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PARASITES AND ASSOCIATED PATHOLOGY OF THE SWAMP WALLABY, *WALLABIA BICOLOR* (MARSUPIALIA)

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ABSTRACT: Thirty-five swamp wallabies from Victoria, New South Wales and Queensland, Australia were examined for parasites. Thirty-nine species of nematodes, five species of cestodes and eight species of arthropods were found. Wallabies from Queensland and northern New South Wales had a less diverse helminth fauna (23 species) than did wallabies from southern New South Wales and Victoria (32 species). *Rugopharynx* spp. and *Cloacina* spp. occurred in large numbers in the stomach but provoked no pathological changes. Known pathogenic species (*Globocephaloides trifidospicularis* and *Hypodontus macropi*) were encountered in small numbers only and did not produce any lesions. Pathological changes associated with parasites were: gastric nodules associated with *Labiostrongylus clelandi* and *Parazoniolaimus collaris*, bronchopneumonia due to *Marsupostrongylus* spp., biliary fibrosis associated with *Progamotaenia festiva* and fibrous peritonitis, pleuritis, pericarditis and eosinophilic splenitis due to *Breinlia mundayi*. *Echinococcus granulosus* was the only parasite found which also occurs in domestic animals.

INTRODUCTION

The swamp wallaby, *Wallabia bicolor* (Desmarest, 1804), is a common macropodid marsupial occurring in forested areas of the Great Dividing Range from western Victoria to north Queensland (Ride, 1970). A number of species of nematodes and cestodes has been reported from swamp wallabies (see Mackerras, 1958) and in recent years, several new species have been described, or amplified descriptions have been published of parasites known to occur in this host (Mawson, 1972, 1973, 1977a, b; Spratt and Varughese, 1975, Beveridge, 1976, 1978, 1979b; Beveridge and Mawson, 1978; Spratt, 1979, 1984; Spratt and Presidente, 1981; Cassone, 1983). There is little information on the prevalence or intensity of parasitic infections in swamp wallabies or on pathological changes associated with them. In this communication, we report the results of post-mortem examination on swamp wallabies from both southeastern and northeastern Australia.

MATERIALS AND METHODS

Thirty-five swamp wallabies were collected either by shooting or as road-killed animals. There were 19 males, seven females, and the sex was not recorded for the others. Collection sites numbered as in Figure 1, and number of animals collected (in parentheses) were: Queensland: 1. Townsville (1), 2. Charters Towers (5), 3. Clermont (1), 4. Rockhampton (1); New South Wales: 5. Dorrigo (1), 6. Kingstown (1), 7. Nowra (1); Victoria: 8. Bonang (6), 9. Dartmouth (4), 10. Cape Conran (1), 11. Bellbird Creek (1), 12. Marlo (1), 13. Orbost (1), 14. Bairnsdale (1), 15. Loch Sport (1), 16. Warby Range (1), 17. Bend of Islands, Yarra Valley (1), 18. Healesville (2), 19. Kamarooka (1), 20. Buangor (1), 21. Djerriwarrh Creek (1), 22. Teesdale (1).

The pelage was examined for ectoparasites and those recovered were preserved in 70% ethanol. As many of the wallabies were road-kills, no attempt was made to assess the prevalence of ectoparasites. The various organs and body cavities were examined for helminth parasites, although in some instances, gunshot or vehicular trauma precluded examination of certain organs. Parasites were preserved in 10% formol saline or 70% ethanol; lesions were excised and fixed in 10% formol saline. The entire gastric content of some animals was preserved, and the number of nematodes estimated by a dilution method. The number of nematodes of each

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TABLE 1. Helminth parasites collected from 35 swamp wallabies in eastern Australia.

