THE PREVALENCE OF GASTROINTESTINAL PARASITES IN A SMALL POPULATION OF CAPTIVE BOAS AND PYTHONS (FAMILY BOIDAE)

STEPHEN J. DIVERS

Elands Veterinary Clinic, Station Road, Sevenoaks, Kent, TN13 2XA
Tel. 01732 452333 Fax. 01732 741614

SUMMARY

This paper reports an investigation of the prevalence of helminth parasites in a population of 24 snakes belonging to the family, Boidae. The degree of parasitism was calculated using the McMaster method, and 10 different helminths were identified from the structure of the ova and the host species. Captive bred animals and wild animals that had received effective anthelmintic treatment were discovered to be free from helminth parasitism.

INTRODUCTION

Reptiles are becoming increasingly popular as "pets" and this is paralleled by increased activity of herpetological organisations arranging meetings and events. New publications, especially of a verterinary nature are also becoming available and reflect the increasing likelihood of the veterinary practitioner to be confronted by the reptilian patient. Despite the successful captive breeding of many species of reptiles within the United Kingdom, many are still imported from their country of origin. In particular, due to their docile nature and graceful appearance, the boas and pythons (family Boidae) are amongst the most celebrated and popular of all reptiles. Until recently the major limiting factor for maintaining reptiles in captivity was poor management, however, advances in our knowledge of reptile husbandry (Mattison, 1987; Mattison, 1989; Mattison: 1991), heating/lighting technology and reptilian medicine (Cooper and Jackson, 1981; Frye, 1991; Beynon, Lawton and Cooper, 1992) has greatly increased the health of captive specimens.

Captive breeding of many species is now commonplace with the mass importation of wild specimens no longer required. Nevertheless, due to the cheapness of imported stock the import trade in reptiles is still widespread. In an attempt to identify potential problems facing captive boas and pythons this study investigated the endoparasitic status of 24 captive bred and wild caught Boidae snakes.

MATERIALS AND METHODS

As is the case in domestic animals, the diagnosis of helminthiasis is made by the microscopic examination of fresh stools (Thienpont, Rochette and Vanparijs, 1986). In order to obtain faecal material from both captive bred and wild caught reptiles it was necessary to contact several herpetologists and reptile retailers for assistance.

The frequency of snake defecation varies with the size of the snake, size of the prey and the frequency of feeding, and therefore it was necessary to allocate a period of three weeks for the collection of stools from 24 snakes. Fresh stools were collected and securely stored in universal containers. Each container was labelled with the date of collection, and the owner and individual snake were identified by a unique alphanumeric code. This code was cross referenced to a record sheet which recorded

the date of stool collection, the species, age and history of the individual. In order to maintain sample viability for up to three weeks it was necessary to store all collected material at 4°C. All participants received an information sheet which described the methodology outlined above, together with universal containers, printed labels and a faeces collection record sheet.

Coprological examination was performed by the author at the Institute of Zoology's Pathology Department. In order to investigate a helminth burden the faeces were examined microscopically for parasite eggs, with the number of eggs considered proportional to the degree of parasitic burden. To obtain a more quantitiative estimate of infection, the number of helminth eggs per gram of faeces was calculated using the McMaster method (Thienpont *et al,* 1986). A saturated sucrose solution, with a density of 1.50 at 20°C, was used as the floatation medium. From each sample, 2.0g of faeces were suspended in 60ml saturated sucrose solution. To remove the hairs and other large particles of debris, the suspension was straned through a fine wire sieve and the residue pressed out. The suspension was stirred to obtain a completely homogenous mixture of any eggs that might be present. Using a Pasteur pipette, one compartment of the McMaster slide was filled, being careful to tilt the slide to allow any air bubbles to escape. The same operation was repeated to fill the second compartment. The slide was then left to stand for 5 minutes to allow any eggs to float up to the surface of the floatation medium and adhere to the cover glass. Under low or medium magnification any eggs present could be easily observed and readily counted. Each counting compartment contained 0.15ml of liquid and both compartments were counted for each sample. The number of eggs per gram of faeces (e.p.g.) was obtained by multiplying the average number of eggs counted from both compartments by 200.

Samples were also viewed under high power to facilitate identification and, when possible, photography.

