine toad and provide addresses which are part of a program to locate additional populations and to increase awareness of this species and other threatened species in Puerto Rico.

CONCLUSION

Regional societies also have a role to play in amphibian conservation. Amphibian declines, once the subject of local concern, are now a global phenomena. The scale and extent of this decline can only be understood by monitoring long term changes in amphibian populations. The value of such programs will be enhanced if data is systematically collected in a similar manner from year to year and from region to region. Anecdotal data makes it very difficult to compare temporal and spatial changes in amphibian populations.

Regional societies are well placed to make a significant contribution to this long term process of data collection, the so-called "frog log". You are knowledgeable in the field. Data collection by regional herp societies is essential if we are to understand the nature of population changes before any declines are irreversible. We all must become involved if the rapid destruction of global biodiversity and disruption of ecological processes is to be halted. Our experience with the crested toad has demonstrated that despite the expertise of captive managers, it is still dedicated individuals who effect change. Conservation of threatened species is as much the result of understanding biological processes as it is the dedication of people in the field who focus public attention on the plight of threatened species and the consequence of their loss to the stability of local ecosystems.

LITERATURE CITED

Devison, D. 1991. Management program and breeding of the Puerto Rican crested toad at Metro Toronto Zoo. Proceedings of the 16th National AAZK Conference.

Johnson, B. 1989. The release of captive bred Puerto Rican crested toads: captive management implications and the cactus connection. AAZPA Regional Conference Proceedings. pp 962-967.

Odum, A. 1985. Water quality, an often overlooked parameter for the amphibian enclosure. IN: McKeown, S. and Caporaso, F. (Eds). Proceedings of the 8th International Herpetological Symposium. 118 pp

Paine, F. 1985. Husbandry, management, and reproduction of the Puerto Rican crested toad. IN: McKeown, S. and Caporaso, F. (Eds). Proceedings of the 8th International Herpetological Symposium. 118 pp

Puerto Rican toadlets (*Peltophryne lemur*) exhibiting "clumping" behavior to avoid dessication. Photo by Bob R. Johnson.

TAILS OF GILA MONSTERS AND BEADED LIZARDS

Robert Applegate

1627 Buckman Springs Road
Campo, CA 91906

INTRODUCTION

The beaded lizard (*Heloderma horridum*) has never reproduced successfully in captivity! The Gila monster (*Heloderma suspectum*) has only been successfully bred in captivity once, at the Oklahoma City Zoo. Now that I have your attention, let me explain why the above are in fact true statements. One of the stated goals of captive propagation of reptiles and amphibians is the establishment of captive breeding colonies so that we will no longer have to remove specimens from the wild, or that species which are extinct in the wild may be successfully maintained and propagated in captivity. A term used by the U.S. Fish and Wildlife Services is "self-sustaining populations". With the Gila monsters and beaded lizards we have so far failed in this task. The Oklahoma City Zoo has made a start with some production of second generation Gilas, but not nearly enough to establish a self-sustaining colony. I offer you a definition of successful reproduction as establishing a self-sustaining colony or colonies of a species in captivity. A supply of animals from the wild is still required to maintain these two species in captivity, therefore I conclude that my lead statements are sadly true.

According to Slavens (1990), there are 127 beaded lizards in 43 collections, however, there was only one reported breeding (mine). Slavens also lists 357 Gila monsters in 82 collections and only 9 (mine included) reported breeding the species that previous year (1989). Because of the laws around the country, most privately held individuals of these venomous species are not reported. It is a shame how well-meaning, but inappropriate laws on most government levels prohibit most of the private sector from contributing much needed knowledge regarding these animals. On the other hand, there are too many long-term captive animals listed for sale on reptile dealer lists for them to all have been legally acquired before they were protected by the various states where they occur. Of course the CITES Appendix II listing regulates the Mexico imports. Many wild-caught animals are also sold as captive-hatched. I know of one instance where several Gilas were wild-caught, smuggled out of Arizona, and offered as captive-hatched animals. One animal had been found on the road with one damaged eye, obviously hit by an auto. The smuggler did not even charge the recipient for that one because the extent of injury was not known and the animal may die. That animal survived and was later sold as captive hatched. The bad eye was explained away as a result of someone accidentally stepping on the animal while it was loose on the floor!! I guess if you want to believe, you will, don't let facts confuse you. Gila monsters are still quite common in parts of their range. Arizona has taken the lead in trying to get some of these animals in to proper hands. They have an urban salvage program where Gilas found in populated areas or displaced by development are distributed to those lucky enough, or attached well enough, to qualify as a recipient. Ask the Arizona Game and Fish Department for details if interested.

My personal involvement with the *Heloderma* dates back over 20 years when I was dealing in reptiles commercially and they were one of the commodities I bought and sold. When I heard that California planned to protect Gilas, I selected a few to use for my school lectures, obtained legal permits for them, and sold the rest. I had no interest in breeding them at that time. My current colony of beaded lizards were acquired in 1987 and 1988.

ENCLOSURES

Heloderma are easy to keep. My adults are in cages with a floor area measuring 1.5 x 3 ft, or 3 x 3 ft. The enclosures are either 1.5 or 2 ft high. Over the years I have tried almost every form of substrate, including sand, gravel, newspaper, ground corn cobs, indoor-outdoor carpet, wood shavings, and wood chips. They all work. *Heloderma* are messy animals, so keeping them dry and clean are the major objectives. You want enough substrate material to absorb the moisture of the feces until you can clean, but not so much that you can not find the feces or any uneaten food item. I provide water bowls large enough to hold the cage occupants without over flowing. I always keep a water bowl in the cage with the adults, but except for this, I keep the cages as dry as possible. I have observed lizards soaking even at 10°C in the winter. I also provide a hide box, usually cardboard boxes which I discard when they are badly soiled.

The adult cages are on two levels, which make up the outer walls of my snake room in the garage of my house. The backs of these cages ar glass and the doors open inside the room. The doors and tops are 1/8 inch mesh. Above the upper level are 8 ft fluorescent lights, and between the levels are 10 regular light sockets, spaced over 18 ft of cages to illuminate the lower level. These lower lights are wired into a thermostat and provide heat for the room, which also houses part of my colubrid snake colony. The temperature of the room is set for about 29°C, but the *Heloderma* can move against the outer glass wall to be cooler or warmer depending on the unregulated garage temperatures. When the lights are on for heat, animals on the upper level can lay on the warm spot above the light. There is no set light cycle, in fact almost the opposite with the lights tending to be on at night when it is cool, and off during the day. The cycle of the fluorescent lights is random, sometimes remaining off or on for days. There are windows in the garage (eastern exposure) so the lizards are exposed to a natural light cycle. Some morning sun hits the corner cages, and the lizards do sometimes bask where it hits. The sunlight, however, has passed through 2 panes of glass, so I do not believe there is any benefit except for a warm spot.

SEX DETERMINATION

Determining the sex of the animals is a major problem. I have tried probing, examining the anal scale differences, and observing behavioral differences. The only animals I was sure of were the ones that laid eggs. There are some subtle differences between the sexes with regard to head size and body configuration, but observing a large group together without any captive fattened individuals would increase the odds of determining the sexes correctly. Older males <u>tend</u> to have larger wider heads and narrower bodies than females. I recently became aware of the technique for sexing using the hypodermic injection of fluids into the lizard's tail, but was afraid to try it for fear of damaging my animals. In 1989, during the Northern California Herpetological Society conference, I had the opportunity to see pictures of how it was done, and talk to a veterinarian who had done it. The procedure was simple, but the fear of damaging valuable animals was still there. Shortly after this, I discussed the technique with my local veterinarian and friend, Dr. David Judy. He was familiar with the procedure and offered to help determine the sex of my specimens. For some reason he did not want 18-20 poisonous lizards in his office, so he came to my house. The technique is so simple it defies description (see Stewart, 1989 for details). Finally, after 18 years I know which ones are the girls! All but one of my original group were male!! Now I know why there were not more eggs.

The behavior of these lizards needs a lot of research before its meaning can be interpreted. I have had what turned out to be males behave aggressively (do not want to assume it was combat) towards each other with and without females present, females towards males, males towards females, females with each other, and in one case a young female looked like she was trying to copulate with her mother! Remember, for 18 years I did not know which animals were what sex, so I tried to figure it out by observing their behavior. Passive animals would sometimes become extremely aggressive when exposed to direct sunlight, but the significance of this is unknown. When housing several males together, it appears that some sort of dominance hierarchy was established. Even when no aggression is observed, one individual would sometimes refuse to eat, regurgitate meals, or just lose weight and become listless. Removing this individual to an isolated cage usually corrected the problem and the animal would regain its vigor and weight. On both instances where I observed this, excess captive hatched males were put with older wild caught males and became the subordinates, losing weight and vigor. After the subordinate animal was isolated and had resumed feeding, I sometimes had to give a small dose of Flagyl (100 mg/kg) to stop regurgitation. I am unsure of what caused the onset of the regurgitation: Flagellate parasites from the wild caught males could have infected the intolerant captive bred males or the stress of constant contact with a dominant animal could have lowered the subordinant's immune system. Another mystery that needs research.

FEEDING

Wild caught *Heloderma* frequently will refuse to eat rodents regularly, or at all. Sometimes dipping a mouse in egg will entice feeding. Also, a small rat may be preferred to a mouse. I had to force fed mice to one female from 1970 until 1989. She laid eggs regularly from 1974 until 1989 (probably a record of some type). The captive-hatched babies usually start on pinkie mice, but sometimes a drop of egg on the head of the pinkie, or the split-head pinkie technique is required for one to two feedings before they will switch to normal mice. All my captive-hatched animals will take rodents from tongs. Captive-hatched animals seem to be more aggressive than <u>most</u> of the wild caught *Heloderma*.

Heloderma can reach adult size in two or three years. I do not know what age they need to be for successful breeding because I have never hatched an egg from my captive hatched females, although some have laid eggs their third year.

REPRODUCTION

I turn the heat off November 1 and hibernate the room until March 1. My Gila colony has been producing eggs fairly regularly since 1974 (I did have a few from wild caught gravid females prior to that). I believe the cooling period is required to stimulate reproduction, because the years I kept the room warm for the boas and pythons were unproductive for the Gilas. Exposing the *Heloderma* to a winter brumation period like that used for colubrid snakes will increase your probability for reproductive success.

I mix the animals, introducing them into different cages from time to time to induce mating, but I have also observed many copulations between long-term cagemates. Neitman (1986) reports 10 copulations between April 18 and May 14 with the resulting eggs laid from May 31 through June. Except for one clutch of 8 eggs laid on July 3, my data correlates well with his. My beaded lizards copulated May 21, with eggs following on June 13 (1 bad), July 4 (1), July 10 (7), and July 11 (1). The two eggs that hatched were from the July 10 group and they hatched on January 29 of the following year. The same male *Heloderma h. horridum* mated with a female *H. h. alvarezi* on July 28 with bad eggs following from October 16-31. The female *alvarezi* seemed to be out of sync with the others. It is possible that even after almost 20 years in captivity, she still had a different biological rhythm than the others because her origin was further south. Unfortunately, she died that following winter, so I was never able to come to a conclusion. Taking sperm samples from *Heloderma* is much more hazardous than from colubrid snakes. Interestingly, *Heloderma* sperm looks more like a fat comma with a tail, instead of like the slimmer colubrid sperm. The movement of the sperm is not as smooth either.

I acquired a small colony of beaded lizards (1 male, 2 females) in late 1987. These animals had originally been purchased from Western Zoological (Monrovia, CA) in 1970. I estimated the age of the group at 20 years, and was concerned about breeding older, sexually inactive animals. They were in excellent condition, so I hibernated them right away and brought them out in March of 1988. One female bred in May and died in late June. She was starting to look larger, but she died while I was away so I was unable to confirm the presence of eggs. The second female laid eggs in 1988 (2 hatched in 89), again in 1989 (none hatched), and again in 1990 (3 hatched in 1991).

In 1990, she laid the eggs on approximately July 24. Sadly, I was away again and can't be sure of the exact date. I had given up on her laying eggs and had taken her out of the egg laying cage and put her back in her regular cage before I left for a week of work. When I returned, I noticed a large, 6-8 inch pile of wood shavings in the center of the cage. She advanced, hissed, and snapped at me as I investigated the pile (could be nest defense, but she is nasty anyway so I cannot be sure). When I got her away from the pile, I found 9 eggs buried in the shavings. They had gone unnoticed in my absence, and were partly desiccated. They all look as if they would have a chance, so I put them into my incubator, but only three survived to hatch between February 28 and March 10, 1991.

Over the years I have had *Heloderma* eggs laid on newspaper, in a collecting bag, in water bowls, etc. If I was present during laying, or soon after, many of these eggs were hatched. However, I have lost many to desiccation, drowning, and being eaten by the adult animals in the cage. Determining when a female is ready to lay eggs is difficult. Usually a puffiness around the rear legs can be observed, but exactly when egg laying should occur is still a mystery. When I suspect the female is close to laying, I put her in a 15 gallon glass aquarium that is 75% full of damp sphagnum moss. Usually, she will burrow down, lay her eggs, then surface without eating the eggs. The eggs stay damp in the moss. I check for eggs by lifting the aquarium up and looking up through the glass bottom. Other nests I have provided have not worked as well because the female usually digs all the material out of the container and scatters it about.

EGG INCUBATION

I put two to four eggs in a one pound plastic butter tub with about one inch of vermiculite under them. I push small indentations in the vermiculite to keep the eggs from rolling around. I add water to the vermiculite until the surface looks damp. I then place this tub inside a plastic sweater box with ventilation holes on all sides and one inch of water in it to float the egg container. Each sweater box will hold six or more tubs of eggs. I then stack these boxes inside my incubator (capacity 16 boxes), and set the temperature at 27.5°C. Once every second week I check the eggs, removing any obviously bad ones and adding water where necessary. This incubator is in the closet of my bedroom snake room, so the background temperatures are usually between 24° and 32°C. By November, when I begin to cool the room for the hibernation period, most of the colubrid eggs have hatched, and any Gila eggs are two to three weeks away. The beaded lizard eggs, however, stay in the incubator when the background temperatures drop down to between 10° and 15°C. The heat source inside the incubator can not compensate for this external drop, so the beaded lizard eggs have dropped to 21°C on occasion. This gradual fluctuation does not seem to effect the eggs as those that are still good by November usually hatch. The incubator has accomplished the three important factors for hatching *Heloderma* eggs; maintaining high humidity (water evaporating all around the egg tubs) without having the eggs too wet, good ventilation (besides air circulation inside the incubator, each time I open it the air is changed), and relatively stable temperatures (the water holds temperature, and any changes are very gradual). I could increase the intensity of the heat source for the eggs that over winter, but the cooler temperatures do not seem to be a problem so I will not try to fix it.

Table 1 summarizes the hatch weight, sex, and incubation period for the eggs and subsequent offspring produced by my colony. The data I have obtained does not vary significantly from that in previous publications. For further reading on *Heloderma*, I suggest the excellent paper on Gilas written by Howard Lawler and Warren Wintin in the 1987 NCHS Conference Proceedings. This paper has an extensive bibliography also.

JUVENILES

The baby lizards are individually housed in plastic shoeboxes, and are transferred into plastic sweater boxes as they grow. I keep the babies on newspaper and provide a small (not large enough to soak in) water bowl. When the bowl is provided they usually get the cage too wet, so one week they get water and the next week it is removed. There is a heat source under the back end of the shoeboxes so the lizard can thermoregulate. Cleaning is simple.

Table 1. Incubation dates, sex, and hatch weights for *Heloderma* offspring.

Oviposition Date	Hatch Date	Sex	Hatch Weight (grams)
Gila Monsters			
June 19	November 5	♂	35.1
June 19	November 13	♂	33.5
June 19	November 8	♀	34.3
June 30	November 14	♀	28.6
June 24	November 18	♀	36.4
July 3	October 25	♀	16.0 (Still alive)
June 30	November 17	♂	27.1
June 30	November 14	♀	26.7
June 24	November 17	?	38.4
June 24	November 22	♀	13.7 (Died at 3 months)
Beaded Lizards			
July 10	January 28	♀	47.0
July 10	January 31	♂	46.8
July 26	February 28	♂	38.0
July 26	March 3	♂	36.6
July 26	March 10	♂	41.5

I offer a mouse on tongs, the lizard attaches itself to the mouse and hangs on while I lift it into a waiting bucket. I clean the cage while the lizard eats the mouse. Then I dump the lizard back into its clean cage. Juveniles and adults often will immediately defecate on the clean substrate.

CONCLUSION

As more of my young female lizards grow to sexual maturity, I expect to produce a generation of captive reproduced animals. My entire 20+ years of experience with the *Heloderma* lizards has been one long learning experience. You can read volumes of material about raising these lizards, but this has little value since no one really knows the answers to the questions. We do not even know all the questions yet. All that I am sure I have learned is that we really don't know much about these lizards. Hopefully someone can use this material to enhance their ability to properly care for and reproduce these animals. That is not science, that is HERPETOCULTURE!!

ACKNOWLEDGEMENTS

I would like to thank Dr David Judy of the Judy Vet Clinic in El Cajon, CA for helping me solve the sex mystery of these lizards and some of the magic he has performed saving the lives of other members of my collection, Marilee Harding for her tireless work feeding the "critters" while I am away, and the herpetological community in general for the spirit of sharing information, such as at these Symposiums.

LITERATURE CITED

Anstandig, L. 1983. The Breeding and Rearing of the Mexican beaded lizard, *Heloderma horridum*, at the Detroit Zoo. IN: Tolson, P. J. (Ed). Proceedings of the 7th International Herpetological Symposium. pp 64-73

-------------. 1987. Management and Breeding of the Mexican beaded lizard, *Heloderma horridum*, at the Detroit Zoo. IN: Rosenberg M.J. (Ed). Proceedings of the 11th International Herpetological Symposium. pp 83-86

Lawler, H. and Wintin, W. 1987. Captive Management and Propagation of the Reticulated Gila Monster *Heloderma s. suspectum*. IN: Gowen, R. L. (Ed). Proceedings of 1987 NCHS Conference on Captive Propagation and Husbandry of Reptiles and Amphibians. pgs 49-56

Neitman, K. 1986. Captive Reproduction of the Gila Monster *Heloderma suspectum* at the Houston Zoo. IN: Peterson, C. H. (Ed). Proceedings of the 10th International Herpetological Symposium. pgs. 119-123

Slavens, F. 1990. Captive Reptile and Amphibian Inventory, current January 1990, Private Publication

Stewart, James. 1989. Techniques for Sex Identification in Reptiles. IN: Gowen, R. L. (Ed). Proceedings of the 1989 NCHS Conference on Captive Propagation and Husbandry of Reptiles and Amphibians. pp 99-104

Beaded lizard (*Heloderma horridum*) hatching. Photo by Robert Applegate.

THE BIOLOGY OF THE CHINESE CROCODILE LIZARD IN CAPTIVITY

Robert George Sprackland

Associate Curator of Herpetology
Department of Biology
San Jose State University
San Jose, California 95192

and

present address
Department of Herpetology
California Academy of Sciences
Golden Gate Park
San Francisco, CA 94118

INTRODUCTION

The Chinese crocodile lizard, *Shinisaurus crocodilurus* Ahl 1930, has long been an enigma to herpetologists. Discovered in 1928, described in 1930, it remained one of the rarest reptiles in museum collections for over half a century (Sprackland, 1977). Then, suddenly, specimens were exported from China to the international pet trade in large numbers. Natural history studies were limited to the original paper by Fan (1931), and though cited in later literature (eg., Pope, 1935, 1955), were not supplemented by new research until quite recently (Shen and Li, 1987; Zhang and Tang, 1985).

In contrast, the availability of crocodile lizards to zoos and private herpetoculturists has lead to an extensive body of data on the biology of this species (De Lisle, 1987; Magdefrau, 1987; Schafer, 1987; Wilke, 1987; Sprackland, 1989a, b; Visser, 1989; Gritis, 1990). This paper reviews these data and new observations made on eight adult and 21 juvenile crocodile lizards in the author's collection, and summarizes the current state of knowledge of successful crocodile lizard husbandry.

HISTORICAL RESUME

Shinisaurus crocodilurus was described from two preserved specimens sent to Berlin by Chinese zoologists (Ahl, 1930). Chinese internal politics effectively prohibited further study by western scientists during the following forty years. The few available shinisaur specimens in western museums were used for a variety of anatomical (Haas, 1960; Hecht and Costelli, 1969) and systematic (Nopcsa, 1932; McDowell and Bogert, 1954; Rieppel, 1980; Estes et al., 1989) studies. A primary focus of study involved determining the relationships between *Shinisaurus* and other lizards. Ahl (1930) proposed a family Shinisauridae to accommodate the unique species, a practice retained by Pope (1935, 1955). More detailed study of morphology allowed McDowell and Bogert (1954) to ally *Shinisaurus* with the Central American Xenosauridae, a relationship reaffirmed by later authors (Rieppel, 1980; Estes et al., 1989).

Herpetologists had come in contact with crocodile lizards by 1976, as evidenced by a pair of specimens donated that year to the Smithsonian's Museum of Natural History, but it was not until 1983 that published reports of studies based on live animals appeared (Huang, 1983). By 1985, crocodile lizards were being offered for sale on animal dealers' price lists in the United States and Europe. Initial experiences with husbandry in the United States met with high mortality (Sprackland, 1989b). In contrast, European herpetoculturists quickly succeeded in keeping lizards alive for a prolonged period (Wilke, 1986, 1987). Success was due, in large part, to recognizing the lower temperature requirements, highly aquatic habits, and territoriality of crocodile lizards. Subsequently, several reports have discussed husbandry (De Lisle, 1987; Magdefrau, 1987; Bartlett, 1989; Sprackland, 1989a, b; Perringaux, 1990; Gritis, 1990). Concurrently, reports of breeding were being published (Murphy, 1987; Schafer, 1987; Sprackland, 1989a; Visser, 1989), however, few reports were optimistic about breeding successes as most juveniles died within the first few weeks after birth.

The crocodile lizard occupies a very limited region of southern China, presently known from four localities in Kwangsi (Quanxi) Province, and even cursory examination of availability of the species to the pet trade during 1987-1990 suggests that continued collection from wild populations cannot long continue. Magdefrau (1987) reported that only 500 individuals were known from these populations, but he suggested that the rough terrain made precise population counts difficult. Nevertheless, such conditions warrant close monitoring by responsible zoologists. In response to population over-collecting, *Shinisaurus* has been placed on CITES Appendix II (Anonymous, 1990) because "although not necessarily threatened with extinction, may become so unless trade...is strictly controlled." Anecdotal, unpublished information gleaned from reptile importers and field researchers suggests that crocodile lizard populations may be seriously threatened. Consequently, serious research with the goal of successfully rearing neonates may be essential for survival of the species.

HOUSING

Shinisaurus crocodilurus is an inhabitant of low, temperate mountain forests, primarily at elevations of 200-400 m (Shen and Li, 1987). The habitat is moist, subject to 2000 mm rainfall per year (Shen and Li, 1987). Typical habitat is adjacent to swift-flowing streams with stony bottoms and some aquatic vegetation near shore. Not surprisingly, the lizard is highly aquatic, and captives may spend more time in the water than on land.

Terraria for crocodile lizards should be large, in part because the animals grow to 46 cm, and are territorial. Custom terraria should include a 50% division for land and pool areas. The pool should be at least 8 cm in depth, and contain water filtered by either an under-gravel or external box (preferred) system. In most areas of the United States, the water need not be heated above room temperature; crocodile lizards display no ill effects in water as cool as 8°C.

Crocodile lizards in my collection are housed in 55 gallon all glass terraria (91 x 43 x 30 cm). A 20 cm tall plexiglass partition is glued at a slight diagonal to the floor of the cage, dividing the floor space about midpoint. On one side is assorted, smooth, aquarium gravel, in which are placed small potted plants. Natural bark half cylinders are used as refugia. The other side is covered with an under gravel filter, 25 mm of aquarium gravel, and 10-12 cm of water. The inclined plexiglas retainer has several large pieces of gravel glued to its surface, providing footholds to allow the lizards easy exit from the pool. Complete details of the construction of this aquaterrarium are given in Sprackland (in press).

This terrarium design allows for very low maintenance housing for crocodile lizards. Crocodile lizards do not burrow, so potted plants (the pots buried in gravel) thrive in the moist environment. Constant filtration of water reduces the need for water changes to one or two changes per year. A gravel (versus peat) substrate (see Sprackland, 1989a) allows water to stay clear, and carries very little odor. High humidity and standing water do promote algal and fungal growths, necessitating frequent cleaning of glass and solid props (e.g., rocks, bark, dishes) in the cage.

Though reported to climb in the wild (Fan, 1931; Shen and Li, 1987), adults rarely do so in captivity (Gritis, 1990). Juveniles, however, are frequently found climbing into and sleeping in the foliage. The use of branches and other climbing props can be restricted to terraria intended for juveniles.

TEMPERATURE AND LIGHT

The natural temperature range for crocodile lizards is 18-35°C (Shen and Li, 1987). Captives therefore do well when maintained at normal room temperatures (Schafer, 1987; Sprackland, 1989a, b; Gritis, 1990). Early reports from animal importers relate high mortality in specimens kept for prolonged periods above 26°C, with mortality increasing as available water area is diminished. Crocodile lizards typically thermoregulate by basking in water, which should be kept between 22-24°C.

Crocodile lizards are reported to enter hibernation when ambient air temperature drops to 10°C for at least "a few days" (Shen and Li, 1987). This normally corresponds with mid-November, and lasts until March or early April. Various herpetoculturists relate that during these months, crocodile lizards typically become lethargic and feed rarely, if at all. My first crocodile lizards were acquired in January 1987, and refused to feed for several weeks. At the time, I interpreted this as a dislike for the offered food items (Sprackland, 1989b). Observations over four years have confirmed that such feeding apathy corresponds to winter low temperatures, and is replaced by very aggressive feeding with the onset of warmer weather (and possibly longer days) commencing in mid-March.

Originally, the shinisaur cage was illuminated by sunlight for 4-6 hours daily. When moved to larger quarters, artificial lights were used, specifically fluorescent lights coupled with either a Grow-Lux® or Vita Lite®. Heat lamps were never used.

FEEDING

Crocodile lizards are primarily insectivorous, feeding on crickets, beetles, waxworms, mealworms, jumbo mealworms, and moths, but taking other invertebrates including blood and earthworms, and snails. Though specimens in my care have never taken fish, other keepers have routinely fed their specimens goldfish (Schafer, 1987; Sautereau, pers. comm.). In fact, a group of 10 guppies introduced into my crocodile lizard pool in 1987 has not only remained intact, but has gone through ten generations (as of January, 1991) of happily breeding fish. Adult crocodile lizards will readily consume live newborn mice (Gritis, 1990; pers. obs.).

Gritis (1990) reported that his *Shinisaurus* would feed on seedless red grapes and strawberries, which may be the only record of these lizards taking non-moving food items.

It seems unlikely that crickets (or other orthopteran insects) form part of the natural diet of *Shinisaurus*. Newly acquired specimens have great difficulty in capturing the swift insects, and lizards must learn how to stalk and corner crickets. If a cricket enters the pool, crocodile lizards often charge the helpless insect, and much feeding activity occurs in the water. Specimens that are too inept at catching crickets can be offered insects that have had the large jumping legs removed.