Parasite	Accession no.	Site in host	Overall prevalence (%)	Regional prevalence (%)	
				Southern	Northern
Nematoda					
Strongyloidea					
Arundelia dissimilis (Johnston and Mawson, 1939)	10699	Stomach	31	4	0
Cloacina annulata Beveridge, 1979	12109	Stomach	17	24	0
C. castor Beveridge, 1979	12110	Stomach	17	20	10
C. cornuta (Davey and Wood, 1938)	7322	Stomach	9	0	33
C. edwardsi Mawson, 1972	12463, 12105	Stomach	71	76	60
C. gallardi Johnston and Mawson, 1940	12106	Stomach	31	32	30
C. hydriformis Johnston and Mawson, 1938	12464	Stomach	6	0	20
C. mawsonae Beveridge, 1979	12467, 12103	Stomach	46	44	50
C. papillata Beveridge, 1979	12466, 12108	Stomach	74	84	50
C. pollux Beveridge, 1979	12102	Stomach	17	16	20
C. wallabiae Johnston and Mawson, 1939	12107, 11109	Stomach	49	68	0
C. sp. (undescribed) 1	13638	Stomach	3	0	10
2	13639	Stomach	3	0	10
3	12104	Stomach	3	4	0
4	7395, 7408	Stomach	17	0	60
Cyclostrongylus wallabiae Johnston and Mawson, 1939	12097, 7387	Esophagus	57	76	10
Hypodontus macropi Moennig, 1929	12093	Caecum, ileum, colon	51	68	10
Labiostrongylus clelandi (Johnston and Mawson, 1939)	12127	Stomach	51	72	0
L. communis (Johnston and Mawson, 1939)	11097	Stomach	3	0	10
Macropostrongyloides baylisi (Wood, 1930)	13010, 12337	Colon	3	0	10
M. dissimilis (Johnston and Mawson, 1939)	12098, 11095	Stomach	26	36	0
Macropostrongylus macropostrongylus Yorke and Maplestone, 1926	7410	Stomach	3	0	10
Parazoniolaimus collaris Johnston and Mawson, 1939	12126	Stomach	57	80	0
Rugopharynx australis (Moennig, 1926)	12100	Stomach	57	80	0
R. epsilon (Johnston and Mawson, 1939)	12469, 12101	Stomach	66	88	30
Trichostrongyloidea					
Austrostrongylus aggregatus Johnston and Mawson, 1940	10966	Duodenum	3	4	0
A. chandleri Mawson, 1973	10966	Duodenum	3	4	0
A. victoriensis Cassone, 1983	12095	Duodenum	46	64	0
Filarinema sp. 1 (undescribed)	12096	Pylorus	20	28	0
F. sp. 2 (undescribed)	12096	Pylorus	9	12	0
Globocephaloides macropodis Yorke and Maplestone, 1926	12334	Duodenum	3	0	10
G. trifidospicularis Kung, 1948	12099	Duodenum	31	44	0

TABLE 1. Continued.

Parasite	Accession no.	Site in host	Overall prevalence (%)	Regional prevalence (%)	
				Southern	Northern
Oxyuroidea					
Macropoxyuris sp.	13002	Colon	9	0	30
Metastrongyloidea					
Marsupostrongylus spp.		Lung	46	86	10
M. dorrigoensis Spratt, 1979	—				
M. longilarvatus Spratt, 1979	12006				
M. wallabiae Spratt, 1984	—				
Filarioidea					
Breinlia (Breinlia) mundayi Spratt and Varughese, 1975	10770 WL N333	Abdominal and thoracic cavities	26	36	10
B. (Johnstonema) annulipapillaum (Johnston and Mawson, 1938)	10198	Sub cutis	3	4	0
Dirofilaria roemeri Linstow, 1905	WL N206	Stifle joint	11	16	0
No. of nematode species			39	27	21
Simpson's Index (nematodes)			—	0.05	0.07
Sorenson's Index (nematodes)			0.43	—	—
Cestoda					
Cyclophyllidea					
Echinococcus granulosus (Batsch, 1786)	—	Lung, liver, abdominal cavity	11	12	10
Progamotaenia ewersi (Schmidt, 1975)	S1516	Ileum	6	4	10
P. festiva (Rudolphi, 1819)	S1498	Bile duct	9	12	0
P. macropodis Beveridge, 1976	S2124	Ileum	20	28	0
Triplotaenia undosa Beveridge, 1976	S962	Duodenum	11	16	0
No. of cestode species			5	5	2
Simpson's Index (cestodes)			—	0.26	0.50
Sorenson's Index (cestodes)			0.57	—	—

species or genus in the stomach was calculated from their proportion in the subsample used to estimate total numbers. In those animals where dilution counts were not carried out, 100–200 nematodes were randomly selected and identified. Feces were collected in 2% potassium dichromate and later examined for coccidian oocysts. Fixed tissues were embedded in paraffin, sectioned at 6 μm and stained with haematoxylin and eosin.