RESULTS

All of the snakes examined were considered healthy by their owners and of the 24 faecal samples examined, 12 contained helminth eggs while the remaining 12 samples contained no eggs and were considered to be clear of any obvious helminth infection. This represents a prevalence of gastrointestinal helminth parasitism of 50% for the population of snakes in this study.

Of the snakes that failed to exhibit helminth eggs in fresh faeces, 9 (75%) were captive bred and the remaining 3 specimens (25%) were long term captives that had been treated with the anthelmintic, ivermectin, within the last twelve months. Of the snakes that were found to be harbouring helminth parasites, 10 specimens (83%) were recently imported or long term captives that had not been treated with any anthelmintics. The remaining 2 specimens (17%), an adult D'Alberts Python *(Liasis albertisii)* and an adult Brazilian Rainbow Boa *(Epicrates cenchria cenchria),* were considered by their owners to be captive bred.

Parasite identification was performed by the author on the basis of host species and the genus of helminth reported in the literature to parasitise a particular species of snake host (Kiel, 1975; Frank, 1981; Bernard, 1986; Frye, 1991). In addition to the recognised parasities an interesting pseudoparasite, probably of plant origin, was identified from the faeces of a Brazilian Rainbow Boa, *Epicrates cenchria cenchria* (Plate 1).

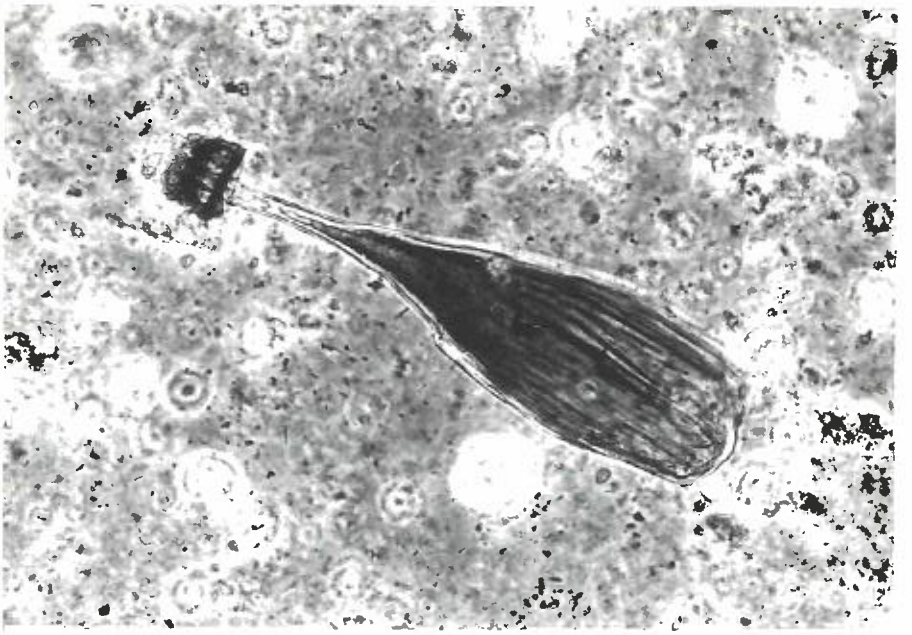

Plate 1. Pseudoparasite (probably of plant origin) from a Brazilian Rainbow Boa *(Epicrates cenchria cenchria)*

