The parasitology of the black stilt (*Himantopus novaezelandiae*)

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Abstract

The parasite faunas of the black stilt *(Himantopus novaezelandiae)* and the pied stilt (*H. h. leucocephalus*) in New Zealand were investigated. The alimentary tracts of eleven black stilts, and three entire pied stilts were examined for the presence of parasites, particularly helminths, and a number of carcasses were surveyed for the presence of ectoparasites. Faecal samples were taken from captive black stilts at the Ruataniwha aviary, and examined for evidence of parasite infection.

Diplophallus polymorphus (Cestoda: Acoleidae), and an unidentified cyclophyllidean cestode (Platyhelminthes: Cestoda) were recovered from black stilt viscera. *Capillaria* sp. (Nematoda:Trichuridae) were found in the intestine of two birds. Faecal sampling indicated that 67% of the enclosures within the aviary contained birds infected with adult *Capillaria* sp. nematodes, a new host record. Furthermore, sampling indicated one or more chicks were infected with this nematode before leaving the brooders in which they were raised. New host records were recorded for the digenean fluke genera *Acanthoparyphium* (Echinostomatidae), *Cotylurus* (Cotyluridae), and *Catatropis* (Notocotylidae) in the black stilt. Microphallids (Microphallidae: Microphallinae) were recovered from two black stilts; in one case they were present in huge numbers. The same host was also infected with the protozoan *Giardia* sp. (Zoo mastigophorea: Polymastigida); there were signs this bird may have been suffering from malnutrition.

Examination of pied stilts produced new host records of the digenea *Catatropis* (Notocotylidae), Psilostominae (Psilostominae), *Uvitellina* (Cyclocoelidae), and the acanthocephalan *Polymorphus* (Palaeacanthocephala: Polymorhidae). *D. polymorphus* (Cestoda: Acoleidae) and an unidentified cyclophyllidean cestode (Platyhelminthes: Cestoda) were also recovered. The parasites *Wardianum* (Cyclocoelidae), *Acanthoparyphium* (Echinostomatidae) and *Capillaria* (Nematoda: Trichuridae) were also recovered.

The acanthocephalans were responsible for severe damage to the host intestine,which would probably have resulted in its death within a short period of time.

Examination of pied stilts for ectoparasites revealed relatively heavy ectoparasite (louse and mite) burdens, although no ectoparasites were detected on black stilts. Lice have been identified as *Austromenopon himantopi, Quadraceps hemichrous,* and *Q. semifissus,* while mites were representatives of the genera *Grallobia* and *Bychovskiata* (Analgoiciea, Analgidae).

The presence of these parasites, particularly large numbers of intestinal helminths, could slow the development of black stilts raised in captivity and may cause illness and increased mortality among stilts released into the wild. It is likely that parasites are shared between *H. h. leucocephalus* and *H. novaezelandiae.*

As stilts released from captivity to the wild are likely to be under some stress, they are susceptible to parasitic infection. It may be necessary to treat birds with an anthelmintic prior to their release in order to improve their ability to survive the initial stage of their release.

1. Introduction

The black stilt, *Himantopus novaezelandiae* is one of the rarest wading birds in the world. Once widespread throughout New Zealand, they are now confined to the Mackenzie Country in South Canterbury. The decline of the black stilt in numbers and range is thought to be due to the effects of predation, loss of habitat,and hybridisation with the pied stilt *(Himantopus himantopus leucocephalus)* (C. Reed, pers. comm.).

To combat the decline in black stilt numbers, the Department of Conservation (DoC) has established an integrated management programme, which includes captive breeding and release of black stilts (Black Stilt Recovery Plan; Reed *et al.* 1993a). From an estimated 38 black stilts in 1984 (Reed *et al.* 1993a), the population reached 70 adult birds in 1993 (Reed *et al.* 1993b). Each year since 1993, up to 35 juveniles have been hand-reared and released directly into the wild. Approximately half of these birds survived for 2 months or more in the wild, and the wild population has been boosted by at least 13 birds (C. Reed, pers. comm.).

Unfortunately, there have also been a number of fatalities; the cause of death in 24 released juveniles whose bodies were recovered and necropsied were general trauma (7), hitting powerlines (6), predation (7), and unknown causes (4) (C. Reed, pers. comm.). Most of the juveniles necropsied in 1994 suffered severe tapeworm infections. It is thought that in some cases these infestations were severe enough to cause the death of the bird examined (C. Reed, pers. comm.).

Although disease and parasitism may affect the survival and reproduction of the host, there has been little research on the role of these agents on native species in New Zealand (Ranum & Wharton 1996, Anonymous 1993). Almost all multicellular organisms host one or more species of parasite - protozoan, helminth, or arthropod, whose effects range from being unnoticeable to being life-threatening.

McCallum & Dobson (1995) illustrated two important epidemiological features of disease in endangered species: firstly that the small population sizes of endangered species mean that they are unlikely to maintain infections by virulent pathogens. Endangered species will therefore tend to suffer from virulent infectious diseases only after exposure to infected hosts of a more common and widespread species. Secondly, because most individuals in the population have never been exposed to a pathogen, there is little acquired immunity to it within the population; if an epidemic does occur, its effects are likely to be severe.

Only two parasite species were known to infect *H. novaezelandiae:* the cestode *Diplophallus polymorphus,* and a similar, but unidentified cestode, possibly *D. coili* (C. Reed pers. comm.). *D. polymorphus* has been previously recorded in black-necked stilts, *H. mexicanus* (Hinojos & Canaris 1988), while *D. coili* has been recorded in the American avocet *Recurvirostra americana* (Ahern & Schmidt 1976, Garcia & Canaris 1987, Edwards & Bush 1989).

Cestodes of the genus *Anomotaenia* have been identified from Australian stilts (*H. leucocephalus* and *H. himantopus).* Some trematodes which belong to the genus *Acanthoparyphium* were also found in these birds; members of both genera are considered to be capable of causing pathology if present in large numbers (I. Beveridge, pers. comm.). It is possible that these parasites could be present in New Zealand populations of both the pied stilt and the black stilt. Specimens of *Anomotaenia* are considered to resemble specimens taken from New Zealand stilts (R. Hobbs, pees. comm.), so this could indeed be the case.

Internal parasites such as trematode and cestode worms, and ectoparasites (ticks, lice, and fleas) are known to have adverse effects on the health of birds, especially nestlings (Loye & Zuk 1991). Although these parasites may have a significant impact on the health of some individuals, it is difficult to say what effect they are having overall on the black stilt population, or the successful survival of captive birds released in the wild. It is likely that juvenile birds released from the captive rearing station at Twizel will be under nutritional stress while they adapt to a natural diet (C. Reed, pers. comm.). In this situation, they may be more vulnerable to the pathological effects of parasitism than usual (McCallum & Dobson 1995).

With an adult wild population of only 70 birds spread over 1 million hectares (Reed *et al.* 1993a), the black stilt population is probably too small and widely spread to successfully maintain parasites; the low population density means that the likelihood of transmission between potential hosts is slight. It is however possible, even likely, that pied stilts act as a reservoir of infection, sharing parasites with the black stilts. This has been observed by Edwards & Bush (1989), where avocets (a close relation of the stilt) collected from bodies of water shared with other bird species were infected mostly by parasites normally associated with those species. Individuals of a rare host species (i.e. black stilt) are more likely to be exposed to parasites of common host species (i.e. pied stilt) than their own "core" species (Holmes 1986).

Edwards & Bush (1989) also point out that all helminth parasites of avocets with known life-cycles require an invertebrate intermediate host. Diet may therefore play an important role in determining the composition and structure of the helminth communities within the black and pied stilt populations.

The primary goal of this study was therefore to enumerate and identify the species composition of the parasite fauna within both pied and black stilts. This could indicate the sources of parasitic infections of the black stilt, the severity of these infections, and possible avenues of remedial action that could be taken to ease the danger, if any, to the black stilt.

2. Methods

The parasite fauna of the black stilt was studied through external examination of dead birds for ectoparasites, analysis of the alimentary tract of birds that died of natural causes, and faecal sampling. Materials from black stilt alimentary tracts have been coded with the banding pattern of the bird to which they belonged. Faecal samples were taken from enclosures at the Ruataniwha aviary, Twizel.

Three pied stilts collected by DoC were examined in order to compare the parasite fauna of the pied stilt to that of the black stilt. Because whole carcasses were provided, it was possible to perform a complete examination of the whole bird, inside and out, for the presence of parasites. Although these birds are referred to as 'pied stilts' or '*H. h. leucocephalus*' throughout this report, the birds examined were in fact 'node C' hybrid stilts, with similar plumage and morphology to pure pied stilts (see Pierce 1982, 1984 for a full description of hybridisation and taxonomy).

2.1 EXAMINATION FOR ECTOPARASITES

Twenty-three frozen black stilts were examined for ectoparasites at the Ruataniwha aviary by rubbing forceps through the feathers. Indicators of ectoparasitec infestation (other than the observation of ectoparasites themselves) may include a generally dishevelled plumage: poor quality plumage, disturbance of feathers, and bald patches.

The three pied stilts available for examination were examined in a similar fashion to the black stilts. Collected ectoparasites were preserved in eppendorfs containing 70% ethanol.

2.2 DISSECTION

Examination of viscera by dissection is the most effective way to enumerate the parasitic fauna of an animal. Unfortunately this requires the death of the host, and is not feasible in the study of birds such as the endangered black stilt, where every bird is potentially important for the survival of the species. Viscera were available from birds that had died of natural causes. The pied stilt is less threatened, and three birds were collected by DoC for the purposes of this study.

Black stilt examination

The intact alimentary tracts of seven black stilts were available for the purpose of dissection and examination. These lacked the oesophagus, which had previously been removed, but retained much of the liver. In addition to these, there were a number of alimentary tracts which had already been dissected during post-mortems, two complete gizzards, a sample of lung and liver, and

the gizzard contents from another bird. All material was preserved in 10% formalin.