Most helminth parasites collected have been deposited in the Australian Helminthological Collection, housed in the South Australian Museum, Adelaide. For each helminth species, representative registration numbers are given in Table 1. Metastrongyles and filarioids were identified by D. M. Spratt. The determinations of the metastrongyles have already been published (Spratt, 1979, 1984). Filaroid nematodes have been deposited in the collection of C.S.I.R.O. Division of Wildlife and Rangelands Research, Canberra, Australia (WL). Arthropod parasites are deposited in the Australian National Insect Collection, Canberra, Australia and hippoboscids in the B. P. Bishop Museum, Honolulu, Hawaii.

For analysis, the wallabies were divided into "northern" and "southern" populations at the parallel of latitude 31°S, where the dividing line between summer rainfall areas (to the north) and uniform or winter rainfall areas (to the south) (Burbidge, 1960) intercepts the eastern

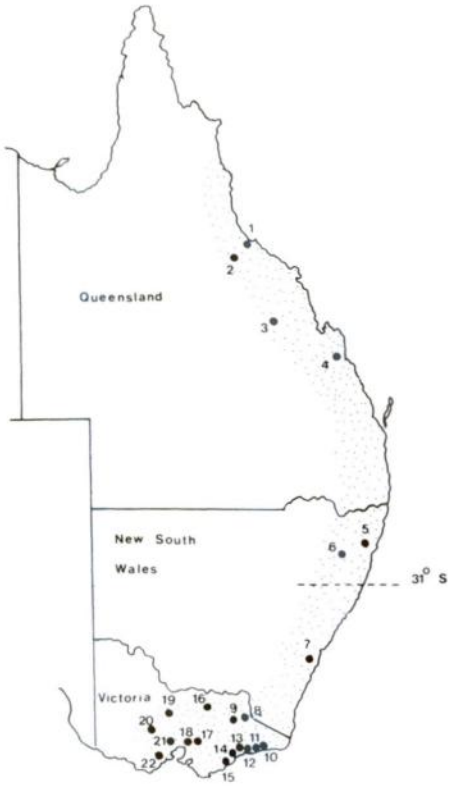

FIGURE 1. Distribution of the swamp wallaby in eastern Australia (stippled area) and collection sites for survey. Localities: 1. Townsville, 2. Charters Towers, 3. Clermont, 4. Rockhampton, 5. Dorrigo, 6. Kingstown, 7. Nowra, 8. Bonang, 9. Dartmouth, 10. Cape Conran, 11. Bellbird Creek, 12. Marlo, 13. Orbost, 14. Bairnsdale, 15. Loch Sport, 16. Warby Range, 17. Bend of Islands, 18. Healesville, 19. Kamarooka, 20. Buangor, 21. Djerriwarrh Creek, 22. Teesdale.

coast. The division corresponds approximately to the eastern Torresian and Bassian zoogeographic provinces as described by Main et al. (1958). For each population, diversity of helminths within the population was expressed using Simpson's Index, and the difference between the parasite faunas was expressed using Sorenson's Index (Greig Smith, 1964).

RESULTS

Prevalence and intensities of parasites

The helminth parasites found, their locations in the host and prevalences are shown in Table 1, except for *Strongyloides* sp. which was specifically sought only in wallabies from Queensland. The parasite may have been present in other wallabies but detailed examinations were not carried out. *Filarinema* spp., *Macropostrongyloides baylisi*, *Cloacina hydriformis*, *Macropoxyuris* sp., *Strongyloides* sp. and *Breinlia mundayi* are reported for the first time in swamp wallabies. Of the 44 species of helminths found in swamp wallabies, 32 species were found in animals from Victoria and southern New South Wales (Table 1). By contrast, 23 species, 11 of which were species of *Cloacina*, were identified in swamp wallabies from northern New South Wales and Queensland.