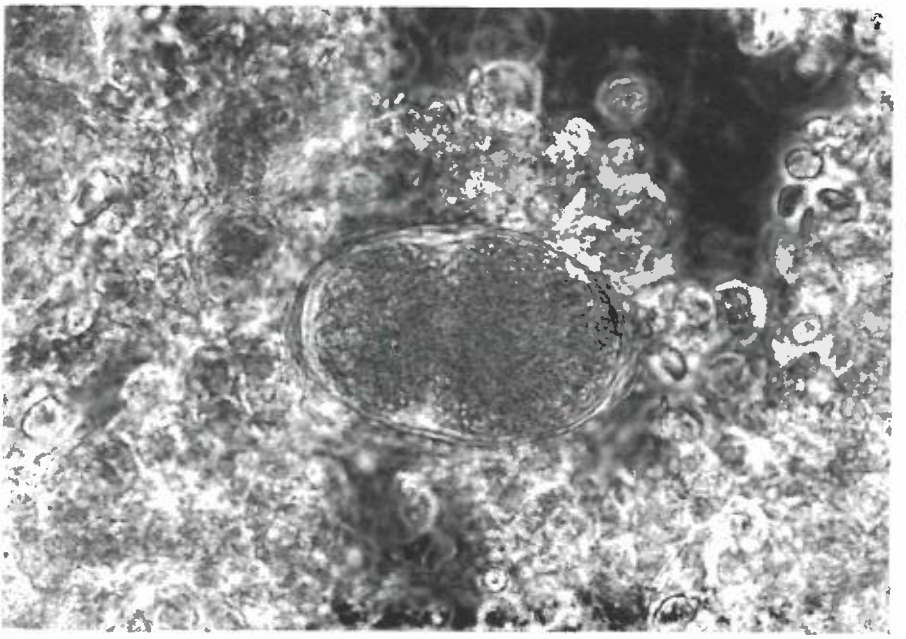

Plate 2. Trematode Ovum (Family Plagiorchiidae) from a **Red-Tailed Boa Constrictor** *(Boa c. constrictor)* X140.

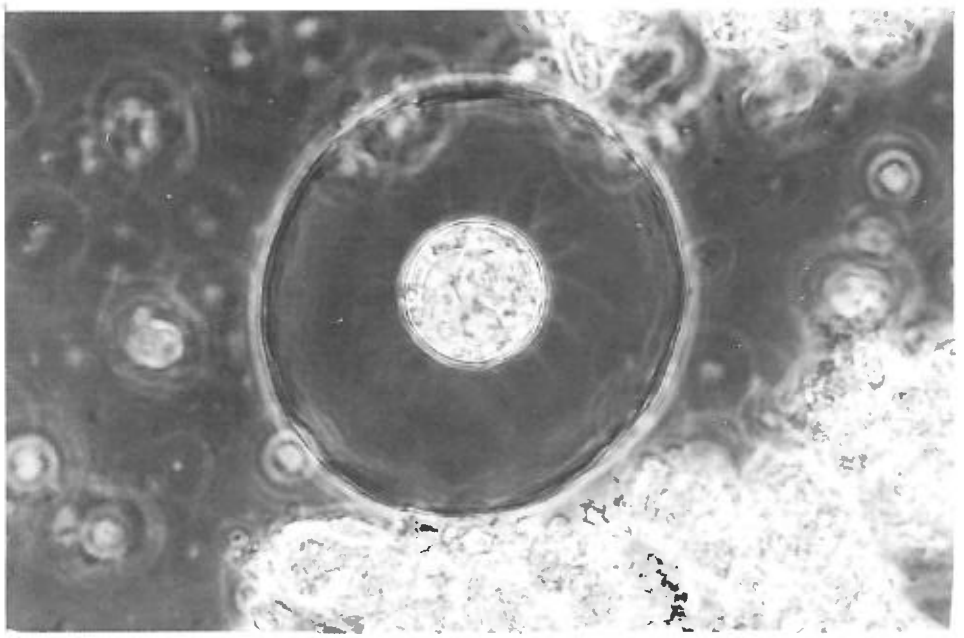

Plate 3. Proteocephalidea Cestode Ovum *(Ophiotaenia* sp) from a Rainbow Boa *(Epicrates c. maurus)* X140.