These were dissected and examined in a glass petri dish under a dissecting microscope capable of 40x magnification (although most dissection was performed at 7x magnification). Prior to dissection, intact alimentary tracts were divided into gizzard and proventriculus, upper intestine (duodenum), lower intestine (ileum), and caeca, bursa, and large intestine (rectum).

In addition to these areas, some parasites were found at the bottom of the container holding the gut, having escaped from or floated free of the gut. These parasites were recorded as having come from an unknown location.

The washings from each specimen were sieved (500 µm aperture) to simplify counting and removal of parasite specimens from the viscera. Each parasite removed was placed in a labelled glass specimen tube containing 10% saline solution. These specimens were stored for staining, mounting and examination.

Pied stilt examination

Pied stilts were carefully dissected after examination for ectoparasites. This involved making an initial incision through the midventral body wall, cutting around the anus and urogenital opening. Before the viscera were removed, the visceral cavity was examined for the presence of parasites free in the body cavity, beneath the peritoneum, or in the mesenteries. The surface of the liver was also examined for the presence of parasite cysts. Each internal organ was removed separately and placed in a dish of physiological saline; these were then carefully teased apart under a Zeiss dissecting microscope at 7x magnification. During the dissection, the mucosal wall of the intestine was scraped using the back of a scalpel blade to remove attached parasites.

The washings from each specimen were sieved (150 µm aperture) to simplify counting and removal of parasite specimens from the viscera. Each parasite removed was placed in a labelled glass specimen tube containing 70% ethanol. These specimens were also stored for staining, mounting and examination.

The methods differed slightly between the black stilt and pied stilt samples because the former material had been fixed in 10% formalin, while the latter material had been frozen, but was otherwise unpreserved.

2.3 CONSTRUCTION OF WHOLE MOUNTS

Parasite specimens were examined for the purpose of identification by staining and mounting on permanent slides. Specimens were washed in distilled water, then dehydrated through a series of ethanol dilutions (35% and 50%, each for one hour) to 70% ethanol and left overnight.

Each specimen was left in 1% acetic acid carmine red stain for approximately 30 minutes. After this period, specimens were destained using 1% acid alcohol solution and then washed in 70% ethanol. Specimens were left overnight in 100% ethanol to dehydrate. Following this, specimens were cleared overnight using cedar oil.

After clearing, the cedar oil was replaced with the mounting agent DePeX (Gurr), and specimens were again left overnight. Specimens were mounted in DePeX on single concave slides.

Nematodes possess a relatively impermeable cuticle, making them more resistant to stains (D. Wharton, pers. comm.). For this reason, nematodes were left in the stain for 1 hour.

Specimens were examined under a Zeiss Axiophot Photomicroscope; photos were taken using 50ASA black and white film at magnifications of up to 100x.

2.4 FAECAL SAMPLING

Faecal samples were taken from both the brooders and outdoor aviaries located at the Ruataniwha aviary in Twizel. Within the brooders, samples were collected by scraping faeces from the floor into a 50 ml plastic specimen container with a scalpel blade. Most of the eight brooders contained chicks incubated at the captive rearing station, with one clutch (four chicks) per brooder. In order to minimise disturbance to the chicks, samples were collected just before brooders were cleaned each day, when chicks are moved into an adjacent chamber. Each of these samples therefore represents the pooled faeces of four chicks over a 24 hour period.

At times, brooders contained single injured stilts or reduced broods of chicks. Although samples were collected in an identical way, each sample represented fewer birds, and there were consequently less faeces collected from these brooders.

Samples were taken from all outdoor aviaries except Near 3, Far 1, and New 2 and 3; all of these but Far 1 were unoccupied at the time faecal samples were taken. The aviary New 1 was only represented by a small number of samples, as it contained young chicks protected by their parents; similarly, no samples were taken from Near 4 and Far 1 because nesting pairs were present. To avoid causing unnecessary stress and possible abandonment of nests, these aviaries were avoided. Near 2 and 3, Far 2, New 2, and New 3 were all unoccupied during the primary sampling period, but samples had been collected from some of these by DoC earlier in the year.

Sampling in the outdoor aviaries consisted of placing two plastic sheets in each aviary. Each sheet was approximately 3500 cm^2 in area. Sheets were anchored in place with rocks and stones to prevent them from being blown about the aviary by the wind. Again to avoid additional stress to the birds, these sheets were placed in the aviaries when the birds were fed in the morning, and samples were scraped off the sheets using a scalpel in the afternoon, when the birds were fed again.

Each aviary contained two to four birds. Although this meant that there was a similar number of birds in each aviary to the number within each brooder, the outdoor aviaries provided significantly fewer, smaller samples because of the small size of the plastic sheets in comparison to the rest of the aviary, and the fact that much of the area of the aviaries was covered by running water, and could not be sampled.

Altogether ninety faecal samples were collected and examined, covering the period 29/9/95 to 7/12/95. Once the faecal samples were collected, they were frozen until processing and examination could take place. Each faecal examination was performed using a modification of the "comprehensive procedure for the enumeration of helminth eggs and protozoan cysts in faeces" (MAFF 1986). This procedure involves the differential centrifugation of faecal samples; samples are placed in relatively dense salt solutions - any object lighter than the salt solution (i.e. possessing a lower specific gravity) will rise to the top of a centrifuge tube during centrifugation, and adhere to a coverslip at the top of the tube. Among these objects should be any helminth eggs or protozoan cysts present in the faeces under examination.

Each faecal sample was weighed on a Sartorius electronic balance, and placed in a glass jar containing approximately 45 glass balls and 42 ml distilled water. The jar was then shaken in order to break up the faeces, and the contents of the jar were poured through a 150 μm sieve. The strained fluid was then poured into two 15 ml centrifuge tubes, which were centrifuged in an IEC model GL Clinical Centrifuge at 1800 rpm for 2 minutes.

The supernatant was discarded, and the remaining pellet was resuspended in saturated NaCl solution. The saturated NaCl solution used in this study was formulated by mixing 31.7 g of NaCl with 88.1 ml of distilled H_2O to produce 100 ml of saturated solution. Each 15 ml tube was filled until there was a convex meniscus at the top of the tube. A 26 mm x 26 mm coverslip was placed on this meniscus in the centrifuge. After centrifuging at 1800 rpm for 2 minutes, the coverslip was removed and placed on a numbered microscope slide for examination. In order to make these slides semi-permanent, each edge was sealed with clear nail varnish.

The supernatant was again discarded, and replaced with saturated $ZnSO_4$ solution; the remaining pellet was resuspended using a vortex mixer, and each tube was filled with saturated solution until a convex meniscus formed; a coverslip was placed over the top of the centrifuge tube. The saturated $ZnSO_4$ solution involved 54.6 g $ZnSO_4$ being added to 94.7 ml distilled H_2O to produce 100ml of solution. Centrifugation again involved speeds of 1800 rpm for 2 minutes. After centrifugation, each coverslip was placed on a numbered microscope slide and sealed with transparent nail varnish.

The faecal egg counts (FECs) of the four slides involved in each sample were summed. To find the number of eggs per gram of faeces, the following formula was employed (based on the equation given in MAFF 1986):
Eggs per gram = FEC x (3 + weight of faeces examined)

Slides were examined under the photomicroscope, using light microscopy, differential interference contrast microscopy, and polarised light. Photos were

taken using 50ASA black and white film. Helminth eggs were identified using Thienpont *et al.* 1986.

3. Results

3.1 ECTOPARASITES

No ectoparasites were found on examined black stilt carcasses. It is likely that any ectoparasites that were present on these birds abandoned the hosts prior to the recovery of its remains, which had then been frozen for some time. Despite the absence of ectoparasites on the birds examined, it is likely that many black stilts are infested by ectoparasites.

Pied stilts suffered relatively heavy ectoparasite burdens of both lice and mites. Mites were more common among the wing feathers of pied stilts, while lice tended to be found on the body, although no quantitative measure of abundance or distribution was made. No ectoparasites were observed on the skin or in the anterior nares of birds, but their existence cannot be excluded on the basis of this study.

These mites were representatives of the genera *Grallobia* and *Bychovskiata* (Analgoidea, Analgidae) (D. Bishop, pers. comm.). All birds examined were infected with these mites, but louse were located on only two. The louse species *Austromenopon himantopi, Quadraceps hemichrous,* and *Q. semifissus* were present on both these birds. There is a fourth louse species recorded from New Zealand stilts, *Saemundssonia platygaster*; this is rarely recorded, but obvious when present (R. Palma, pers. comm.).

3.2 VISCERAL PARASITES

Parasite fauna of black stilt viscera

Examination of the gizzard contents of birds YR-BkW and WR-RR failed to detect the presence of parasites. Similarly, no parasites were found within the gizzard and contents of the WR-RR. The lung and liver of WBk-WG were also examined without detecting any parasites.

Examination of the complete black stilt alimentary tracts resulted in the recovery a variety of parasites, summarised in Appendix 1. The prevalence, intensity, and abundance of these infections have been recorded in Appendix 2.

Prevalence is the frequency with which a parasite was found within the host population. Infection intensity describes the average number of parasites within infected hosts, while parasite abundance is the average number of parasites within each member of the entire population (including uninfected hosts) (Anderson 1993).

Many of the parasites recovered were poorly preserved, making complete identification very difficult. No attempt was made to identify parasites beyond the level of the genus, because of the difficulty of the task considering the state of the specimens, the poor state of the systematics of most of these taxa, and the likelihood that many of the specimens collected may constitute new species.

The single cestode infecting WY-RBk is almost certainly *Diplophallus polymorphus.* The scoleces of both the reference sample of *Diplophallus polymorphus* provided by Massey University and the cestode taken from WY RBk had retracted into the body of the parasite, making identification difficult, and the state of preservation meant that the use of other distinguishing characteristics (such as the number of testes) was virtually impossible. Comparison of the specimen taken from WY-RBk with the reference sample suggested that they were of the same species, both samples possessing extended cirri on mature and gravid segments, which form a conspicuous bilateral fringe, an effective analytical taxonomic feature (P. Mason, pers. comm.). At least one of the cestocles inhabiting the duodenum of WBk-BkG was also *D. polymorphus.*

Examination of these samples was unable to give any hints to the morphology of the mature fertilised egg; some eggs in the mature proglottids were spherical, some elliptical, and there was considerable variation in size. This degree of variation is rare within helminth species (although not in *D. polymorphus*; see Burt 1980) and complicated potential identification of *D. polymorphus* eggs in faecal samples.