The intensities of infection with several gastrointestinal nematode species are given in Table 2. Nematodes omitted either occurred at a low prevalence (e.g., *Labiostrongylus communis*), could not be counted accurately (e.g., *Marsupostrongylus* spp.), or because of their localization in one part of the stomach, were not estimated accurately by the techniques used (e.g., *Arundelia dissimilis*). *Rugopharynx australis* occurred in the greatest numbers, followed by *Cloacina* spp. and *R. epsilon*. Mean intensities of all other helminths were less than 1,000.

Coccidian occysts were not detected by flotation method in any of the seven fecal samples examined. On histological examination of kidneys from eight wallabies, small numbers of macrogamonts and occasional sporoblasts of *Klossiella serendipensis* Barker, Munday and Harrigan, 1975 were seen in epithelial cells of the proximal convoluted tubules. Sarcocysts in skeletal and cardiac muscle were detected in the tissue of one of two animals from which tissues were examined. The sarcocyst walls were thin, with small zoites, corresponding to "Type B" cysts of Munday et al. (1978). No pathological changes were associated with either of these protozoans.

TABLE 2. Intensity of infection of common gastrointestinal nematodes from swamp wallabies.

Parasite	No. wallabies examined	Intensity*	
		Mean	Range
Strongyloidea			
Cloacina spp.	7	7,400	580–14,200
Labiostrongylus clelandi	4	230	2–600
Parazoniolaimus collaris	4	920	200–2,000
Rugopharynx australis	5	21,300	650–56,000
R. epsilon	5	4,300	1,150–6,400
Hypodontus macropi	11	8	1–27
Trichostrongyloidea			
Austrostrongylus spp.	3	80	40–110
Filarinema spp.	8	9	1–17
Globocephaloides trifidospicularis	9	2	1–3

* No. parasites/infected wallaby.

The following ectoparasites were found: the hippoboscids (Diptera) *Ortholfersia macleayi* Leach, 1816, *O. phaneroneura* Speis., 1902; the lice (Phthiraptera) *Heterodoxus ualabati* Plomley, 1940, *Latumcephalum lesouefi* Harrison and Johnston, 1916, *Boopia* sp.; the ticks (Ixodoidea) *Amblyomma triguttatum* Koch, 1844, *Haemaphysalis bancrofti* Nuttall and Warburton, 1915; and the mite (Acari) *Thadeua serrata* Domrow, 1976.

Pathological changes associated with parasitic infections

In nine swamp wallabies from Victoria, one to three raised firm nodules, up to 2 cm in diameter (Fig. 2), were located in the sacciform fore-stomach, anterior to the esophageal inlet, close to the junction of the squamous and cardiac mucosa (for terminology of stomach anatomy see Langer et al., 1980). The tails of numerous nematode larvae protruded from the rough surface of the nodules. These nematodes were 3rd-stage larvae of either *Labiostrongylus* or *Parazoniolaimus* based on a description of the related parasite *Labiostrongylus eugenii* by Smales (1977). Specimens of *Arundelia dissimilis* found in small numbers on the surface of nodules did not appear to cause the lesion. On histological examination, the nodule was composed of granulation tissue heavily infiltrated with eosinophils. The surface of the nodule was necrotic and many larvae penetrated deep into the lesion. Focal hyperplastic changes were found in the adjacent gastric mucosa with eosinophil infiltrations extending into the lamina propria and submucosa. In a single animal from Townsville, a small plaque of granulation tissue was found (2 cm × 1 cm) containing nematode larvae, but not elevated into a protruberant nodule. The larvae were 3rd-stage strongyloid larvae but could not be identified further.

Marsupostrongylus spp. provoked mild to severe reactions in the lung. In one animal infected with *M. dorrigoensis* and *M. wallabiae* (see Spratt, 1979, 1984) there were scattered focal granulomatous lesions characterized by eosinophil and mononuclear cell infiltrations, with smooth muscle hyperplasia and mild bronchiolitis. Extensive and severe bronchopneumonia was found in two animals examined at Bonang. Peribronchiolitis with hyperplasia and inflammation of bronchiolar epithelium and exudation of eosinophils was evident. There was diffuse in-