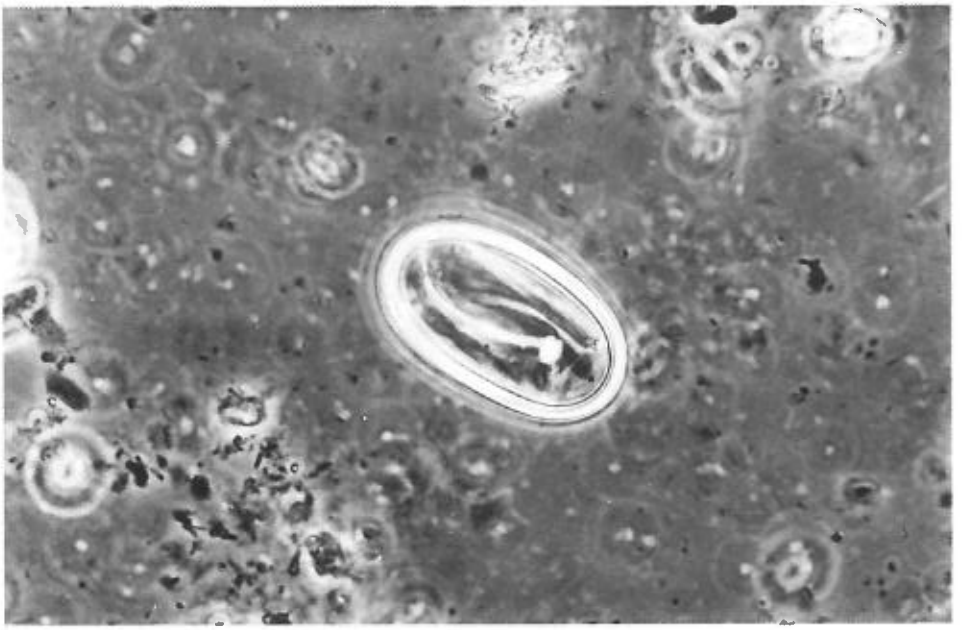

Plate 4. Spirurid Nematode Ovum *(Dracunculus* sp) from a Brazilian Rainbow Boa *(Epicrates c. cenchria)* X70.

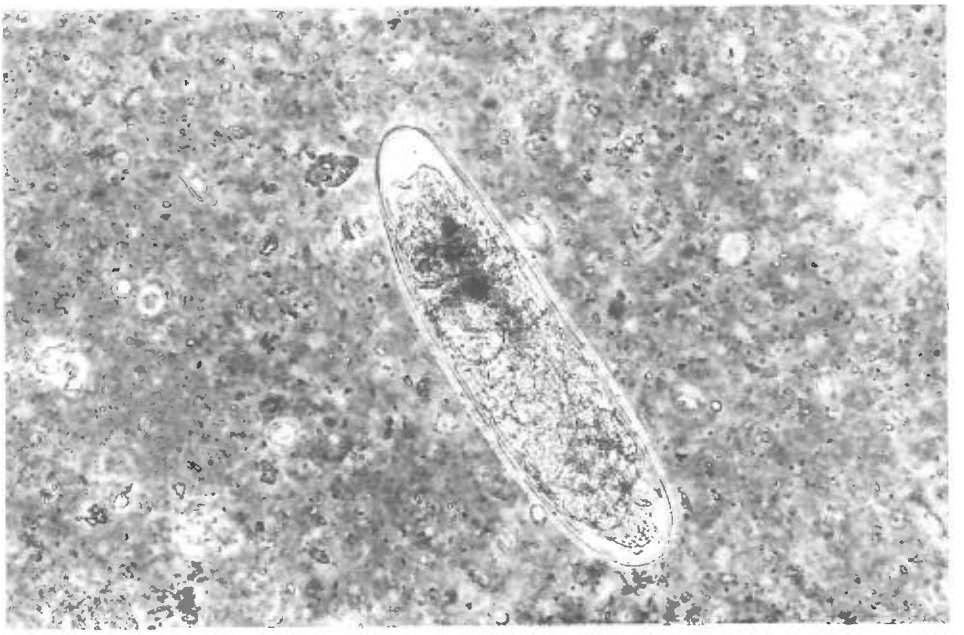

Plate 5. Oxyurid Nematode Ovum *(Spironoura* sp) from a D'Alberts Python *(Liasis albertisi)* X70.