During feeding, crocodile lizards become aggressive towards each other, and one lizard may take food out of the mouth of another. In some cases, lizards will bite other specimens, particularly tails and feet. It is essential to offer numerous food items at any given time into a terrarium containing more than one lizard to circumvent such attacks. (I may add that the one time I was bitten by a crocodile lizard--unquestionably the most painful bite I have every received from any reptile in the past 30 years--was when I tried to separate two lizards that had grabbed the same earthworm, and parts of each other. Anyone experiencing such a bite can understand how these lizards can easily consume snails and, probably, fresh-water clams.)

All invertebrates offered as food items are housed in small containers partially filled with dietary supplements of vitamins (Vitalife®; Terrafauna, Inc.) and calcium (Rep-Cal®; Rep-Cal Research Labs). Additionally, prey items are "dusted" with one of these supplements every third feeding, though at this time there are no well-researched studies of vitamin/mineral supplementation requirements for lizards.

During the hibernation/lethargy months, lizards are fed once every fortnight. From April to early November, food is provided 3-4 times per week, and during this time the lizards quickly hunt and consume all offered food items.

Over the course of 4 years, I have housed crocodile lizards with alligator lizards (*Elgaria*), emerald skinks (*Dasia smaragdina*), and caecilians (*Ichthyophis glutinosus*), all of which were smaller than the crocodile lizards. Never did a shinisaur attack any of these other species, though they would regularly take food from them if possible (caecilians often have to be hand-fed to insure they are getting food).

BEHAVIOR

Crocodile lizards are generally lethargic, slow-moving, foraging lizards. They nevertheless display a great many behaviors associated with territoriality, mating, and foraging that belie their lazy appearance.

TERRITORIALITY, MATING, AND SEXING.

Shinisaurus do not do well in crowded terraria. Specifics of territorial behavior from nature are lacking, but captive males will relentlessly chase and attack other males in the same enclosure. Intrusion into guarded territory is signaled by the resident male by raising the forebody well off the substrate, elevating the head, and raising the neck almost perpendicular to the deck. I have referred to this as "periscope" posture (Sprackland, 1989a). Initial display involved short, slow bobbing of the head, but not the neck. If this display is returned by the intruder, the resident male makes quicker, deeper jerking motions. If the intruder remains in position or advances, the defender may open the mouth slightly, while continuing jerky, deep movements. Both lizards may come very close, and if neither retreats, one may lower the neck and bite the opponent, often on a forelimb or shoulder region. At this point, 1) the attack may be returned in kind, 2) the bitten animal tries to move away, sometimes dragging the attacker before the bite is disengaged, or 3) the bitten animal is quickly released, at which point it retreats. Unless a terrarium is particularly large, the retreating lizard cannot move far enough away to avoid further attacks. On one occasion, the resident male crushed the bones of the knee of the intruder, though neither bleeding nor infection ensued.

The periscope posture is also a prelude to mating. The male initiates head-bobs as noted above. If a female lowers her head, the male may rush forward, bite her neck, and attempt mounting. If the female is unreceptive to mounting, she may lunge forward, or turn around and chase the male. Females defend their lack of receptivity by biting the hip and tail of the male, forcing his retreat. In contrast, if copulation occurs, it may last only for one or two minutes (pers. obs., two occasions).

After copulation, females are no longer receptive to further mating, and will often rush a male while he is in initial display. This behavior is particularly pronounced in gravid females, who become more aggressive as time passes. Females nearing parturition often chase males not involved in display, or, for that matter, otherwise interacting with the female.

Though males appear to initiate mating behavior year-round (Schafer, 1987), copulation probably occurs in March or April (Schafer, 1987; Visser, 1989), though wild populations are reported to mate from July to August (Zhang and Tang, 1985).

Sexing of crocodile lizards is difficult. Males that I have studied were more conspicuously marked with orange-red lateral blotches, and the throat is black with orange markings during displays. However, some females may be as brilliantly marked, and some males are drab brown with no orange markings (Sautereau, pers. com.). Various individuals relay that males are darker ventrally, have higher snouts, and more swollen tails, but I have been unable to corroborate any of these claims with either living or museum specimens of *Shinisaurus*.

METABOLIC REDUCTION.

Shinisaurus has the ability to reduce bodily functions for a considerable period of time. In January 1987, I observed a female who was apparently foraging in the terrarium (temperature 23°C) suddenly stop in mid step, and remain fixed in that position for 36 hours. I have noted elsewhere (Sprackland, 1989a) that they can remain submerged for hours, raising the nostrils for air at 18-45 minute intervals. Subsequently, I have had numerous opportunities to observe lizards for several hours uninterrupted, and have recorded submerged crocodile lizards staying completely submerged for 6 hours (possibly longer, but that was as long as I had to continuously observe). This behavior is not seasonal, occurring virtually all year.

LEARNING

The subject of learning in lizards is still not well researched. Crocodile lizards did learn to accept food offered by hand, and would take such offerings very gently (this practice was halted after being bitten). Despite early reports that handling induced stress resulting in lizards fasting, I found *Shinisaurus* easy to handle, usually quite docile, never offering to bite, except as noted above.

Meal worms and waxworms were kept in white plastic or styrofoam containers, and lizards would rush to the front of their terrarium upon seeing me approach with the container in hand. Lizards were not allowed to see moving prey until it was dropped into their enclosure, suggesting some mental link between seeing the container and knowing food was forthcoming. On two occasions I had left the container near the cage when I had to leave the room, only to return later to see a lizard frantically trying to reach the container through the glass.

REPRODUCTION

Crocodile lizards are prolific in captivity, with reports of reproduction coincident with the first reports on husbandry (Huang, 1983; Wilke, 1986; Murphy, 1987; Schafer 1987). Equally prolific have been reports of high juvenile mortality. Schafer (1987) reported that two female lizards at San Diego Zoo produced a combined total of 18 neonates, only eight remaining alive at the time she wrote her report (age: five months). Gritis (1990) reports on one stillborn birth, which by his account was premature. Visser (1989) records the birth of two live and three stillborn lizards from a June, 1987 birth, and six live lizards ("two of which died shortly after birth") from an April 1989 birth. Wilke (1985) records a birth of five crocodile lizards, all of which died.

My work with crocodile lizards began with two adult females acquired on January 14, 1987. Neither accepted food until February 1, and on February 14 the larger lizard delivered a single stillborn lizard in the pool at 6:00 pm, followed by a second stillborn at 10:00 pm (Sprackland, 1989b). A third neonate, still encased in the chorion, was deposited on February 20, and the fourth and final neonate delivered on March 10. The extremely long spread in delivering these neonates is remarkable, and I find no other record of a similarly prolonged delivery. On February 20, I also learned of a large female crocodile lizard at the California Academy of Sciences that had that week delivered six stillborn lizards and then died.

On November 23, 1988, another female crocodile lizard delivered eight live young. These were immediately removed to a small holding tank, then settled into a 30-gallon aquaterrarium, with a pool area of 13 x 21 x 5 cm.

The substrate was fine, smooth aquarium gravel, and moist sphagnum moss. By November 30, five were taking small mealworms; by December 3, all but one were taking food. On December 15, all eight were still alive, and had begun to accept waxworms. At night, most lizards would burrow into the substrate, but one would perch on a small branch suspended over the pool. A heat tape was used under the terrarium, and temperature was kept at 18-23°C. The first juvenile died on January 9, 1989, followed by another on January 27; two on February 4, two one February 26, and 2 on March 5. The two that died on February 4 were covered with a white fungal growth, not seen in any other lizards. All had been eating, sometimes within hours before death. On February 4, I divided the remaining 4 juveniles, so two were in a drier terrarium (substrate of paper towels, a small dish, 10 x 10 x 3 cm, for water), the others remaining in the large enclosure. By February 26, the two survivors had increased the diet to include small crickets.

On March 11, 1989, five stillborn were found from the smaller of the original females. On March 12, she was found dead in the terrarium.

On March 11, 1990, another female delivered six live, but very frail-looking, young. Half were left with the adults, half removed to a smaller terrarium. All died by May 20, 1990.

During November 9-11, 1990, the original (largest) female delivered another seven juveniles (see Figure 3). One was much smaller than its siblings, but the remaining six were larger and stouter than any previous young. Two lizards were given to two other herpetoculturists; one died on November 22, the other on January 16, 1991. The remaining lizards were kept in the large terrarium with the three adults. The first of these died on December 16, 1990. The others were regularly taking waxworms and small mealworms. They also took small redworms, but only once; afterwards, the worms appeared distasteful to the lizards. The crocodile lizards would seize a worm, then quickly release it and move away. Despite eating and looking robust, all juveniles died by January 2, 1991. Three had enlarged, open lesions on the dorsum. These lesions had been treated with topical antibiotics (Polysporin), and have subsequently been tentatively linked with tuberculosis.

JUVENILES

Young crocodile lizards resemble the adults, with the following exceptions: the head is proportionately larger and narrower across the parietal region; the top of the head is a tan or straw color, much lighter than the dorsum; and there is virtually no sign or orange-red coloring (two juveniles from the November 1990 birth had tiny orange markings just posterior to the axilla). The head coloration begins to darken after about 20 days.

Juveniles measure 11-15 cm total length at birth. Clutch size ranges from 1 to 10 (Sprackland, 1989b).

Juveniles are not notably more active than adults, but they are much more likely to climb, frequently found in live plants, on air line tubing to the filter, and on the backs of adults. Adult *Shinisaurus*, rarely inclined to tolerate the proximity of other adults, routinely allowed juveniles to perch on their backs, necks, and heads. Though adults are quite aggressive when feeding, they never were observed to take food in the vicinity of juveniles. Neither did they feed unless no juveniles were in proximity (e.g., juveniles in bushes or pool, adults on land). While no evidence of overt parental care was observed, crocodile lizards are obviously tolerant of the presence of juveniles which are clearly small enough to be considered possible prey items.

A single juvenile with aggressive, territorial behavior was observed in each of two litters. These individuals would chase any and all siblings from any position in the terrarium. Biting behavior was only noted once, despite long hours of observation for just such behavior. Juveniles did not engage in any display, nor assume the characteristic periscope posture seen in adults.

DISEASES AND MISCELLANEOUS

Crocodile lizards in my care showed few signs of disease. The large female obtained in January 1987 developed a small sore on the dorsum in February 1987. This was treated topically with antibiotic cream (Polysporin) daily for seven days, and the sore disappeared with no later recurrence.

Similar sores were apparent on juvenile crocodile lizards from the November 1990 clutch. These did not respond favorably to topical treatment, and it is possible they are related to cause of death.

There is no reliable information on longevity of crocodile lizards, and the first report to address the subject is apparently Gritis (1990), who noted that adults live for at least one year. The larger female that I obtained in January 1987 was 30.5 cm in total length on arrival, and measured 32 cm in January 1991. A sexually mature female obtained in November 1988 measured 26.1 cm, and gave birth two days after arrival. She measured 27.5 cm in January 1991. Private breeders report that juveniles attain small adult size in three years (Hudak, pers. com.).

Extrapolating from these data, the smaller female noted above was at least four years of age on acquisition, and was presumably 5 years old as of January 1991, while the larger female was at least seven, and possibly as much as nine years of age by the same date.

REARING OF JUVENILES

At this time there is no consensus on the methods to employ to successfully rear crocodile lizards. Private herpetoculturists are having better successes than those reported by zoos. David Martin (pers. com.) relates one keeper who has bred the animals several times over several generations, a feat apparently unparalleled elsewhere. According to Martin, this breeder keeps his neonates cooler than the adults, in separate terraria, and with large pools. However, another successful breeder relays that he keeps his juveniles warmer, with less water and lower humidity, and housed with the mother (Webber, pers. com.).

DISCUSSION

As of this writing, the Chinese crocodile lizard is a potentially endangered species, occupying a very limited range in mountainous Kwangsi Province. The huge number of lizards exported from China strongly suggests serious deleterious impact on native populations. Because of the ease of obtaining the lizards through 1989, many unverifiable reports have been circulated by importers and breeders to the effect that the species is extremely difficult to find at this time in the wild, and that China will drastically or completely limit export of *Shinisaurus* effective February 1991.

The number of animals available to private sources and zoos should be more than enough to initiate large scale, well documented captive breeding programs that can allow supply for researchers and breeders and produce animals for reintroduction in China. In the United States, Audubon Zoo is initiating a Studbook for *Shinisaurus* as part of a comprehensive breeding program (Snider, 1990). A second program, directed by the author, will utilize facilities of private individuals and The Reptile Breeding Centre in order to develop a successful breeding program and studbook, provide animal exchange with other breeders, and to publish viable breeding and rearing data so it is available to the herpetological community. At present, too much valuable and successful information is in too few hands. Our project welcomes any useful information from successful breeders of *Shinisaurus*. This unique species has shown itself to be long-lived and hardy in captivity. The remaining hurdle is to reduce juvenile mortality and thus eliminate the need to exploit wild populations while simultaneously providing stock to re-establish the lizard in damaged populations.

ACKNOWLEDGEMENTS

For help in acquiring specimens, I thank Kathy Hudak (Reptile Breeding Centre, Rochester, NY), Doug King and Ron Roper (Atlantis Pets, Los Gatos, CA). For sharing unpublished information on experiences with crocodile lizards, I thank Paul Gritis, Kathy Hudak, Ken Howell (Steinhart Aquarium), James B. Murphy (Dallas Zoo), Wanda Olson, Loic Sauterau, Jens Vindum (California Academy of Sciences), and Eric Webber. David Martin (San Jose State University) obtained valuable anecdotal information and has assisted in the ongoing breeding project. Harold De Lisle provided several interesting articles.

The following curators allowed me access to preserved *Shinisaurus* in their care, and their hospitality is greatly appreciated: Robert Drewes and Jens Vindum (California Academy of Sciences); George Zug (U.S. National Museum of Natural History); and Richard Zweifel (American Museum of Natural History).

LITERATURE CITED

Ahl, E. 1930. Beitrage zur Lurche- und Kriechtiere Fauna Kwangsi. 5. Eidechsen. Sitzungsberichte der Gesellschaft naturforschenden Freunde zu Berlin, 326-331.

Anonymous. 1990. New trade regulations. Tropical Fish Hobbyist, 38(9):118.

Bartlett, R. 1989. Digest for the Successful Terrarium. Tetra Press, NJ.

De Lisle, H. 1987. The Chinese crocodile lizard, *Shinisaurus crocodilurus* Ahl. Herpetology, 17(2):27-32.

Estes, R., K. deQueiroz, and J. Gauthier. 1989. Phylogenetic relationships within Squamata. IN R.Estes and G. Pregill, eds., Phylogenetic Relationships of the Lizard Families; 119-281. Stanford Press.

Fan, T. 1931. Preliminary report of the reptiles from Yaoshan, Kwangsi, China. Bull. Dept. Biol. Coll. Sci. Sun Yatsen Univ., Canton, 11:1-154.

Gritis, P. 1990. Observations on the lizard *Shinisaurus crocodilurus* in captivity. Herpetological Review, 21(4):81-83.

Haas, G. 1960. On the trigeminus muscles of the lizards *Xenosaurus grandis* and *Shinisaurus crocodilurus*. American Museum Novitiates, 2017:1-54.

Hecht, M., and J. Costelli. 1969. The postcranial osteology of the lizard *Shinisaurus*. 1. The vertebral column. American Museum Novitiates, 2378:1-21.

Huang, Q. 1983. A preliminary observation on the ovoviviparous habit of *Shinisaurus crocodilurus* in captivity. Acta Herpetologica Sinica, 2(2):77-79.

Magdefrau, H. 1987. Zur situation der Chinesischen Krokodilschwanz-Hockerechse, *Shinisaurus crocodilurus* Ahl, 1930. Herpetofauna, 51:6-11.

McDowell, S., and C. Bogert. 1954. The systematic position of *Lanthanotus* and the affinities of the anguinomorphan lizards. Bull. American Museum of Natural History, 105(1):1-142.

Murphy, J. 1987. Chinese crocodile lizards at the Dallas Zoo. AAZPA Newsletter, 28(1):12

Nopcsa, F. 1932. Zur systematischen Stellung von *Shinisaurus*. Zoologischer Anzeiger 97:185-187.

Perringaux, P. 1990. 100 Reptiles en terrariums. D.P.Perringeaux, Montmirail, France.

Pope, C. 1935. The Reptiles of China. American Museum of Natural History, NY.

-------. 1955. The Reptile World. A.A. Knopf, NY.

Rieppel, O. 1980. The Phylogeny of Anguinomorph Lizards. Birkhauser-Verlag, Basil, Switzerland.

Schafer, S. 1987. Breeding the Chinese crocodile lizard, *Shinisaurus crocodilurus*, at the San Diego Zoo. IN: Rosenberg, M.J. (Ed). Proceedings of the 11th International Herpetological Symposium, pp 103-105.

Shen, L., and H. Li. 1987. Notes on the distribution and habits of the lizard *Shinisaurus crocodilurus* Ahl. Bull. Chicago Herp. Soc., 22(1):4-5.

Snider, A. 1990. Information request. Herpetological Review, 21(4):77.

Sprackland, R. 1977. All about lizards. TFH Publications, Neptune, NJ.

-------------. 1989a. The Chinese crocodile lizard. Tropical Fish Hobbyist, 38(2):102-109.

-------------. 1989b. An enigmatic dragon brought to light; the Chinese crocodile lizard in captivity. The Vivarium, 2(2):12-24.

--------------. In Press. Aquaterrariums. TFH Publications, Neptune, NJ.

Visser, G. 1989. Chinese crocodile lizards *Shinisaurus crocodilurus* in Rotterdam Zoo. International Zoo Yearbook, 28:136-139.

Wilke, H. 1986. Eingewohnung, Haltung und Geburt der seltenen Krokodilschwanz-Hockerecsen, *Shinisaurus crocodilurus*. DGHT- Rundbrief, 88:12-13.

--------- 1987. The crocodile-tail lizard in Darmstadt Vivarium. Bull. Chicago Herpetological Society, 22(1):6.

Zhang, Y., and Z. Tang. 1985. The biology of *Shinisaurus crocodilurus*. Acta Herpetologica Sinica, 4(4):340.

A large group of *Cyclura c. carinata* on Little Water Cay, Caicos Islands. Photo by David W. Blair.

Bradypodion thamnobates
Photo by Bert Langerwerf.

Oustalet's chameleon, *Chamaeleo oustaleti*, captive-hatched young adult male.
Photo by W. Hines.

Chinese crocodile lizard, *Shinisaurus crocodilurus*, eating a waxworm.
Photo by Robert George Sprackland.

Puerto Rican crested toad, *Peltophryne lemur*, female.
Photo by Bob R. Johnson.

Yosemite toad, *Bufo canorus*; 6+ year old female.
Photo by David Lamar Martin.

Yosemite toads, *Bufo canorus*, breeding.
Photo by David Lamar Martin.

Cyclura n. nubila **on Isla Magueyes, Puerto Rico.**
Photo by David W. Blair.

Cyclura ricordi **in captivity.**
Photo by David W. Blair

Cyclura cychlura inornata **on Allen's Cay.**
Photo by David W. Blair.

Cyclura c. cychlura **in captivity on Andros Island.**
Photo by David W. Blair.

Cyclura r. rileyi **on Green Cay, San Salvador.**
Photo by David W. Blair.

Cyclura cychlura figginsi **in captivity.**
Photo by David W. Blair.

Eastern water dragon, *Physignatus lesueri*.
Photo by Bert Langerwerf.

Red-headed basilisk, *Basiliscus galeritus*.
Photo by Bert Langerwerf.

Beaded lizard, *Heloderma horridum*, enclosure.
Photo by Robert Applegate.

Gila monster, *Heloderma suspectum*, hatchlings.
Photo by Robert Applegate.

Pygmy spiny-tailed *Egernia depressa*.
Photo by R. Peter Yingling.

Tree skink, *Egernia striolata*.
Photo by R. Peter Yingling.

Blood pythons, *Python curtus*, hatchling.
Photo by Ron Goellner.

Blood python, *Python curtus*, female with clutch of newly laid eggs.
Photo by Jeff Ettling.

Baja mountain kingsnake, *Lampropeltis zonata agalma*.
Photo by D. McCloud.

Blood pythons, *Python curtus*, hatching at the St. Louis Zoological Park.
Photo by Jeff Ettling.

Tent tortoise, *Psammobates tentorius*.
Photo by Sean McKeown.

***Philodryas baroni*.**
Photo by Harold De Lisle.

WEST INDIAN ROCK IGUANAS: THEIR STATUS IN THE WILD AND EFFORTS TO BREED THEM IN CAPTIVITY

David W. Blair

Cyclura Research Center
316 West Mission, #117
Escondido, CA 92025

INTRODUCTION

Rock Iguanas of the genus *Cyclura* are among the largest and most impressive lizards in the Western Hemisphere. Unfortunately, they are also among the fastest disappearing lizard species on Earth. Historically they have been restricted to islands in the Bahamas, Greater Antilles, and Virgin Islands, although their former range was certainly much greater than it is today. On most of the islands where they still occur, they are the largest of the surviving native land vertebrates. In fact, adult iguanas have virtually no known natural predators. Their decline in numbers probably began in Pre-Columbian times when the Arawak and Lucayan Indians sought them as food items to supplement their meager diets. Bones of *Cyclura* have been found in caves and Indian kitchen middens on several islands where they no longer occur: Saint Thomas, New Providence, Great Exuma and Puerto Rico. The pressures of hunting by these native peoples may have initiated the process of decline of the rock iguanas, however, the real devastation began with the arrival of Europeans to the islands. Not only was much habitat lost as the human population increased but, with him, man brought his domesticated animals, many of which turned feral. Today the greatest threats to the rock iguana are probably the direct predation by or competition with these introduced animals. The list is extensive, with cattle, goats, pigs, rats, mongooses, dogs, and/or cats present on most inhabited islands throughout the West Indies.

These large diurnal lizards inhabit subtropical areas of Antillean xerophytic thorn forest which is typical of West Indian islands. The substrate is usually heavily eroded limestone composed of many crevices which the iguanas utilize as retreats; several species sometimes dig their own burrows in sandy areas. Adults usually establish home ranges around these retreats which they actively defend against intruders. Often the home range of a dominant male will overlap with that of one or more females. Most wild *Cyclura* are generally polygamous, but individuals may also be promiscuous or monogamous.

Rock iguanas are chiefly herbivorous, ingesting the leaves, flowers or fruits of dozens of different plants. They also consume some animal matter in the form of very easily attainable items such as insects, land crabs and carrion, including dead sea birds, fish and, occasionally, other iguanas. Iguanas are good climbers and have been observed browsing in trees more than five meters above the ground.

BREEDING

Mating virtually always occurs during the same two week period each year, regardless of climatic conditions. This would indicate that it is triggered by seasonal changes in the photoperiod. Depending upon the species, mating occurs from late May until mid June, with oviposition occurring approximately forty days later. Females often travel outside of their normal activity ranges to find areas that are suitable for nesting. Several exploratory nests are sometimes initiated before one is actually completed. Nest burrows average 0.4-1.5 m in length, with an enlarged chamber at its terminal portion to allow the female to turn around. Smaller species of rock iguanas lay from 2-6 eggs, while some of the largest forms may lay as many as twenty-three. Egg size is dependent upon the size and age of the nesting female. *Cyclura* eggs are among the largest laid by any lizard in the world. After laying, females often guard nest sites for up to one month, repelling any intruder that ventures too close. The temperature within the nest remains a surprisingly constant 30-33°C degrees throughout the entire incubation period of 80-90 days. It takes the combined effort of many hatchlings to dig out of a nest chamber, sometimes not emerging for as long as two weeks after hatching. Neonates disperse quickly and lead solitary lives. Unfortunately, mortality rates for juveniles are extremely high and on at least some islands, it approaches one-hundred percent.

In the wild, rock iguanas reach sexual maturity at 6-9 years of age. It is believed that they are also among the longest lived of any lizard, reaching ages of at least twenty-five to forty years. Some researchers believe that they may actually live to be closer to eighty years of age!

TAXONOMY and STATUS

The most recent revision of the genus *Cyclura* was conducted by A. Schwartz and M. Carey in 1977. They examined 378 preserved specimens and named seventeen forms representing eight species. Of these recognized forms, at least one is thought to be extinct, two others nearly so, and many have scattered populations of various status. I have spent the last fifteen years observing many populations in the wild and searching all available literature in an attempt to determine the status of all extant *Cyclura*. I would like to review and update each one of them here:

Cyclura carinata carinata. Turks and Caicos Rock Iguana

Distribution: Turks Islands (Big Sand Cay, Long Cay); Caicos Islands (Pine Cay, Ft. George Cay, North Caicas, Big Iguana Cay, Ambergris Cays, Water Cay, Little Water Cay).
Size: Males up to 360 mm snout-vent length and 1.864 kg. Females up to 242 mm snout-vent length and 1.135 kg.
Status: Of the twenty or more populations, some are still dense, others definitely declining, and several recently extirpated (Pine Cay, Long Cay off South Caicos, and Six Hills Cay)
Number Remaining: Probably several thousand
Most important areas to protect: Ambergris Cays, Big Iguana Cay, and Little Water Cay

Cyclura carinata bartschi. Bartschi's Rock Iguana

Distribution: Known only from Booby Cay off Mayaguana Island, Bahamas
Size: Males up to 335 mm snout-vent length. Females up to 285 mm snout-vent length
Status: Approximately 200-300 with juveniles still present
Protection needed: All of this very small uninhabitated Cay should be protected. Herds of goats now present should be removed. Much of the Cay shows signs of over-grazing

Cyclura collei. Jamaican Rock Iguana

Distribution: The Hellshire Hills on the island of Jamaica
Size: Over 490 mm snout-vent length
Status: Thought to be extinct for many years, a live specimen was recently brought to a zoo in Kingston, Jamaica. Unfortunately, it died after several months in captivity. A search of the area where it was found

brought twenty-three additional sightings of what was thought to be fifteen individual animals. Probably fewer than one hundred still remain.
Protection needed: The entire Hellshire Hills area should be set aside as a preserve with a resident warden. All additional development and hunting in the forty-four square mile uninhabitated area should be excluded. Charcoal burners have now moved to within 1.5 km of the iguanas' only remaining habitat.

Cyclura cornuta cornuta. **Rhinoceros Iguana**

Distribution: Hispaniola; Isla Beata; Ile de la Petite Gonave; Ile de la Tortue; Ile Grande Cayemite; reported from Isla Saona and Isla Cabritos in Lago Enriquillo
Size: Males up to 545 mm snout-vent length and 6.8 kgs. Females up to 510 mm snout-vent length and 5.4 kgs.
Status: Must certainly be declining in impoverished Haiti and the Dominican Republic. Because of its large distribution, there are probably several thousand left in the wild. Nobel visited Isla Cabritos in 1923 and found none present.
Protection needed: All uninhabitated offshore islands where the iguanas are still present and the Cul-de-Sac - Valle De Neiba plain and the Peninsula de Barahona. In recent years hatchlings produced at both Toranga Zoo in Australia and Zoo Dom in Dominican Republic have been released into the wild.