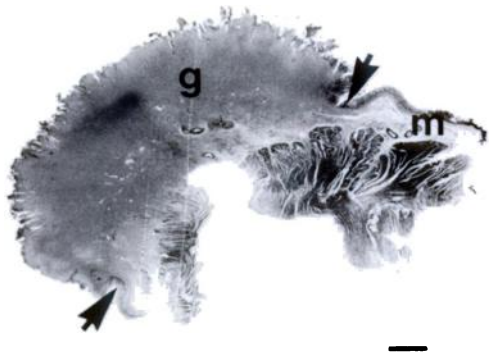

FIGURE 2. Granulomatous nodule (g) from gastric epithelium caused by larvae of *Parazoniolaimus collaris* and/or *Labiostrongylus clelandi*. Arrows mark junction of granulomatous tissue with mucosa (m). (Scale bar 5 mm).

terstitial thickening and smooth muscle hyperplasia was widespread. Dead and degenerating worms were surrounded by masses of eosinophils and foreign body giant cells. Parasites were not recovered from the lung tissue but were probably *M. longilarvatus* (Spratt, 1984).

The filarioid nematode *Breinlia mundayi* provoked a mild focal reaction in the peritoneal and thoracic cavities and in the pericardial sac, characterized by mild eosinophil infiltration and fibrosis on the surface of many organs, including lungs, heart, spleen, liver and kidneys, often with the formation of fibrous tags. Calcified nematodes were also found encapsulated in the omentum. Eosinophil infiltrations around blood vessels in the heart, hepatic portal tracts and in the cortex of kidneys were considered to be reactions to circulating microfiliariae. Focal granulomatous lesions in red pulp along the margin of the spleen were associated with sequestered microfilariae. Another filarioid, *Dirofilaria roemeri*, was found encapsulated in a thick fibrous capsule in the region of the stifle joint.

The cestode *Progamotaenia festiva* provoked a mild cholangitis with thickening of the walls of the major bile ducts as described previously in other macropodid species (Presidente and Beveridge, 1978).

Pulmonary cysts of *Echinococcus granulosus* ranged from a large viable cyst 7 × 4 cm in size located on the cardiac lobe of the lung, to a small immature cyst less than 1 cm in diameter in another animal; a degenerate cyst in the liver was identified in a third animal. A disseminated hydatid infection was found in a single wallaby at Dorrigo, New South Wales; it had numerous viable cysts 1 to 3 cm in diameter scattered on viscera throughout the peritoneal and thoracic cavities.

Apart from mild localized dermatitis at points where ticks were attached, no significant lesions were associated with any of the ectoparasites found. Likewise, no pathological changes were associated with any of the protozoa.

DISCUSSION

The largest nematode intensities in swamp wallabies were those of *Rugopharynx australis*, *R. episilon* and *Cloacina* spp. in the stomach. In spite of their large numbers, there were no associated pathological changes. *Cloacina* spp. were abundant in all collection areas, whereas *R. australis* and *R. epsilon* were absent or infrequent in animals collected in northern New South Wales and north Queensland. The absence of *R. australis* in macropodid marsupials from wet tropical areas has been reported previously (Beveridge, 1982).

Northern and southern populations of swamp wallabies differed markedly in their cestode and nematode parasites as shown by relatively low Sorenson's Indices (Table 1). The southern population of wallabies had a more diverse fauna of cestodes and nematodes as shown by the larger number of species recovered (32 species from southern areas; 23 species from northern areas) and lower Simpson's in-

dices. However, the nematode fauna in each population was still relatively diverse, without any single species dominating, and hence Simpson's index was lower than values reported from studies on the parasites of other mammals (Holmes and Podesta, 1968; Stone and Pence, 1978; Pence and Meinzer, 1979).

Gastric nodules containing larvae of *Labiostrongylus eugenii* have been described in the tammar wallaby, *Macropus eugenii*, by Smales (1977). In swamp wallabies *L. clelandi* and *Parazoniolaimus collaris* occur in mixed infections, and because the life histories of both parasites are unknown, it is not possible to assign the larvae in gastric nodules to any one species. Munday (1971) reported larvae of *Labiostrongylus* spp. associated with a squamous cell carcinoma of the stomach in the pademelon, *Thylogale billardierii*, but no neoplastic changes were associated with lesions induced by *Labiostrongylus* in swamp wallabies.

