SECTION 2

PARASITES

OF

GAMEBIRDS
Introduction

In 1937 R.J. Ortlepp described the first worms from South African guineafowls. Since then, seven publications have appeared, approximately one every ten years. When Dr. Junker joined the Department of Veterinary Tropical Diseases, she started examining the helminths of guineafowls that had been collected over the years by Prof. I.G Horak, mainly from the KNP. The opportunity arose to extend the geographical range for this project to include hosts from Musina, Limpopo Province, in the northern part of the country, otherwise the date from these hosts would have been lost.

Infections with thorny-headed worms, tapeworms and roundworms are common in guineafowls and their helminth fauna is diverse. A total of 22 species were recovered from the alimentary canal, comprising eleven tapeworms, ten roundworms and a single thorny-headed worm. A single trematode (fluke) species was present in the liver.

I funded most of the project, and was also intimately involved with the collection of the helminths from Musina, and the preparation of the manuscripts. This part is divided into two chapters, one dealing with the descriptions of new species or re-descriptions of known ones, and the other dealing with the population dynamics of the worms. A check list of the parasites of guinea fowls is included in this section. The publications in the first chapter are listed in chronological order and those in the second one by subject and then chronologically.

**DESCRIPTIONS AND RE-DESCRIPTIONS OF PARASITES OF GAME BIRDS (P 429)**


**POPULATION DYNAMICS (P 463)**


JUNKER, K. & BOOMKER, J. 2007. A check list of the helminths of guineafowls (Numididae) and a host list of these parasites. *Onderstepoort Journal of Veterinary Research*, 74, 315-337.
CHAPTER 1

Descriptions and re-descriptions of parasites of gamebirds
**Mediorhynchus gallinarum** (Acanthocephala: Gigantorhynchidae) in Helmeted guineafowls, *Numida meleagris*, in the Kruger National Park, South Africa

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**ABSTRACT**


*Mediorhynchus gallinarum* was recovered from the small intestines of 36 of 50 Helmeted guineafowls sampled from August 1988 to May 1989. The intensity of infection ranged from 1–141 worms per host, with a mean intensity of 23.2 (± 34) and a median intensity of 5. The Wilcoxon-Mann-Whitney test revealed no significant differences between the mean worm burdens of male and female birds at the 5% level (P > 0.05). Slightly more female than male acanthocephalans were collected. The majority (63.4%) of females had eggs with fully-developed embryos, 9% had immature eggs, 21.2% had no eggs and the egg status of 6.4% could not be determined. No seasonal pattern of intensity of infection emerged from the data, but worm burdens were markedly higher after good rains in February 1989. South Africa constitutes a new geographic record for *M. gallinarum*.

**Keywords**: Acanthocephala, Helmeted guineafowls, *Mediorhynchus gallinarum, Numida meleagris*

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**INTRODUCTION**

The guineafowl family Numidiidae is widespread and common in the Afrotropical region, where they utilize a wide variety of habitats ranging from dense rainforest to semi-desert. Of the four genera of guineafowls, *Agelastes, Acryllium, Guttera* and *Numida*, the last-named’s helmint fauna has been studied the most extensively. There are few references to cestodes and nematodes from *Guttera* (Crested guineafowl) and even fewer from *Acryllium* (Vulturine guineafowl) (Yamaguti 1959, 1961, 1963; Ortlepp 1963; Schmidt 1986). The authors are aware of only one publication pertaining to acanthocephalans from guineafowls other than *Numida*, namely *Mediorhynchus taenia-tus* (syn. *Empodius segmentatus*) from *Guttera pucherani edouardi* in the former Belgian Congo (Southwell & Lake 1939).

Many of these studies were conducted in North and West Africa, where guineafowls are commercially reared as a source of protein and necessitated a more detailed knowledge of the birds and their parasites (Hodasi 1976). The possibility of wild guineafowls as alternative or reservoir hosts for helminths of domestic chickens and vice versa, also required investigation (Fatunmbi & Olufemi 1982). In South Africa three studies concerning the gastrointestinal worms of Helmeted guineafowls have been conducted, one each in the Eastern Cape Province, the Kimberley area in the Northern Cape Province and in the surroundings of Pretoria in Gauteng Province (Saayman 1966; Crowe 1977; Verster & Ptasinska-Kloryga 1987).
The present paper describes a survey in which the acanthocephalan burdens of free-ranging guinea-fowls in the southern part of the Kruger National Park (KNP) were determined, as well as those of “scavenger” guinea-fowls frequenting the refuse dump at the Skukuza tourist rest camp. Some scanning electron micrographs and measurements intended to supplement the descriptions of Mediorhynchus gallinarum given by Bhalerao (1937) and Nath & Pande (1963) are included.