Cyclura cornuta onchioppsis. **Navassa Island Rhinoceros Iguana**

Distribution: Known only from Navassa Island, now thought to be extinct.
Size: Males up to 420 mm snout-vent length. Females up to 378 mm snout-vent length
Status: Navassa Island was visited in 1966 and 1967 and no animals were present. An entomologist visited the island again in 1986 and saw no signs of any iguanas although he was not specifically looking for them. Military occupation of the island prior to the 1960's was apparently responsible for its extirpation.
Protection needed: A thorough search should be made of the entire island to determine if any iguanas remain. A very slim possibility exists that some specimens may remain in captivity somewhere in the world but they would certainly be aged. Unfortunately sub-species of *C. cornuta* are very difficult to distinguish from one another.

Cyclura cornuta stejnegeri. **Mona Island Rhinoceros Iguana**

Distribution: Mona Island (off Puerto Rico)
Size: Males up to 535 mm snout-vent length and 6.4 kgs. Females up to 490 mm snout-vent length and 5.2 kgs.
Status: Mona Island probably still supports 2500 to 3000 iguanas. They are reproducing successfully but the mortality rate for hatchlings is extremely high.
Protection needed: The Puerto Rican government has recently restricted visitation to the island. If a program to remove feral animals is initiated, and successful, its continued survival is hopeful.

Cyclura cychlura cychlura. **Andros Island Rock Iguana**

Distribution: Andros Island, Bahamas
Size: Males up to 411 mm snout-vent length. Females up to 465 mm snout-vent length.
Status: Estimated at 5,000 in 1961; probably many fewer remain today.
Protection needed: Presently animals are most abundant on the scattered uninhabitated cays of western South Andros. This entire area should be set aside as a preserve with a resident warden to enforce protection.

Cyclura cychlura figginsi. **Exuma Island Rock Iguana**

Distribution: Exuma Cays, Bahamas; Guana Cay, Prickly Pear Cay, Allen Cay, Guana Cay off Norman's Pond Cay, Ozie Cay, Bitter Guana Cay, Gauling Cay)
Size: Males up to 355 mm snout-vent length and 1.4 kgs. Females up to 320 mm snout-vent length and 0.85 kg
Status: Of the seven known populations, five were in good shape in 1980, while two were definitely declining. I visited Guana Cay near Great Exuma in 1985 and found a dense colony on the small cay with juveniles present. Probably fewer than 1000 exist.
Protection needed: All uninhabitated cays with iguanas present should be incorporated into the Exuma Land and Sea Park system and afforded protection.

Cyclura cychlura inornata. **Allen's Cay Rock Iguana**

Distribution: U Cay, Leaf Cay, and Allen's Cay, Exuma Islands, Bahamas
Size: Males up to 380 mm snout-vent length. Females up to 220 mm snout-vent length.
Status: Two of the three small cays support fairly dense populations of iguanas. Fewer than 500 remain.
Protection needed: The islands where these iguanas occur are private property and a popular yachting destination from Nassau. There are often a dozen or more boats anchored in the small harbor. Hopefully these cays can be incorporated into the Exuma Land and Sea Park. Introduced non-native trees are threatening nesting areas and should be removed.

Cyclura nubila nubila. **Cuban Rock Iguana**

Distribution: Cuba and Isla de la Juventid; Archipielago de los Canarreos; Cayow de san Felipe; Jardin de la Reina; Archipieago de Sabana-Camaguey; Cayo Cinco Leguas, and many other islets and cays; introduced on Isla Magueyes off southwestern Puerto Rico.
Size: Males up to 745 mm snout-vent length. Females up to 623 mm snout-vent length.
Status: This very large iguana has by far the largest distribution of any member of the genus. As a result, it probably exists in greater numbers in the wild than any other *Cyclura*. Servicemen report many individuals on the grounds of the U.S. military base at Guantanamo Bay. Density studies on small undisturbed cays have shown up to twenty-five iguanas per hectare. The introduced population on Isla Magueyes off Puerto Rico is thriving. These animals escaped or were released from a small zoo in the mid 1960's, but we have been unable to ascertain from what specific area of Cuba this small group originated. Interestingly, most of the Puerto Rican animals have predominantly rusty orange and brown coloration, while those from mainland Cuba are described as gray or "greenish" with tan.
Protection needed: A thorough population study should be conducted. Apparently the Cuban government has expressed a willingness to cooperate in such an endeavor. Any uninhabitated cays with viable populations of *C. n. nubila* should be set aside as reserves and afforded protection.

Cyclura nubila caymanensis. **Cayman Island Rock Iguana**

Distribution: Cayman Islands: Little Cayman Island, Cayman Brac.
Size: Males up to 483 mm snout-vent length. Females up to 420 mm snout-vent length
Status: Only a very small population exists on Cayman Brac. Little Cayman still supports a reproducing population, but feral cats take a great number of juveniles. Once more numerous, there are probably less than 500 individuals remaining today.
Protection needed: Recently, plans to construct the world's largest oil transshipment terminal on Little

Cayman have fortunately been "indefinitely postponed". All remaining uninhabitated areas of the island should be protected from further development and have a warden in residence. Attempts should be made to bring the feral cat population under control. Additional imports of domestic animals to the island should be prohibited.

Cyclura nubila lewisi. **Grand Cayman Blue Rock Iguana**

Distribution: Cayman Islands; Grand Cayman Island
Size: Males up to 515 mm snout-vent length. Females up to 410 mm snout-vent length
Status: Very seriously endangered. There are thought to be less than 50 animals remaining in the wild on Grand Cayman Island.
Protection needed: Captive breeding projects have been quite successful both in the U.S. and recently on Grand Cayman. The Cayman National Trust has established two reserves on the island into which, ultimately, captive produced animals will be released. The present concern is to be certain that all animals destined to be released are purebred lewisi. There is now evidence that some of the captive stock may have been inadvertently mixed with caymanensis. Current work with blood haplotypes should clear up this confusion in the near future.

Cyclura pinguis. **Virgin Islands Rock Iguana**

Distribution: British Virgin Islands; Anegada; introduced on Guana Island
Size: Males up to 550 mm snout-vent length and 7.75 kg. Females up to 500 mm snout-vent length and 5.25 kg.
Status: Endangered. Probably less than 300 remain. Although this iguana is still reproducing successfully on Anegada, survivorship of juveniles is very low due to the large population of introduced cats, dogs, goats, and cattle. Between 1985 and 1986, at least eight adult iguanas were captured on Anegada and transported to Guana Island near Tortola. Indications are that at least some successful reproduction has taken place on that island.
Protection needed: Additional studies are required to identify areas frequented by *C. pinguis* on Anegada, particularly the eastern uninhabitated portion of the island. Reserves should be established along the northern coastal areas and on the peninsulas and cays associated with the interior ponds. Guana Island is a small, privately owned resort hotel island whose owner considers it a "nature reserve".

Cyclura ricordi. **Ricords Rock Iguana**

Distribution: Hispaniola; Valle de Neiba, the Peninsula de Barahona, Isla Cabritos in Republica Dominicana; probably the Plaine de Cul de Sac in Haiti.
Size: Males up to 460 mm snout-vent length and 3.18 kg. Females up to 365 mm snout-vent length
Status: *C. ricordi* and *C. c. cornuta* are the only sympatric forms of *Cyclura*, although the range of the former is much more restricted than that of the latter. Estimated at near 5,000 individuals in 1970, the population must certainly be greatly reduced today.
Protection recommended: Isla Cabritos in Lago Enriquillo is quite difficult to reach and very isolated. This lake also harbors one of the largest concentrations of salt water crocodiles in North America. The entire area should be established as a national park with resident wardens. Surprisingly, *C. ricordi* (along with *C. c. cornuta*) is not listed by the United States as threatened or endangered.

Cuclura rileyi rileyi. **San Salvador Rock Iguana**

Distribution: San Salvador Island, Bahamas; Low Cay, "Guana" Cay, Green Cay, Man Head Cay, Pidgeon Cay, and Goulding Cay
Size: Males up to 400 mm snout-vent length and 1.25 kg. Females up to 320 mm snout-vent length and 1.25 kg
Status: Endangered. Probably less than 500 animals remain. Populations are dense on some of the cays of San Salvador while others are very sparse. They have been extirpated on several cays which supported substantial populations of iguanas in recent years. Contrary to some earlier reports, *C. r. rileyi* does still inhabit mainland San Salvador.
Protection needed: Access to cays where iguanas still occur should be limited to prevent the introduction of feral animals. Land and sea parks could be established to incorporate the most important of these cays. At least "Guana" Cay, Green Cay and Low Cay should have routine visits by wardens to ensure protection is enforced.

Cyclura rileyi cristata. **Sandy Cay Rock Iguana**

Distribution: Sandy (White) Cay at the southern end of the Exuma Islands, Bahamas. Possibly introduced on a small cay within the Exuma Cays Land and Sea Park.
Size: Males up to 280 mm snout-vent length and 0.634 kg. Females up to 254 mm snout-vent length
Status: Vulnerable. The population on tiny Sandy Cay is quite dense but I saw few juveniles present in 1984. Historically, this was the only known habitat of *C. rileyi cristata* but recently an introduced population was discovered on a small cay within the protection of the Exuma Cays Land and Sea Park, which may prove to be *C. r. cristata*. The combined populations of these iguanas probably total less than 400 animals.
Protection needed: Sandy Cay should be officially designated a wild life reserve with a warden present to enforce protection and to alert visiting yachts not to bring dogs or cats ashore. The cay is apparently sometimes used as a trash dump by passing boats.

Cyclura rileyi nuchalis. **Crooked Island Rock Iguana**

Distribution: Fortune (Long) Island, Fish Cay, and North Fish Cay in the Crooked-Acklins group, Bahamas
Size: Males up to 263 mm snout-vent length. Females up to 240 mm snout-vent length
Status: The population on Fortune Island has been greatly reduced if any remain at all. Fish Cay and North Cay, although very small in area, still support fairly dense populations of iguanas. There are probably only several hundred animals remaining.
Protection needed: Officially all iguanas in the Bahamas are protected by law, but regrettably enforcement throughout the islands is almost nil. Certainly these small cays will remain very vulnerable to disturbances without a warden present to enforce protection. As this is a popular area for fishing boats, there is always the danger of cats or rats being introduced by visiting yachts.

CAPTIVE HUSBANDRY

Up until the last twenty years, *Cyclura* have done very poorly in captivity. The average longevity in zoos worldwide before 1981 was less than 2.5 years. Apparently the private sector has done somewhat better. We currently maintain many animals that are 10 to 20 year captives. There are reliable records of animals that have been captive for more than 35 years. Most of these older animals show little evidence of their advanced ages and, in fact, appear to be in their reproductive prime.

We believe our success in breeding these iguanas at Cyclura Research Center (C.R.C.) is due to several factors. The first of these is a high quality balanced diet, heavy on green leafy vegetables. We feed the following mix on a daily basis to hatchlings and 2-3 times per week to adults. Percentages of each item used are in the exact quantities listed:

- one bunch kale
- one bunch collard greens
- one bunch mustard greens
- one bunch beet greens
- one bunch parsley
- one head romaine lettuce
- one head endive

(Seasonal substitutes for any of the above: one bunch chard, one bunch turnip greens, one head escarole)

- one lb. bean sprouts (or soy bean sprouts)
- one lb. squash, grated (banana or zucchini)
- 2/3 lb. carrots, grated
- 2/3 lb. beets, grated
- 2 lbs. fruit, chopped (bananas, melons, apples, plums and pears)
- 1/2 cups dry low fat premium dog food soaked in water to soften (Science Diet, Iams, Pro-Plan, Nutro, or primate or monkey chow)
- 1 heaping teaspoon vitamin and mineral supplement (Vita-Life, Reptivite, Super Preen, etc.)
- 1 heaping teaspoon cooked bone meal
- hatchlings and juveniles are also offered crickets, super meal worms, or pinkies once per week
- fresh water for drinking is available at all times

All of our iguanas are kept in outdoor enclosures constructed of a wood framework covered by 14 gauge galvanized, after welded wire screen; 1/2" x 1" mesh wire is used on cages housing juveniles, while 1" x 2" mesh is adequate to confine adults. Two basic designs have been used. Originally, we built cages around a central building constructed on a concrete slab and heated by a thermostatically controlled space heater. Individual inside cages within the structure are lined with formica and have large access doors to facilitate easy cleaning. Openings are cut through the building's walls to allow access to the exterior cages. There are locking doors on these openings so that animals can be restricted to either section while cleaning or maintenance is being conducted.

The outdoor portion of the enclosure is begun by digging a 4" wide by 18" deep trench into which concrete is poured to form a footing. A wood frame is constructed and is attached to the footing either with lag-bolts or with spikes shot by a stud gun. The welded wire is then attached to the wood frame with 0.25" wide by 1.5" long staples from a pneumatic staple gun. A 1.5 x 1.5 m section in one corner of the outdoor enclosure is dug and soil removed to a depth of about 0.4 m. This hole is filled with sand to be used as the nesting area for the iguanas. The remainder of the enclosure is planted with grass and landscaped with piles of rocks and small palm trees. While this is our preferred enclosure, it has several disadvantages. The building and concrete footing and slab are expensive and time-consuming to construct. Also, in many areas of the U.S. such structures require building permits and must be inspected periodically during construction, resulting in several delays.

A second design is now used on all newly constructed enclosures. First, a base is assembled out of 2" x 6" clear redwood which is laid upside down on a level area. PVC coated hex mesh wire is stapled across the frame and individual strips are overlapped and secured to one another with hog ring clips. This coated wire mesh prohibits any iguanas from burrowing out of the enclosure. The entire base is then flipped over and filled with 6 inches of sand or decomposed granite. A 2" x 4" wood frame is constructed on this base and covered with the galvanized welded wire. A 24" wide strip of 5/8" outdoor siding is added along the lower sides of the enclosure to prevent nervous animals from charging into the wire or rubbing their noses on it. Cages are normally at least 6 feet tall so they may be serviced comfortably by an average height adult.

Within each cage, a 1 m long x 0.5 m wide x 0.3 m high wooden box is placed on concrete blocks. This box is constructed with 2" thick sides and 0.75" plywood top and bottom, coated several times with a non-toxic sealer. The top is hinged so the interior is easily accessible and a lockable door is cut in the front side to allow animals entry. 110 volts A.C. is run in plastic conduit to a grounded outlet box in each container. A rigid heating pad designed specifically for outdoor use with animals is used as a heat source in each box. These are available from several manufacturers (Kane, Lectro-Kennel, Osborne, etc.) in a variety of sizes. Most draw about 75 watts and can be used with a rheostat to vary the heat output. Heat pads should fit the container so that there is approximately 10 cm of space between its edges and sides of the box. This allows the animals to lie mostly off the pad if there temperature becomes too warm, but does not allow them to get so far away from the pad that they become chilled when temperatures are quite low. This heating method has proved successful for more than 5 years at our inland Southern California location, where outside winter temperatures sometimes drop to -5°C. All heaters are wired to a time clock so they may be run for only a few hours at night, or used continuously during inclement weather. Supplemental heat is not used at all during six months of the year. It is first turned on when nighttime temperatures begin to drop below 12.5°C. Animals are then checked every night to be sure they are inside the heated boxes. When temperatures drop lower than 10°C the doors to all the boxes are closed every evening and opened up again every morning. This not only helps to retain heat within the unit but prohibits any animal from leaving the boxes when outside temperatures are dangerously low.

Mid-summer temperatures can sometimes reach 44°C. To help moderate such heat, 70% shade cloth is installed over two thirds of each enclosure. A drip irrigation line runs down the center of each roof with spray nozzles every few meters. These misters are controlled by an automatic watering timer and can be programmed to come on as often as needed or operated manually. Fresh water faucets are present in each cage for washing and refilling water receptacles.

REPRODUCTION

Previous authors have listed lack of sufficient space as the primary reason that rock iguanas fail to thrive and reproduce in captivity. While I would certainly agree that enclosures provided should be as large as possible, access to natural sunlight and adequate diet is at least as important. We have had young adult *C. nubila* produce viable eggs in cages with as little as 3.25 square meters of floor space. A more appropriate size, however, for a large adult pair would be at least 9 square meters supplied with several basking sites. Visual barriers are not usually necessary with compatible pairs, but can be helpful in reducing stress during breeding and nesting season. We have tried several different arrangements for establishing breeding groups, including pairs, trios, and quads. Dominant adult males can rarely be kept together except in extremely large enclosures that are usually impractical to construct. Females are often quite aggressive toward one another and nearly always establish a pecking order. Submissive females often do not do well and always remain smaller and exhibit a slower growth rate than their dominant counterparts. Relationships sometimes change very quickly in an apparently compatible trio or quad. Even after many peaceful months together, a subordinate female may suddenly be found with missing digits or more serious lacerations. This often occurs immediately after females have oviposited. The most compatible group has proven, not surprisingly, to be one male and one female. We arrange cages so males can see one another and display back and forth. There is some evidence that these displays may help to stimulate breeding activity.

Copulation can occur at various times throughout the year in captivity, particularly when iguanas are introduced to each other for the first time. Normally, however, breeding takes place for only a few weeks each year during May and June. I have only observed successful copulation approximately twenty times in fifteen years. It is usually preceded by numerous head bobs by the male, who then circles around behind the female grasping the nape of her neck in typical iguanid fashion. Intromission generally lasts from 1-3 minutes and a pair normally mates no more than once or twice per day. Our captive *C. nubila* are always the first species to oviposit beginning in the first week of July. *C. cornuta* are usually somewhat later, nesting in mid-August. Eggs laid after October 1 have always been infertile.

Gravid females normally begin the nesting process with a reduction in their food intake and an increase in activity level about two weeks before laying. Abdomens often become distended to the point where the outline of individual eggs can easily be seen. Several days before actual laying, females begin to dig all over the cage, excavating many shallow depressions. In the cages with only 6 inch deep sand substrate, a triangular piece of plywood is installed in one corner. This enables even the largest females to excavate adequate nests under the plywood in sand which would otherwise be too shallow. It usually takes most of one day to excavate a complete nest burrow. Females often remain in burrows overnight rather than returning to their regular retreats. Occasionally females will remain underground up to two days after laying, but more often emerge the following morning and begin to fill in the nest burrow with sand. Huge quantities of sand are removed from around the nest site until a large mound is formed over the spot where the eggs were laid. Eggs are removed immediately after oviposition and artificially incubated, but females will continue to guard nest sites for up to two months. If the mound of sand is raked flat after egg removal, the female will immediately move large quantities of sand to attempt to re-cover the area. This behavior normally lasts for one or two weeks, but can be induced for months afterward by turning on the sprinkler system or by a moderate rain.

After removal, eggs are placed into plastic sweater or shoe boxes containing 5 cm of vermiculite. This vermiculite has been moistened by adding one part distilled water to one part vermiculite by weight. Eggs are placed into the substrate with one half buried and the other half exposed to the air. Care is taken not to turn the eggs over and to keep them in the same position as they were laid. The loose fitting covers are set onto each container and they are placed into an incubator set at 30°C. Containers are opened one minute every three days to allow for some air exchange. A pan of water in the bottom of the incubator helps to maintain a fairly high humidity. Fertile eggs are usually full, turgid, and white in color. They are distinguishable from infertile eggs which are often soft, flabby and somewhat yellow in color. Eggs which are retained too long by a female may also be hard and misshapen. All eggs are incubated, but ones appearing to be infertile are kept separate from "good" eggs and usually collapse and mold very quickly and are subsequently discarded.

During incubation, fertile eggs increase substantially in size as they absorb moisture from the substrate. At 30°C, eggs begin to hatch in as soon as 80 days, often dimpling in slightly several days before the first slits appear. After successfully slitting the egg shell with their tiny egg tooth, hatchlings will often sit with only their heads protruding for up to two days. Once free of the eggs, they are left in the container for three days to allow the "umbilical" scar to heal and because the movement of the first ones out crawling over the others probably stimulates them to hatch in unison. This is necessary in the wild to assure that they can burrow to the surface from the underground nest chamber, a process which requires a group effort. Once hatching is observed, covers of the containers are set ajar to allow for more air circulation. They must, however, be taped securely in place as the active youngsters can easily knock them off and scatter about the incubator. At these temperatures, all hatchlings are out by 90 days. For this time period, it is not necessary to add additional moisture to the substrate, but if incubation exceeds 100 days, more distilled water should be added. Even a few degrees reduction in temperature will result in prolonged incubation times of as much as 120 to 140 days with a larger percentage of deformities. Some eggs containing fully developed embryos fail to hatch at all, apparently unable to properly assimilate the yolk. We've had no cases of "twinning" in our *Cyclura* eggs, but are aware of two other documented incidences of twins at other institutions.

I normally wait until all the hatchlings in each plastic box have been out of their eggs for three days before removing them from the incubator. Four or five are placed into 30 gallon terrariums with a bottom substrate of butcher paper. An under-tank heating pad is placed under one end of the terrarium. Two light hoods are used; one holds 25 watt red bulbs for nighttime use; the other is a round, reflective hood with a 75 watt Phillips Grow Lite® plant bulb. Both lights are controlled by timers and each is turned on and off automatically for alternating 12 hour periods. By this method, nighttime temperatures are maintained at 21-27°C degrees (75-80°F), rising to 32-35°C (90-95°F) during the day. A hide box is placed in the end of the terrarium over the heat pad and a shallow container of purified water is added for drinking.

Baby iguanas shed almost immediately in tiny flakes as their skin dries out. Their bellies are distended with yolk and they often do not begin to feed for 7-10 days, when they also completely shed again, this time in large patches. We have seen no differences in growth or survivorship between hatchlings fed feces from adult iguanas and those which are not. Sufficient microflora for proper digestion is probably obtained from the crickets and pink mice fed at an early age. Interestingly, *C. nubila* seem to really relish crickets, while *C. cornuta* youngsters consumed them rather sparingly. Hatchlings are kept indoors through the first winter and are moved into indoor/outdoor cages when the weather begins to warm in spring. Growth is rapid and we have seen iguanas attain over 20 inches in overall length within the first year.

The most reliable method for sexing *Cyclura* is by probing. We do not recommend probing iguanas under two months of age, for the membranes within the cloaca are very delicate. Probes may puncture through the membranes and slip under the skin of the tail even with very light pressure. At three months of age, females probe about 5 mm in depth, while males are close to 12 mm deep. The smallest probe (1 mm) is generally used on hatchlings. By two to three years of age, the primary sexual characteristics are becoming evident and the presence of the male hemipenes can usually be detected visually. Adult *C. cornuta* females will probe under 15 mm in depth while adult males probe almost 50 mm. Males (of most species) will also have enlarged and exuding femoral pores on the underside of the thighs.

Growth rates are often accelerated in captivity as compared to that of wild iguanas. This is certainly due to the richer diet they receive on a continuous basis. Consequently, sexual maturity is reached at a younger age than their wild counterparts. Female *C. nubila* have laid fertile eggs in their third year and *C. cornuta* have reproduced successfully in their fourth or fifth year.

We have seen very few medical problems with our animals. Newly acquired *Cyclura* sometimes are infested with mites and these are treated by spraying with 0.03% pyrethrin spray (as used for birds). The spray is allowed to sit on the iguana for one or two minutes and is then thoroughly rinsed off. Care should be taken to avoid spraying into the eyes. A second treatment is applied two weeks following the first. Containers housing iguanas may be treated with the same spray allowing it to remain on surfaces for ten minutes before rinsing.

Wild *Cyclura* harbor huge swarms of nematodes in the caecum. It is thought that these may present an almost symbiotic relationship, helping to break down the roughage consumed in their natural habitat. We have never treated for these worms in healthy captive animals, even though they are occasionally seen in fresh stools. Weak or injured animals may need to be treated, however, as heavy loads of nematodes can further debilitate an animal already under stress. *Salmonella* and *Arizona* are sometimes seen in fecal analysis of *Cyclura*. These are considered to be part of the normal intestinal microflora but may become pathogenic when animals are under great stress. As these organisms can be transferred to humans and other animals, sound hygiene practices should always be observed.

Only one injury has ever required stitches in our iguanas. This was inflicted on an adult male's leg by an unreceptive female as he attempted to mate with her. Minor cuts and scrapes usually heal on their own, although we often apply an antibiotic spray to the afflicted area to help reduce the chance of infection. We have only suffered the loss of one adult *Cyclura* at C.R.C. in the last ten years. This animal appeared perfectly healthy up until the day that it died. A necropsy revealed that this gravid female had become egg-bound from several ruptured eggs. Yolk material coated all internal organs.

CONCLUSION

We are very pleased with the advances made in captive propagation at Cyclura Research Center and other facilities around the world. Captive-produced animals provide a source for zoos, research, and the private sector without exerting any pressures on wild populations. Although still controversial, more and more of these animals may be recycled back into the wild to restock depleted areas. This has been done for years in the Dominican Republic with *C. cornuta* and a program begun in the Cayman Islands will attempt to do the same with *C. n. lewisi* in the near future. Ultimately, the continued survival of *Cyclura* iguanas will depend on how aggressive we are in preserving their natural habitat and attempting to reverse much of the damage that has already been done to it. Until, and unless, these goals are accomplished, captive breeding programs will remain their most important hedge against extinction.

ADDENDA

I have recently received word from Dr. Peter Vogel, head of the *Cyclura collei* Research and Conservation Group, that they now hold thirty one Jamaican iguana hatchlings in captivity in Kingston. These animals will be reared for captive breeding stock and eventual re-release into the wild. Unfortunately, the Jamaican Ministry of Mining has requested to have half of the Hellshire Hills declared as a limestone quarry zone. This would almost certainly spell disaster for the remaining population of iguanas. A major campaign is now underway to have the entire forty four square miles declared a protected area under a recent Jamaican law. We wish Dr. Vogel great success in this endeavor. Perhaps Goat and Little Goat Islands should also be considered as possible sanctuaries.

Edward Lewis of Texas A&M University has just released results of his blood haplotype study of *Cyclura n. lewisi* in captivity. Unfortunately, most of the specimens tested are apparently intergrades with the closely related *C. n. caymanensis*, including numerous animals held in the captive breeding program on Grand Cayman Island. This is cause for concern as many of these animals had been scheduled for eventual release into the wild. It may be appropriate to sterilize all hybrid animals, at least on Grand Cayman Island, to reduce the chance of contaminating the few remaining wild iguanas.

Finally, a visit in June, 1992 to Isla Magueyes, Puerto Rico has revealed that the previously reported "thriving" population of introduced Cuban rock iguanas, *Cyclura n. nubila*, apparently now consists mostly of older adult animals. Although my visit was very brief and only a portion of the island was explored, not a single hatchling or juvenile was observed. In fact, the youngest animal observed was at least three years of age. Long term employees on the island confirmed that the number of iguanas has declined in recent years. Several cats seen on the island may be responsible since their prowess as efficient lizard predators has been well documented.