What is not shown by these statistics is that the majority of the unidentified cestocles consisted of the scolex and just a few segments. The representatives of *D. polymorphus* recovered would represent a similar biomass to all other cestocles combined.

Although some echinostomes were poorly preserved, they could be identified as belonging to Sub-family Echinostomatinae, and a tentative generic classification was possible. The cligenean genus *Acanthoparyphium* accounted for the majority of the echinostomes examined. These were often difficult to detect and remove from among the intestinal villi. Many of the echinostomes examined were juveniles (~6 days old), suggesting infections were recent.

The microphallids (Microphallidae: Microphallinae) were mature, with two distinctive egg sacs. These face into the lumen of the host, and are the most prominent morphological feature under the dissecting microscope. The small size of the microphallids, their location among the villi, and the large numbers present made accurate counts difficult.

Cotylurus sp. (Digenea: Strigeidae) were found in the caeca and intestine of infected birds. In WW-BkBk those inhabiting the intestine were all located in a band between one and two centimetres below the openings to the caeca. In the more heavily infected WBk-BkG the area of infection was more widespread. These parasites were firmly anchored to the mucosal wall, and had to be carefully prised or scraped off. On many occasions when the parasite was removed, it took at least one villus with it. A similar situation occurred when

members of the genus *Catatropis* (Digenea: Notocotylidae) were removed from the host mucosa; a circular imprint sometimes remained at the site of attachment.

The intensity of the infection found in WBk-BkCT is illustrated by the fact that one section of intestine 6 mm long contained 94 strigeids and 158 microphallids. The duodenal loop (~3 cm) from the same bird produced 4 cestodes (at least one of which was *D. polymorphus*), 1033 echinostomes, 90 microphallids, and one *Catatropis* sp.

The only organ examined which contained parasites was the liver of the black stilt WW-BkBk, which contained a digenean and a cestode. This is considered a post-mortem artifact, because cestodes cannot survive in the liver, and the digenean appeared to be a representative of the genus *Acanthoparyphium*, otherwise found in the intestine and caeca of the host.

Parasite fauna of pied stilts

Examination of pied stilt material produced an interesting and varied parasite fauna. There were two species of cestode, one species of nematode, five species of digenean, and one acanthocephalan species. The size and composition of this parasite fauna is summarised in Appendix 3 and Appendix 4.

The only gross physical pathology observed within examined pied stilts was damage to the small intestine of pied#2, caused by penetration of the intestine by Acanthocephala (*Polymorphus* sp.). Removal of acanthocephalans from the gut wall was a time-consuming task due to the presence of the hooks on the proboscis, which burrows into the gut wall, and holds the parasite in place. This may affect innervation of the intestine, as well as secretory and motor function (Petrochenko 1971).

A number of Acanthocephala appeared to have penetrated the gut wall and protruded into the abdomen. If this occurs before death, potentially lethal peritonitis may result (Clark *et. al.* 1958). It is likely that this bird would have died sooner rather than later, given the damage caused by the acanthocephalans within the gut and body cavity.

In addition to these parasites, there were five genera of digenean parasite. Three of these (*Acanthoparyphium, Catatropis,* and a genus of the family Psilostominae) were found within the digestive system. The former two genera were also present in black stilts. The other two genera were located in the body cavity; these have been identified as *Wardianum* and *Uvitellina* (Cyclocoelidae: Cyclocoelinae). These are parasites of the air sacs of birds; both have been recovered from stilts before (*H. leucocephalus* and *H. candidus* respectively; Yamaguti 1958).

3.3 FAECAL SAMPLES

During this study, 31 of the 90 samples were found to contain *Capillaria* sp. (Nematoda: Trichuridea) eggs, representing 14 of the 21 aviaries tested; an exact prevalence is difficult to calculate, as some birds were transferred be-

tween aviaries as the need arose. Average FECs (faecal egg counts; the number of capillarid eggs per gram of faeces) of infected aviaries are summarised in Appendix 5.The results of all faecal samples are recorded in Appendix 6.

Flotation with saturated $ZnSO_4$ solution detected capillarid eggs in thirty-one faecal samples. Centrifugation and flotation in saturated NaCl solution detected eggs in only six of these cases, proving to be less sensitive than $ZnSO_4$.

Far 2 was only tested once, and only 0.03 g of faeces were collected; this is not enough to indicate if the resident was infected. Three samples from the aviary New 1 yielded only 0.96 g of faeces, which is also a small amount from which to draw any conclusions. New 2 was not tested, as no birds were present during the study period; it is possible that this enclosure could also be infected, as the eggs of capillarid nematodes can survive in the environment for some time.

An important point to note from Appendix 5 is that chicks in Brooder 7 were infected with adult *Capillaria* nematodes. These chicks have never left the confines of the building in which they were born. The only possible sources of infection are aquatic invertebrates fed to the young chicks, or direct infection from eggs shed by the juvenile held in that Brooder on 29 Sept, although the likelihood of the latter occurring is somewhat reduced by daily scrubbing and disinfection of each Brooder.

No protozoan cysts were identified from faecal material. A number of objects within these faecal samples remain unidentified; some may be parasitic in origin.

4. Discussion

The primary goal of this study was to identify and enumerate the parasite fauna of black and pied stilts. An important and obvious difficulty of working with endangered species such as the black stilt is that it is not feasible to cull members of the population for parasitological study. This means that only non-invasive methods of investigation are possible, and the number of hosts involved must be small.

The situation is little better in relatively more common species such as the pied stilt. Given the stringent ethics requirements now in operation, it is difficult to receive permission to collect a significant number of vertebrates for an accurate parasitological survey. Low sample sizes prevent researchers from being able to confidently claim that the parasite fauna observed in a study is representative of the whole population. It has been shown that the parasite richness of a host species is often a function of the number and depth of studies devoted to that animal rather than any ecological, behavioural, or physiological factor (Walther et al. 1995).

The study of parasites can be difficult, and the death of the host may be necessary to confirm parasite presence and identity; often the presence, abun-

dance, and effects of parasites can only be inferred. The magnitude of this problem is somewhat lessened in the study of parasites of the alimentary tract, as live birds can be examined through non-destructive methods such as faecal sampling and stomach pumping (Ranum 1993).

4.1 ECTOPARASITES

The absence of ectoparasites on any of the black stilts examined was not surprising. The examined birds had been frozen for up to two years, and stored in plastic shopping bags. Many of these birds were found only after some time had passed; it is likely that any ectoparasites that were present would have abandoned their hosts before they were recovered.

The best ways to determine if an animal is suffering ectoparasitec infestation is to either capture and examine a live bird, or to kill a bird, and to immediately place it in a bag, which is then sealed. This ensures any ectoparasites present remain, unable to escape. These can then be removed and identified (MAFF 1986).

The major impact of ectoparasites on host fitness is through debilitation of otherwise enfeebled birds, and as vectors for infectious disease. Ectoparasite populations can increase rapidly on sick birds (Wobeser 1981), while nesting birds may also be heavily. infested, as ectoparasites may become established in nesting material. Generally ectoparasites will not be a major cause of concern if the host is healthy.

4.2 VISCERAL EXAMINATION

The use of direct counts is generally considered to be the most accurate method of determining helminth burdens. Only direct counts enable researchers to determine exactly how many parasites are present, their age, sex, and precise identity. This process is easier still if the material examined is fresh.

Examination of parasites within the viscera was complicated by poor preservation of some material, despite being fixed and stored in 10% formalin. The primary difficulty was that the scoleces of recovered cestodes were often severely degraded, had retracted into the body, or were missing altogether. This made accurate counting difficult and identification impossible. This was compounded by the deterioration of the internal structure of the cestodes, which made effective staining problematic; specimens failed to clear adequately and internal structures could not be discerned.

There are three likely causes of this problem: (1) Some of the birds were not found until some time had passed after their death; this means that the gut and the parasites within had time to decay. (2) Each gut and its associated parasites was stored in a small (120 ml) container of formalin; in some cases insufficient formalin may have been present to properly preserve the contents of the container. (3) Even though there was sufficient formalin, the parasites within the gut may not have been preserved properly due to poor pen-

etration of formalin through the gut. Although the gizzard of each intestinal tract was cut to aid penetration, this may have been inadequate.

It is best to extract the parasites from the gut as soon as possible post-mortem. Nematodes should be placed in full-strength glacial acetic acid or hot 70% ethanol; this kills and fixes the worm uncoiled. Once this has occurred, worms should be transferred to glycerin alcohol (9 parts 70% ethanol, 1 part glycerin) for storage. Cestodes should be relaxed in refrigerated tap water for 2-4 hours, then fixed in AFA (8.5 parts 70% ethanol, 1 part formalin, 0.5 part glacial acetic acid). Digenean trematodes should be relaxed in refrigerated tap water for 30-60 minutes, then fixed in AFA. Acanthocephala should be fixed in AFA; if necessary they may be placed in refrigerated tap water overnight to allow the proboscis to extend (Greiner & Ritchie 1994). This would increase the likelihood that parasites would be in suitable condition for identification.

Comparison of Appendix 2 and Appendix 4 shows that *H. novaezelandiae* appears to suffer more intense levels of digenean parasitism than *H. h. leucocephalus*, which in turn suffered heavier cestode burdens.

Some 10 parasite species were identified in this study; seven new host records were noted from *H. novaezelandiae* (including *Giardia)* (John &John 1949), while five more were recorded from *H. h. leucocephalus.* The number of parasite species found in this study (using fourteen birds) is lower than those found in similar studies of related birds, which utilised larger sample sizes; *R. americana* has 50 known helminth parasites (Edwards & Bush 1989 (n=22), Garcia & Canaris 1987 (n=33)) and *H. mexicanus* has 19 known helminth parasite species (Hinojos & Canaris 1988 (n=35)).