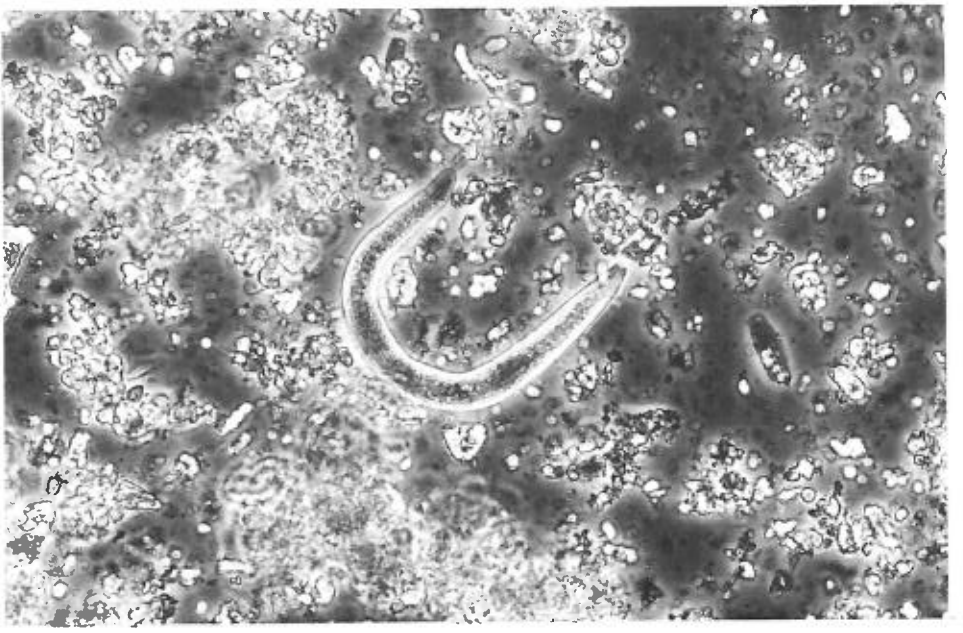

Plate 6. Strongylid Nematode Larva *(Kalicephalus* spp) from a Blood Python *(Python curtus)* X70.

For a fuller description of the parasites identified, including life-cycle, epidemiology, pathology, diseases and tretments, the reader is directed towards the comprehensive literature on reptilian parasitology (Kiel, 1975; Frank, 1981; Barnard, 1986; Frye, 1991; Bone, 1992).

Table 1. Helminth Eggs Identified from the Faeces of Boas and Pythons

ID Code	Species of Snake	Helminth Parasites Identified	Eggs per gram of faeces
PO8	*Boa constrictor constrictor*	Cestodes; *Ophiotaenia* sp. Nematodes; *Ophidascaris* sp.	650 950
P10	*Boa constrictor constrictor*	Trematodes; *Plagiorchiidae* sp. Nematodes; *Ophidascaris* sp.	275 750
B01	*Epicrates cenchria cenchria*	Nematodes; *Ophidascaris* sp. Pseudoparasite (plant origin)	3100
BO4	*Eryx colubrinus loveridgei*	Nematodes; *Hexametra* sp. *Spironoura* sp.	950 850
R13	*Python molurus bivittatus*	Nematodes; *Ophidascaris* sp.	250
R14	*Liasis albertisi*	Nematodes; *Ophidascaris* sp. *Spironoura* sp.	1200 700
R15	*Boa constrictor constrictor*	Nematodes; *Dracunculus* sp.	400
H02	*Python molurus bivittatus*	Nematodes; *Capillaria* sp.	450
	Python molurus bivittatus	Nematodes; *Capillaria* sp.	350
H06	*Boa constrictor imperator*	Nematodes; *Capillaria* sp. *Ophidascaris* sp.	250 200
V04	*Python curtus*	Nematodes; *Hexametra* sp. *Kalicephalus* sp.	450 275
V08	*Epicrates cenchria maurus*	Cestodes; *Ophiotaenia* sp. Nematodes; *Ophidascaris* sp. *Capillaria* sp. *Drancunculus* sp.	250 1000 550 275

DISCUSSION

Helminth identification was made on the appearance of the egg and their host species distribution (Kiel, 1975; Barnard, 1986; Frye 1991). In most cases identification of the family and genus was possible, but species identification was not attempted due to the taxonomic difficulties involved.