LITERATURE CITED

Auffenberg, W. 1976. Rock Iguanas, part II. Bahamas Naturalist 2:9-16

Blair, D.W. 1983. Dragons of Cayman. Rock iguanas cling to their islands. Oceans 16(2):31-33

Carey, W.M. 1975. The rock iguana, *Cyclura pinguis*, on Anegada, British Virgin Islands with notes on *Cyclura cornuta* on Hispniola. Bull. Florida State Mus. Sci. 19(4):189-234

Duval, J.J. 1982. Recommendations for the captive management of West Indian rock iguanas, (*Cyclura*). IN: Proceedings of 6th International Herpetological Symposium. pp 181-196

Iverson, J.B. 1979. Behavior and ecology of the rock iguana *Cyclura carinata*. Bull. Florida State Mus., Bio. Sci. 24(3):175-358

Noble, G.K. 1923. Trailing the Rhinocerous Iguana. J. Amer. Mus. Nat. Hist. New York 23:541-558

Perera, A. 1985. Datos sobre la dieta de *Cyclura nubila* (Sauria:Iguanidea) en los alrededores de cayo Larga del sur, Cuba. Poeyana 291:1-12

Schwartz, A. and W.M. Carey. 1977. Systematics and evolution in the West Indian iguana genus *Cyclura*. Stud. fauna Curacao and Carib. Isl. 53(173):15-97

-------------- and R. Thomas. 197?. A checklist of West Indian amphibians and reptiles. Special publs. Carnegie Mus. Nat. Hist. No. 1:1-216

Wiewandt, T.A. 1977. Ecology, behavior, and management of the Mona Island ground iguana *Cyclura stejnergeri*. Ph.D. dissertation, Cornell University, Ithaca, N.Y.

---------------- 1982. Known and extinct forms of *Cyclura* iguanas. IN Burghardt and Rand (eds), Iguanas of the World, pp392-394, Noyes Publication, Park Ridge, N.J.

Cyclura c. cornuta in captivity. Photo by David W. Blair.

MANAGEMENT OF CHAMELEONIDAE AT THE OKLAHOMA CITY ZOOLOGICAL PARK

David Grow

Curator of Herpetology
Oklahoma City Zoological Park
2101 N E 50th
Oklahoma City, OK 73111

INTRODUCTION

Chameleons are wonderful examples of nature's capacity to produce animals that are finely adapted to their environment. The chameleons' adaptations to its arboreal lifestyle are very distinctive and, more importantly, are readily observable by the zoo visitor. These seemingly exaggerated adaptations facilitate the interpretation of various aspects of biology for the public. These features combine to make chameleons desirable exhibit subjects.

The very adaptions that make the chameleons good exhibit animals may be contributing to the demise of some species. While some species like the Oustalet's chameleon are known to thrive in areas of disturbance, many other species may be very sensitive to disturbance in their habitat. The current rate of deforestation is an obvious threat, but it has been shown, however, that moderate mosaic deforestation in the rain forest, such as with slash and burn agriculture, can alter the internal environment of the remaining forest. Significant changes in the humidity, temperature, and composition of invertebrate species have been documented in forest gaps and in slash and burn study areas (Brokaw 1985; Ewel *et, al.*1981; Kircher 1989). Most species of chameleons have demonstrated very specific temperature and humidity requirements in captivity. It is not unreasonable to believe that these changes negatively affect these highly specialized animals.

The IUCN (1987) reports that slash and burn agriculture is the dominant threat to the remaining forests of Madagascar. It further states that in recent years as much as 35,000 hectares of previously undisturbed closed canopy forests were destroyed annually. This process continues with terrible impetus.

Over half of all chameleons are endemic to Madagascar (Welch, 1982.), many requiring rain forest habitat. The IUCN has provisionally identified 18 species that are under some degree of threat. Some of these species are know only from their type locality. With further study in the wild and given the rate of deforestation, a number of these species will possibly be recognized as endangered in the near future. As the ruination of Madagascar continues, the development of self-sustaining captive populations of these species will become increasingly important.

Unfortunately, at this point, establishing such populations in a variety of institutions seems unrealistic. Chameleons have proven notoriously delicate in captivity (Wagner, 1958; Bustard, 1959; Bustard 1963; Mattison, 1982). While many zoos have worked with chameleons, only a few have reported any success with them and were limited to very few species (Castle, 1990; ISIS, 1990; Slavens, 1990).

Chameleons have developed a reputation that has caused zoos to avoid them. They are, however, rapidly approaching a time when they will need the help zoos can offer. Before captive propagation can become a realistic option in conserving this group, however, we must develop better husbandry techniques.

In 1985, chameleons were again being considered for exhibition at the Oklahoma City Zoo because of their educational value. Their reputation for ephemeral health reinforced by previous failures caused us to carefully evaluate the feasibility of exhibiting chameleons. The available literature lacked specific husbandry information, and communication with other workers was not encouraging. It was apparent that there was a need to improve zoos overall performance with chameleons. The staff was challenged to teach themselves.

The formal goal of the program that evolved out of these deliberations was to solve problems with husbandry and reproduction so that captive management could become a realistic option in the conservation of chameleons. Knowing that some losses were inevitable, an objective oriented program was implemented in late 1985, addressing increasingly difficult or rare species, only after specific measures of success were achieved with each. The goal has not been to breed as many species of chameleon as possible, but rather to learn from each species selected so that we would be more successful with the next. The Oklahoma City Zoological Park is endeavoring to position itself so that it is able to help these species and other zoological institutions that will participate in captive conservation programs.

During this period the zoo has worked with nine species of chameleon: *Bradypodion damaranum; B. gutturale; Chamaeleo dilepis; C. jacksoni; C. melleri; C. montium; C. oustaleti; C. parsoni;* and *C. willsi*. Three of these species have reproduced and five species have been raised from hatchling or birth. This paper presents current management techniques that have facilitated this success.

Two species were initially employed to establish basic husbandry parameters. The flap-necked chameleon, *Chamaeleo dilepis*, was the initial teaching species because they were very common, inexpensive, and appeared to inhabit a broad range of environments. Much was learned from this species which reproduced in 1987. Progeny were raised which subsequently reproduced. Flap-necked chameleons were replaced by Jackson's chameleons, *Chamaeleo jacksoni*, in early 1987. Over 150 offspring were produced over the next two years, including F_3 young.

These successes encouraged us to address the next objectives by adding to the collection over the next three years, beginning in 1987 with three species of Malagasy chameleons: Oustalet's, Parson's, and most recently Wills' chameleons. Oustalet's chameleon reproduced in 1988 with only four hatches occurring in August in spite of several apparently fertile clutches being laid. Four more eggs were manually slit and hatched in 1990 but did not survive. A clutch of 54 eggs sired by one of the 1988 offspring is currently incubating under three separate thermal regimes. Castle (1990) presents a more detail account of reproduction in this species. Wills' chameleon copulated shortly after arrival and laid 9 eggs on December 12, 1990, which appear fertile.

The initial environmental parameters provided chameleons at the zoo are greatly influenced by what was learned from the literature. Busack (1969) provides a bibliography of the Chameleonidae of over 500 citations between 1864 and 1964. Brygoo (1971), while in French, has been an important source of information on Malagasy chameleons. Parcher (1974) provides some behavioral data on Parson's and Wills' chameleons in the wild. DeWitt (1988) describes husbandry requirements of the Jackson's chameleon. An excellent review of the needs of oviparous chameleons in captivity is presented by Bustard (1989). de Vosjoli (1990) provides a very helpful account of general husbandry for chameleons. Fitzsimons (1943) and Branch (1988) provide helpful accounts of the dwarf chameleons (*Bradypodion*). Authors that have published general works in reptile husbandry which include comments on the care of chameleons in captivity are Schifter (1984), Zimmerman (1986), Mattinson (1987), and Obst (1988). Recent personal communication with other workers has been extremely helpful.

Almost as important as species accounts has been data on the local climate and environmental cycles. General information for broad areas is available but specific data for local climates is less common. The Department of Commerce's publication Climates of the World has provided some information, but Annuare Statistique de Madagascar published in 1953 has been much more helpful with data on specific localities.

Other significant components in the development of management programs or husbandry decisions are our own experiences, and the physical environment of the herpetarium itself. The building was built in 1925 and is the oldest active animal exhibit in the zoo. We are allowed much latitude in adapting particular areas of the building to specific needs of husbandry programs. An important component is a greenhouse attached to the rear of the building where most of the chameleons, not on exhibit, are housed. In this we are able to produce a wide variety of environments.

ENCLOSURES

Three basic types of enclosures are employed: exhibits within the building varying widely in size and environment, aluminum framed screen walled enclosures which because of their size are very mobile and wood or steel framed wire walled enclosures that are not very mobile. Individual enclosures are made as large as possible with due regard to space constraints, adequate prey density and providing the zoo visitor with some hope of seeing what we are trying to exhibit.

Four species of chameleons are on exhibit: Knysna dwarf chameleon, sail-fin, Oustalet's and Parson's chameleons. The Parson's chameleon exhibit is the largest exhibit in the building measuring 5 x 1.8 x 3 m tall. This exhibit contains a tempered water stream and rain sprinkler system. It is the only sky lit exhibit and is thickly planted. The sail-fin chameleon exhibit is about 1.2 m in each dimension and is likewise thickly planted. The Oustalet's chameleon exhibit is also large, 2.5 x 2 x 2.5 m tall, but the propping is more open with fewer plants. The dwarf chameleon exhibit is being expanded to 2 m long x 1 m x 1.5 m tall, and will also contain the South African areolated tortoise, *Homopus areolatus,* and tent tortoise, *Psamobates tentorius.*

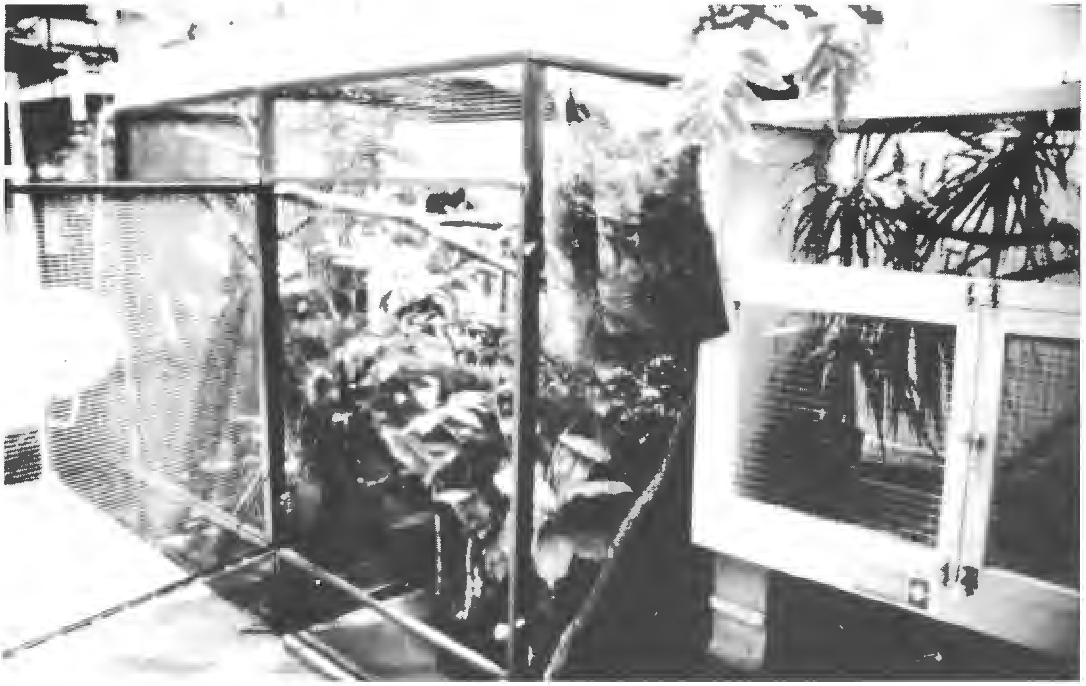
There are three basic types of reserve enclosures. The aluminum framed screen walled enclosures are of three sizes: 60 cm in each dimension; 45 cm in each dimension; and 30 x 30 x 60 cm long. They are inexpensive and easy to construct. In Oklahoma these very light enclosures are advantageous as they are easily moved outdoors as weather permits. The wood framed wire enclosures are of two sizes: 122 x 60 x 100 cm tall and 122 x 122 x 213 cm tall. They are not very mobile at all, nor are they very weather proof. The most recent design involves angle steel framed panels of vinyl coated wire which can be bolted together to form a single independent enclosure or configured to form a bank of enclosures. These enclosures are 122 x 122 x 182 cm tall. This most recent design, while more expensive, may prove most versatile because it is mobile, interchangeable, and should be the most durable of the enclosures.

LIGHTING AND TEMPERATURE

Lighting is provided in a variety of ways, however, we provide natural sunlight whenever possible. Sky lights are the next best thing, however, we do supplement with fluorescent and incandescent lighting even in the greenhouse. The artificial lighting includes 40 and 80 watt fluorescent Durotest Powertwist Vitalite® lamps often augmented by Sylvania BL40® black lights. The incandescent bulbs used include 75 and 150 watt reflector growlites with 275 watt infrared heat lamps when necessary during the cold weather of winter. While there may be some differences in the illumination preferences between species that inhabit closed canopy rain forest and open brush savanna, an attempt was made to provide illumination as bright and as of broad spectral composition as possible.

Incandescent lighting is provided primarily for thermoregulatory opportunities. The thermal opportunities provided chameleons are based on information from previously mentioned literature, but are often significantly compromised by the environment of the physical setting.

The wide ranging climate of Oklahoma and our limited ability to control its effects have shown the chameleon species we have worked with to be somewhat more thermally tolerant than we expected. Cooling has proved more



Steel and wood framed wire chameleon reserve enclosures in the herpetarium greenhouse.
Photo by David Grow.

Outdoor reserve enclosures, collapsible aluminum framed screen, 7 m one side.
Photo by David Grow.

difficult than heating, but one technique has allowed even Jackson's chameleons to be kept outside during the hot Oklahoma summer. A garden soaker hose is draped or coiled over one half of the outdoor enclosure providing a localized spray or drip of water significantly cooler than the air temperature. The water is turned on when the temperature rises above 28°C. The chameleons typically use this opportunity to moderate their body temperature. During an extended period of 38°C+ temperatures during the summer of 1990, it became necessary to install a programmable garden water timing system as the chameleons reacted poorly to constant rain during the extended periods of heat. The system provided several periods of rainless reprieve during the day and they seemed to respond well to the change. Black plastic 40% shade cloth stretched over the outdoor reserve area aided in reducing the rate of heating during the rainless periods.

The design of our enclosures and the special efforts made to keep the chameleons outside are results of our desire to provide as fresh and buoyant an atmosphere around the chameleon as possible. It is widely agreed that chameleons do poorly in stagnant damp atmospheres. Electric fans in strategic locations help maintain air circulation. The two largest chameleon exhibits are capable of ducting outside air directly into the exhibit using thermostaticly controlled fans.

WATERING AND FEEDING

A special concern is to provide adequate drinking water while allowing the enclosure to dry between waterings. The screen or wire enclosure, whether in or outside, facilitates adequate drying even after being drenched. Somewhat greater care must be exercised when watering exhibits. An intravenous drip system placed so that the dripping water is caught in a water proof container in the substrate is helpful in reducing extraneous wetting of the exhibit.

Watering is gauged to the needs of the species or individuals. A pneumatic pressure sprayer has proved a very handy tool, allowing a spectrum of both spray intensity as well as configuration ranging from a fine mist to a steady stream. Our experience shows that small chameleons like *Bradypodion* prefer a mist of water accumulating on surfaces in the exhibit so drinking is facilitated over a period of time, while larger chameleons will readily drink from a gentle stream of water. Water temperature also influences a chameleon's inclination to drink. Water near 28-30°C has proven most commonly acceptable, but may vary with species or individuals. Techniques of this nature require individual attention.

The composition of the diet for chameleons is comparatively simple. Gray crickets of various sizes make up the basic diet, with giant mealworms and waxmoth larvae. The chameleons are fed daily using four techniques: Hand casting prey throughout the enclosure; placing prey in smooth round bottom bowls hung at appropriate heights in the enclosures, hanging a container in the enclosure that allows only a gradual release of prey, and individual hand feeding. Again, the technique is gauged to the species, characteristics of the enclosure, or individual animal.

Chameleons at the Oklahoma City Zoo are supplemented with a wide spectrum vitamin powder, a calcium/phosphorus and Vitamin D_3 supplement. We recognize the narrow nutritional content of the diet and the likely possibility that artificial lights, regardless of their reputation or reputed spectral content lack adequate ultra violet radiation in the 280-320 nm range. Neckton-Rep® and Osteform® powder are sprinkled on prey items 3-5 times per week with Neckton-Rep occasionally being provided in drinking water.

DISEASES

Chameleons coming into the collection are quarantined from exhibit animals. The limitations of our facility sometimes requires that new specimens are often times housed in the same rooms as resident animals. It is very difficult to say, based on supporting evidence, whether this has caused problems.

New arrivals, however, are promptly evaluated for parasitism as the larger enclosures share a common floor. Parasitism is addressed only after the potential impact of additional stresses in handling any particular individual for treatment is carefully evaluated. Parasites that have been observed include strongles, ascarids, coccidia, flagellates, flukes, and one tapeworm. Oral treatments for these parasites have included ivermectin, mebendazole, fenbendazole, sulfathamethazine and sulfanilamide. A review of health records shows that these treatments have not been entirely successful and a new treatment protocol for parasitism is being developed.

Other health problems have been myriad and can only be briefly reviewed within the scope of this paper. Various gut problems generally associated with discolored poorly formed stools surrounded by mucoid fluid have been treated with a combination of amikacin and caribinicillin over 14 days. Prior to the decision to administer antibiotics, microscopic examination of the suspect stool is made to determine to what extent parasites are contributing to the problem. Stomatitis has been successfully treated with this antibiotic regime with the addition of regular debridement and daily topical application of betadine solution. Cultures have most commonly revealed *pseudomonas* and *salmonella* as the pathogens.

Post mortem and histopathological examinations have not revealed a dominant cause of death. Various types of hepatitis have been suggested in several deaths. Parasitism has been implicated in at least two deaths while gastritis has been involved in a single death. A number of deaths occurred with no identifiable cause.

Typically, one male and one to three females are maintained in an exhibit. The enclosure must provide all of the inhabitants with proper cover and adequate thermoregularity opportunities. We have yet to work with a species of chameleon where the males will tolerate the other's presence. Similarly, females sharing a single enclosure must be monitored as a dominant female can force the other females to the lower reaches of the enclosure. This will often depress the displaced females' appetites. An important part of our management plan is to identify subtle social stress and remove affected individuals to less stressful environments.

REPRODUCTION

With the possible exception of Parson's chameleons, chameleons will readily reproduce if they are maintained in a supportive captive environment. It is not always necessary to observe copulation to determine if a female has been bred. Females of the four species which have reproduced at the Oklahoma City Zoo have all displayed a change in coloration once they became gravid. Other indications include an increasing girth of the abdomen as gestation progresses, and noticeable lumps on the flanks produced by the eggs developing in the body.

As parturition or oviposition approaches, changes in basking behavior, feeding and drinking habits, and intolerance of enclosure mates will become more readily observable. Adequate thermoregulatory opportunities and proper hydration are extremely important during this time.

Birth in Jackson's chameleons and Kenya dwarf chameleons have been similar to that described for Jackson's by DeWitt (1988) and Zimmerman (1986). No special modification of propping have been necessary to support parturition.

Accommodating oviposition for egg laying species has been a much more problematic effort. Six females, representing three species, have died containing near or full term eggs or died shortly after oviposition. While the specific etiology of this syndrome maybe diverse and complex, our experience suggest two general causes. The production of viable eggs requires a tremendous investment of energy on the part of the female. The nutrition that is applied to the developing eggs is taken from, both the female's diet and her own body. If she becomes gravid while she is even slightly under weight, disinclined to eat, parasitized, or unhealthy in any way, her chances of survival are severely compromised.

We have often observed females moving about the floor of an enclosure prior to oviposition in what appears to be a search for just the right spot to dig her nest. Lack of adequate nesting sites may cause a female to postpone oviposition because she continues searching for an acceptable site. Females that do not find an acceptable site die. There are several considerations that should be addressed when providing a site for oviposition:

1. The substrate should be deep enough and wide enough to allow the female to dig a completely subterranean burrow at least as long as she is.

2. The substrate should be of a consistency that does not collapse during the nesting process. It should be only moist enough to facilitate burrowing.

3. The substrate should not contain any hard obstructions like pots, rocks or sticks. We have found that some females often dig a burrow up to the root ball of a plant and oviposit next to it.

4. The female should have time to become familiar with the substrate and enclosure where oviposition is to occur. We try to provide adequate nesting opportunity as early as possible.

Incubation of the eggs has also proven problematic. The hatch rate of the species that have reproduced at this zoo has been very low. Flap-necked chameleon eggs were incubated in plastic wrap covered gallon jars set in a water bath to maintain the temperature at 22-23°C. The incubation substrate was vermiculite, 5 cm deep moistened with water at a ration of 1:1 by weight. Hatching occurred in seven clutches of eggs with an incubation period of 220-230 days.

The initial incubation parameters for Oustalet's chameleon eggs were the same as for flap-neck chameleons. However, after 268 days of incubation in this regime, no development could be observed in the first clutch of eggs upon candling. It was felt that the incubation temperature was to cool. Only four young hatched from one of four clutches after the incubation temperature was warmed to room temperature, 26-28°C. Incubation was 245-248 days. Only after an accidental four-day cooling to 22°C was any development observed in the remaining clutches of eggs. None of these eggs hatched. Several eggs of one clutch were observed to be split at approximately 483 days of incubation. After a period they were opened and were found to contain full term dead embryos. The remainder of three clutches of eggs were opened with similar results, although, some eggs contained living embryos. Four of these survived for several days. Currently, the most recent clutch is being incubated in three separate thermal regimes to hopefully further identify incubation parameters in this species.

The husbandry of neonates is similar to that of the adults but there is far less room for error. They are housed in a variety of containers, the two smaller sizes of aluminum framed screen enclosures being most satisfactory. They are housed individually or in groups of six to ten. Our experience indicates that it is more important for neonates to have adequate opportunity to space themselves on a horizontal plane rather than a vertical plane. When horizontal space is restricted individuals space themselves in a vertical orientation, and the bottom individuals loose their health rapidly. Vestigial winged *Drosophila* and newly hatched gray crickets are provided at least once a day in the morning. A second feeding is often provided in the afternoon. Enclosures are sprayed with water two to three times a day. During weather above 32°C water is constantly provided. Neonates are much less thermally tolerant than the adults. Care is taken to observe feeding habits and interactions between individuals on a daily basis. Young are separated into additional enclosures when aggression and depressed appetites are observed.

CONCLUSION

Chameleons are wonderful animals providing an exciting challenge in captivity. There are several general factors that must be addressed if any chameleon management program is to be successful.

1. A buoyant and fresh atmosphere surrounding the chameleons is critical.

2. A wide variety of species specific thermal requirements must be accommodated.

3. Stress arising from a wide variety of sources must be promptly recognized in individuals and promptly reduced or eliminated.

4. Adequate space and enclosures must be available for the management of social behavior.

5. Chameleons need individual attention on a daily basis requiring serious dedication on the part of the keeper.

We believe captive management can make a contribution to the conservation of chameleons. As more information on the needs of captive chameleons is revealed and shared, broader success at a greater variety of institutions should occur.

LITERATURE CITED

Anonymous. 1977. Climates of the World. U.S. Government Printing Office. Number C52.2:C61/3

Brain, C.K. 1961. *Chamaeleo dilepis* a study on its biology and behavior. J. Herpet. Assn Rhod. No. 15: 15-20.

Branch, B. 1988. Bill Branch's Field Guide to the Snakes and Other Reptiles of Southern Africa. Ralph Curtis Books, Sanibel Island, FL

Brokaw, N.V.L. 1985. Gap-phase regeneration in tropical rainforest. Ecology 66: 682-87.

Busak, S.D. 1969. Bibliography of the Chameleontidae 1864-1964. Smithsonian Herpetological Information Services, U.S. National Museum, Washington DC.

Bustard, R. 1959. Chameleons in captivity. Br. J. Herpet. 2: 163-165.

------------- 1963. Growth, sloughing, feeding, mating, gestation, life-span, and poor health of chameleons in captivity. Copeia 1963: 704-705.

------------- 1989. Keeping and breeding oviparous chameleons. Brit. Herp. Soc. Bull. 27:18-33

Brygoo, E.R. 1971. Reptiles. Saurien Chameleonidae. Genre *Chameleon. Faune de Madagascar.* 33, 318 pp. Paris, Orstom/CNRS.

Castle, E. 1990. Husbandry and breeding of chameleons, *Chamaeleo* spp., at the Oklahoma City Zoo. International Zoo Yearbook. 29: 74-84.

de Vosjoli, P. 1990. The General Care and Maintenance of True Chameleons, Part I Husbandry. Advanced Vivarium Systems, Lakeside, CA., 36 pp.

--------------- 1990. True Chameleons, Part II, notes on popular species, diseases and disorders. Advanced Vivarium Systems, Lakeside, CA., 28 pp.

DeWitt, C. 1988. Jackson's chameleon, *Chamaeleo jacksoni*: captive behavior, care and breeding. Vivarium 1(2):17-20.

Ewel, J.,C. Berish, B. Brown, N. Price, and J. Raich. 1981. Slash and burn impacts on a Costa Rican wet forest site. Ecology 62: 816-29.

Fitzsimons, V.F. 1943. The Lizards of South Africa. Transvaal Museum. Petoria, South Africa.

ISIS Species Distribution Report Abstract: Reptiles as of June 30, 1990. ISIS, Apple Valley, MN.

IUCN/UNEP/WWF. 1987. Madagascar, an environmental profile. M.D. Jenkins (ed.) IUCN, Gland, Switzerland, Cambridge, U.K., 374 pp.

Kircher, J. 1987. A Neotropical Companion. Princeton, JF: Princeton Univ. Press.

Mattison, C. 1982. The Care of Reptiles and Amphibians in Captivity. Sterling Publishing Co., New York, NY

Obst. F.J., K. Richter, U. Jacob, 1988. The Completely Illustrated Atlas of Reptiles and Amphibians for the Terrarium. J. G. Walls (ed.) TFH Publications, Neptune City, NJ., 830 pp.

Parcher, S.R. 1974. Observations on the natural histories of six Malagasy Chameleontidae. Z. Tierpshychol. 34. pp 500-524. Verlag Paul Parey, Berlin und Hamberg.

Schifter, H. 1984. Chameleontidae in Encyclopedia of Animal Life. Vol. 6. Reptiles. B. Grzimek (ed.) Van Norstrand Rheinhold, NY., 589 pp.

Slavens, F.L. and K. Slavens. 1990. Reptiles and Amphibians in Captivity, Breeding Longevity and Inventory, January 1, 1990. Slaveware Seattle, WA.

Struyf, H. 1979. *Chamaeleo jacksoni, Chamaeleo hoehnelli*, en hun jongdieren. Lacerta 39: 202-207.

Wagner, V.A. 1958. The chameleon's breeding habits. African Wild Life 12: 285-293.

Welch, K.R. G. 1982. Herpetology of Africa: A Checklist and Bibliography of the Orders Amphisbaenia, Sauria and Serpentes. Robert E. Krieger Publishing Co., Malabar, Florida.

Zimmerman, E. 1986. Breeding Terrarium Animals. TFH Publications, Neptune City, NJ.