From the data available from this study, it appears that the primary concern for conservation managers is the high abundance of digenean parasites in the black stilt. These may cause considerable harm to the host when present in large numbers. The difficulty for conservation managers lies in preventing these infections. This appears to be impossible to manage in the wild, as most parasites are naturally acquired by the avian host through invertebrate intermediate hosts, such as crustaceans or snails.

Dosing birds within the aviary will probably prevent any real threat to the birds during this time, and for a short time following their release. Dosing birds effectively once they are released into the wild could prove difficult. At the Ruataniwha aviary, every attempt has been made to ensure that the birds' surroundings are as natural as possible once they leave the brooders. This is a double-edged sword, however, as the more natural an aviary setting, the more likely an infection is to occur.

4.3 FAECAL SAMPLING

The presence of worm eggs in faecal samples provides positive evidence that an animal is infected. Unfortunately the size of worm burdens cannot be accurately estimated from faecal egg counts. These must be interpreted cautiously; it is very important to note that although the presence of large num-

bers of eggs or larvae in the faeces may indicate the presence of heavy parasitic infestation, their absence, or presence in small numbers does not necessarily mean such infections are not present (MAFF 1986).

Faecal egg counts are affected by several factors. Among these is the fact that regular diurnal fluctuations have been found to occur (Chappell 1993); these fluctuations are significant enough to require consideration in experimental design and analysis of results. In the case of the brooder samples, each representing a 24-hour period, any fluctuations have been fully controlled. In the outdoor aviaries, any eggs released at night will not be detected, as samples were collected during the clay. Faecal egg counts are also influenced by the uneven distribution of eggs throughout the faeces (although the impact of this variation is comparatively small) and variation in the quantity of faeces passed, which affects the number of eggs per unit weight (MAFF 1986).

There are also a number of factors which limit the significance of these counts. The immune response of a resistant host can reduce or suspend ovulation of adult worms within the host; the resistance of the host can greatly increase the pre-patent period, and some infections can cause disease, but fail to become patent (produce eggs) (MAFF 1986). Other factors (such as intraspecific competition for resources) can affect the relationship between female worms and the number of eggs released in the faeces. Immature parasites do not lay eggs, and therefore cannot be detected by faecal egg counts; however, some juvenile stages are capable of causing serious damage to the host (Whitfield 1993).

Another problem with relying on faecal egg counts for the identification and enumeration of the parasite burden of a host is the difficulty associated with identifying species, particularly amongst nematodes, whose eggs are not usually easily distinguishable. Despite the similar appearance of the eggs of many species, the effects of the larvae and adults differ widely in pathogenicity and fecundity (Whitfield 1993). This problem is somewhat reduced in diagnosis of *Capillaria* spp. infections, as the presence of polar plugs at each end of the egg are a distinctive feature. Differentiating between individual *Capillaria* species on the basis of egg morphology alone, however, is difficult.

For the above reasons, faecal egg counts are best used as an indicator of infection or as a method of comparison, rather than as a quantitative measure of infection. The presence of eggs indicates the presence of a particular parasite, but the absence of those eggs does not necessarily indicate the absence of that parasite.

There have been a number of techniques devised to minimise these problems with the use of faecal egg counts, probably the most effective of which are differential centrifugation methods. These involve the use of flotation solutions, such as saturated solutions of NaCl, $ZnSO_4$, or $MgSO_4$. The types of eggs recovered through such techniques are related to the specific gravities of the solutions involved; for example saturated $ZnSO_4$ (SG 1.364) is recommended for the recovery of Fasciola eggs from faeces, as these will not float in saturated NaCl solution (SG 1.204) (MAFF 1986), the higher the specific gravity of a solution, the greater its density.

In this study, flotation in NaCl solution achieved unsatisfactory rates of recovery in comparison to those achieved using $ZnSO_4$ solution. Any further studies should therefore use saturated $ZnSO_4$ solution during centrifugation and flotation. Another possibility is the use of sedimentation techniques, which may be superior when searching for large fluke eggs (Greiner & Ritchie 1994).

The same factors which affect the accuracy of helminth egg counts also affect counts of protozoa. Most species of wild or domestic birds host one or more species of coccidial protozoa, which may or may not be pathogenic. Intestinal coccidia have been extensively studied due to their effect on commercially important species such as fowl and turkeys. The presence of helminth eggs or protozoan oocysts in the faeces has little clinical significance. High oocyst counts can be recovered from apparently healthy animals, while animals may appear diseased even when very few oocysts are present (MAFF 1986).

Diagnosis of protozoan infection such as coccidiosis should be based on the clinical history and examination of post-mortem material of infected animals. Protozoan oocyst counts help to support diagnosis, but it is difficult to reliably identify parasites from the oocysts. The infection may be identified from the nature and location of the lesions and the appearance of the developmental stages of the parasites in tissue smears (MAFF 1986).

Despite the relatively high prevalence of capillarid infection within the aviaries (67%), only two capillarids were recovered from dissected black stilts. There are three possible reasons for this: (1) the black stilts dissected had been deceased for more than two years in some cases, and capillarid infections may have been rare at that time; (2) *Capillaria* spp. are notoriously difficult to detect, as they tend to burrow into the wall of the gut (Anderson 1992); (3) the small sample size dissected, and an aggregated distribution of parasites in the host population may mean that although *Capillaria* spp. may have infected members of the population, at the time these birds died, the birds examined were not infected by them. Positive identification of the nematode species infecting birds currently in the aviary will have to await dissection of those birds (following their death from natural causes).

No protozoan oocysts were identified from faecal smears, but their absence, especially in chicks and juveniles, cannot be confirmed until collected material is examined further.

4.4 SOURCES OF INFECTION

Usually tapeworm eggs must be eaten by an intermediate host before the first stage of development can occur. This intermediate host could be an earthworm, snail, insect, fish, or other animal edible to the definitive host. Once within the intermediate host, the parasite quickly migrates from the gut into muscles or other body tissues, where development occurs. If the intermediate host is eaten by a suitable bird host, digestion of the tissues releases the infective worm into the gut, where it develops into an adult (Arnall & Keymer 1975).

This life cycle is often short; infective cysticercoids are present in the intermediate host within two or three weeks of ingestion of an oncosphere (hexacanth embryo), and tapeworms can reach maturity within the definitive host within two weeks (Wobeser 1981).

Ahern & Schmidt (1976) believed that waterboatman species acted as a possible intermediate host of *D. polymorphus* found in the duodenum of North American avocets, but were unable to demonstrate transmission from waterboatman to the definitive bird host. Edwards & Bush (1989) asserted that all parasitic helminths with known lifecycles in avocets require an invertebrate intermediate host for transmission. However, several species of capillarid which infect avocets and stilts are known to utilise direct lifecycles, or use earthworms as transport hosts in a facultative indirect lifecycle (Anderson 1992). Thus, there is a distinct possibility that parasite transmission of some species occurs directly through the environment.

Acanthocephalans (Crompton & Nickol 1985), flukes (Cheng 1986) and cestodes (Whitfield 1993) do require one or more invertebrate intermediate hosts to complete their lifecycles. The most common invertebrate species in lotic stilt habitats (flowing water) are *Deleatidium lillii, D. myzobranchia, Aoteapsyche, Pycnocentrodes,* Chironomidae, and Tubificidae (Pierce 1982). In lentic (still water) habitats, *Sigara arguata, Anisops assimilis, Xanthocnemis zelandica, Chironomus* spp., Tubificididae, *Potamopyrgus* spp., and *Lyninaea* are more common (Pierce 1982). The feeding methods of stilts are not selective in nature, and can, for example, involve random sweeps of the beak, catching a large number of invertebrates at once (Pierce 1985), so all of these species and many more besides are potential vectors of parasites.

Parasites can be transmitted between bird species sharing a body of water. Avocets from permanent bodies of water had parasite communities composed largely of species that are normally considered to be specialists in various duck species (Edwards & Bush 1989); ecological conditions were thought to be able to over-ride phylogenetic host specificity. This suggests that black stilts may be capable of being infected by the parasites of other bird species such as the pied stilt, or indeed some of the other birds with which it shares waterways. This possibility is enhanced by the wide array of feeding methods and broad feeding niche utilised by the black stilt (Pierce 1985), which exposes birds to a wide variety of potential intermediate hosts of parasitic helminths.

This possibility was examined to some extent in this study through the dissection of pied stilts. The close phylogenetic relationship between these bird species, as evidenced by their ability to hybridise, means that parasite species should be able to switch between the two species with relative ease.

Comparison of the parasite fauna of the two species showed that this hypothesis may be partially correct. All pied stilts were infected by the cestode *Diplophallus polymorphus.* Only two black stilts were infected with this parasite, but prior to this study *D. polymorphus* was the only positively identified parasite species known to infect black stilts. Similarly, both bird species were infected by the same cyclophyllidean cestode, *Capillaria* sp. nematodes, and the same species of digenean fluke (*Acanthoparyphium* and

Catatropis). It is likely that cross-infection involving these parasites does occur, because the low density of black stilts (less than one per 10 000 ha) means transmission solely between black stilts is unlikely to allow the parasite species to be maintained within that host population.

Any apparent differences in the parasite fauna may in part be due to the small number of birds examined; in their study, Ahern & Schmidt (1976) continued to find new parasite species until the 26th bird was examined. Some differences should not be unexpected, however, as the two birds preferentially occupy different habitats (although there is some overlap). Pied stilts prefer swampy terrain, while black stilts prefer open braided rivers. As a result, birds may be infected at differing rates by certain parasite species, either through differences in habitat occupied, invertebrate prey, or the variety of bird species with which the habitat is shared.