MATERIAL AND METHODS

Study site

The KNP is situated in the eastern part of Limpopo Province and the north-eastern part of Mpumalanga Province. It encompasses an area of 1948528 ha. The survey region in the southern part of the park (South of 24°50' S; Skukuza 24°50 S, 31°35' E) comprises vegetation classified as Lowveld Sour Bushveld and Arid Lowveld (Acocks 1975). Helmeted guinea-fowls are present throughout the study area. The refuse dump at Skukuza tourist rest camp offers easy foraging and attracts hundreds of birds (Horak, Spickett, Braack & Williams 1991).

Survey birds

Each month from August 1988 to May 1989, two Helmeted guinea-fowls on or near the refuse dump at Skukuza, and three at other sites in the southern part of the park were shot. An effort was made to shoot only adult birds, but two of the total of 50 birds were 7 to 10-month-old sub-adults. No birds were collected in March 1989, but of the ten guineafowls that were examined in February 1989, five were sampled in the beginning of the month and five were shot on 28 February. The latter birds are listed as hosts examined in March 1989.

Parasite collection

After the birds had been shot their carcasses were transported to the laboratory at Skukuza. The entire viscera were removed and placed in separate labelled bottles in which they were stored in 10% buffered formalin. During 2005 and 2006 the lungs, crop, small intestine (SI) and the caecum-colon (CC) were removed from the bottles and separated. Macroscopically visible helminths were recovered from each of the organs and transferred to 70% ethanol and examined under a stereoscopic microscope for the presence of endoparasites.

Following the procedures described by Gibbons, Jones & Khalil (1996) some acanthocephalans were stained with aqueous aceto alum carmine and mounted in Canada balsam, while others were cleared in Hoyer’s medium.

Specimens for scanning electron microscopy (SEM) were dehydrated through graded ethanol series and critical point dried from 100% ethanol through carbon dioxide. They were mounted on viewing stubs and sputter-coated with gold. The photography was done using a Hitachi S-2500 scanning electron microscope.

In order to investigate differences in the worm burdens of male versus female hosts, the Wilcoxon-Mann-Whitney test for independent samples was used to compare the mean worm burden of the two groups at the 5% level (P > 0.05) (Thrusfield 1995).

RESULTS

Mediorhynchus gallinarum (Bhalerao, 1937) van Cleave, 1947 (Fig. 1 & 2)

MORPHOLOGY

Mediorhynchus gallinarum is characterized by a so-called acanthopseudoannelid holdfast, an attachment mechanism involving proboscis hooks as well as pseudo-segmentation of the body, considered typical for Moniliformidae and some of the Giganthorhynchidae (Petrochenko 1956).

The trunk is elongate and tapers slightly towards the posterior end. The prominence of the pseudo-segmentation is influenced by the extent of muscle contraction: it can be conspicuous, as in craspedote cestodes or nearly smooth as in sebekiid pentastomes. Pseudo-segmentation also appears to be more pronounced in older, larger specimens. The most anterior part, and in some specimens the caudal tip, is usually unsegmented. Annulus counts range from 52 in a 48-mm-long male to 76 in a 61-mm-long female. In some specimens muscle contraction creates a neck-like zone behind the proboscis, which is absent in relaxed specimens. The protoboscis is almost conical in shape and the teloboscis is trapezoid.

The hooks on the protoboscis are arranged in 18–20 roughly longitudinal rows of 4–5 hooks each. The total length of the hooks, including their roots, ranges from 0.048–0.076 mm, with the hooks in the top
row usually the shortest. Two longitudinal grooves extend from the base of the hook blade to its tip. The rootless spines on the teloboscis vary in length from 0.032–0.047 mm.
The lemnisci are slender and approximately 2.5–2.9 times longer than the proboscis receptacle. In some specimens up to six nuclei, possibly more, per lemniscus were counted. Lemniscus length ranged from 2.09 mm in a 13-mm-long male to 3.47 mm in a 50-mm-long male, but the length of the lemnisci did not necessarily increase with body length. The lemnisci ranged from 0.191–0.343 mm in width. No obvious differences were evident between males and females.