Both *Globocephaloides trifidospicularis* and *Hypodontus macropi* infections cause pathological changes, including death, in other macropodid marsupials (Arundel et al., 1977, 1979), but intensities of these parasites in swamp wallabies were low (<50) and were not considered pathogenic.

Likewise, *Filarinema flagrifer* Moennig, 1929, is known to cause gastric lesions in the red kangaroo, *Macropus rufus*, (Mykytowycz, 1964; Arundel et al., 1979), but the two species of *Filarinema* in the swamp wallaby appear to be nonpathogenic. Some species of *Macropostrongyloides* feed on the blood of the host (Beveridge and Mawson, 1978). If *M. dissimilis* ingests blood it could be pathogenic when present in larger numbers than we encountered (always <10).

Mild inflammation or an extensive verminous bronchopneumonia was seen in wallabies infected with *Marsupostrongylus* spp., similar to an earlier report by McColl and Spratt (1982). Pulmonary function was probably impaired in one animal that had concurrent plumonary hydatidosis and *M. longilarvatus* infection.

Fibrous peritonitis, pleuritis and pericarditis associated with *Breinlia mundayi* infection have not been described previously in the swamp wallaby, but the association of granulomatous splenitis with sequestered microfilariae has been reported from the mountain possum, *Trichosurus caninus*, (Presidente et al., 1982). The occurrence of *Dirofilaria roemeri* in a fibrous capsule of host origin indicates that the swamp wallaby is an "abnormal" host for the parasite. In "normal" hosts such as the wallaroo, *Macropus robustus*, the parasite lies free in intermuscular connective tissues without encapsulation and microfilariae are present in the circulation (Spratt, 1972).

Echinococcus granulosus was the only parasite found that also occurs in domestic animals. Our findings support those of Coman (1972) who found *E. granulosus* in the swamp wallaby in eastern Victoria and proposed that it was important in a sylvatic macropodid-dingo cycle. The liver fluke *Fasciola hepatica* of domestic ruminants occurs in swamp wallabies that graze contaminated agricultural land (Spratt and Presidente, 1981) but was not found by us.

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BOOK REVIEW . . .

Chemical Immobilization in Urban Animal Control Work, Leon Nielsen. Wisconsin Humane Society, IR Publications Ltd., 35 West 38th Street, #3W, New York, New York 10018, USA. 1982. 94 pp. $6.70 US.

"Chemical Immobilization in Urban Animal Control Work" is a basic introductory handbook for individuals involved in animal control work and for others contemplating chemical immobilization of domestic and wild animals. Published by the Wisconsin Humane Society this compact 5½ × 8¼″ handbook contains 31 easy to understand sections which concisely present information including the historical aspects of chemical capture and tranquilization in animal control work, animal behavioral considerations as they relate to chemical restraint, review of chemical immobilization compounds, calculation of drug dosages, immobilization related complications, legal considerations, and personnel safety. The text of the book contains good quality black and white photographs, line drawings and charts containing baseline information such as body temperature, pulse and respiration rates for species of domestic and wild animals. There are actually few errors in the text. Rather there are a few deletions of information which are pertinent. As examples, on page 35 it is stated that "there is no known antidote for xylazine." Currently there are two known alpha-2 adrenergic antagonists in use which antagonize the effects of xylazine (e.g., yohimbine, tolazaline). In the section titled "Immobilization Related Complications" it is suggested that as part of the protocol to assist in alleviating circulatory failure in an immobilized animal that "one person may start mouth to nose or mechanical resuscitation at a rate of fifteen inhalations/exhalations per minute." The reviewer is aware of a specific instance in which a wildlife biologist was exposed to rabies in attempting such a procedure with a bobcat. Thus, the second alternative, such as the use of an Ambu resuscitation bag, would be most appropriate.

Overall Dr. Nielsen should be commended for writing a very practical, unique, and informative introductory handbook for individuals entering into work which includes chemical immobilization as a tool for animal control. Additionally the handbook would make an excellent adjunct to workshops or introductory courses on this subject. As Dr. Nielsen stresses throughout the book, in total agreement with the reviewer, the procedures and techniques discussed should be under the direct supervision of a veterinarian with expertise in the manual and chemical restraint of animals.

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