All of the reptiles that were found to harbour helminth parasites had low/medium egg counts and appeared to their owners to be healthy. No specimen exhibited a significantly high level of helminth parasitism as determined by the faecal egg counts. The lack of obvious disease amongst the afflicted individuals is attributable to the ability of wild reptiles to withstand a low degree of parasitism. In captivity improper husbandry, inadequate hygiene and "captive stress" can significantly depress this ability and result in disease (Frank, 1981; Frye 1991). For example, *Kalicephalus* sp is the principal strongylid nematode of snakes with the transmission of infective third stage larvae by drinking and through skin penetration. If maintained in humid and dirty conditions, larval numbers can build up with massive skin penetration, secondary bacterial infection, and focal dermatitis. The majority of helminths have indirect life cycles and require an intermediate host, however certain nematodes may have a direct life cycle and therefore overcrowding and poor hygienic practices can result in a multiplication of infection and very large burdens and possibly obstruction of the gastro-intestinal tract. On the basis that captive bred reptiles fail to become infected by the vast majority of helminths due largely to lack of contact with intermediate hosts, specimens R14, an adult d'Alberts Python *(Liasis albertisii),* and B01, an adult Brazilian Rainbow Boa *Epicrates c. cenchria)* must have their captive-bred status questioned. On further investigation, the Brazilian Rainbow Boa was imported from an unknown origin in the U.S.A., and therefore its captive bred status is difficult to verify.

CONCLUSIONS

This investigation demonstrates that wild caught and imported snakes are commonly infected with various species of helminth parasites, but captive bred snakes or wild caught specimens treated with an effective anthelmintic, such as ivermectin, can be clear of helminth parasitism.

Snakes can play host to a wide variety of helminths from all the major groups (cestodes, trematodes and nematodes), but individuals can appear healthy if the level of parasitism is low and the captive husbandry is adequate. On the basis of this study, captive bred boid snakes, being less likely to carry helminth parasites, are to be preferred over wild caught specimens as captives. On those occasions when wild caught boids are presented a faecal examination and appropriate treatment performed by a veterinary surgeon should be sought, even if the snakes appear healthy.

The cost of a faecal examination and effective worming is likely to be in the range of £10-£20 for a medium to large boid. Furthermore, a snake that has been wormed, kept in a clean vivarium and isolated from any unwormed snakes is likely to remain free from helminth parasites once treated. Considering this with the fact that certain metazoan parasites, namely the pentastomes, can potentially infect humans, it becomes clear that the costs of worming are relatively small compared to the long term health benefits available to both the snake and its owner.

ACKNOWLEDGEMENTS

The author is indebted to Howletts Zoo Park (near Canterbury, Kent), The Reptile House (London Zoo, Regent's Park, London), The Vivarium (55 Boundary Road, London E17), The Reptile-arium (Culvers Nursery, Enfield), and several BHS members for providing material. In addition, the author would like to thank Mr. Andrew Cunningham (Pathology dept., Institute of Zoology, London) for providing laboratory facilities and equipment and Ms. Angie Poole (Anatomy Dept., Royal Veterinary College, London) for developing and printing the photographs.

REFERENCES

Barnard, S.M. (1986). Colour atlas of reptilian parasites Part II. Flatworms and roundworms. *Compendium of Continuing Education* **8** (4): 259-262.

Beynon, P.H., Lawton, M.P.C. and Cooper, J.E. (1992). *Manual of Reptiles.* BSAVA, Cheltenham

Bone, R.D. (1992). Gastrointestinal System. In: *Manual of Reptiles.* (Eds. P.H. Beynon, M.P.C. Lawton and J.E. Cooper). BSAVA, Cheltenham.

Cooper, J.E. and Jackson, O.F. (1981). *Diseases of the Reptilia.* Academic Press, London.

Frank, W. (1981). Endoparasites and ectoparasites. In: *Diseases of the Reptilia,* Vol. 1. (Eds. J.E. Cooper and O.F. Jackson). Academic Press, London.

Frye, F.L. (1991). *Biomedical and Surgical Aspects of Captive Reptile Husbandry,* 2nd Edition. TFH Publications, New Jersey.

Kiel, J.L. (1975). A review of parasites in snakes. *The Southwestern Veterinarian* **28** (3).

Mattison, C. (1987). *The Care of Reptiles and Amphibians in Captivity.* 2nd Edition. Blandford Press, Poole.

Mattison, C. (1989). *Keeping and Breeding Snakes.* Blandford Press, Poole.

Thienpont, D., Rochette, F. and Vanparijs, O.F.J. (1986). *Diagnosing Helminthiasis by Coprological Examination.* 2nd Edition. Janssen Research Foundation, Belgium.