**Females:** The average body length is 32 ± 17 mm \((N = 423)\), ranging from 4–110 mm, with a median of 35 mm. The maximum body width varies from 0.6–4 mm \((\text{mean} = 1.4 \pm 0.6 \text{ mm})\), with large gravid females, especially when the body was contracted, the widest.

The length of the proboscis receptacle ranges from 0.701 mm in a 6-mm-long female to 1.19 mm in a 48-mm-long female \((\text{mean} = 1.0 \pm 0.162 \text{ mm})\). The width of the proboscis receptacle varies from 0.296–0.554 mm, with an average of 0.399 ± 0.072 mm. Eggs with a compact, granular outer shell and fully developed embryos measure on average 0.049 mm \((\text{range:} 0.043–0.052 \text{ mm})\) in width and 0.079 mm \((\text{range:} 0.070–0.086 \text{ mm})\) in length. The embryo itself is 0.054 mm \((\text{range:} 0.047–0.058 \text{ mm})\) long and 0.025 mm \((\text{range:} 0.021–0.028 \text{ mm})\) wide.

**Males:** The mean body length is 25 ± 14 mm \((N = 284)\) with a range of 3–70 mm. The median is 25 mm. The average maximum width ranges from 0.5–2.8 mm \((\text{mean} = 1.1 \pm 0.4 \text{ mm})\) and the measurements taken from 14 males are presented in Table 1.

**FIG. 2** *Mediorhynchus gallinarum*

A. Anterior part displaying the arrangements of hooks on the protoboscis and smaller spines on the teloboscis; x 150. B. Same specimen rotated 180°; x 130. C. En face view. The two apical pores of the apical organ are visible (arrows); x 500. D. Close-up of a hook partially retracted into the surrounding pouch. Note the grooved surface of the hook; x 2000.
TABLE 1  Morphological data of *Mediorhynchus gallinarum* males recovered from Helmeted guineafowls in the Kruger National Park. All measurements given in micrometer unless otherwise indicated.

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<th>TBL</th>
<th>TBA</th>
<th>TBP</th>
<th>RLL</th>
<th>LLL</th>
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<td>nm</td>
<td>nm</td>
<td>nm</td>
<td>2708</td>
<td>258</td>
<td>nm</td>
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<td>nm</td>
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</tbody>
</table>

ATL = Anterior testis length  
CBL = Copulatory bursa length  
CGL = Cement gland area length  
LLL = Left lemniscus length  
LLW = Left lemniscus width  
nm = Not measured  
PBL = Protoboscis length  
PRL = Proboscis receptacle length  
PRW = Proboscis receptacle width  
PTL = Posterior testis length  
RLL = Right lemniscus length  
RLW = Right lemniscus width  
SVL = Seminal vesicle length  
TBA = Width of anterior border of teloboscis  
TBL = Teloboscis length  
TBP = Width of posterior border of teloboscis
Mediorhynchus gallinarum in Helmeted guineafowls, Numida meleagris, in South Africa

Measurements of the proto- and teloboscis were only taken from specimens in which these features were fully extended.

The oblong shaped testes are located in the posterior third of the body. In young males the sexual organs are clustered in the caudal region. Testes move anteriorly and the gap between the anterior and posterior testis widens as the males grow larger.

TAXONOMIC REMARKS

Harris (1973) described Mediorhynchus selengensis from Francolinus leucoscepus in Kenya. In their revision of the genus Mediorhynchus Schmidt & Kuntz (1977) classified this species as a junior synonym of M. gallinarum to the description of Harris (1973). Vercruysse, Harris, Bray, Nagalo, Pangui & Gibson (1985) chose to retain the name M. selengensis for acanthocephalans collected from guineafowls in Burkina Faso until such time as Asian and African material could be more thoroughly compared.

The main difference between our specimens and those of Harris (1973) is the number of proboscis spines. Harris (1973) described only two to three spines per row, whereas our specimens carry five to seven spines per row. Nevertheless, Harris’ (1973) illustration suggests that more spines per row may be present. The remaining measurements overlap to a large extent. Not having examined Harris’ specimens we would tend to agree with Schmidt & Kuntz (1977) and assign our specimens to M. gallinarum.