ALTERING THE CLUTCH SIZE AND OFFSPRING SIZE OF REPTILES

Barry Sinervo

Department of Integrative Biology
University of California
Berkeley, CA 94720

INTRODUCTION

Clutch size and offspring size are two aspects that are of central importance in any captive breeding program. The trade-off between the number and the size of individual offspring a female can produce is a general feature of reptilian reproduction (Ballinger, 1978). Because of functional limitations on the total mass of the clutch (e.g., Sinervo and Licht, 1991a), a female cannot increase offspring number without decreasing the size of individual offspring. Thus, these two reproductive attributes are intimately related. Here, I describe a few techniques (Sinervo and Licht, 1991a,b) for altering both clutch size and offspring size in lizards -- presumably, these techniques have more general applicability to other reptiles.

Such issues are not strictly in the esoteric realm of research scientists. The benefits accrued to a captive rearing program from increasing the number of eggs in a clutch are straightforward to enumerate -- count them off one by one. Although the general importance of clutch size to amateurs and professionals is fairly obvious, the significance of offspring size is not as well characterized. Nevertheless, the effects of offspring size may be critical in the rearing of offspring that are very small at hatching or birth. Owing to the problems of hatchling size, finding food items of the right size could be a grave problem.

In addition to these issues, much of my research involves the large-scale maintenance (300+ females) of a captive breeding program that produces up to 2000 hatchlings per year. The methods that we have developed to streamline housing requirements and care may be useful to other captive breeders of small (or large) lizards.

LARGE SCALE HUSBANDRY

We rear side-blotched lizards (*Uta stansburiana*) and fence lizards (*Sceloporus occidentalis*) in 14 l terraria with wire screen tops. A thermal gradient (25-45°C) is maintained during the light cycle using an incandescent bulb (60 watts) placed over one side of the container. Three females and one male can be housed in each terrarium (copulatoria). Background illumination using fluorescent ultraviolet lighting (FBL 40, Philips®) is a must for any lizards that spend an inordinate amount of time under the UV-intense desert sun. Under these conditions it is imperative that the lizards are provided water and ad libitum food daily (crickets (*Achaeta sp.*), dusted periodically with Vionate® or Nekton-S® commercial vitamin supplement and calcium) and mealworms (*Tenebrio sp.*) larvae or waxworms (*Galeria sp.*) larvae on a weekly basis. Adequate feeding is a must because the females will develop dominance hierarchies in the restricted space of the copulatoria (30 x 45 cm) if food is limiting. We have found that typical 5 g side-blotched lizard (~60 mm snout vent length) requires about 3+ crickets per day. These crickets are close to the maximum size that they can choke down. Keeping lizards as breeders requires much more food than the typical "pet" which is usually maintained at slightly above maintenance requirements. Remember, female lizards can produce half their body weight in eggs every 3 weeks and this requires a tremendous quantity of crickets, mealworms and calcium.

Females with oviducal eggs (the feel of round hard follicles changes to a soft mushy oblong shape) are transferred to ovipositoria consisting of 14 l terraria partitioned into four compartments with aluminum sheeting (Figure 1). An incandescent bulb (40 watts) over the center of the partitions provides an adequate thermal gradient (25-45°C) for behavioral thermoregulation. A square pan filled with sand that fits snugly into the partitioned areas is placed over a 5 cm thick layer of moistened sand/peat moss. Access to the sand/peat layer is provided via a 8 cm slot located away from the heat source. Females readily construct burrows under the pan and into the sand/peat layer and in most cases they lay their clutch within the sand/peat layer. The sand/peat layer needs to be moistened 2-3 times per week.

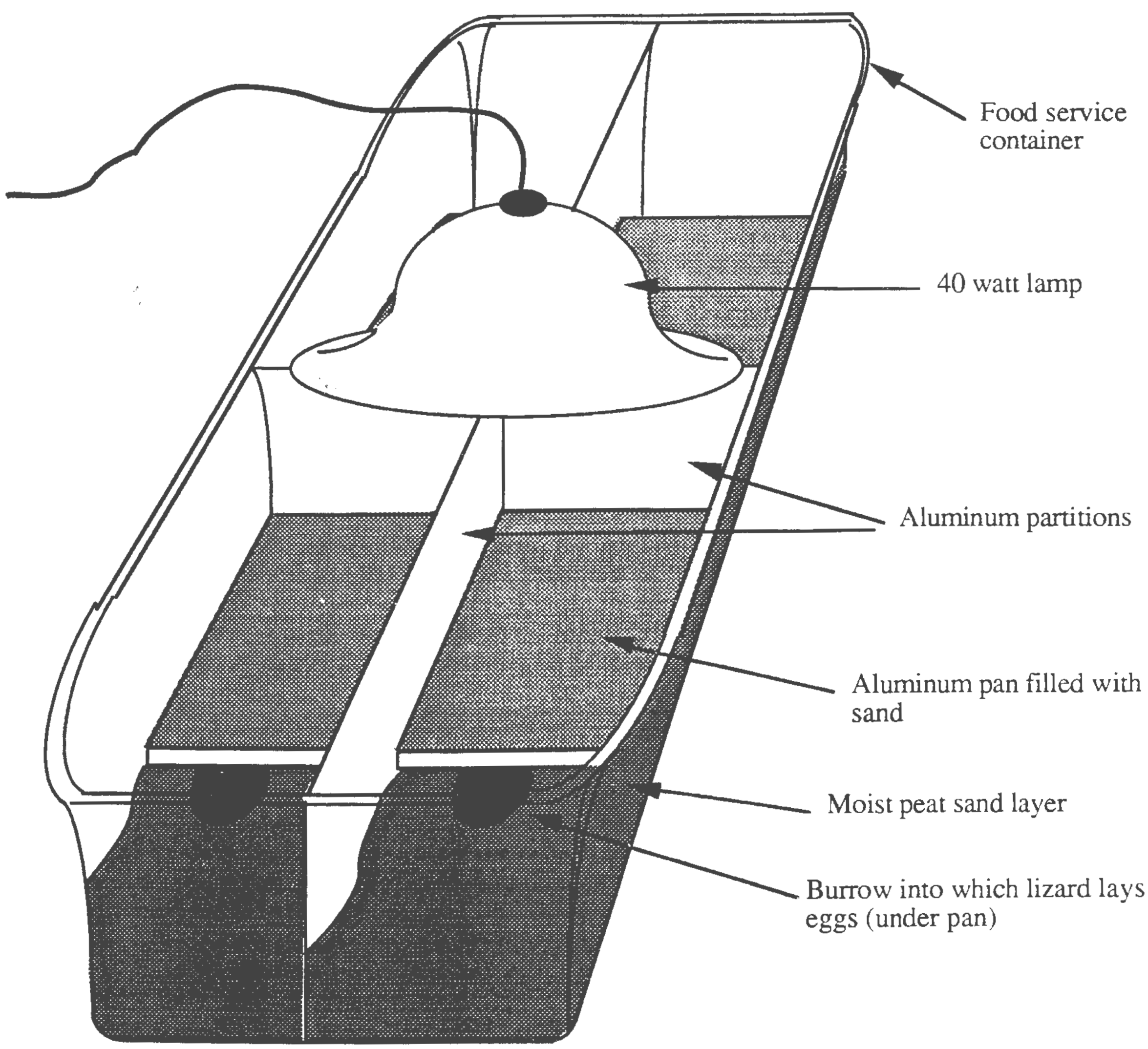


Figure 1. Set up for ovipositoria that houses four small, 5-20 g lizards. The 14 liter terraria are food service containers measuring 15 cm X 30 cm X 45 cm.

The conditions I have described are optimal for lizards in the 5-20 g range (*Uta*-sixed up to *Sceloporus*-sized lizards). These housing requirements balance the needs of the lizards against the limited space in any rearing room. Using this set up, it is possible for us to produce > 2000 hatchlings per season from a lab colony of 200-300 female side-blotched lizards. These females are maintained in a room measuring 8 x 14 ft (cages are on 4-shelf racks -- a veritable Herp Hilton). If larger lizards need to be maintained, just scale all containers up by the appropriate snout vent length.

INCREASING AND DECREASING THE NUMBER OF EGGS IN A CLUTCH

To increase the clutch size of female *Uta* (Sinervo and Licht, 1991a), they are first palped and categorized as either early (follicles < 3 mm) or late (> 3 mm). Over the first six days, females receive 10 μg of ovine follicle stimulating hormone (FSH) in 100 μl saline by intra-abdominal injection every other day. Thereafter and until ovulating, females received 2 μg in 100 μl saline every other day. The lizards receiving FSH typically produce larger clutches of viable eggs (Figure 2A). Other studies on the green anole (*Anolis carolinensis*) have demonstrated that administration of a daily 10 μg dose increases the number of yolking follicles, but that daily doses of 1 μg are not sufficient to illicit a response (Jones et al., 1976). Larger doses of FSH are apparently pharmacological and can result in a dramatic proliferation of vitellogenic and previtellogenic follicles (Licht, 1970; Jones, 1976) and block ovulation (Licht, 1970).

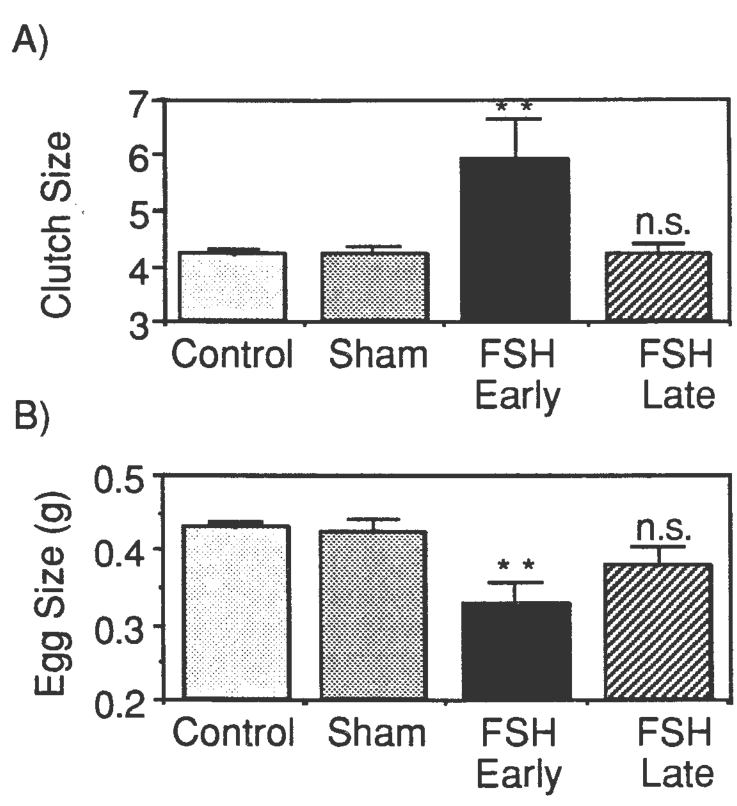


Figure 2. The effect of FSH injections on clutch size (A) and egg size (B). The effect of FSH injections for females early in the yolking cycle (follicles < 3-4 mm) or late (follicles > 3-4 mm) are considered separately. Sham-injected females received injections of the saline and unmanipulated females received no injections. The ** indicates results that were statistically significant from controls, n.s. indicates results that were not statistically significant from controls.

It is important to note that the FSH treatment is only effective if it is initiated very early during the reproductive cycle. A typical cycle for uta is 24 days and the 3 mm follicle size is reached within the first half a week. If treatment is initiated after this point there is no effect on clutch size. Also noteworthy, is that the FSH treatment has the concomitant effect of reducing offspring size (see Figure 2B). This is because the size of eggs that a female produces is dependent on the number of eggs that she lays.

The effects that clutch size has on offspring size can be illustrated by a second technique for changing clutch size -- decreasing the number of eggs (Sinervo and Licht, 1991a,b). This is a relatively straightforward surgical procedure that is much simpler than the routine neutering or spaying practiced by veterinarians. When follicles are relatively small (for uta this is about 3-5 mm, *Uta* follicles are ovulated and fertilized at ~8 mm in diameter) they are surgically exposed by making a small incision in the abdomen (female is anesthetized using metafane). The follicles on one ovary are poked with a syringe needle and the yolk is gently squeezed out of the follicle (yolkectomy). The now flaccid ovary is tucked back inside and the female is sutured closed. Females undergoing this surgery typically produce a smaller clutch of eggs (from the intact ovary) consisting of much larger eggs (see Figure 3).

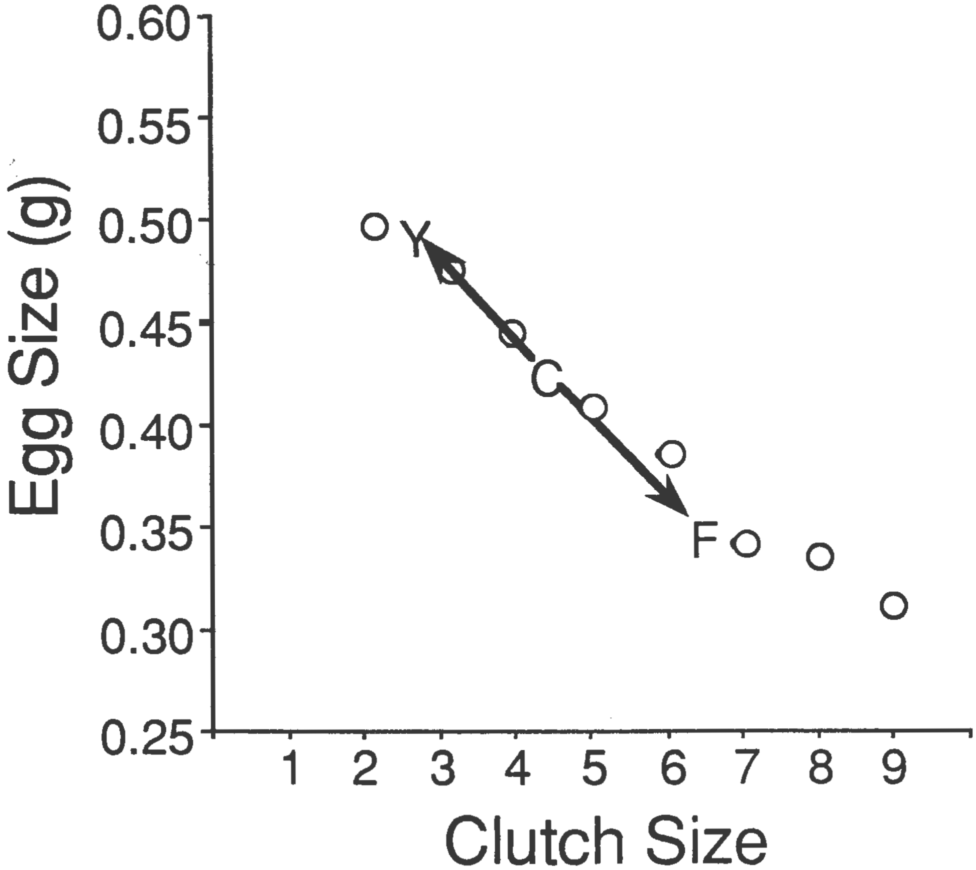


Figure 3. The effects of clutch size on egg size (g) for female *Uta*. Egg size decreases with increased clutch size. This pattern for unmanipulated utas also hold true for female *Uta* that have the clutches either increased relative to controls (C) by FSH injections(F) (i.e., C→F) or decreased by "yolkectomy" (Y) of follicles C→Y.

The two manipulations that increase and decrease clutch size demonstrate that clutch size is involved in the regulation of offspring size. In fact it is very difficult to get around this trade-off between size and number of offspring in reptiles. Nevertheless, we can use this to an advantage in increasing the efficiency of any captive program. The biology of the animal dictates which direction of clutch size manipulation would be most profitable.

IMPLICATIONS FOR CAPTIVE BREEDING PROGRAMS

Although the techniques for increasing offspring size by surgically ablating follicles do require a surgery, it is relatively straightforward requiring only about 15 minutes. In fact it is a much more simple surgery than removing necrotic eggs from egg bound lizards and snakes. The benefits to a captive rearing program are measured in terms of offspring health. Larger offspring are usually much more vigorous (Sinervo, 1990; Sinervo and Huey, 1990). These giant hatchlings are indeed robust relative to unmanipulated hatchlings. They may in fact start off feeding better and usually have higher survival in the wild. Increasing offspring size would be very useful for those animals that produce very small offspring that are very difficult to rear (e.g., some of the dwarf boas (*Ungaliophis sp.*) and the smaller day geckos (*Phelsuma sp.*))

The techniques for increasing clutch size are even more straightforward. It only requires injections of FSH every other day and the results on clutch size can be quite dramatic. For example, in *Uta* I increased clutch size from an average clutch size of 4.3 to 6.0 eggs -- almost a 50% increase in clutch size. Although the FSH injections does increase clutch size it does so by reducing the size of the offspring. This may or may not be much of problem if the offspring are very easy to rear. The FSH injections may also be an effective hormonal treatment that primes the reproduction of animals that are difficult to breed. FSH alone is sufficient to illicit growth of ovaries and follicles even without overwintering (Licht, 1970). FSH treatment may be the reptilian analog of GNRH/LH treatments used on amphibians. More research is needed on this intriguing possibility. For example, although females injected with FSH that had already started their reproductive cycle (i.e. naturally) do produce viable eggs and hatchlings it is unclear whether FSH alone with no overwintering can result in viable eggs and hatchlings.

Increasing clutch size and reducing offspring size has an additional added benefit, it apparently reduces the risk of female lizards becoming egg bound or eggs bursting when they are laid (Sinervo and Licht, 1991a). If this is generally true, it might be quite important for lowering the risk of reproductive mortality of breeding stock. However, the exact causes of eggs (or neonates) getting stuck in the female are unclear (see DeNardo in these proceedings). If the risk of becoming egg bound increases because offspring are too large then increasing clutch size and decreasing offspring size lowers this risk (as was the case for female utas). However, if eggs become lodged because the female becomes exhausted during laying, then increasing clutch size might in fact exhaust the female further and increase the likelihood of these events. This may be the case for many snakes which can have very large clutches (Dale Denardo, D.V.M., personal communication). Thus, the reasons for female reptiles becoming egg bound may differ depending on whether females are producing very large offspring or very large clutches.

It is still unclear whether these techniques will work on other reptiles such as snakes and turtles. Given the similarity of the hormonal regulation of reproduction among reptiles (Licht et al. 1970), it seems likely that comparable methods could be found for altering clutch size (and offspring size) in these groups as well.

LITERATURE CITED

Ballinger, R. E. 1978. "Variation in and evolution of clutch and litter size", pp. 789-825. In Jones, R. E. (eds.), The vertebrate ovary, Vol. New York, Plenum.

Jones, R. E., R. R. Tokarz, F. T. LaGreek and K. T. Fitzgerald. 1976. Endocrine control of clutch size in reptiles VI. Patterns of FSH-induced ovarian stimulation in adult *Anolis carolinensis*. Gen. Comp. Endo. 30: 101-116.

Licht, P. 1970. Effects of mammalian gonadotropins (ovine FSH and LH) in female lizards. Gen. Comp. Endocrinol. 22: 463-469.

------------, H. Papkoff, S. W. Farmer, C. H. Muller, H. W. Tsui and D. Crews. 1977. Evolution of gonadotropin structure and function. Recent Prog. Horm. Res. 33: 169-248.

Sinervo, B. 1990. The evolution of maternal investment in lizards: an experimental and comparative analysis of egg size and its effects on offspring performance. Evolution. 44: 279-294.

--------------- and R. B. Huey. 1990. Allometric engineering: an experimental test of the causes of interpopulational differences in locomotor performance. Science. 248: 1106-1109.

--------------- and P. Licht. 1991a. The physiological and hormonal control of clutch size, egg size, and egg shape in *Uta stansburiana*: Constraints on the evolution of lizard life histories. J. Exp. Zool. 257: 252-264.

--------------- and ----------. 1991b. Proximate constraints on the evolution of egg size, egg number, and total clutch mass in lizards. Science. 252: 1300-1302

AGAMA INTERNATIONAL, INC.
A LIZARD BREEDING INSTITUTE

Bert Langerwerf

President
Agama International, Inc.
Rt. 2, Box 285
Montevallo, Alabama 35115

INTRODUCTION

As a seven year old boy I captured and kept my first lizard, which died within a month. That was forty years ago when I started as the only terrarium keeper in my native Dutch village. Since then I have learned much about keeping lizards and after all that time, I even began to think like a lizard. During a rare thunderstorm on my favorite beach in Spain, I dug myself into the sand. Only my nose and eyes were sticking out so I could breath and see people running by, just like *Phrynocephalus mystaceus* or a *Phrynosoma* species.

Since the fourteenth century, my ancestors were all farmers who lived in the same region. I studied physics and mathematics at the University of Amsterdam, and spent 15 years teaching these subjects. The combination of long-term lizard watching, my farming background, and my studies in physics are the basis for my success in breeding lizards.

FEEDING

Around 1970, I started keeping lizards on a larger scale inside small greenhouses. At this time lizard keeping was rare and if people kept them, it was almost exclusively in an indoor terrarium. Since this time, I have discovered that lizards need a great amount of space to avoid stress and to thermoregulate as they desire, instead of the forced constant temperature indoor terrarium. Right away I made an important discovery which I easily solved using my background in physics. If lizards are kept behind glass, their eggs never hatched, except maybe for part of the first clutch laid in captivity. Lizards kept behind screen and profiting from the full exposure of the sun did much better -- almost all of their eggs hatched.

The glass acts as a filter and removes the 300 nm ultra violet light, so no vitamin D_3 can be synthesized by the lizards. Therefore, the calcium in the intestinal tract of the lizards, mainly in the stomach contents of the ingested insects, cannot be absorbed. The first clutch of eggs may hatch, but after that the female lizards run out of calcium and the embryos tend to die right before hatching, unable to cut the egg shell. Sometimes a helping hand at the right moment could save a very low percentage of the weak hatchlings. At that time, I believed that I had solved the entire problem by replacing the 300 nm ultra violet light with vitamin D_3 aquosum in the drinking water; being by far the cheapest solution. Other options, such as special plexiglass or artificial ultra violet lamps are more expensive.

A problem is never as simple as it appears. Basically I have discovered that lizards which bask for longer periods of time and/or live further south require more vitamin D_3. This is why a lizard like the leopard gecko (*Eublepharus macularis*) which is nocturnal and from relatively high latitudes is so easy to breed for many people.

Another problem was that even though vitamin D_3 is water soluble, it still has many fat-like properties. Lizards suffering from stress or parasitism such as flagellates can not digest fat very well (the tail gets dirty from sticky feces), so even if a supplemented diet is offered, viable eggs will not be produced. The optimal level of vitamin D_3 required has not been determined yet for the various lizard species. Roughly, I give 5,000-10,000 i.u. per liter of drinking water. I plan to do some research in this area next autumn for two completely different species -- a basilisk and a lacerta.

Insects are poor in calcium as they do not require it to build there own body. Therefore, the only calcium provided by an insect is contained in the food which passes through their stomachs. Insects will never become richer in calcium, even if they are fed a rich diet over a long period of time. Feeding the insects a calcium rich diet only immediately before the insects are offered to the lizards will prove more efficient . This is what occurs in nature where no one is dusting the insects for the lizards. Kale is a food that many insects prefer and which is rich in calcium. My lizards get an excess of insects at a single feeding, ensuring that even the weaker individuals are fed, and these insects feed at night from kale leaves that I have placed in the terraria to replace the water dishes. Now I never have to clean water dishes as I put fresh kale leaves into the terraria every second day. This is also quicker than cleaning dishes.

Today, there are several quality powders on the market with which to dust your insects. It is important that the powder contains a form of highly digestible calcium (insects tend to contain sufficient phosphorous) and vitamin D_3, not D_2. Two examples are Rep-Cal® from Rep-Cal Research labs in Los Gatos, CA and Formula C/P® from Mardel Laboratories Inc. in Glendale Heights, IL. I could not determine how many i. u. of vitamin D_3 are present in both of these powders. Adding this to the fact that different lizards require varying amounts of vitamin D_3 demonstrates that we are still mostly guessing and that there is still much research required.

REPRODUCTION

Like most animals, lizards are also opportunistic. This must be considered when establishing an optimal breeding program. In nature, the more food and space that are available, the quicker the lizards will reproduce to fill that space before another competitive species takes the opportunity. To increase your volume at breeding, more space and food is essentially required.

The space problem does not simply imply more cubic meters, space for lizards also requires areas with different temperatures, different humidities, and enough basking spots for all individuals. When fewer cubic meters are available, it becomes more difficult to increase the loading capacity of the space. If you have a nice terrarium in which one male and two female basilisks live and reproduce, one should not expect to double the offspring by adding two females. Instead, one can even experience a decrease in the number of offspring produced and at higher food costs. In addition, young lizards will grow faster in a less crowded the terrarium. The hard work and effort needs to be done at the beginning when you are planning and building all these spacious terraria.

Another factor that will effect production is food; the more food, the more eggs. The food for insectivorous animals should be of high quality. I mainly feed crickets and super mealworms (*Zophobas morio*), but have also used locusts, regular mealworms, and flies. Feeding a rich diet to your insects right before you feed them to the lizards is especially important. In the early eighties, five years before I helped import them into the United States, I discovered that the super mealworm was a better food for lizards because it is easy to breed, has a more digestible chitin skin than the regular mealworms, and is preferred by many lizards. Also, the mealworms are easy to feed from a dish, unlike crickets which jump away and hide. The super mealworm is now the bulk food for my lizards, and once such lizards as basilisks, lacertas, and *Sceloporus* reach adult size, super mealworms become their only food. Their nutritional requirements are simply supplied through the stomachs of the mealworms, instead of by feeding a large variety of insects.

ENCLOSURES

At Agama International, I have now built about 200 terraria that measure approximately 10 square meters each. Since we are working with 80 different species, I have built various types of terraria so that the most desirable microclimates can be provided for the varying needs of the different species. A terrarium should contain enough space and enough possibilities that the lizards can find the most favorable microclimate. One can do this indoors with computers, etc., but this would be so expensive that the captive bred offspring could never be sold. My basic idea is to start with the local climatological conditions, then build adaptations inside the terraria to establish microclimates which meet the needs of the species.

My personal desire would be to just breed endangered species of lizards in order to maintain large (over 100 individuals) breeding groups inside the United States as long as their natural habitat abroad remains threatened. Experience has taught me though, that nobody wants to fund this type of work, and I do not have the time to wait another 20 years for the thinking to change. So now I have moved on to the other option: At Agama International we plan to breed non-endangered and mainly common species for the pet trade. If one looks at the pet trade in Europe and the United States, it becomes clear that many lizards collected in the wild die between capture and purchase by the terrarium keeper. So raising 1000 lizards for the pet trade will probably save nearly 10,000 wild animals. In Europe, there is so much public pressure on the bad situation of dealing wild-caught reptiles that new laws will be enacted very soon. These laws will be extremely drastic: Dealers may no longer keep reptiles in stock, only members of reptile societies may still keep the animals. In the end, no wild caught reptiles will be available and the future of the terrarium keeper can only be assured through captive breeding. This public pressure and the proposed laws are the result of the poor care that captured animals receive, both in the country of origin and by some irresponsible dealers. As a result, breeding reptiles here is necessary so that we will not be reliant on wild populations when the laws change.