4.5 THE PARASITES OF *HIMANTOPUS NOVAEZELANDIAE*

Studies of the Recurvirostridae (the family consisting of stilts and avocets) show a high prevalence of helminth parasites within sampled populations. No *R. americana* collected by Ahern & Schmidt (1976) (n=37) or Garcia & Canaris (1987) (n=33) were free of parasites. Nor were *H. h. himantopus* collected by Ukoli (1965) (n=6), or *H. leucocephalus* examined by Mawson (1968) (n=5), only one of the 35 *H. mexicanus* examined by Hinojos & Canaris (1988) was free of helminthological infection. All of the black stilt viscera and pied stilts examined in this study were infected to a greater or lesser extent, and all pied stilts were infested with ectoparasites.

At the outset of this project, all that was known about the parasite fauna of *H. novaezelandiae* was that it was infected by the cestode *Diplophallus polymorphus,* and one other, similar cestode (possibly *D. coili*) (C. Reed, pers. comm.). Shortly after the beginning of the current investigation, Australian veterinarians identified cestodes from the black stilt as being similar to members of the genus *Anomotaenia*, found in Australian stilts. There were also echinostome flukes present, probably from the genus *Acanthoparyphium* (S. Haigh, pers. comm.). It is thought that large numbers of the latter genus may be capable of causing some pathology if they are present in large enough numbers (I. Beveridge, pers. comm.).

All known helminth parasites of *Himantopus* spp. are summarised in Appendix 7, including all new host records from this study.

Diplophallus polymorphus

Diplophallus polymorphus (Platyhelminthes: Cestoda) has been found in the black stilt before this study (T. Charleston, pers. comm.); in fact it is the only parasite whose identity has been confirmed to the species level. Cestodes are generally not known to cause pathological effects on their hosts. In situations where the host is under physiological or nutrient stress, however, there can be a negative impact on the health of the host (Loye & Zuk 1991).

D. polymorphus (Rudolphi 1819) has been recorded in the avocets *Recurvirostra avosetta* and *R. americana*, and the stilts *Himantopus himantopus* and *H. mexicanus* (Burt 1980). As its name suggests, *D. polymorphus* shows great intraspecific variation; these differences are related neither to host species or host locality (Burt 1980). Indeed, *D. polymorphus* is likely to be synonymous with the species *D. coili* Ahern & Schmidt 1976 and *Himantocestus blanksoni* Ukoli 1965 (Burt 1980).

D. polymorphus is found in the duodenum, and occasionally the anterior region of the small intestine. Adult worms vary in size from 80 to 280 mm, with a corresponding width of 6.2 to 3.5 mm, depending on the degree of contraction upon fixation. The scolex is clearly marked off from the strobila without a neck. The four suckers are forward directed, measuring 0.21-0.29 mm in diameter. The rostellum is pear-shaped, with a length of 0.29-0.39 mm; there are 20-25 rostellar hooks of varying size (Burt 1980).

The segments of the strobila are usually much shorter than broad. In most strobila the extended cirri of mature and gravid segments form a conspicuous bilateral fringe, which can be observed with the naked eye. The number of testes cannot accurately be determined on a whole mount, but is approximately 120 to 160 per segment (Burt 1980).

Eggs containing hexacanth embryos are present in the posterior segments where the number of segments in the strobila is over 200. They have two envelopes, both light-refracting; the outer one is ellipsoidal 72-92 µm in length and 39-59 µm in diameter, and the inner one is an embryophore enclosing the hexacanth embryo - this is the least variable feature in size and shape, with a length of 35.5-37 µm and a breadth of 10-11 µm (Burt 1980).

Other cestodes which appear to belong to the genus *Diplophallus*, but which do not entirely agree with the above description, are *Himantocestus blanksoni*, from *Himantopus himantopus himantopus*, *D. coili* from *Recurvirostra americana*, and *D. andinus* from *R. andina* (Voge & Read 1953). There is considerable confusion concerning the classification of these parasites in relation to each other, and indeed there has been considerable argument regarding placement of a number of genera including *Diplophallus* in the families Acoleidae Ransom 1909, Diploposthidae Poche 1926, and Hymenolepididae Raillet & Henry 1909 (Voge & Read 1953, Yamaguti 1959, Schmidt 1970, Ahern & Schmidt 1976, Burt 1980).

All species of *Diplophallus* inhabit the duodenum and small intestine of their host; commonly these infections occur in pairs, an unusual situation in monoecious cestodes. The ecological explanation advanced by Ahern & Schmidt (1976) is that the size and nutrient requirements of these worms would cause great stress to the host through absorption of nutrients and possibly even causing blockage of the intestine, reducing the chances of survival of both the host and the parasite. It appears that initially there are a number of parasites present, but that as time passes, competition between the worms increases as they grow, until only two worms survive (Ahern & Schmidt 1976).

Attempts to determine the intermediate host of *D. coili/D. polymorphus* by Ahern & Schmidt (1976) involved starving a number of invertebrates in a beaker, and feeding them a gravid proglottid. Active oncospheres were found

in the gut of the waterboatman (Hemiptera, Corixidae, Sigata sp.) 3-4 hours after they were fed eggs. Although this suggests the waterboatman is an intermediate host, Wetmore (1925; cited in Ahern & Schmidt 1976) believed the life cycle of *Diplophallus* was direct, and reported observing avocets eating the cast-off terminal segments of worms.

Cyclophyllidean cestode sp.

The scolex of this cestode possesses four suckers, and an armed rostellum possessing ten hooks. There is a noticeable neck, and the strobila possesses an opening every second segment, probably a genital opening. Unfortunately the reproductive system of this cestode is insufficiently defined in the available specimens to greatly narrow the range of possible cestode families.

Although large numbers of these parasites were recovered, especially from the pied stilts, most were immature and/or fragmented, missing either the scolex or strobila, and making identification difficult.

Capillaria sp.

There are over three hundred described species in *Capillaria sensu lato*, and classification of the subfamily Capillariinae to which *Capillaria* belongs is considered to be one of the most difficult and unsatisfactory of the Nematoda (Anderson 1992). Many of the species found in *Himantopus* spp. have been assigned to new genera (e.g. *Aonchotheca*, *Baruscapillaria*, *Eucoleus*) to reduce the size and complexity of the genus *Capillaria*. These have not yet found their way into common use, however, and the term *Capillaria* is therefore used throughout this dissertation.

Capillaria vary in length from one to eight centimetres, with most tending towards the lower end of the scale, males being smaller than females. Most *Capillaria* spp. utilise a direct life cycle; eggs are passed out in the droppings of the host, developing into a stage infective to other birds over a period of about one month. Some species require an intermediate host to complete their life cycle (Arnall & Keymer 1975).

The more "natural" an aviary, the more likely its inhabitants are to be infected due to the build-up of parasites in the soil and vegetation (Arnall & Keymer 1975). The Ruataniwha aviary is about as "natural" as humans can make it, containing running water, vegetation, and nearby wetlands. The area surrounding the outdoor aviaries is frequented by a number of bird species, including juvenile black stilts released from the aviary in the previous year, which may fly over the aviaries and interact with stilts within the aviary.

Members of this nematode genus have been found in stilts and avocets in North America. Garcia & Canaris (1987) found that five of 33 *R. americana* examined were infected with the nematode *Capillaria recurvirostrae*, with an average of seven worms infecting each bird. Hinojos & Canaris (1988) identified three species of *Capillaria* nematodes in *H. mexicanus*, and found one member of the genus which they were unable to further identify; these are summarised in Appendix 7.

Nine members of this genus have been found to infect New Zealand birds, although none of these hosts was of the Order Charadriiformes to which *H. novaezelandiae* belongs (Weekes 1982). The only two *Capillaria* species recovered from *R. americana* and *H. mexicanus* found in New Zealand birds are *C. contorta* and *C. obsignata*. *C. contorta* has been recovered from the grey partridge (*Perdix perdix perdix*) by McKenna (1976), while *C. obsignata* has been recovered from the small intestine of the common fowl *Gallus domesticus* (Rickard & Pohl 1969) and the rock pigeon *Columba livia* (McKenna 1976).

Acanthoparyphium sp.

The incidence of fluke infestation is relatively high in aquatic bird species. Unlike tapeworms, which tend to be limited to the host intestine, flukes can be found in many organs (Whitfield 1993). Like tapeworms, most flukes are hermaphrodites, possessing both male and female reproductive organs. Many flukes, with the exception of blood flukes, require two intermediate hosts for their development; the first of these must be a mollusc, while the second is usually another cold-blooded animal, often an invertebrate, but sometimes a fish (Whitfield 1993).

Cotylurus sp.

Only one strigeid (Platylaelminthes: Digenea) has been previously recorded in either the avocets or stilts. The strigeid *Parastrigea mexicanus* has been detected in *Recurvirostra americana* (Coil 1957), but no mention was made of its location within the host. Members of the family Strigeidae parasitise aquatic birds, reptiles, and mammals that feed on aquatic amphibians, fish, and invertebrates (Yamaguti 1958, Cheng 1986); *Cotylurus* spp. are found in aquatic birds such as ducks, geese, and plovers (Cheng 1986).

Strigeids possess an indirect life cycle: free-swimming ciliated miracidia actively penetrate snails, where they undergo two sporocyst generations. These give rise to free-swimming furcocercous cercariae which penetrate a variety of aquatic animals (for example molluscs, amphibians, and reptiles), where they develop into metacercariae. The lifecycle is completed when the definitive host consumes the second intermediate host (Cheng 1986).

Pathology of the helminth parasites of *H. novaezelandiae*

Obviously these parasites provide a variety of potential sources of pathology. A summary of some effects of these parasites is outlined below.

Tapeworms are rarely considered to be pathogenic to their definitive host, although they can cause considerable harm to their intermediate host. The clinical signs of tapeworm infestation can include dullness, loss of appetite, excessive thirst, loss of weight, anaemia, and leg weakness (Arnall & Keymer 1975). The amount of nutrients absorbed by tapeworms is considered to be negligible, as is host absorption of parasite waste products and toxins, although the effect of these on the host is unknown (Whitfield 1993).