EPIDEMIOLOGY

Small numbers of acanthocephalans were recovered from the CC of six guineafowls, and these have been included in the SI counts.

The prevalence of infection with M. gallinarum was 72%, i.e. of the 50 hosts examined 36 harboured parasites. A total of 846 worms were recovered from the 36 hosts. Worm burdens were usually low, with a median intensity of 5, and the intensity of infection ranged from 1–141, with a mean intensity of 23.2 ± 34. Hosts infected with less than 10 acanthocephalans accounted for 58% of the total host population, hosts with a burden ranging between 10 and 20 parasites comprised 14% and in 28% of the guineafowls the worm burden exceeded 20. The mean intensity of infection of male and female birds was 19.8 ± 36.4 and 27 ± 31.8, respectively. No significant differences between the mean intensities of infection at the 5% level, with a two-tailed P value of 0.2892, were observed with the Wilcoxon-Mann-Whitney test.

The mean intensity of infection with male and female acanthocephalans was 9 and 13, respectively, and the sex ratio favoured females (55.9% versus 37.7%). The small number of males and females recovered from the majority of hosts did not provide an adequate sample size for statistical testing. However, in nine of 10 hosts in the group harbouring more than 20 acanthocephalans, female parasites outnumbered males and constituted 60% of the adult parasites in this group. Immature M. gallinarum comprised a mere 0.4% of the infrapopulation, and the gender of 6% of the acanthocephalans could not be determined because they were poorly preserved.

The uteri of the majority of the females (63.4%) contained mature eggs, 9% only immature eggs and 21.2% contained no eggs. The status of eggs in the uteri of 6.4% of females could not be determined.

The mean intensities of infection during the various months of collection are presented in Table 2, and the seasonal variation in infection in Fig. 3. Infection peaked during late summer and autumn, but because the sampling period did not cover a full year the seasonality of infection cannot be determined with certainty.

TABLE 2 The mean numbers of Mediorhynchus gallinarum recovered from 50 Helmeted guineafowls in the Kruger National Park

<table>
<thead>
<tr>
<th>Collection date</th>
<th>Mean intensity of infection (range)</th>
<th>No. of birds infected/examined</th>
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<tr>
<td>Aug. 1988</td>
<td>7.8 (1–14)</td>
<td>4/5</td>
</tr>
<tr>
<td>Sep. 1988</td>
<td>3.5 (1–5)</td>
<td>4/5</td>
</tr>
<tr>
<td>Oct. 1988</td>
<td>2.5 (1–4)</td>
<td>2/5</td>
</tr>
<tr>
<td>Nov. 1988</td>
<td>4.0 (4)</td>
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<td>Jan. 1989</td>
<td>4.3 (2–8)</td>
<td>4/5</td>
</tr>
<tr>
<td>Feb. 1989</td>
<td>41.0 (3–67)</td>
<td>5/5</td>
</tr>
<tr>
<td>Mar. 1989</td>
<td>74.4 (5–141)</td>
<td>5/5</td>
</tr>
<tr>
<td>Apr. 1989</td>
<td>26.5 (4–52)</td>
<td>4/5</td>
</tr>
<tr>
<td>May 1989</td>
<td>25.0 (2–48)</td>
<td>2/5</td>
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</tbody>
</table>
Few hosts were examined from the different localities in the southern part of the KNP on the various collection dates. Consequently, data pertaining to differences in the mean intensities of infection at the different sites versus the dump at Skukuza should not be overinterpreted. However, in February and March 1989 the worm burdens of all six guineafowls sampled along the Lower Sabie Road were markedly larger than the overall mean intensity of 23.2, with individual burdens consisting of 51, 37, 48, 141, 86 and 119 worms. In contrast, the acanthocephalan burdens of four guineafowls sampled at the dump at Skukuza at the same time varied from far below to far above average, namely 76, 3, 5 and 21.

DISCUSSION

We have not been able to establish whether the grooves on the surface of the hooks of *M. gallinarum* are a unique feature of this parasite or genus or whether it is a characteristic with a wider taxonomic significance.