Although we are currently working with 80 species, I believe this will be reduced to 30-40 species which we are the most successful at breeding. I am still constructing terraria and expanding my colonies at the same time. To mention one species: In November 1989, I purchased 12 *Basiliscus plumifrons* and as of March, 1991 I have about 150 animals from breeding. By mid 1992, I expect to produce about 100 each month. I don't want to mention all the species that we are working with as these can be referenced in Slavens (1990). Our goal is to produce 500-1000 individuals of each species yearly. I expect that within a few years this will translate into a yearly output of 10,000 to 20,000 lizards. The genera *Basiliscus, Eublepharus, Pogona, Chamaeleo, Bradypodion, Lacerta, Sceloporus,* and *Physignathus* will be well represented. With the revenue generated, I can turn back to my first wish; breeding endangered species of the genera *Uromastix*, *Cordylus*, *Cyclura*, *Shinisaurus*, among others.

CONCLUSION

I hope my work will make it possible for other seven year old boys and girls in the future to learn about nature by keeping a lizard from my captive breeding program. For this reason I have started breeding many affordable species such as *Lacerta strigata, Prammodromus algirus, Leiocephalus carinatus, Eublepharus macularius,* and *Basiliscus basiliscus.* In the future, conservation and protection of wild areas will very much depend on the fact that our youth can still have contact with nature. Strict laws, forbidding us to keep anything wild, or the unavailability of lizards for terrarium keeping will in the long run work against conservation, as apathy arises from ignorance.

LITERATURE CITED

Slavens, F. 1990. Inventory of Live Reptiles and Amphibians in Captivity, Current January, 1990. Slavens, Seattle, WA

ACKNOWLEDGEMENTS

Dr. Mark Paris, Secretary, Agama International Inc.

Vitaly Charny, Animal Keeper, Agama International Inc.

Over half of the terraria at the Agama International, Inc. facility. Photo by Bert Langerwerf.

CAPTIVE REPRODUCTION AND HUSBANDRY OF THE AUSTRALIAN EGERNINE SKINK *Egernia striolata*

R. Peter Yingling

8437-84 Via Mallorca
La Jolla, CA 92037

INTRODUCTION

Skinks (family Scincidae) are the largest group of lizards, with approximately 73 genera and 800 species. They are found in most of the warmer regions of the world, but the majority occur in sub-Saharan Africa, Southeast Asia and Australasia. Most species are rather small, slender, smooth-scaled, and semi-fossorial; a number show a pronounced tendency toward reduction and even loss of the limbs.

Greer (1970) classified skinks into four subfamilies (Acontinae, Feylininae, Scincinae and Lygosominae) based on a correlation of several skull features and external characteristics. The first two groups are specialized limbless burrowers from small areas of central and southern Africa. Most of the Scincinae are found in Africa and southwest Asia, with several representatives in the New World. The Lygosominae are the most advanced and widespread skinks, occurring in South America, Africa, Asia and Australasia. Included in this subfamily are the "egernines", *Egernia*, *Tiliqua*, and *Corucia*; Hutchinson (1980) gives a comprehensive overview of these genera. The first two are found mostly in Australia, although one species of each also occurs in New Guinea and Indonesia. *Corucia* is monotypic and restricted to the Solomon Islands.

Egernia is the most morphologically and ecologically varied group in the entire family; the genus includes about 26 species (Wilson and Knowles, 1988) that are found in all areas of Australia except the highest mountains. These are mostly medium-sized to large skinks with well-developed limbs, each with 5 digits. The lower eyelid is scaly and moveable, and the ear opening has one or more lobular scales. Body scalation may be smooth, weakly striated, keeled or heavily spinose. All but one species are diurnal; the smaller forms are largely insectivorous, while the larger ones are omnivorous. Several exhibit quite unusual behavior, such as colonial living and regular defecation sites. All members of the genus are live-bearing, with rather small litters (usually 2-8); a well-developed chorioallantoic placenta has been demonstrated in some forms (Cogger, 1986).

Wilson and Knowles (1988) divided *Egernia* into 7 species groups, based on differences in scalation, size, feeding preferences and general habits. The tree skink, *Egernia striolata*, is a small species, reaching an adult total length of about 20 cm (8-9 in), about half of which is tail. Coloration varies from light gray to brownish, frequently with darker flecking that forms a pattern of narrow broken lines; a dark brown or black longitudinal band is also present on each side of the body. Juveniles closely resemble adults in both coloration and pattern. This form occurs from the interior of east-central Queensland south through New South Wales (west of the Great Dividing Range) to northern Victoria and southeastern South Australia in dry sclerophyll forest and associated woodland.

MATERIALS AND METHODS

The author obtained his first tree skink (21.0 cm total length and presumably an adult male) in March, 1985 from a private breeder. In August, 1988, a pair of individuals was obtained in trade with the Blijdorp Zoo, Rotterdam, Holland; these were both about 17.0 cm (7.75 in) and were presumed adults. The first animal was kept separately from the pair; all were housed in a custom-built unit consisting of four 70 x 30 x 30 cm (26 x 12 x 12 in) wooden boxes contained in a modular rack. A 15-watt fluorescent fixture was placed directly above a screened rectangular cutout in each cage top, and a 10-watt heater cable was stapled to a piece of thin plywood and mounted directly underneath each box. For more information on this design, see Yingling, 1988.

All cages were provided with about 1.3 cm (0.5 in) of gravel substrate, a small ceramic water dish, and two curved pieces of eucalyptus bark. No branches were included, but gravel glued to the cage walls served as a vertical substrate. An electronic timer was set to switch on both the cage light and heater for about seven hours daily; no efforts were made to vary either the photoperiod or temperature seasonally or to synchronize them to then-prevailing conditions in the Southern Hemisphere. Both Vita-lite® and ordinary cool white® fluorescent tubes were used at various times with no apparent change in the lizards' behavior. During the off-cycle the animals were effectively at ambient room temperature, which varied from about 20-30°C.

However, the tree skink has been shown experimentally (Bennett and John-Alder, 1986) to have a preferred mean body temperature of about 33°C., so an attempt was made to attain this condition. The timer and heater were both connected to a "bi-metal" thermostat, which was calibrated with a thermometer to maintain the rear half of the cage substrate at 30-33°C. (the thermostat is accurate to about 2-3°C.). One of the bark pieces was positioned in this heated portion and the other piece was located in the unheated section.

All three lizards are very secretive, emerging from cover infrequently, even during feeding. One animal seemed to tame more than the others and soon learned to take food from my hand. All individuals, however, remained nervous and "wriggly" when handled. They fed readily on crickets, mealworms (*Tenebrio* larvae), waxworms (*Galleria*) and also accepted small bits of fruit, cat food, and mixed vegetables. Apparently, they had no difficulty digesting the chitinous mealworm cuticles. Food was dusted with Super Preen® vitamin powder with no change in the acceptability of the food. Aggression during feeding was minimal between the pair; sometimes one animal took food from another, but no overt fighting or injuries occurred.

Courtship or mating activity was not observed between the paired animals, but in July, 1989, two young were born. The right eye of one juvenile was missing, possibly a congenital defect. Even though no aggression was displayed by the parents, the young were immediately moved to a 5-gallon terrarium. After about a week, the juveniles began feeding on small crickets and waxworms. No aggression was noted between these two animals, but several months later, the two-eyed juvenile was found dead, possibly killed by its one-eyed congener. The latter animal was then moved back into the cage with its parents, and was soon missing part of its tail. It was then placed in a 5-gallon tank with a subadult pygmy spiny-tailed skink (*Egernia depressa*), and both animals seemed to adjust without incident.

In late June, 1990, two more young appeared, and both were physically normal. They were removed from the parents' enclosure to a 5-gallon terrarium and soon began taking food (mainly small waxworms). After several weeks, however, one individual stopped feeding and soon afterward the tip of its tail was apparently eaten by its more aggressive congener. The cage was then divided in half by a wooden partition and in about a week, the subordinate animal resumed feeding. Over the next several months, it gained weight and its tail tip slowly regrew. At present (January, 1991), these juveniles are about half-grown; they measure 12.0 cm (4.5 in) total length and both appear to be thriving.

DISCUSSION

Most of the literature on the tree skink consists of taxonomic accounts, thermoregulatory studies, and isolated captive breeding records. At least two captive care books (Weigel, 1988; Zimmermann, 1986) also mention this species.

Bustard (1970) surveyed a naturally-occurring population of *Egernia striolata* in Australia for 6 years and found them to be mostly arboreal, occurring beneath the bark of dead trees. Adults were solitary, although juveniles were often seen on the same tree. The species was found to be strictly diurnal and quite shy, basking intermittently, but never far from cover. Individuals apparently have small home ranges, as most recaptures were from the same location. The diet was largely insects, 2-6 young were produced once annually and adulthood was reached by the second or third year. These data correlate well with the above captive observations.

The few captive breeding records for the tree skink (Olney, 1978-1983; Slavens, 1988) give only the location and number of young produced. Reproduction has occurred in England (Zoological Society of London), Holland (Blijdorp Zoo), Australia (Taronga Zoo, Sydney), and Germany (Max Planck Institute, Wuppertal); no previously published records have been located for the United States. The author's pair of *E. striolata* apparently were offspring from parents originally produced at the Max Planck Institute, but the origin of the single individual could not be ascertained.

CONCLUSION

In summary, the tree skink appears to be an ideal terrarium animal, adjusting well to captivity and breeding quite readily. However, this species seems to have a high degree of intraspecific aggressiveness (at least in captivity), therefore caution should be exercised when pairing individuals. The author will make future efforts to breed the single individual with one or more of the offspring, since it is likely that these animals are unrelated.

LITERATURE CITED

Bennett, A. F. and H. John-Alder. 1986. Thermal relations of some Australian skinks. Copeia 1986(1): 57-64.

Bustard, R. 1970. Australian lizards. Collins, Sydney and London.

Cogger, H. G. 1986. Reptiles and amphibians of Australia. 4th edition. Reed Books Pty., Ltd. Sydney. 688 pp.

Greer, A. E. 1970. A subfamilial classification of scincid lizards. Bull. Mus. Comp. Zool. 139(3): 151-184.

Hutchinson, M. N. 1980. The systematic relationships of the genera *Egernia* and *Tiliqua* (Lacertilia: Scincidae). A review and immunological reassessment. IN: Banks, C.B. and A. A. Martin (eds.). Proceedings of the Melbourne Herpetological Symposium, May 19-21, 1980. pp. 176-193.

Olney, P. J. S. (ed.). 1975-1983. (*Egernia striolata*). Reptiles bred in captivity and multiple generation births. Section 3. IN: Internat. Zoo. Yearbook, vols. 17-25.

Slavens, F. L. (ed.). 1988. Inventory, longevity and breeding notes - reptiles and amphibians in captivity, current January 1988. Privately printed, Seattle, WA. 401 pp.

Weigel, J. 1988. Care of Australian reptiles in captivity. Reptile Keepers Association, Gosford, New South Wales. 144 pp.

Wilson, S. K. and D. G. Knowles. 1988. Australia's reptiles; a photographic reference to the terrestrial reptiles of Australia. Collins, Sydney. 435 pp.

Yingling, R. P. 1988. Beyond the sweater box; cage construction techniques for amphibians and reptiles. IN: Gowen, R. (ed). Proceedings of the Northern California Herpetological Society's Conference of Captive Propagation and Husbandry of Reptiles and Amphibians. pp. 155-161.

Zimmermann, E. 1986. Breeding Terrarium Animals. TFH Publications, Inc., Neptune City, NJ. 384 pp.

PRODUCTS MENTIONED IN TEXT

Bi-metal thermostat. Custom-built by Lyon Electric Co., San Diego, CA (no longer available).

Cat food. Friskies Buffet® (various flavors). Carnation Co., Los Angeles, CA.

Electronic timer. Micronta LED programmable clock/timer, cat. no. 63-888. Radio Shack, division of Tandy Corp., Fort Worth, TX.

Fluorescent tubes. Vita-lite®, Duro-test Corp., Bergen, NJ.; cool-white® tubes, General Electric Corp.

Heating cable. Drier®, part no. 5905354, Frigidaire Division, General Motors Corp., Dayton OH (used to eliminate damp spots in refrigerator cabinets).

Vitamin-mineral powder. Blair's Super Preen®, RHB Laboratories, Inc., Santa Ana, CA.

OPERATIVE TEMPERATURE FOR THE HERPETOCULTURIST

Thomas J. Greek

2582 Chesapeake Dr.
Madison, WI 53719

INTRODUCTION

Organisms are sensitive to temperature during all stages of their lives, from a developing embryo to an adult. They are forced to live within a maximum and minimum temperature span, outside of which they will perish. Between these temperatures there is a smaller range within which they are normally active and smaller still is the range for optimal physiological functioning. Despite these thermal constraints placed upon them, animals survive in a wide range of habitats from cold mountain peaks to hot dry deserts. This includes ectothermic animals such as reptiles and amphibians, whose body temperature is determined primarily by the environment.

In captive environments, the keeper is responsible for providing the proper temperature regime. The literature, including these proceedings, are full of examples that include maintenance temperatures. In this paper, I will introduce the concept of operative temperature (T_e). By understanding the components of temperature, the keeper can better interpret the literature and provide an improved habitat for his animals.

DEFINITION OF OPERATIVE TEMPERATURE

The term operative temperature was first used in 1937 (Winslow et al.) to describe the temperature an animal feels. To help in understanding what I mean, let me give some examples. We've all heard the television weather man describe - "it's 40 degrees outside, but with the wind chill it feels like 10 degrees" or "it's 90 degrees in the shade, but it feels much hotter in the sun." What he is saying is that despite what the air temperature is, your body feels something different.

The equation for T_e is:

$$T_e = T_a + \frac{(R_{abs} - \epsilon\sigma T_a^4)}{\rho c_p(1/r_a + 1/r_r)}$$

where T_a is the air temperature, R_{abs} is the absorbed long and shortwave radiation, ϵ is the emissivity, σ is the Stefan-Boltzmann constant, ρ is the air density, c_p is the specific heat of air, r_a is the aerodynamic resistance to convective heat transfer, and r_r is the resistance to radiative heat transfer. (For an in depth evaluation of this and other operative temperature equations, see Paw U, 1987). Some of these components are related to the more detailed physics of the equation. Others have more practical applications to the herpetoculturist and I will limit my discussion to these.

Absorbed radiation is divided into two components, short and longwave radiation. Shortwave radiation is what we see as light and can be either direct or diffuse such as when it is reflected from clouds, the ground, or other structures (Figure 1). The effect of shortwave radiation on operative temperature is dependent upon the animals position with respect to the direct solar beam. A lizard lying on a rock perpendicular to the sun can maximize the radiation hitting its body and have the greatest increase in operative temperature. Turning so that only the top of its head receives direct sunlight will minimize the effects of direct radiation. By getting out of the direct sunlight it can eliminate that factor all together, while still leaving the diffuse shortwave radiation component.

Longwave radiation can also be called thermal radiation. This can come from the sky or any neighboring structures (Figure 2).

Both short and longwave radiation are affected by a factor called absorptivity. This is a term that describes how much of the radiation hitting the animal is actually absorbed and how much is reflected. Darker colors absorb more radiation while lighter colors reflect more. Not all species of reptiles and amphibians have the ability to alter absorptivity.

Notice that $\epsilon\sigma T_a^4$ is subtracted from the absorbed radiation value. This is because the heated animal, just like any other heated object, will emit radiation as well. If the animal is loosing more radiation than it is gaining, its operative temperature can be lower than the air temperature (Figure 3).

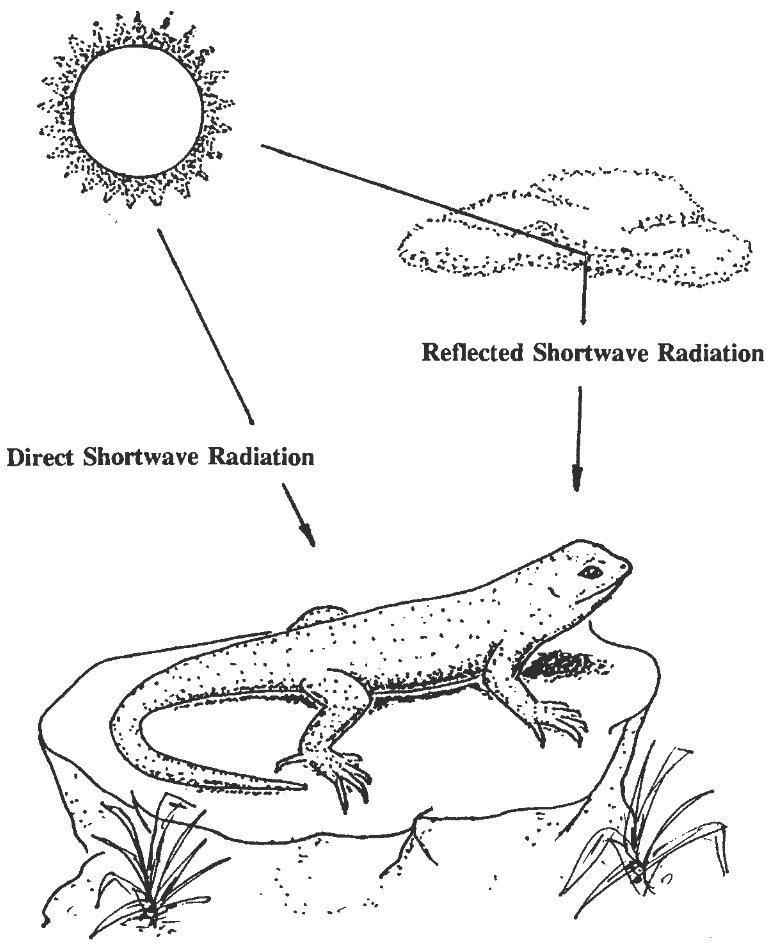


Figure 1. The components of shortwave radiation in a lizard's operative temperature.

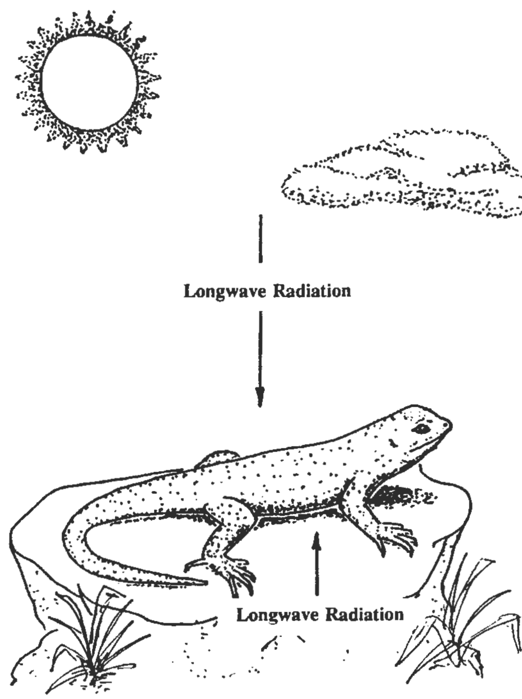


Figure 2. The components of longwave radiation in a lizard's operative temperature.

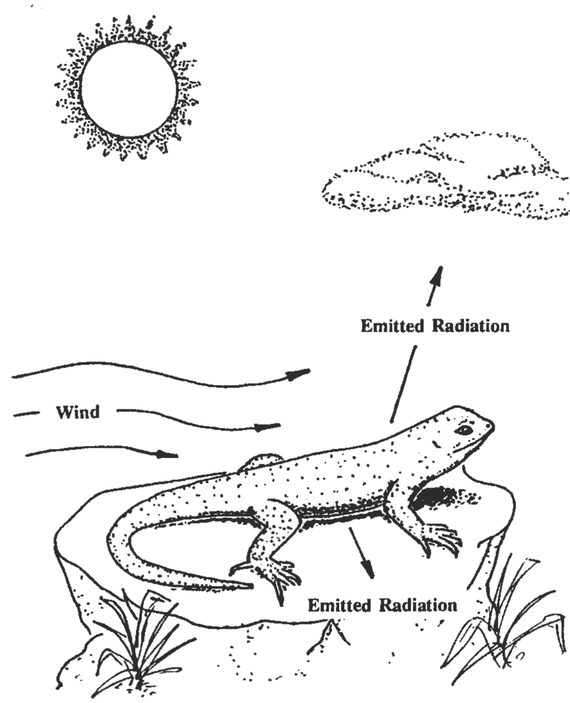


Figure 3. Sources of potential energy loss in a lizard's operative temperature.

How would a lizard in the wild use these components of absorbed radiation? When the lizard emerges in the morning it will lie in the direct sunlight with its body as flat as possible and perpendicular to the sun. In addition, the animal will turn to its darkest possible coloration to absorb as much radiation as possible. During the heat of the day, the lizard may spend all of its time in the shade and be displaying its lightest coloration thereby decreasing the amount of radiation absorbed. This behavior is an attempt to maintain its preferred body temperature.

The influence of r_a (boundary layer resistance) and r_r (resistance to radiative heat transfer) on operative temperature is shown by the rates of heat gain or loss. The boundary layer of air around the body acts as insulation and protects from the loss of heat. Wind will decrease this boundary layer so that heat is lost more readily. Most of us would know this as the wind chill factor (see Figure 3). Birds and mammals will fluff up their feathers or fur to trap air as insulation, but reptiles do not have this capability.

Body size has an effect on heat gain. Large animals may take a relatively long time to heat up to their preferred body temperature, while a smaller species will heat up quickly. Conversely, a small animal will cool quickly while the larger will cool much slower. This effects the animals behavior in the wild. A small lizard on a cold day may be able to heat itself up to its preferred body temperature, but after a short time of hunting in the cooler shade it will have to return to the sunlight to heat up. A large lizard may spend a longer time basking, but then can spend a longer time hunting as well.

The complex nature of operative temperature allows ectothermic animals to be active during air temperatures that are below or above their preferred body temperature. Sensitivity analysis of the operative temperature equation can be used to correlate behavior with environmental components (Greek et al., 1989). Some species can change absorptivity to affect the amount of absorbed radiation. Animals can change body position with respect to direct sunlight or wind direction and thereby change operative temperature. Simply by moving between direct sun and shade changes the amount of incoming shortwave radiation and will influence operative temperature.

RELATION TO HERPETOCULTURE

Since the operative temperature equation is so complex, the same value can be achieved in numerous ways. But not every species uses each operative temperature factor the same way. An arboreal diurnal species would most likely use solar radiation as its primary means of heating. Captive maintenance using only a hot rock for conductive radiation could lead to stress, poor health, or lowered reproductive success of the animal. In the same fashion, a nocturnal species heated with only a spotlight would have the same adverse outcome. Of course some species are more generalists and the method of heating the cage may not be critical, but it should be included when reporting your captive husbandry methods.

Determining what temperatures you should provide for your herptiles can be an arduous task. Some herpetoculturists have gone to climatic tables to determine temperature ranges, humidity and rain fall for the geographic area of the species. This is a great start but the data must be analyzed with the natural history of the animal in mind. Within the habitat, an animal will only utilize a small portion which is termed the microhabitat. Because of the various factors affecting operative temperature, these microhabitats can be much warmer or cooler than the air temperature.

Remember that reptiles and amphibians will alter their preferred body temperature in response to various physiologic stimuli. After a meal, snakes are often noticed to have one coil closer to the heat source. And the medical benefits of allowing ectotherms to create a behavioral fever have been well documented. Therefore, a temperature gradient to allow these changes is essential.

CONCLUSION

Operative temperature is a complex equation that includes air temperature, solar radiation, wind speed and body characteristics to determine the temperature that an animal will feel in its environment. Scientists have used operative temperature for physiological and ecological studies. Herpetoculturists can apply operative temperature by understanding the components and comparing them to the natural history of the animal in order to provide the proper type of heating for their species.

LITERATURE CITED

Greek, T.J., Paw U, K.T., Weathers, W.W. 1989. A comparison of operative temperature estimated by taxidermic mounts and meteorological data. J. Therm. Biol. 14:19-26.

Paw U, K.T. 1987. Mathematical analysis of the operative temperature and energy budget. J. Therm. Biol. 12:227-233.

Winslow, C.E.A., Herrington, L.P., Gagge, A.P. 1937. Physiological reactions of the human body to varying environmental temperatures. Am. J. Physiol. 120:1-22.

CAPTIVE REPRODUCTION AND BIOLOGY OF THE ARGENTINE SNAKE *Philodryas baroni*

Harold F. De Lisle

Department of Biology
Moorpark College
Moorpark, CA 93021

INTRODUCTION

The genus *Philodryas* is composed of seventeen species of colubrid snakes inhabiting the tropical and temperate regions of South America (Peters and Orejas-Miranda, 1970; Vanzolini, 1986). Most species are arboreal or semiarboreal and are characterized by having the last two maxillary teeth enlarged and channeled, making them truly rear-fanged.

Among the species I have studied, the most remarkable is *Philodryas baroni* Berg, a species from northern Argentina, where it is variously known as "la serpiente voladora" (flying snake) or "culebra verde" (green snake) (Freiberg, 1982). It is an active diurnal species that does not hesitate to glide from branch to branch through the small trees of the Chaco. *P. baroni* is one of the largest members of the genus, reaching at least 165 cm snout-vent (SV) and 208 cm total length.

This species has two color phases: one is green (yellow-green to bright leaf green); a second phase is yellowish to reddish brown with dark brown spots and bars. Both phases occasionally have a black vertebral stripe.

HABITAT

Philodryas baroni is found in the provinces of Chaco, Santiago del Estero, Salta, Tucuman, and Catamarca in Argentina (Peters and Orejas-Miranda, 1970). This range is in the western part of the Gran Chaco region and the foothills of the Andes.

The Gran Chaco is a hot, semiarid region covered mostly by thorny scrub with some grassy savannas and seasonally flooded plains in the north and east. Mostly subtropical, the region has pronounced seasonality with a long hot summer and a mild, dry winter. The rainy season comes in the late summer months of February, March, and April (Rudolph, 1985). This climate produces a habitat that in many ways resembles the southern Sonoran Desert of northern Mexico. *P. baroni* inhabits the more arid, western parts of the Gran Chaco.

HABITS

Although *P. baroni* possesses the typical streamlined morphology of an arboreal snake, it is not especially active, for all its "racer-like" appearance. It spends many hours resting motionless on branches or dangling among the leaves. It usually holds its head above the resting body. The function of the nasal appendage has not been determined, although one hypothesis is that it is used to turn and probe leaves in search of resting tree frogs. This species has a moderately nervous temperament and resists being handled. However, it is totally non-aggressive, unlike several other species of *Philodryas*. It has never been known to bite in defense. It is easily disturbed, always making an attempt to flee, even when cornered.

Although considered an arboreal snake, it does not hesitate to travel across open ground as it moves about its home range.

Most members of the genus *Philodryas* are omnicarnivorous. *P. baroni* follows the generic food preferences. Food in the wild is reported to consist of birds, tree frogs, and small mammals (Freiberg, 1982). This species readily accepts mice in captivity. Mice are actively pursued around the cage and are killed by constriction and paralysis by the mild venom of this snake.

Like most members of the genus *Philodryas*, *P. baroni* is diurnally active. It is active over a surprisingly wide range of temperatures (15-35°C in the laboratory). If maintained above 20°C, these snakes will feed all year round. Seasonal activity in the wild is unknown, although most species of reptiles in the Chaco become dormant for a couple of months in mid-winter (July-August), when night-time temperatures can approach freezing. Vitt (1980) found that the breeding season of two species of *Philodryas* in the semiarid Caatingas region of Brazil was quite short.