Capillaria spp. nematodes are more of a threat to their host. A light infestation is generally well-tolerated by the adults of most bird species, but can

result in signs of indigestion, dullness, poor appetite, and regurgitation of food. However, heavier infestations can result in inflammation of the gut so severe that parts of its lining mucous membrane may separate, producing slimy, yellow or bloody diarrhoea. Birds afflicted in this manner rapidly become mopy, emaciated and anaemic, and may die.

In fowl species, capillarids have been associated with high mortality rates in heavily infected flocks, while chronic light infestations have been associated with decreased weight gains and lower fecundity (Rickard & Pohl 1969). There is also indirect evidence to suggest that capillarids are also predisposing agents of other diseases in chickens. The removal of heavy infestations of *Capillaria* spp. can increase egg production and fertility, and reduce disease and mortality rates (Rickard & Pohl 1969).

Clinical signs of fluke infection may include general malaise; lack of appetite, thirst, and diarrhoea, to anaemia or jaundice. When the parasites infest the rectum and/or cloaca they may interfere with egg-laying. Heavy burdens may even obstruct the gut, killing the host (Arnall & Keymer 1975). *Acanthoparyphium* spp. are thought to be pathogenic if present in large numbers (I. Beveridge pers. comm.).

Some strigeid flukes are known to be harmful to the definitive host. The metacercariae of some species are known to cause pathology including meningeal brain tumours and blindness (*Diplostomum* spp.), while adults of *Cyathocotyle bushiensis* are known to damage the lining of the host caeca (Cheng 1986). *Cotylurus* spp. also damage the caeca and intestinal villi of their host.

The bird WBk-BkG appeared to suffer from the effects of the heavy digenean burden it carried. Often when parasites were removed there remained a circular imprint in the host mucosa at the site of attachment. There were signs of villial resorption in some areas, indicating the host was suffering from malnutrition.

4.6 THE PARASITES OF *HIMANTOPUS HIMANTOPUS LEUCOCEPHALUS*

The pied stilt *H. h. leucocephalus*, as already explained, shares the parasites *D. polymorphus* (Platyhelminthes: Cestoda), *Catatropis* sp. and *Acanthoparyphium* sp. (Platyhelminthes: Digenea), as well as the unidentified cyclophyllidean cestode (Platyhelminthes: Cestoda) with *H. novaezelandiae*. These will not be mentioned further here. In addition to these species, *H. h. leucocephalus* also possesses a number of parasites which were not found in *H. novaezelandiae*.

Polymorphus sp.

This is the one of the first records of an acanthocephalan being recovered from a stilt. Acanthocephalans are elongate endoparasites of the vertebrate alimentary tract, generally under 35 mm in length; the examples found in pied#2 were 16 mm or less. In most forms the body is cylindrical, and ta-

pered at both ends. The main characteristic of the Acanthocephala is the protrusible, armed proboscis with which they burrow into the wall of the host alimentary tract.

Acanthocephalans are dioecious (separate sexes), readily distinguishable on the basis of the copulatory apparatus; in addition females are larger than males and possess less well-developed spines on the body. Like cestodes, acanthocephalans lack a digestive tract, absorbing nutrients across the body wall. Fresh specimens show pronounced coloration. This is probably due to the absorption of lipochromoid taken up by the worm from the food of the host animal (Yamaguti 1963).

The classification of the Acanthocephala is an area of dispute (Petrochenko 1971, Amin 1985). These specimens recovered from the pied stilt were identified as *Falsifilicollis* sp. following Yamaguti (1963). Petrochenko called this genus *Parafilicollis*, and placed it in a different order from Yamaguti. More recently both were subsumed into the genus *Polymorphus*, subgenus *Profilicollis* (Amin 1985).

Acanthocephalans are highly fecund, and expelled eggs are highly resistant to environmental stresses, able to survive for several months in harsh conditions (Cheng 1986). The presence of an acanthocephalan infection can be diagnosed by examination of faecal smears.

All known Acanthocephala possess indirect life cycles. Eggs are passed out from the vertebrate host in the faeces.These eggs contain a partially developed acanthor; once ingested by an invertebrate intermediate host, development within the egg continues until the acanthor is fully developed. At this stage the acanthor ruptures from the egg, perforating the gut wall, and metamorphoses in the haemocoel. Metamorphosis involves transformation into a juvenile via an 'acanthella' stage. Once a recognisable adult morphology is achieved, the juvenile encysts (referred to as a cystacanth) until the intermediate host is eaten by an appropriate vertebrate host. At this time, the cystacanth exsheaths and develops to full sexual maturity, although in some cases a second intermediate host is required.

Acanthocephalans possess relatively low host-specificity (Hoberg 1986), can utilise a wide variety of paratenic hosts (both vertebrate and invertebrate), and are able to use the same host as both an intermediate and definitive host, depending on the conditions at the time of infection (Cheng 1986).

Pathology occurs through the physical damage caused by the proboscal hooks as the worm pierces and ruptures the lining of the host's intestine. In heavily infected hosts, the amount of nutrients absorbed by the parasites can be significant, and may harm the host. The number of Acanthocephala required to kill a host varies widely, from as few as one or two (Webster 1943) to more than 600 (Clark et at. 1958). In this case there were 21 acanthocephalans infecting the host.

The damage inflicted by the proboscis often elicits an inflammatory response, followed by the deposition of fibrous material (Crompton 1973); in the infected bird examined, nodules had formed on the serosa of the intestine. This may inhibit innervation and secretory and motor function of the intestine

(Petrochenko 1971). It also appears that in this bird the acanthocephalans have ruptured the gut and entered the abdomen; this is likely to cause peritonitis, and quite possibly the death of the host.

Crompton (1973) stated the belief that acanthocephalans and cestodes are likely to be competitors because both groups are generally restricted to the small intestine, and both share the same feeding mechanism, providing the potential for significant niche overlap. This is supported somewhat by the results of this study: the two pied stilts uninfected by Acanthocephala possessed 117 and 708 cestodes, respectively; the bird which was infected by Acanthocephala contained only 3 cestodes. However, more animals will have to be examined before this can be confirmed.

Cyclocoelinae

These parasites were recovered from the body cavity of pied stilt#2, on or near the air sacs. The two genera recovered, *Uvitellina* and *Wardianum* each measured approximately 8 mm in length and 2 mm in width. Being monostomes they lack a ventral sucker. It is unclear what effect, if any, these parasites have upon the host.

4.7 KNOWN PARASITES OF *HIMANTOPUS* SPP.

A list of the known helminth parasites of *Himantopus* spp. follows (Appendix 7); it cannot be considered an exhaustive list. In addition to the helminth parasites recorded below, Hinojos & Canaris (1988) identified six species of lice (*Actornithophiulus* spp., *Quadraceps* spp., and *Austromenopon himantopi*), and one Acarid nasal mite (*Rhinonyssus himantopus*) from *H. mexicanus.* This study recovered mites of the genera *Grallobia* and *Bychovskiata* (Analgoidea:Analgidae), and the louse species *Austromenopon himantopi, Quadraceps hennichrous,* and *Q. semifissus* from *H. h. leucocephalus.* There is a fourth louse species recorded from New Zealand stilts, *Saemundssonia platygaster*; this is rarely recorded, but obvious when present (R. Palma, pers. comm.). One black stilt suffered from an infection of *Giardia* (Mastigophorea: Zoomastigophorea).

The study undertaken by Hinojos & Canaris (1988) showed that the helminth fauna of *H. mexicanus* was more similar to that of *R. americana* than *H. h. himantopus.* Garcia & Canaris (1987) and Edwards & Bush (1989) provide useful checklists of the metazoan parasite species inhabiting *R. americana* in North America.

4.8 FUTURE RESEARCH

This report cannot be considered a complete examination of the parasite fauna of the black stilt. Further examination of viscera from black stilts would probably result in the.discovery of new parasite species, or may at least provide better specimens for the purposes of identification. For superior results, fresh material must be examined, preferably immediately after a natural mortality.

Experimental studies could be carried out to determine the path of infection for these parasites. Other research could be carried out to determine possible forms of treatment for parasites, and their effect on host condition, survival, and fecundity (bearing in mind the possibility of side-effects and the likelihood of reinfection).

It may also be possible in the future to determine if there are genetic or phenotypic markers which are linked with parasite and/or disease resistance. Some DNA analysis is already under way to determine the relationships between "pure" black and pied stilts, and their hybrids, and work has been planned to determine if genetic factors exist which affect the impact of parasites on their host.

5. Conclusions

Both the black stilt *H. novaezelandiae,* and the pied stilt *H. h. leucocephalus* possess a diverse parasite fauna. In most cases the effect on the host is minimal. In large numbers, however, the host bird can suffer damage to the intestinal villi, and villial resorption due to malnutrition and/or starvation.

Most, if not all the parasites infecting these birds possess an indirect life cycle, generally passing through one or more invertebrate intermediate hosts. Preventing infection in the wild is probably not feasible, nor is prophylactic treatment such as removal of the intermediate hosts. In captivity, unnaturally crowded conditions increase the possibility of infections occurring, but these infections can be treated.

It appears that trematodes, and possibly Acanthocephala, pose the greatest parasitic threat to the continued health and survival of the black stilt in the wild. The possibility of closing captive-reared birds against these parasites prior to release should be carefully considered.

Another concern is the presence of capillarid nematodes in the Ruataniwha aviary, especially in chicks who have never left the brooder enclosures. These infections can be successfully treated by anthelmintics, but the source of infection must also be isolated, or the birds will immediately become reinfected. This will be difficult in the larger outdoor aviaries, but should be relatively simple in the brooders. It is probable that the chicks were infected by eggs released in the faeces of an injured bird previously kept in that enclosure.

Managers could consider using particular brooder enclosures for the treatment of adult birds, once the bird is released, these enclosures could be cleaned more thoroughly than is usually the case. Particular attention should be paid to the outdoor part of the enclosure; eggs would be more likely to survive in gaps between rocks or the moister environment of the water bath.

Regular monitoring of faecal samples should detect the eggs of all the parasites mentioned in this report; note, however, that it will detect only reproducing adults. Different techniques of examining faeces may also be neces-

sary; different densities of solutions favour the recovery of different types of eggs using centrifugation, while some recommend using sedimentation techniques to detect the larger fluke eggs.