No grooves were seen on SEM photographs taken by Taraschewski, Sagani & Mehlhorn (1989, cited by Taraschewski 2000) of hooks of *Echinorhynchus truttae* and *Moniliformis moniliformis*. The function of these structures is open for speculation. They might simply improve the holdfast of the hooks in the surrounding host tissue. Alternatively, the increased surface area could assist in the uptake as well as secretion of substances. Polzer & Taraschewski (1992, cited by Taraschewski 2000) discuss the discharge of penetration enzymes through the hook pores of *Pomphorhynchus laevis*.

The majority of acanthocephalans in this study were recovered from the SI and only a small number were found in the CC. While the caecum is a predilection site of some acanthocephalans (De Buron & Nickol 1994), we are not sure whether our findings represent a true distribution pattern or are the result of contamination during the processing of the hosts. There is also the likelihood of post mortem migration. *Mediorhynchus gallinarum* parasitizing domestic fowls in Papua and New Guinea were confined to the mid and lower small intestine (Talbot 1971), and Crowe (1977) recovered *Mediorhynchus taeniatus* only from the small intestine of Helmeted guineafowls. We are not aware of any controlled studies concerning the site preferences of any members of the genus *Mediorhynchus*.

Morphologically *M. gallinarum* falls into the category of acanthocephalans with a short neck and the associated shallow mode of attachment as described by Taraschewski (2000). Histological examination of *M. gallinarum* in domestic fowls revealed that their attachment rarely penetrated below the muscularis mucosa (Nath & Pande 1963; Talbot 1971). Taraschewski (2000) states that non-perforating species remain mobile and can alter their point of attachment. They do not occupy extra-intestinal sites within their hosts. According to Kennedy & Lord (1982) acanthocephalans can successfully utilize a much larger region of the digestive tract than their predilection site, and at high levels of infection are known to expand their distributional range within the alimentary canal (Taraschewski 2000). The hosts from which acanthocephalans were collected from both the CC and the SI in the present study carried relatively low worm burdens (4, 4, 8, 36 and 67) and infections involving considerably higher intensities were restricted to the SI in some of the other hosts. In view of the above, post mortem migration appears the more probable explanation for the specimens we found in the CC.

Only a small percentage of *M. gallinarum* were immature, and this can be attributed to the short period of time required by the cystacanth, once ingested by a final host, to develop into an adult. In experimental infections of several species of woodpeckers with cystacanths of *Mediorhynchus centurorum* the mean prepatent period was 35 days (Nickol 1977).

More than 60% of the female *M. gallinarum* examined during this study contained eggs with shelled embryos. This is contrary to Van Cleave’s (1947a) report that fully grown female specimens of *Mediorhynchus* spp. recovered from a variety of birds invariably lacked embryonated eggs. His speculation that sterility might be seasonal, is not supported by
**Mediorhynchus gallinarum** in Helmeted guineafowls, *Numida meleagris*, in South Africa

Our data in the case of *M. gallinarum*. We do, however, accept his view on sterility possibly being due to the absence of males or an indication of the unsuitability of a certain bird species as final host.

One of the hosts examined in this study was infected by a single large (6.5 cm) female containing only immature eggs, which in view of the many gravid females recovered from other guineafowls, we interpret as lack of fertilization. A relatively large female in another bird contained sterile eggs, despite the presence of a male. In the latter case it is possible that the male was acquired during a more recent infection. In pentastomid parasites copulation occurs when the uterus of the female is undeveloped and the sexes are of approximately equal size. As the uterus develops it becomes impossible for the male to deposit sperm in the female spermathecae (Riley 1986). As in pentastomes insemination in the acanthocephala is possibly restricted to a short critical period during female development. Riley (1986) suspects that the absence of male pentastomids retards female development. This does, however, not seem to be the case in the Acanthocephala.

Van Cleave (1947a), who examined collections of the genus *Mediorhynchus* from various parts of the world, found the intensity of infection to be extremely low in many avian hosts. Often a single worm was present. He saw this as an indication of the absence of reservoir hosts, reasoning that the normal final hosts of *Mediorhynchus* would not feed on possible reservoir hosts, i.e. animals large enough to consume the intermediate host (Van Cleave 1947a). Van Cleave (1947a) saw this as an indication of the absence of reservoir hosts, reasoning that the normal final hosts of *Mediorhynchus* would not feed on possible reservoir hosts, i.e. animals large enough to consume the intermediate host (Van Cleave 1947a).