No data are available as yet on the life span of this rarely kept species (Bowler, 1975). My oldest specimen was acquired as a large adult and has been in captivity 2 years 10 months at the time of this symposium.

HUSBANDRY

Philodryas baroni is easily maintained in captivity. It needs a fairly large cage with tree branches. Active snakes never use a hide box, retreating to it only for winter dormancy. In my experience they do not care to soak, and prefer low to moderate humidity (<50%). A small water dish is sufficient for drinking purposes. A fluorescent light seems to stimulate activity, but is not essential. A basking lamp or hot rock is necessary only if one wishes to maintain activity where temperatures fall below 15°C.

Temperatures below 15°C stimulate dormancy, which lasts three months in southern California ambient temperatures. Snakes tolerate dormancy well with little weight loss. Snakes maintained above 15°C will continue to feed weekly.

As mentioned previously, this species readily accepts mice as food in captivity, and specimens in my study have been maintained on one or two laboratory mice per week. They are able to take relatively large mice for the body diameter of the snake.

Ecdysis occurs at 2-3 month intervals, and has always been problem free in my group.

Diseases and parasites are not a major problem with this snake. I have had only specimen die from unknown causes, and that was within the 60 day quarantine period after importation. One other specimen acquired an external abscess of the lower jaw, but continued to feed well.

This species seems to readjust its internal mechanisms to northern hemisphere seasons quite readily, as is apparent from the following account on reproduction.

REPRODUCTION

The usual method of using probes to determine the sex of snakes is not reliable for this species, and also, I suspect, for many non-colubrine colubrid snakes. The two snakes in this report of reproduction were originally identified as males by probing. But on June 20, 1990, during a spell of very hot weather (+37°C), two snakes housed in the same cage were observed mating at 1630 hrs. Courtship was not observed; mating was in typical colubrid fashion, and continued for only five minutes after it was first observed. The male is 105 cm SV, tail 44 cm; female 112 cm SV, tail 27 cm. The snakes had been in captivity for eight months, and had not experienced winter dormancy (cool temperatures).

On July 26, nine eggs were laid during the day over an eight hour period. The eggs were measured and averaged 60.8 x 26.5 mm for the fertile eggs and 56.3 x 23.3 mm for the infertile eggs. The eggs were placed on vermiculite (60% to 40% H_2O by volume) and incubated at ambient temperatures (13-37°C). Six of the eggs hatched on October 14-15. Hatchlings remained in the egg for 10-12 hours after pipping the shell.

Clutch size among the species of *Philodryas* is reported to range from 6 to 14 (Vitt and Vangilder, 1983; do Amaral, 1976). There are no literature reports for *P. baroni*.

Size of hatchlings at birth ranged from 372-446 mm, and first ecdysis occurred from two to four weeks after birth. Hatchlings then began to feed on "pinky" mice, but only sporadically during the California winter months under maintenance temperatures of 15°-21°C. Growth during the first three months is contained in Table 1. The average growth rate for the first three months was 10%; the largest snake at birth is the best feeder and grew 15%.

Table 1. Growth of *P. baroni* hatchlings.

BIRTH	3 MONTHS
446 mm	520 mm
428	475
419	459
395	450
392	421
372	403

VENOM

All members of the genus *Philodryas* are opisthoglyph snakes. They possess a large Duvernoy's gland with ducts leading to the last two maxillary teeth which are long, curved, and contain grooves through which the venom flows (Figure 1) (De Lisle, 1982).

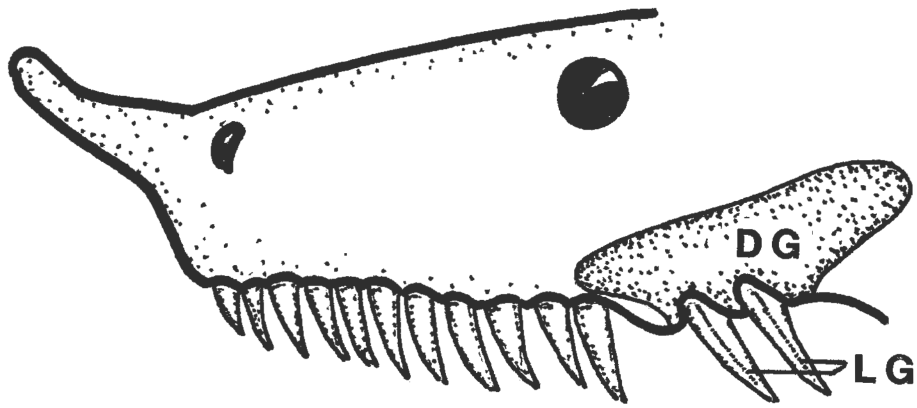


Figure 1. The Duvernoy's gland (DG) and the laterally grooved maxillary teeth of *P. baroni.*

Duvernoy's gland is a modified salivary gland which produces a variety of lytic enzymes (Rosenberg et al, 1985). No secretions of any *Philodryas* species have been analyzed to date. The process is difficult, expensive, and requires the resources of a large physiological and biochemical laboratory.

Even the secretory mechanism of Duvernoy's gland is not entirely known. There is thought to be a prolonged resting stage when the gland is inactive, and a short active stage stimulated by hunger or feeding reflexes which lasts several hours. This active phase is probably controlled by parasympathetic innervation as is salivary secretion in most vertebrates. The serous secretions leave the cells during this active stage and enter the many tubules of the gland. When the snake engages the maxillary teeth in a bite the secretions enter the ducts and are squeezed by surrounding muscles into the channels of the enlarged teeth (fangs).

The venom of *P. baroni* is highly effective. Ten trials were conducted with 12-15 gm mice, and resulted in an average of 62 seconds to death. Similar sized gopher snakes (*Pituophis catenifer*) were used for comparative purposes. Without the Duvernoy's secretion, by constriction alone, it took an average of 90 seconds to kill the mouse.

The venom of *P. baroni* certainly aids in quick immobilization of prey, but it may also have a digestive function as well. In those few species of snakes with Duvernoy's gland where secretions have been analyzed, some tissue digesting enzymes were present which would begin the breakdown of the food item from the inside while the usual digestive enzymes work from the outside. The digestive process is fairly rapid in *P. baroni*, taking as little as 48 hours at 28°C for complete passage through the alimentary canal.

As mentioned previously, *P. baroni* is very mild-tempered, no human envenomations have occurred, and the possible effects of the venom on humans is unknown. A closely related species, *P. olfersi*, a very aggressive species, was reported by Nickerson and Henderson (1976) to have caused human envenomation as a result of a four second bite to the thumb. Swelling occurred within 10 minutes, and the entire hand was swollen within seven hours. No treatment was given and symptoms subsided in 48 hours.

ACKNOWLEDGEMENTS

I wish to thank Rudolfo Ruibal of U.C. Riverside for his help in understanding the ecology of the Gran Chaco, and Christine Roscher for providing specimens for the research.

Note: The same female mentioned in the text produced 13 eggs on July 10, 1991 and 10 eggs on July 1, 1992.

LITERATURE CITED

Bowler, J.K. 1975. Longevity of reptiles and amphibians in North American collections. SSAR Herp. Circ. #6, 35pp.

De Lisle, H.F. 1982. Venomous colubrid snakes. Bull. Chi. Herp. Soc. 17:1-17.

do Amaral, Afranio. 1976. Serpentes do Brasil. Univ. Sao Paolo, 246pp.

Freiberg, M. 1982. Snakes of South America. T.F.H. Publ., 189pp.

Nickerson, M.A. and R.W. Henderson. 1976. A case of envenomation by the South American colubrid *Philodryas olfersi*. Herpetologica 32:197-198.

Peters, J.A. and B. Orejas-Miranda. 1970. Catalogue of the Neotropical Squamata: Part 1. Snakes. U.S.N.M., Washington DC, 347pp.

Rosenberg, H.I., and A., Bdolah, E. Kochva. 1985. Lethal factors and enzymes in the secretion from Duvernoy's gland of three colubrid snakes. Jour. Exp. Zool. 233:5-14.

Rudolph, J.D. (ed). 1985. Argentina, a country study. Amer. Univ. Foreign Area Studies, Washington DC.

Vanzolini, P.E. 1986. Addenda and corrigenda to the catalogue of neotropical squamata. Smithsonian Herp. Info. Serv. no. 70., 25pp.

Vitt, L.J. 1980. Ecological observations on sympatric *Philodryas* (Colubridae) in northeastern Brazil. Papeis Avulsos Zool. (Sao Paolo) 34:87-98.

------------, and L.D. Vangilder. 1983. Ecology of a snake community in northeastern Brazil. Amphibia-Reptilia 4:273-296.

THE CAPTIVE CARE AND HUSBANDRY OF THE BAJA MOUNTAIN KINGSNAKE

Roger J. Klingenberg, D.V.M.

6297 W. 10th Street
Greeley, CO 80634

INTRODUCTION

The Baja mountain kingsnake (*Lampropeltus zonata agalma*) is the southern most subspecies of the group of snakes commonly known as the California mountain kingsnakes. *L. z. agalma* is commonly known as the San Pedro mountain kingsnake or the Baja mountain kingsnake. These are small secretive kingsnakes that show many traits common to montane species. By understanding these characteristics, captive propagation has become possible in the last couple years.

The 25 to 30 specimens viewed by the author have ranged from hatchlings to adults of several years of age. Hatchlings are 15-18 cm at birth and average 3-4 gm. The largest adult was approximately 71 cm long and weighed 195 gm. These small snakes are generally red, white, and black banded, although the white often takes on a yellowish hue. One young female was a vivid orange instead of the typical bright red. Distinctive characteristics include 40 or more triads from head to vent, red on the nose varying from small dots to liberal amounts, and varying amounts of black dorsally invading or splitting the red especially in the rear third of the body. The nose is a black background with varying amounts of red and bordered to the rear of the head by a variable white band before the normal triads begin. Perhaps the most attractive feature besides the many triads is the zig-zag pattern of the white within the triads.

L. z. agalma is indigenous to the Baja California mountains. They are found primarily in the northern range of the Sierra Juarez and the Sierra Pedro Matif. These snakes are found at higher elevations, starting at 1,800 m, usually at the point where conifer growth begins. Like the other subspecies, *L. z. agalma* seems to occupy a sub-terranean existence in the extensive rock formations present at these elevations. As mentioned previously, these small snakes seem to be quite secretive. Very few have been found "cruising" in the open, but those which were, were found during the late afternoon and early evening at sub-optimal temperatures. As will be expanded on later, these snakes seem to frequent and tolerate lower temperatures than most kingsnakes, with some individuals found in the wild reported to be ice cold to the touch. Therefore, I postulate that these snakes are bi-urnal and are likely to be active from early evening until early morning.

These snakes have been found as early as late April and as late as September. Due to the high elevations, it would seem that hibernation would begin as early as late August and possibly extend to the middle of March or later.

The major prey in their normal sub-terranean habitat is lizards. However, they will also eat small rodents and have been speculated to occasionally eat one of the numerous species of tiny night and ground snakes. To my knowledge there are no proven accounts of feeding on other snakes.

CAPTIVE HUSBANDRY

An enclosure needs to be able to provide for the secretive behavior of *L. z. agalma*. In the coming year my colony will be caged in the Herpetat Modular cages (Merker and Mackin, 1987) with a drawer system. Wooden cages with drawers would also be appropriate. However, in 1990 my colony was housed in Sta-in-Pet® glass cages with a sand substrate. A 10 x 15 x 5 cm black plastic reptile hide box was included in each cage. A liberal amount of dark aspen chips were provided in one half of the cage for the snakes to burrow and hide in, and an additional hide box was provided by a piece of PVC pipe that was cut in half and placed face down. Water was always available in a 5 x 5 cm ceramic crock.

Heat was provided by two sources, one of which, a heating pad designed to accompany the Sta-in-Pet cages, provided an excellent heat gradient as the heat was dissipated in the sand substrate. The black plastic hide box was placed half on and half off the heat, as were the wood chips. The PVC house was on the opposite, cooler side of the cage. The other source of heat was the background heat provided in the reptile room. A heater was turned on each morning and provided a background of 25-26°C for 8 hours daily. In the early evening the heat automatically turned off and the room slowly dropped to a nighttime low of 20°C.

The majority of these snakes would readily accept pinkie or small fuzzy mice. Only one snake required the mice to be lizard scented for a couple weeks. Over two seasons of working with these snakes I observed that they feed readily after hibernation and through the summer, but would abruptly go off feed in late July to early August despite temperature or light cycle provided. Since the feeding season was relatively short, the snakes are fed liberally unless regurgitation is seen.

Light is provided to the reptile room in general and not to the individual cages. Two banks of Vita-lights® are suspended in the room, so illumination of each cage is good. A timer will provide 8-10 hours of light when the snakes are brought out of hibernation. After two weeks, the light is increased to 12 hours daily to possibly promote hormonal changes and encourage breeding.

Hibernation is initiated in early September by discontinuing feeding (they usually have stopped themselves), unplugging the individual cage heat sources, and decreasing the light cycle back to 8 hours daily. After 2-3 weeks the background heater is discontinued. As weather permits, natural cold is allowed into the room by a controlled opening until the temperature is in the 10-13°C range. This temperature is maintained until the end of hibernation in mid to late February. The light cycle is reduced to 2 hours daily during the majority of hibernation. In mid to late February, the process is reversed with the light cycle gradually increased, background heat initiated, and within 1-2 weeks the in-cage heaters turned on. Feeding is initiated approximately 7-14 days post hibernation depending on the activity and interest of the individual.

This subspecies of the California mountain kingsnakes seems to be very cold tolerant. At 10°C, they are much more alert and active than are other snakes, such as grey banded kingsnakes (*Lampropeltus alterna)*, Pueblan milksnakes (*L. triangulum campbelli*), and California kingsnakes (*L. getula californiae*), at the same temperature. For example, a grey banded kingsnake at 10°C will exhibit extreme sluggishness, and ataxia (jerky, uncoordinated movements). A Baja mountain kingsnake will not exhibit ataxia and while slightly sluggish will still musk and strike in defense.

BREEDING

Most other *Lampropeltis* species are put together <u>after</u> the female's first shed of the season. Females are transferred to the male's cage and the pair is kept together except during feeding. Typically, the females are kept with the males for 3-4 days, then separated for 1-2 days for feeding, then back together again. It has been noted by other persons working with this species that waiting for the female to complete her first shed of the season prior to introducing the male has resulted in decreased interest and infertile eggs. Sometime later when the female turns opaque (hopefully for her pre-egg laying shed), the pair is separated into their own cages for the remainder of the season.

While shy and secretive in their normal day to day activities, these snakes appear to be very aggressive breeders. Males tend to pursue females within minutes of being introduced, and breeding behavior is repeated frequently. Males tend to refuse food during the breeding season. Also, since their active season is so short, the males should be offered food frequently to facilitate feeding prior to the breeding season.

In 1990, one of the two females bred became gravid. The gravid female weighed 120 gm prior to breeding and 165 g at pre-egg laying shed. Three weeks post breeding she refused all food and after 45 days, she shed. Ten days post shed she laid 7 eggs which were incubated at 28°C in vermiculite. After 62 days of incubation, all 7 eggs hatched within a 24 hour period.

After hatching, the babies were separated into their own separate shoe boxes with a sand substrate, shelter, and a water dish provided. No food was offered until shedding was complete. Only one neonate ate a pinkie when first offered. The others were induced to eat by lizard scenting procedures similar to those used on grey banded kingsnakes. These hatchlings seemed only slightly easier to start feeding than grey banded hatchlings.

There was marked disparity in the amount and location of red on the head and the shape and amount of white in the band at the back of the head. The triads varied tremendously as well, ranging from a low of 38 to a high of 58 triads. Obviously, there is more red in the low triad animals.

From previous observations, the author feels that most of these snakes need to be approximately 3 years old before being bred. While early growth is rapid, the hatchlings, like the adults, seem to have a short feeding season and quit abruptly near early fall.

DISEASES AND MALADIES

The author feels that he has noted three main problems including parasitism, enteritis, and regurgitation. The common denominator is involvement with the gastrointestinal tract.

Regurgitation was an initial problem seen not only in my colony but also in some colonies of my clients. The most common cause of regurgitation is related to environmental temperatures. As stated before, these snakes seem to be fairly tolerant of cooler temperature, however, they do not seem to thrive at higher temperatures. Cage temperatures higher than 27°C resulted in frequent regurgitation. On the other hand, holding them at temperatures consistently below 19°C resulted in regurgitation, possibly a result from improper digestion. The best results were obtained when heated to 27°C maximum during the day, and dropped to around 20°C at night. Obviously, a heat gradient in the cage is useful for thermoregulation.

Over-eating is another common cause of regurgitation in some individuals, and is easily remedied by smaller and more frequent meals. Additional reasons for regurgitation were gastroenteritis and parasitism, usually bacterial or protozoan in nature. The bacterial forms were accompanied by mucoid stools, anorexia, and listlessness in addition to regurgitation. Simple changes in temperature and feeding patterns did not remedy these more complex cases. Cloacal cultures resulted in gram negative bacteria as would be expected, however, *Pseudomonas* was not identified.

Instead, cultures revealed *Aerobacter* and *Proteus* species. Treatment was initiated with either one of the following drugs:

1) Trimethoprim-Sulfa (Tribrissen) elixir: 15 mg/lb. once daily x 3 days orally

2) Chloromycetin palmitate: 20 mg/lb. once daily x 3 days orally

Protozoal gastroenteritis presents much the same as bacterial, but fecal examinations reveal motile trichomonads. I feel these protozoans are normal flora in small quantities, but in large numbers cause a clinical gastroenteritis. *Giardia* was never identified. Treatment consists of oral metronidazole (flagyl) at 25 mg/lb given on days 1, 3, and 10. In either type of enteritis, it is also very important not to feed during treatment, and when starting back on food to go very slowly with small meals.

Two common intestinal worms have been found; *Strongyloides* and *Kalicepilus* (hookworms). Both of these parasites have direct life cycles which means that they can build up in the captive environment. Both parasites produce larvae that can penetrate skin, enabling these parasites to increase to alarming numbers in captive specimens. Both parasites have responded well to treatment with fenbendazole (Panacur) at 10 mg/lb given on days 1, 3, 14. Repeat fecal examinations should be performed to verify successful treatment. As with the bacterial and protozoal gastroenteritis, food should be withheld while treating and then restarted very slowly.

LITERATURE CITED

Applegate, Robert. 1984. Feeding Baby Snakes (rodent eating species). Handout from author.

Merker, G. and Mackin, B. 1987. Herpetat Modular Cages: Innovative Snake Housing. IN: Gowen R.L. (Ed) Proceedings of the Northern California Herpetological Society's Conference on Captive Propagation and Husbandry of Reptiles and Amphibians. pp. 137-145

CAPTIVE HUSBANDRY AND REPRODUCTION OF THE BLOOD PYTHON AT THE ST. LOUIS ZOOLOGICAL PARK

Jeff Ettling

St. Louis Zoological Park
Forest Park
St. Louis, MO 63110

INTRODUCTION

The blood python (*Python curtus*) is a brightly colored, moderate size (1.5 - 2.0 m) python native to the central and southern Malay Peninsula, Sumatra, Borneo, Sarawak, Brunei and Sabah (Mehrtens, 1987). Three subspecies of blood python are currently recognized; *P. c. curtus*, *P. c. breitensteini* and *P. c. brongersmai* (Welch, 1988). The preferred habitat is aquatic rainforest areas such as swamps, marshes and streams (Mehrtens, 1987). They lie in vegetation or bury themselves in the mud near the water's edge with only the head exposed waiting for their favorite prey of birds or rodents (Opferman, 1987). The geographic range of *P. curtus* is near the equator, where environmental conditions such as temperature and humidity are fairly constant. The temperature averages 27°C during the day and 22-24°C at night. Seasonal temperatures only fluctuate 1-2°C (Opferman, 1987). Due to high rainfall, the humidity usually remains above 80% and rarely falls below 70% during the dry season. The photoperiod throughout the year is 12L:12D. The coolest part of the year corresponds to the monsoons of November and December (Opferman, 1987).

Sixty six (21 males, 26 females and 19 individuals of unknown sex) *P. curtus* were reported to be held in North American collections as of January 1, 1990 (Slavens, 1990). Bowler (1977) reported a longevity record of 27 years 9 months for a female *P. c. curtus*. Information regarding the captive reproductive biology of *P. curtus* is scanty. Ross (1978) reported on mating, egg laying, incubation and hatching based on information he had compiled from amateur and professional herpetoculturists. The most inclusive account of captive reproduction is by Opferman (1987), who reported temperature cycling, breeding, incubation, and hatchling care. Ross and Marzec (1990) analyzed data submitted by zoos and private breeders in an attempt to determine the husbandry practices necessary for successful reproduction. Three zoological institutions and one private breeder reported reproduction during 1989 (Slavens, 1990).

MATERIALS AND METHODS

The St. Louis Zoo received a trio (2.1) of adult *P. curtus*, from a local herpetoculturist, on August 12, 1987. These snakes were long term captives with no reproductive history. The "tan" male and the female were imported in 1972 and 1983, respectfully. The "red" male was captive bred at the Catoctin Mountain Zoological Park, Thurmont, MD in 1979.

The snakes are housed together on exhibit in a concrete block enclosure measuring 213 x 198 x 226 cm. Logs, bark slabs, a ficus tree and a water bowl are provided on a shredded-bark mulch substrate. Four 40 watt Vita-Lite® full spectrum fluorescent tubes and three 150 watt Plant-Gro® incandescent bulbs provide a photoperiod of 12L:12D. Skylights provide natural photoperiod for the St. Louis, Missouri area. Ambient temperature varies from 23.8-

30°C. The humidity for the tropical rainforest biome section varies from 33-57%, but is higher within the cage due to frequent mistings and moisture retention by the shredded-bark mulch. The snakes are fed 1-2 freshly killed laboratory rats weekly, biweekly, or monthly depending on the body weight of the individual snake.

In 1988 and 1989, a triple beam balance and a vernier caliper were used to weigh and measure the eggs and hatchlings (to the nearest 0.1 g and 1.0 mm, respectively). In all three years the eggs were placed in plastic sweater boxes (41 x 28 x 18 cm), containing vermiculite as an incubation substrate at a water:vermiculite ratio of 1:1 (w:w). The boxes were covered and placed in a human infant incubator in 1988 and 1989. In 1990, an aquarium with a submersible aquarium heater was used to incubate the eggs due to temperature malfunction of the human infant incubator. Temperatures in both incubators were kept at 31-32°C with relative humidity of 100% throughout incubation. The major difference is that heavy condensation is produced within the "aquarium" incubator.

Each hatchling was housed individually in plastic shoe boxes (32 x 18 x 10 cm) provided with a newspaper substrate and a small water bowl. Holes were drilled around the perimeter of the boxes to provide ventilation. Most neonates began feeding on fuzzy mice between three and four weeks of age. Neonates which had not eaten by the seventh or eighth week were assist fed. The neonate was lightly held behind the head while a dead fuzzy mouse was placed in its mouth. Once the mouse had made contact with the mouth, the neonate would usually bite down and attempt to constrict it. Some neonates had to be assist fed for several weeks before they would feed on their own.

OBSERVATIONS AND REPRODUCTION

The female was introduced into the exhibit with the "red" male on March 16, 1988. Copulation was observed for the first time on the morning of March 20, 1988 at 0800 hours. Copulation was also noted on the mornings of March 22, 26, and 28, 1988. Courtship was not observed.

A clutch of sixteen adherent eggs (total mass = 1914.3 g) and one nonadhesive egg (mass = 103.1 g) were laid on July 2, 1988. Table 1 summarizes data from this clutch and two others laid by *P. curtus* at the St. Louis Zoological Park. Fifteen days after egg deposition four of the adherent eggs proved to be infertile, as was evident by discoloration and fungal growth. Eleven of thirteen eggs hatched 61-64 days later. On September 7, 1988 the remaining two eggs were cut open and found to contain fully developed dead embryos.

The "tan" male was introduced into the exhibit with the "red" pair on December 7, 1988. Intermittent copulation was noted from October 3, 1988 through January 21, 1989. A clutch of 12 adherent eggs and four nonadhesive eggs were laid on April 19-20, 1989. Fifteen snakes hatched 61-63 days after egg deposition.

Courtship and copulation were observed from November 20, 1989 through January 9, 1990. Courtship was noted for the first time on December 16, 1989. The "tan" male had his body aligned with that of the female and was prodding her with his spurs. Tactile - alignment (Gillingham et al, 1977) was also observed. Seventeen eggs and one infertile egg mass were laid on March 7, 1990. Egg measurements were not recorded. None of the eggs had hatched 75-80 days following egg deposition, so the eggs were cut open and found to contain fully developed dead embryos.

It has not been determined whether the excess moisture produced by the "aquarium" incubator had a detrimental effect on the eggs.

The Fort Worth Zoological Park, Fort Worth, Texas hatched *P. curtus* in 1987 and 1989. Dave Blody, Curator of Reptiles, graciously provided reproduction data from the two clutches. Table 1 summarizes data from two clutches of *P. curtus* eggs laid at the Ft. Worth Zoological park.

The Fort Worth Zoo acquired a captive bred female on June 17, 1987. Fourteen days later on July 1, she laid eight adherent eggs. The eggs were incubated in a vermiculite:water ratio of 1:0.75 at 31-32°C. Four eggs hatched 70-72 days after deposition.

The Fort Worth female laid thirteen eggs on June 20, 1989. No egg measurements were recorded. Eleven eggs hatched 67-69 days later.

Table 1. Average egg and hatchling measurements for four clutches of *P. curtus* eggs. The weights were measured in grams and the lengths and widths were measured in millimeters.

Clutch	Date laid	# eggs	Eggs			Hatchlings		
			weight	length	width	#	weight	length
STL[1] #1	7-2-88	17	118.7	75.7[2]	53.3[2]	11	78.7	410.9
STL #2	4-19,20-89	16	130.8	81.5[3]	52.2[3]	15	90.2	412.0
STL #3	3-7-90	18	----	----	----	0	----	----
FTW[4] #1	7-1-87	8	146.3	----	----	4	110.6	452.0
FTW #2	6-20-89	13	----	----	----	11	84.3	400.1

[1] STL = St. Louis Zoological Park female.
[2] n = 10
[3] n = 4
[4] FTW = Fort Worth Zoological Park female.

By comparing the reproduction data from the St. Louis clutches with Fort Worth's clutches, several differences can be noted. Although both institutions incubated their eggs on a vermiculite substrate at ca. 32°C, the ratio of vermiculite:water was slightly different. St. Louis used a 1:1 (w:w) ratio whereas Fort Worth used a 1:0.75 (w:w) ratio. The 1987 (FTW) clutch is significant with regard to the incubation time and hatchling size. Hatching usually takes place after an incubation period of 58-65 days (Ross et al, 1990). Fort Worth had four eggs hatch after 70-72 days of incubation. The mean measurements for the 1987 (FTW) hatchlings were larger (+26.2 g, +44.4 mm) than the combined mean measurements of the 1988 (STL) and 1989 (STL, FTW) hatchlings.