When a bird dies, efforts should be made to collect and correctly store parasites for future identification as soon as possible.

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8. Appendices

Appendix 1: The parasite burden of individual *Himantopus novaezelandiae.*

BIRD	LOCATION	PARASITE TYPE	IDENTITY	NUMBER
WY-RBk	Duodenum	Cestode	*D. polymorphus*	1
RW-WW	Unknown	Cestode	Unknown	1
WY-WY	Gizzard	Digenean	*Acanthoparyphium* sp.	1
	Jejunum	Cestode	Cyclophyllidean	1
		Digenean	*Acanthoparyphium* sp.	2
	Unknown	Cestode	Cyclophyllidean	4
WBk-RBk	Small intestine	Cestode	Cyclophyllidean	1
RY-WBk	Duodenum	Cestode	Cyclophyllidean	1
	Jejunum	Cestode	Cyclophyllidean	9
		Digenean	Echinostomatinae	1
WBk-WG	Large Intestine	Cestode	Cyclophyllidean	20
		Digenean	*Acanthoparyphium* sp.	10
WG-YR	Proventriculus	Cestode	Cyclophyllidean	22
		Digenean	*Acanthoparyphium* sp.	3
	Jejunum	Cestode	Cyclophyllidean	5
		Digenean	*Acanthoparyphium* sp.	1
WBk-BkW	Jejunum	Digenean	*Acanthoparyphium* sp.	1
	Unknown	Cestode	Cyclophyllidean	29
		Digenean	*Acanthoparyphium* sp.	20
WW-BkBk	Liver	Cestode	Cyclophyllidean	1
		Digenean	*Acanthoparyphium* sp.	1
	Gizzard	Cestode	Cyclophyllidean	4
	Proventriculus	Cestode	Cyclophyllidean	4
		Digenean	*Acanthoparyphium* sp.	3
	Duodenum	Cestode	Cyclophyllidean	10
		Digenean	*Acanthoparyphium* sp.	2
	Large Intestine	Digenean	*Cotylurus* sp.	8
	Caeca	Cestode	Cyclophyllidean	4
		Digenean	*Cotylurus* sp.	7
		Digenean	*Catatropis* sp.	3
	Unknown	Cestode	Cyclophyllidean	50
		Digenean	*Acanthoparyphium* sp.	24
		Digenean	*Cotylurus* sp.	6

WBk-WBk	Small Intestine	Digenean	Echinostomatinae	40
		Digenean	Microphallinae	66*
		Digenean	*Catatropis* sp.	4
	Unknown	Cestode	Cyclophyllidean	35
		Digenean	Echinostomatinae	331
		Digenean	*Catatropis* sp.	11
		Digenean	*Cotylurus* sp.	2
		Nematode	*Capillaria* sp.	1
WBk-BkG	Duodenum	Cestode	*D. polymorphus*	1
		Cestode	Cyclophyllidean	3
		Digenean	Echinostomatinae	1033
		Digenean	Microphallinae	90*
		Digenean	*Catatropis* sp.	1
	Small Intestine	Cestode	Cyclophyllidean	10
		Cestode	Unknown	1
		Digenean	Echinostomatinae	2986
		Digenean	Microphallinae	14615*
		Digenean	*Cotylurus* sp.	132
		Digenean	*Catatropis* sp.	35
		Nematode	*Capillaria* sp.	1
		Protozoan	*Giardia* sp.	**
	Caeca	Digenean	*Catatropis* sp.	43
		Digenean	Microphallinae	370*
		Digenean	*Cotylurus* sp.	20
		Digenean	Echinostomatinae	1
	Washings	Digenean	Echinostomatinae	6
		Digenean	Microphallinae	8
		Digenean	*Cotylurus* sp.	28
		Digenean	*Catatropis* sp.	1

* estimated figure
** no estimate made

Appendix 2: Summary of parasite fauna of *Himantopus novaezelandiae*.

Parasite Type	Prevalence (proportion of infected hosts)	Intensity (avg no. of parasites per infected host)	Abundance (avg. no. of parasites per potential host)
Cestoda*	0.91	21.50	19.55
Diplophallus polymorphus	0.18	1.00	0.18
Echinostomatinae**	0.73	557.625	405.55
Microphallinae	0.18	7574.50	1377.18
Catatropis sp.	0.18	47.50	8.64
Cotylurus sp.	0.27	67.67	18.46
Capillaria sp.	0.18	1.00	0.18
Giardia sp.	0.09	***	***

* not including *D. polymorphus*
** including *Acanthoparyphium* sp.
***no figures available

Appendix 3: The helminth fauna of *Himantopus himantopus leucocephalus*.

Bird	Location	Parasite Type	Parasite Identity	Parasite Number
Pied 1	Duodenum	Cestode	*D. polymorphus*	1
	Small Intestine	Cestode	Cyclophyllidean	65
	Intestine	Nematode	*Capillaria* sp.	1
	Large Intestine	Cestode	Cyclophyllidean	52
		Digenean	Psilostominae	2
	Caeca	Digenean	*Catatropis* sp.	6
Pied 2	Duodenum	Cestode	*D. polymorphus*	2
	Body Cavity	Digenean	*Wardianum* sp.	2
		Digenean	*Uvitellina* sp.	1
	Small Intestine	Cestode	Cyclophyllidean	3
		Acanthocephalan	*Polymorphus* sp.	21
Pied 3	Duodenum	Cestode	*D. polymorphus*	2
	Small Intestine	Digenean	*Acanthoparyphium* sp.	7
		Cestode	Cyclophillidean	702
	Large Intestine	Digenean	*Acanthoparyphium* sp.	6
		Cestode	Cyclophillidean	6
	Caeca	Digenean	*Catatropis* sp.	7

Appendix 4: Summary of parasite fauna of *Himantopus himantopus leucocephalus*.

Parasite Type	Prevalence (proportion of infected hosts in popn)	Intensity (avg. no. of parasites/infected host)	Abundance (avg. no. of parasites/potential host)
Cyclophyllidean Cestode	1.00	276.00	276.00
Diplophallus polymorphus	1.00	1.67	1.67
Acanthoparyphium sp.	0.33	13.00	4.33
Psilostominae	0.33	2.00	0.67
Catatropis sp.	0.67	6.50	4.33
*Polymorphus*sp.	0.33	21.00	7.00
Cyclocoelinae	0.33	3.00	1.00
Capillaria sp.	0.33	1.00	0.33

Appendix 5: Faecal egg counts (FECs) Indicating capillarid infection in captive black stilts in Ruataniwha Aviary, 1995,

Aviary Designation	Average FEC (eggs/g of faeces)
Brooder 3[1]	85.71
Brooder 6	68.75
Brooder 7[2]	18.12
Brooder 7[3]	0.21
Brooder 8[4]	38.24
View 1	3.09
View 2	203.26
Near 1	1.54
Near 2	5.34
Far 3	45.53
New 3	19.38
New 4[5]	3.99
New 4[6]	53.89
New 5[7]	23.19
New 5[8]	4.56
New 6	10.68
New 7	11.69

Explanatory notes: (1) Average FEC was calculated solely from 17/11 when 1 adult bird was present; later samples were taken when there were 4 chicks present (producing no capillarid eggs). (2) Refers to 29/9 and 17/11 when 1 juvenile was present. (3) Involves all subsequent samples, when 3 chicks were present. (4) The average FEC was calculated solely from 29/9 when 1 juvenile was present (later samples from other birds were negative). (5) Refers to 1/10 when 4 juveniles were present. (6) 2 juveniles present. (7) Refers to 1/10 when 3 juveniles were present. (8) 2 juveniles present.

Appendix 6: Results of faecal samples from Ruataniwha Aviary, Twizel.

Location	Date	Faecal Weight (g)	Egg Count	Capillarid FEC (eggs per gram)	No. of Birds	Bird Age
B 1	17/11	1.45	-	0	4	Chick
B 1	22/11	0.13	-	0	1	Chick
B 2	17/11	1.28	-	0	4	Chick
B 2	22/11	4.23	-	0	4	Chick
B 2	23/11	3.41	-	0	4	Chick
B 2	5/12	2.44	-	0	4	Chick
B 2	6/12	3.03	-	0	4	Chick
B 3	17/11	0.14	4	85.71	1	Adult
B 3	5/12	2.48	-	0	4	Chick
B 3	6/12	2.60	-	0	4	Chick
B 4	17/11	1.54	-	0	4	Chick
B 4	21/11	3.90	-	0	4	Chick
B 4	22/11	6.16	-	0	4	Chick
B 4	5/12	3.43	-	0	4	Chick
B 4	6/12	4.07	-	0	4	Chick
B 5	29/9	0.47	-	0	1	Adult
B 5	23/11	3.82	-	0	4	Chick
B 5	24/11	5.28	-	0	4	Chick
B 5	5/12	1.41	-	0	4	Chick
B 5	6/12	2.53	-	0	4	Chick
B 6	29/9	1.25	-	0	2	Juvenile
B 6	17/11	0.22	23	313.64	1	Juvenile
B 6	6/12	1.22	10	24.59	1	Juvenile
B 7	29/9	0.76	9	39.47	1	Juvenile
B 7	17/11	0.73	-	0	1	Juvenile
B 7	20/11	2.52	-	0	4	Chick
B 7	22/11	4.68	-	0	4	Chick
B 7	5/12	3.40	-	0	4	Chick
B 7	6/12	3.70	1	0.81	4	Chick
B 8	29/9	2.04	26	38.24	2	Adult
B 8	21/11	4.19	-	0	4	Chick
B 8	22/11	4.79	-	0	4	Chick
B 8	24/11	4.48	-	0	4	Chick
B 8	5/12	1.11	-	0	2	Chick
B 8	6/12	0.67	-	0	1	Adult
Vw 1	17/11	0.03	-	0	2	Adult
Vw 1	21/11	0.01	-	0	2	Adult
Vw 1	23/11	0.14	-	0	2	Adult
Vw 1	24/11	0.16	-	0	2	Adult
Vw 1	5/12	0.14	-	0	2	Adult
Vw 1	6/12	1.08	-	0	2	Adult
Vw 1	7/12	3.30	5	4.55	2	Adult
Vw 2	17/11	0.62	10	48.39	1	Adult
Vw 2	5/12	0.12	2	50	1	Adult
Vw 2	6/12	0.74	33	133.78	1	Adult
Vw 2	7/12	1.59	163	307.55	1	Adult

Nr 1	?	3.45	-	0	4	Adult
Nr 1	20/11	0.90	1	3.33	4	Juvenile
Nr 1	24/11	0.10	-	0	4	Juvenile
Nr 1	6/12	0.83	-	0	4	Juvenile
Nr 1	7/12	0.56	2	10.71	4	Juvenile
Nr 2	?	2.81	5	5.34	4	Juvenile
Nr 4	20/11	1.12	-	0	2	Adult
F 2	?	0.03	-	0	2	Juvenile
F 3	?	1.20	38	95	4	Juvenile
F 3	17/11	0.10	-	0	2	Adult
F 3	21/11	0.10	1	30	2	Adult
F 3	24/11	0.32	-	0	2	Adult
F 3	5/12	0.38	-	0	2	Adult
F 3	7/12	0.47	-	0	2	Adult
Nw 1	5/12	0.15	-	0	2/2	Ad./Juv
Nw 1	6/12	0.38	-	0	2/2	Ad./Juv
Nw 1	7/12	0.43	-	0	2/2	Ad./Juv
Nw 3	1/10	2.16	2	19.38	2	Adult
Nw 4	?	0.79	-	0	2	Adult
Nw 4	1/10	3.01	4	3.99	4	Juvenile
Nw 4	17/11	0.10	5	150	2	Juvenile
Nw 4	5/12	0.23	4	52.17	2	Juvenile
Nw 4	6/12	0.29	12	124.14	2	Juvenile
Nw 4	7/12	1.05	9	25.71	2	Juvenile
Nw 5	1/10	2.07	16	23.19	3	Juvenile
Nw 5	17/11	0.06	-	0	2	Juvenile
Nw 5	20/11	0.11	-	0	2	Juvenile
Nw 5	5/12	0.17	-	0	2	Juvenile
Nw 5	6/12	0.53	2	11.32	2	Juvenile
Nw 5	7/12	1.15	2.61	1	2	Juvenile
Nw 6	1/10	1.26	16	38.1	2	Juvenile
Nw 6	4/10	2.70	-	0	2	Juvenile
Nw 6	17/11	0.14	-	0	2	Juvenile
Nw 6	24/11	0.06	-	0	2	Juvenile
Nw 6	5/12	0.27	3	33.33	2	Juvenile
Nw 6	6/12	0.28	-	0	2	Juvenile
Nw 6	7/12	1.19	2	5.04	2	Juvenile
Nw 7	?	0.73	8	32.88	2	Juvenile
Nw 7	1/10	0.82	3	10.98	1	Adult
Nw 7	17/11	0.09	-	0	1	Adult
Nw 7	5/12	0.13	-	0	1	Adult
Nw 7	6/12	0.43	7	48.84	1	Adult
Nw 7	7/12	2.42	-	0	1	Adult

Appendix 7: The helminth fauna of *Himantopus* spp.

Parasite	Host Species	Reference
Class Cestoda		
Acoleus vaginatus	*H. h. meriodinalis*	Hinojos & Canaris 1988
	H. mexicanus	*In* Yamaguti 1959
	H. spinosus	*In* Yamaguti 1959
Acoleus hedleyi	*H. leucocephalus*	*In* Yamaguti 1959
Acoleus rugosus	*H. leucocephalus*	*In* Yamaguti 1959
Davainea himantopodis	*H. mexicanus*	Hinojos & Canaris 1988
	H. leucocephalus	*In* Yamaguti 1959
Dicranotaenia himantopodis	*H. melanopterous*	*In* Yamaguti 1959
	Himantopus spp.	*In* Yamaguti 1959
Dicranotaenia tsengi	*H. himantopus*	*In* Yamaguti 1959
Dilepis australiensis	*H. leucocephalus*	*In* Yamaguti 1959
Diplophallus polymorphus	*H. h. himantopus*	Ukoli 1965
	H. h. leucocephalus	This study
	H. h. meriodinalis	Hinojos & Canaris 1988
	H. mexicanus	Burt 1978
	H. novaezelandiae	T. Charleston pers. comm.
Eurycestus avoceti	*H. mexicanus*	Hinojos & Canaris 1988
Gidhaia (?) meridionalis	*H. rufipes*	*In* Yamaguti 1959
Gyrocoelia perversa	*H. himantopus*	*In* Yamaguti 1959
Gyrocoelia albaredai	*H. himantopus*	*In* Yamaguti 1959
Gyrocoelia australiensis	*H. leucocephalus*	*In* Yamaguti 1959
Hymenolepis himantopodis	*H. mexicanus*	Hinojos & Canaris 1988
Hymenolepis sp.	*H. mexicanus*	Hinojos & Canaris 1988
Infula burhini	*H. h. himantopus*	Ukoli 1965
	H. leucocephalus	*In* Yamaguti 1959
Infula macrophallus	*H. mexicanus*	Coil 1955
Pseudoshipleya farrani	*H. himantopus*	*In* Yamaguti 1959
Thomasitaenia nunguae	*H. h. himantopus*	Ukoli 1965

Class Digenea		
Acanthoparyphium sp.	*H. himantopus*	I. Beveridge pers. comm.
	H. h. leucocephalus	This study
	H. leucocephalus	I. Beveridge pers. comm.
	H. novaezelandiae	This study
Apophallus muehlingi	*H. himantopus*	*In* Yamaguti 1958
Athesmia heterolecithodes	*H. himantopus*	*In* Yamaguti 1958
Catatropis sp.	*H. h. leucocephalus*	This study
	H. novaezelandiae	This study
Cloacitrema michiganensis	*H. mexicanus*	McIntosh 1938
Cotylurus sp.	*H. novaezelandiae*	This study
Cyclocoelum lanceolatum	*H. mexicanus*	Yamaguti 1971[1]
Haematotrephus lanceolatum	*H.c andidus*	*In* Yamaguti 1958
	H. melanopterus	*In* Yamaguti 1958
	H. rubropterus	*In* Yamaguti 1958
Haematotrephus simile	*H. atropterus*	*In* Yamaguti 1958
Haematotrephus(?) adelphus	*H. leucocephalus*	*In* Yamaguti 1958
Hofmonostomum himantopodis	*H. mexicanus*	*In* Yamaguti 1958
Hyptiasmus magnoproles	*H. himantopus*	*In* Yamaguti 1958
Microphallinae	*H. novaezelandiae*	This study
Neivaia mutabile	*Himantopus* sp.	*In* Yamaguti 1958
Notocotylus sp.	*H. mexicanus*	Hinojos & Canaris 1988
Parastrigea mexicanus	*H. mexicanus*	Dubois & Macko 1972[1]
Promptenovum vanbeneoleni	*Himantopus* sp.	*In* Yamaguti 1958
Psilostominae	*H. h. leucocephalus*	This study
Spelotrema excellens	*Himantopus* sp.	*In* Yamaguti 1958
Stomylotrema ali-ibrahimi	*H. himantopus*	*In* Yamaguti 1958
Stomylotrema bijugum	*H. melanopterus*	*In* Yamaguti 1958
Tanaisia fedtschenkoi	*H. mexicanus*	Hinojos & Canaris 1988
	H. candidus	*In* Yamaguti 1958
Tanaisia (Tamerlania) valida	*H.h.melanurus*	*In* Yamaguti 1958
Uvitellina pseudocotylea	*H. candidus*	*In* Yamaguti 1958
Uvitellina sp.	*H. h. leucocephalus*	This study
Wardianum taxorchis	*H. leucocephalus*	*In* Yamaguti 1958
Zygotyle lunata	*H. mexicanus*	Yamaguti 1971[1]

Phylum Acanthocephala		
Centrorhynchus lancea	*Himantopus* sp.	*In* Yamaguti 1963
Polymorphus sp.	*H. h. leucocephalus*	This study

Phylum Nematoda

Amidostomum chevreuxi	*H. himantopus*	*In* Yamaguti 1961
	H. leucocephalus	Mawson 1968
Capillaria anatis	*H. mexicanus*	Hinojos & Canaris 1988
C. contorta	*H. mexicanus*	Hinojos & Canaris 1988
C. mergi	*H. mexicanus*	Hinojos & Canaris 1988
C. obsignata	*H. mexicanus*	Barus & Hernandez 1971[1]
C. triloba	*H. leucocephalus*	Mawson 1968
Capillaria sp.	*H. mexicanus*	Hinojos & Canaris 1988
	H. novaezelandiae	This study
	H. h. leucocephalus	This study
Chevreuxia americana	*H. mexicanus*	Hinojos & Canaris 1988
Chevreuxia revoluta	*H. himantopus*	*In* Yamaguti 1961
	H. candidus	*In* Yamaguti 1961
	H. mexicanus	Barus & Hernandez 1971[1]
Contracaecum spiculigerum	*H. leucocephalus*	Mawson 1968
Eustronglydes mergorum	*H. mexicanus*	Hinojos & Canaris 1988
Porrocaecum ensicaudatum	*Himantopus* sp.	*In* Yamaguti 1961
Porrocaecum heteroura	*Himantopus* sp.	*In* Yamaguti 1961
Splendidofilaria sp.	*H. mexicanus*	Hinojos & Canaris 1988
Strongyloides turkmenicus	*H. candida*	*In* Yamaguti 1961
Tropisurus nouveli	*H. himantopus*	*In* Yamaguti 1961
	H. mexicanus	Barus & Hernandez 1971[1]
	H. leucocephalus	Mawson 1968

Note: References marked with[1] are cited in Hinojos & Canaris 1988.