CONCLUSIONS

P. curtus will thrive and reproduce in captivity if specific conditions are provided. In the wild, these snakes inhabit aquatic environments with warm temperatures. Therefore, it is essential to provide them with temperatures of 24-30°C and a relative humidity above 60%. With favorable environmental conditions, minimal stress and proper nutrition reproduction should occur. Based on my temperature data as well as that of others (Opferman, 1987; Ross et al, 1990), only minimal temperature reduction is needed to induce mating. Standard techniques used for egg incubation and hatchling care of other pythons works well for *P. curtus*.

LITERATURE CITED

Bowler, K.J. 1977. Longevity of reptiles and amphibians in North American collections as of 1 November 1975. S.S.A.R. Herp. Circular. 6:32.

Gillingham, J.C., Carpenter, C., Brecke, B.J. and Murphy., J.B. 1977. Courtship and copulatory behavior of the Mexican milksnake *Lampropeltis triangulum sinaloae* (Colubridae). Southwest Nat. 22:L 185-194.

Mehrtens, J.M. 1987. Living Snakes of the World in Color. Sterling Publishing Co., Inc. New York.

Opferman, R. 1987. Observations on the maintenance and reproduction of *Python curtus*. Litteratura Serpentium 7(3):104-113.

Ross, R.A. 1978. The Python Breeding Manual. Institute for Herpetological Research. Stanford, CA.

------------ and Marzec, G. 1990. The Reproductive Husbandry of Pythons and Boas. Institute for Herpetological Research. Stanford, CA.

Slavens, F. 1990. Inventory of Live Reptiles and Amphibians in Captivity, Current January 1, 1990. Slavens, Seattle, WA.

Welch, K.R.G. 1988. Snakes of the Orient: A Checklist. Krieger Publishing Co.

ACKNOWLEDGEMENTS

I would like to thank Ron Goellner and Ann Day for reviewing the manuscript and for their help with taking slides and collection of temperature data. I also thank Dave Blody for supplying reproductive data on *Python curtus* hatched at the Fort Worth Zoo.

PRODUCTS MENTIONED IN TEXT

Vita-Lite® full spectrum fluorescent tube, Durotest Co., Fairfield, NJ 07007.

Plant-Gro® bulbs, Philips Lighting Co., Somerset, NJ 08873.

MANAGING AND BREEDING TORTOISES IN CAPTIVITY

Sean McKeown

Curator of Reptiles
Chaffee Zoological Gardens of Fresno
894 West Belmont Avenue
Fresno, CA 93728-2891

INTRODUCTION

Land tortoises (family Testudinidae) are a diverse group of chelonians with living requirements that vary greatly on a species by species level. This paper will highlight techniques for the proper management of taxa from Africa, Indian Ocean islands, Southern Europe, South America, and Southeast Asia. Information regarding proper facilities, quarantine procedures, diet and dietary supplementation, methods of minimizing stress, parameters for encouraging courtship and mating, egg incubation, and care and nutritional requirements of offspring will all be presented. Representative species will be highlighted to show the diverse management techniques that are advisable.

OVERVIEW OF COLLECTION TECHNIQUES

Tortoises that have been collected from the wild are typically put through tremendous stress. Most often they are captured by people with little knowledge of their basic needs. Often they are held for long periods of time in small pens in extremely high numbers. They are seldom fed or watered, receive no vitamin/mineral supplements, and rarely, if ever, have their enclosures cleaned. Their journey to the regional wholesalers may take several days under less than ideal conditions. After weeks or months at the wholesaler, where they may be exposed to large amounts of bacterial and respiratory diseases, and to fecal material containing parasites, the luckier ones may be shipped to wholesalers in a developed country. At least here they will be sold for pets instead of for the stew pot. Even at these wholesalers, though, they may be overcrowded, kept at improper temperatures, housed on inappropriate substrate, fed an unbalanced diet, and exposed to additional diseases. It is essential, therefore, to quarantine any new tortoise coming into your personal collection. Even if the specimen has been captive bred, it or its parents may have been exposed to other captive tortoises that have a contagious disease or internal parasites.

SELECTION PROCESS

Whenever possible, obtain tortoises directly from breeders and, if possible, examine their facilities and animals personally. Select only species that are well suited to the climate where you live. Do not obtain tortoises unless you are prepared to house them outdoors during major portions of the year when the weather and climate are suitable. Remember that tortoises are highly susceptible to pesticides, herbicides, snail poisons, and toxic plants. Tortoises are also at great risk when maintained near unfenced swimming pools and dogs.

Before purchasing a tortoise, observe it very closely. If its nose is at all moist or has any discharge, pass on that tortoise and any other tortoises at that place of business or facility as the tortoise most likely has a deadly, highly contagious, and probably incurable respiratory infection. Also pass on any tortoises that have swollen eye lids, that do not have adequate weight for their size, that are not alert, and that will not readily feed in front of you.

The final consideration is to remember that tortoises are vertebrate organisms that typically have a potential life span longer than man. I say potential because they will perish rather quickly, unless they receive the correct care. Nothing could be further from the truth than the general belief that tortoises are easy animals to care for. In this time of severe deforestation and habitat destruction throughout the world, it is critical that you are committed to properly care for any of these chelonians you obtain. Limit the number of species in your collection and concentrate on setting up viable breeding groups of the taxa you are working with. Identify each of your individual tortoises and keep detailed records on each animal.

QUARANTINE PROCEDURES

Quarantine any newly obtained tortoise in an entirely separate area from the rest of your collection for at least six weeks. Check them daily for even the slightest sign of fluid or bubbles coming from the nose. Fluid coming from the nostrils most likely indicates that this tortoise has a severe, highly contagious respiratory disease comparable to what in humans might be termed acute pneumonia. Do not keep any tortoise that shows any signs of a respiratory infection or that has had any past history of respiratory problems as it may be a carrier. Keeping such an animal could lead to the death of every other tortoise in your collection.

Most other health problems that might be encountered during the quarantine period can be cured or eliminated if properly detected and treated. Pre-arrange with a veterinarian who regularly works with reptiles to analyze a fresh fecal sample. If internal parasites are found, work closely with the veterinarian to treat and eliminate these parasites.

Even though the tortoise is in quarantine, make sure that its enclosure is not bare and that the enclosure is relatively large. There should be ultra-violet light fixtures overhead, and the temperature in the enclosure will probably need to be 26°-30°C. Include a shallow water dish that the tortoise can both drink out of and soak in. There also must be areas that afford cover. The broadest range of food, including chopped and grated vegetables, fruits, and moistened dog chow must be offered daily during the quarantine period. Observe which foods are actively eaten and provide suitable amounts of those foods with a vitamin/mineral powder lightly sprinkled on top. The primary goal during the quarantine period is to expose the tortoise to as little stress as possible, while allowing the tortoise to fully adjust to its new environment.

HOUSING

Have a suitable outside area ready for your tortoises before they come out of quarantine. Generally, either individual species or closely related species from the same geographic area should be kept together. Tortoises should be kept on grass and they should have access to both sunny and shady areas. Flowerbeds and other places to hide need to be available. Species that live in forests will also require large areas to soak, preferably in mud bottomed pools. Desert species may need areas suitable for digging deep burrows.

When the weather is not suitable for living outdoors, tortoises must either be deep in their burrows or brought indoors. *Gopherus* and *Xerobates* taxa should be brought in at night if they emerge early in the spring to minimize the chance of enteritis. These North American taxa can brumate during the winter in a box kept in a closet or in the garage away from washer/dryers. <u>All non-desert species</u> will need to be provided with lukewarm water to drink at two to three week intervals during brumation. Tropical species should not be hibernated and should be maintained above 26°C indoors on either alfalfa, newspaper, or rabbit pellets. Do not use a substrate like gravel or sand which can be accidently ingested and can clog up the intestinal tract. Corn cob products are also not recommended.

DIET FOR ADULTS

In addition to grazing on natural grasses outdoors, adult tortoises can be supplementally fed every other day with higher grades of lettuce, spinach, kale and other cabbage family plants, alfalfa, green beans, squash, grated carrots, other vegetables, hibiscus flowers, and prickly pear cactus. Fruits may be given regularly to more specialized taxa and once or twice a week to more generalized species. Small amounts of dog chow or primate chow may also be given once or twice a week to fully adult tortoises.

DIET FOR JUVENILES

Neonates, juveniles, and sub-adults require a very different nutritional regime. The nutrition of the neonate will play a key role in proper or improper shell growth. Tortoises are designed by nature to grow very slowly and not to reach sexual maturity for many years.

If the daytime temperatures are between 21° and 32°C, the young tortoises should be allowed to graze out side in an area offering both sun and shade, and which is enclosed by shade cloth, screen, or hardware wire to prevent predation by birds. When the weather is not suitable outdoors, young tortoises should be housed inside on newspaper or rabbit pellets. Small juveniles are especially prone to ingesting items such as sand, gravel, or bird seed which can lead to intestinal blockages, infection, and death. Juvenile tortoises should be soaked in plastic containers in shallow luke-warm water twice a week for about 30 minutes. Several drops of vitamins may be added to the water if desired. Use separate soaking containers for each species.

When housed indoors, juvenile tortoises may be offered romaine, butter lettuce, spinach, and leaves of alfalfa daily or every other day. Twice a week a variety of grated and chopped vegetables, hibiscus flowers, and small amounts of papaya and other fruits should be added to the feeding. Avoid feeding dog chow or primate chow to juveniles as they will grow too fast and their shells will pyramid, giving them an unnatural appearance and later making successful courtship and mating less likely.

DIETARY SUPPLEMENTS

The food for adults may be lightly sprinkled with a powdered vitamin/mineral supplement on a routine basis during feedings. In the case of neonates, juveniles, and sub-adults, too much vitamin/mineral supplementation can contribute to shell pyramiding. For this reason, vitamin/mineral supplements should be lightly sprinkled on the food only once or twice a week when feeding non-adult tortoises.

PREVENTATIVE MEDICINE

Good husbandry techniques and daily inspections of each tortoise will go a long way towards keeping your animals healthy. Work to recognize any potential problems when the problem first begins. Routinely weigh your tortoises. Quarantine any tortoise that is unusually light. Observe this animal and offer it extra food. Quarantine any tortoise that becomes lethargic or begins to rub its eyes with a foot. Periodically monitor feces for signs of heavy internal parasites.

Construct soaking pools for large tortoises that are mud bottomed to prevent injury to soft body parts by concrete edges as the tortoise enters and leaves the pool. Do not keep lava and other sharp rocks, or heavily spined cacti in the enclosures. Alert your immediate neighbors to your pets and be sure that they do not use pesticides and poisons along shared fences. If your neighbor has a dog that shows unusual interest in your tortoises by barking or digging along the fence, you must have the common sense to either move the tortoise or put in a one foot deep section of concrete under the fence to keep the dog from digging under.

Tortoises should be outdoors only during suitable weather periods. Weather conditions need to be strictly monitored for unexpected changes. If in doubt, bring your tortoises into a protected area.

COURTSHIP AND BREEDING

Whenever possible go to your local university library and review the literature on the species of tortoise you are working with. Bring some dimes and nickels and photocopy any useful pages covering natural history in the wild and care in captivity for that species. Publications of special interest to you are Pritchard, Encyclopedia of Turtles; Ernst and Barbour, Turtles of the World; IUCN The Conservation Biology of Tortoises; Olney and Ellis, ed. International Zoo Yearbook 28 (Reptiles), and Beaman, Caporaso, McKeown, and Graft, eds. Proceedings of the First International Symposium on Turtles and Tortoises: Conservation and Captive Husbandry.

Determine how much area a given species of tortoise needs for proper courtship and mating to be successful. How elaborate is the courtship for that species? Does it involve chasing or circling behavior? Some more generalized species of tortoises do not need to be separated during the year and will mate successfully. Southern European tortoises (*Testudo sp.*) and North African *Geochelone sulcata*, New World rainforest red-footed tortoises (*Geochelone carbonaria*), and yellow-legged tortoises (*Geochelone denticulata*) are examples of species that do not need to be separated. Other taxa including Galapagos tortoises (*Geochelone elephantopus*) and Aldabra giant tortoises (*Geochelone gigantea*) may breed if left together all year, but may copulate with greater efficiency if separated during the winter months and at other intervals.

Many male tortoises seem especially stimulated to mate if they are exposed to a less dominate male for a relatively short time, then put in with a female. Under these conditions the more dominant male may be more apt to go for actual penetration instead of only mounting. The latter behavior is seen so commonly and does not necessarily lead to successful copulation.

Environmental cues should be closely monitored. A majority of tortoises are more interested in mating during periods of weather change such as spring or fall, overcast days, or light rain. Sometimes light rain can be simulated to bring about completion of a courtship sequence that would otherwise prematurely end.

In a number of species including but not limited to radiated tortoises (*Geochelone radiata*), Asian forest tortoises (*Manouria emys*) and red-footed tortoises (*Geochelone carbonaria*), females may occasionally court and mount other females, even going so far as to rock forward and vocalize while mounting. Whether this indicates the female may be ready to breed or whether these activities are in the "vicarious thrill" range behavior, we simply do not know. We can conclude, though, that mounting alone in tortoises does not always indicate copulation. Adult tortoises can almost always be correctly sexed by looking at the degree of concavity of the plastron, the combined length and width of the tail, and the size and shape of the anal scutes.

NESTING AND EGG LAYING

Prior to oviposition, a female tortoise may become more active than usual. She may pace parameter fences or generally roam around the yard occasionally stopping to sniff the ground for temperature and humidity cues. Sometimes a female may even attempt to push out a particular section of fencing. Nesting behavior may start several days before oviposition or may begin that very day. Females may partially dig shallow nest holes or even proper flask-shaped nest holes and cover them up. This behavior may reflect testing for proper incubation conditions or it may be done to confuse egg predators. False nest hole digging varies both by species and individual females.

When a female is finally ready to lay she will typically dig a flask-shaped hole with her rear feet, alternating with one, then the other. She will usually urinate on the spot she is prepared to dig out to loosen the soil and facilitate the digging process. Some desert species, such as a desert tortoise (*Gopherus agassizii*), may lay eggs near the mouth of her burrow. Some forest species may dig nest holes in leaf litter. One taxon, the Asian forest tortoise (*Manouria emys*) actually collects leaf litter from as far as forty feet away and constructs a large mound of earth and leaf litter debris through backwards sweeping motions with her front feet. Once the eggs are laid she guards the nest from predators for up to a week or more.

It is important not to interrupt or disturb a female tortoise during her search for a nesting spot or while she is digging. Such actions may cause her to abandon the nesting procedure and/or may even cause her to go into a state of depression.

Once a female tortoise has finished digging out a nest hole she may or may not lay eggs. Many female tortoises in captivity do not lay, or only rarely lay. The reason for this is unknown and there is no information on whether comparable numbers of females exhibit similar behavior in the wild. If a captive female does in fact lay eggs, she will position herself over the nest hole so that each egg drops into the opening. With great dexterity she positions the eggs using her rear feet at the base of the flask-shaped nest hole, thereby minimizing any chance for breakage. The first eggs have the longest drop from her cloaca to the base of the nest hole and rarely hit anything but soft earth. When a female tortoise begins to lay, she typically goes into a trance-like state. At this point, it does not disturb her to quietly move in from behind, reach under the turtle's cloaca, and gently catch each egg as it is dropped into your hand. After the female has completed the process, move unobtrusively away and allow her to fill in the nest hole. Her rear feet will alternately move earth into the cavity and she may use her plastron to pack the spot down. Some females even use their rear feet to break off grass and pack it on top, thereby concealing the nest and making it extremely difficult to visually detect.

Artificially incubating tortoise eggs will guarantee the highest hatch rate. Tortoise eggs should be marked at the top with a lead pencil to indicate the position in which each was laid. The eggs are best incubated in vermiculite or a vermiculite/potting soil mix. The incubating medium should be briefly soaked in luke-warm water, then squeezed out to a consistency of two parts vermiculite to one part water by weight for more xeric species. For tropical taxa which require more moisture, mix one part vermiculite with one part water by weight. Plastic shoe boxes with tiny holes drilled in the top and one inch of moistened vermiculite in the bottom make excellent egg containers. Add as many eggs as will comfortably fit in the container without touching and add additional moistened vermiculite until just a nickel-sized portion at the top of each egg is visible. Secure the lid, label each container as to species, the laying female, number of eggs incubating and date the eggs were laid. Place the plastic container in the incubator and adjust the temperature for the species; 25-29°C for forest species and 27-30°C for other taxa. For the species studied, tortoise eggs have been shown to be sexually determined by temperature. Higher temperatures typically produce females and lower temperatures yield males. Temperatures which are too high or two low so that they are just at the edge of the incubatable range may produce some live individuals, but typically with major shell deformities.

The eggs should be checked twice a week for the first two months and daily after that period. Eggs may be taken out and candled to check for development from time to time without harm as long as the top portion always remains in the same position. If individual eggs smell foul and appear to be rotting, remove them. Always open bad eggs to determine if they were simply infertile or whether there is some development. If development is present, preserve the embryo in alcohol for rare species, and attempt to determine if there is anything in your incubation regime that might be responsible for the dead embryos. From time to time small amounts of lukewarm water will need to be added to the vermiculite along inside edges of the egg containers to maintain moist conditions. The exact degree of moisture is not generally critical in tortoise eggs, as long as the eggs are not kept too wet.

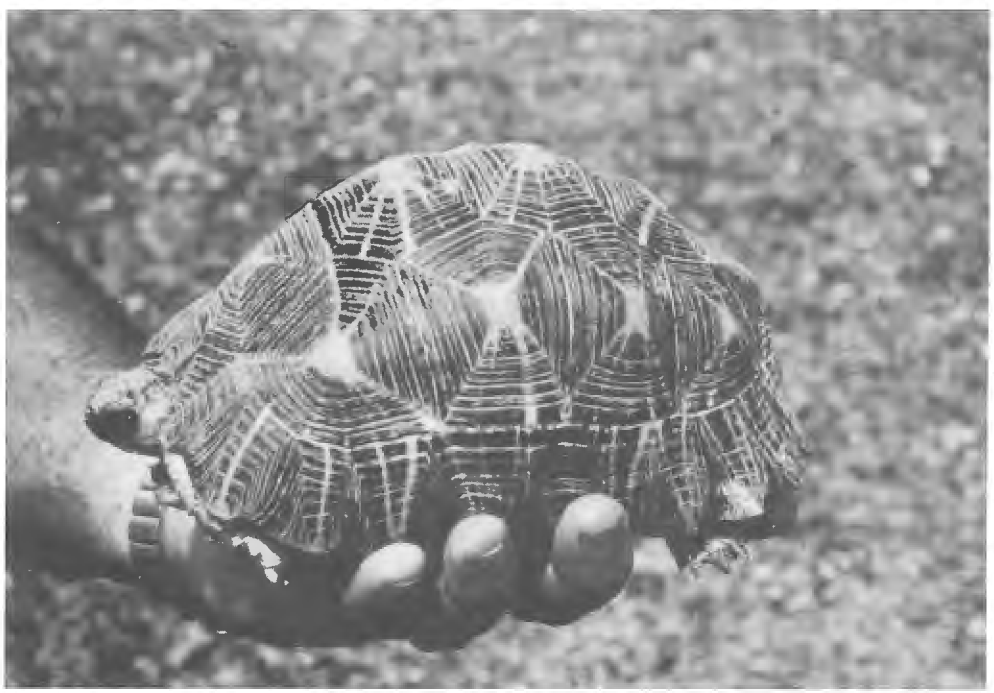
In geographic areas with wet/dry seasons, tortoise eggs may develop more slowly during the drier months and more rapidly during the wet months so that the young hatch when there is an abundant food supply. In some cases, tortoises that hatch before the wet season may go days, weeks or months waiting for the rains to soften the top plug of earth at the top of the nest hole before they are able to make their way to the surface.

Eggs artificially incubated at cooler temperatures generally incubate more slowly than eggs incubated at warm temperatures. When incubated under the regime outlined, eggs from most taxa hatch in three to five months, although in several species the time can be as short as just over two months. In species I have managed, the eggs all hatched the same day or within several days of each other. Several colleagues have told me that occasionally they get one or more eggs that hatch weeks or even months after all the others. I do not know the evolutionary significance of this or whether such an individual or individuals would still have the correct temperature and moisture regime to successfully hatch in the wild.

CONCLUSION

The material presented represents an overview of tortoise management and breeding. Due to the broad scope of the paper, each topic was covered in its most generalized sense. Nevertheless, it is my hope that even such a rudimentary work will in some way benefit those who keep tortoises, and ultimately, the tortoises themselves.

In closing I would like to again emphasize that tortoises both domestically and around the world are under tremendous pressures from mankind. Many are eaten, others are killed merely because they exist. As human populations continue to burgeon and as more wild areas are destroyed, life for tortoises will become even more precarious. We benefit from the wise management of captive populations.



A large juvenile radiated tortoise (*Geochelone radiata*) with excellent shell conformity from having been fed a proper captive diet. Photo by Sean McKeown.

SUMMARY OF WORKSHOP ON EGG INCUBATION

Edited by Richard Staub

Workshop Facilitators
Dave Blody, Curator of Herpetology, Fort Worth Zoo
Vince Scheidt, Private Breeder
Gary Ferguson, Professor, Texas Christian University
David Grow, Curator of Herpetology, Oklahoma City Zoological Park

CHAMELEON EGGS

Gary Ferguson incubated panther chameleon (*Chamaeleo pardalis*) eggs at temperatures ranging from 20-30°C and in a water/vermiculite substrate ranging from 0.4-2 parts water to 1 part vermiculite. Best success was found at 25-28°C with less than 1 part water to 1 part vermiculite. Panther chameleon eggs generally go through a 3 month resting phase, then begin to absorb water and develop. Some eggs will absorb a tremendous amount of water and become very large. These eggs have never hatched. At higher incubation temperatures, the length of the resting phase is increased. At higher water ratios, the eggs absorb too much water, and some even burst.

Sometimes the eggs hatch in unison after a light misting. David Grow has experienced this phenomenon with other species at the Oklahoma City Zoological Park. This might be an adaptation where the eggs are synchronized to hatch with the beginning of the rainy season.

FULLY FORMED EMBRYOS THAT DIE IN THE EGG

Three hypotheses were offered as possible causes for embryos that go to term, but die shortly before they should hatch.

1. Poor nutrition in the female while she was gravid could lead to weakened embryos which are not strong enough to cut their way out of the egg. Better husbandry of the female's nutritional requirements might correct this problem.

2. As the developing embryos approach their hatching date, their rate of metabolism increases. If air circulation within the incubation container is insufficient, a layer of carbon dioxide can form and cover the eggs. Carbon dioxide gas is heavier than air, so it could effectively blanket the eggs and suffocate them. Increasing air circulation around the eggs, especially late in the term, will provide the eggs with the oxygen they require.

3. Herpetoculturists incubate their eggs with more available moisture than eggs incubated in the wild. Soft-shelled eggs incubated with excess moisture will swell until the shell becomes pressurized. If the temperature is now increased, the egg shell can no longer dispel excess water or effectively transport oxygen. Therefore, the embryo essentially suffocates. Allowing the eggs to dry out slightly, especially as they get close to term, should permit them to properly breath.

Many opinions were offered about which incubation substrate was preferred. Vermiculite was commonly used, but both there was no agreement as to whether fine grade or course grade worked better. In Europe, perlite is the more common substrate. They believe the larger particle size increases the gas exchange around the eggs.

Phelsuma EGGS

Phelsuma species commonly lay clutches of two eggs; however, one of these eggs frequently fails to hatch. David Grow has had better success at getting both eggs to hatch if they are removed from the enclosure and incubated artificially. This is often difficult, though, as *Phelsuma* will firmly affix their eggs to the wall of the enclosure. Taping a thin layer of plastic to the upper 3-4 inches of the walls often helps. If the eggs are attached to the plastic, it can easily be cut and removed with the attached eggs.

PROVIDING A HOT SPOT FOR PREGNANT BOAS

Pregnant boas will often seek a basking spot where the temperature exceed 95°F. If sufficient heat is not supplied, litters containing stillborns or neonates with kinked spines are common. Ron Tremper has an old female Baja rosy boa (*Lichanura trivirgata*) that sits on a 118°F hot spot for 4 hours a day, and always produces healthy young. Dave Blody pointed out that simply providing a hot spot may be insufficient if the animal refuses to use it. Snakes will sometimes choose security over a preferred temperature. A hot spot should provide adequate security for the animal and it is the herpetoculturist's responsibility to make sure the animal is thermoregulating properly.

SUMMARY OF WORKSHOP ON THE ROLE OF THE AMATEUR HERPETOLOGIST

Edited by Richard Staub

Workshop Facilitators
Sean McKeown, Curator of Reptiles, Chaffee Zoological Gardens of Fresno
Bob Johnson, Curator of Amphibians and Reptiles, Metro Toronto Zoo
Robert Sprackland, Department of Herpetology, California Academy of Sciences
Brett Stearns, California Academy of Sciences

PROFESSIONAL VERSUS PRIVATE

The herpetological community is currently separated into the professional (zoos and universities) and private parties. The professionals are fearful of the profit-seeking, inscrupulous collectors, while the private herpetoculturists feel the professionals are snobs who do not want to share their animals. The inscrupulous collectors and professional snobs probably comprise less than 5% of the herpetological community; however, this 5%, along with very restrictive laws, is effectively polarizing the parties. The large number of threatened species and the scope of habitat destruction makes it imperative that the private and professional parties join to share their knowledge and resources. The level of interaction between zoos and private individuals varies from zoo to zoo. The AAZPA is slowly beginning to involve more private individuals, especially with stud books on species survival plan (SSP) animals.

RECORDS

The most important role the private herpetoculturist can provide is data collection. Private herpetoculturists need to record all their observations and then publish the data so it can enter the scientific community. Private individuals throughout history have been at the leading edge of science. Just by sheer numbers, private herpetoculturists will make more useful observations than their professional counterparts. These observations, though, need to be recorded and published to have value.

Private individuals are encouraged to stay with a set number of species for many years. Do not constantly change which animals you keep. Long term commitment to a species will give your observations credibility. Anecdotal observations on a few animals for a couple years have less value.

Each regional society should have a coordinator responsible for data collection. Each society should have a standardized form for recording roadkills and animals observed in the wild. This data needs to be tabulated and presented to the scientific community. With the current worldwide amphibian decline, this type of survey data is invaluable. The regional societies represent a wealth of untapped resource. Often, all they require is to be asked by a university or zoo to help or give assistance on a project.

Private individuals need to become more involved with conservation. The animals you keep are not separate from their habitat. An individual who is truely dedicated to the animals he keeps will also want to have that animal protected in the wild. Speak up and voice your concerns about the habitat and its destruction.

PRESERVING ANIMALS FOR SPECIMENS

A responsible scientist will remove a representative sample of a population for preserving. These preserved animals often are not studied for many years, but are eventually used to assess populations. New species are constantly being discovered from specimens preserved many years ago. Preserving samples has a high scientific value as long as it is done responsibly.

Museums are often funded by the number of preserved specimens they manage. This procedure needs to be changed so that museums do not unnecessarily preserve specimens just to gain more funding. Many museums are also moving toward loaning out the animals they collect. When the animal dies, it is then returned to the museum for preserving. Many museums will take your roadkill or deceased animals as long as you have sufficient records on the animal.

Every university and institution has an ethics committee or an animal care and use committee for investigating irresponsible research. These committees have a lot of power and should be notified of any wrong doing.