Given the catholic diet of guineafowls, this argument would not be valid for this particular final host. Since nothing is known about the intermediate hosts of *M. gallinarum* in South Africa, it would be difficult to speculate whether the higher mean intensity of infection is due to the inclusion of reservoir hosts in the life-cycle, or is due to a wide range of possible intermediate hosts, or both.

According to Petrochenko (1956) most individual hosts harbour a single acanthocephalan species only, even if the particular host species serves as host for several different species of acanthocephalans. Our own data and the literature pertaining to guineafowls support this. *Mediorhynchus taeniatus* was the only acanthocephalan present in 42 guineafowls from Nigeria and 13 guineafowls from South Africa (Fabiyi 1972; Verster & Ptasinska-Kloryga 1987). Saayman (1966) recovered *Mediorhynchus* *numidae* (syn. *Empodisma numidae*) from 14 guineafowls, and Vercruysse *et al.* (1985) report only *M. selengensis* Harris, 1973 from guineafowls in Burkina Faso.

**Mediorhynchus taeniatus** has also been recorded from South Africa by Verster & Ptasinska-Kloryga (1987). This species differs from *M. gallinarum* in that it has less than 40 hooks and that the lemnisci are not much longer than the proboscis receptacle (Meyer 1932; Schmidt & Kuntz 1977). *Numida meleagris* shot in the Pretoria area (Gauteng Province) had burdens reaching up to 22 worms per bird, with a mean of 1.7. The prevalence of *M. taeniatus* was 27% (Verster & Ptasinska-Kloryga 1987).

Saayman (1966) recovered *M. numidae* from Helmeted guineafowls in the Eastern Cape Province. This parasite is characterized by the absence of pseudo-segmentation and possesses only three hooks per row (Schmidt & Kuntz 1977). Intensity of infection ranged from one to 27 worms (mean = 11.5) and the prevalence was 39%. It is interesting that in three different geographical regions in which guineafowls were examined in South Africa the genus *Mediorhynchus* is represented by three different species and that only one species was recovered per region. This, as well as the differences in prevalence and intensity of infection, might be the result of different climatological conditions, vegetation types and resulting differences in the arthropod fauna, suspected of being intermediate hosts, present at the three study sites.

While no pattern of seasonal abundance emerged from our data, worm burdens were markedly higher in guineafowls collected during February, April and May 1989. This coincides with the exceptionally high rainfall of 286.3 mm in February (Penzhorn, Horak, Spickett & Braack 1991). The annual mean rainfall for Skukuza recorded by Gertenbach (1980) is 546.3 mm. The high rainfall probably resulted in a rapid increase of insect and other arthropod populations ensuring a ready supply of intermediate hosts for *M. gallinarum* and a convenient source of infection for the final hosts.

All guineafowls are highly terrestrial and feed exclusively on the ground. They are omnivorous oppor-
tunists and the composition of their diet at any given moment is determined by the local abundance of the various food items (Del Hoyo, Elliot & Sargatal 1994). The overall diet is very varied and consists of plant matter such as leaves, roots, bulbs, seeds, fruits and flowers, as well as grit and animal food (Saayman 1966). The latter, while including a few vertebrates like small frogs, toads and lizards, is mainly made up of a wide array of insects, small moluscs, arachnids and millipedes.

About 12% of the annual volume of food consumed by guineafowls consists of invertebrates, but Helmeted guineafowls, in particular, prefer to feed on insects when these are sufficiently abundant. The crop of a single Helmeted guineafowl yielded 5 100 insects when these are sufficiently abundant. The latter, while including a few vertebrates like small frogs, toads and lizards, is mainly made up of a wide array of insects, small moluscs, arachnids and millipedes.

There is a marked individual variation in feeding intensity of guineafowls, and crop contents have been observed to vary considerably between individual members of the same flock (Saayman 1966). This might explain why some of the hosts from the same locality examined at the same time carried very low worm burdens while others harboured large numbers of acanthocephalans. It was especially evident in the guineafowls collected in February/March 1989 from the dump in Skukuza. Overdispersion is a well described phenomenon in parasitology, and amongst others, it is thought to reflect certain traits of individual hosts, such as behavioural differences or immune reactions (Horak & Boomker 2000).

Penzhorn et al. (1991) observed that the guineafowls foraging at the dump were able to maintain good body condition despite the fact that the mass of food-intake compared with veld-collected birds was low. They concluded that the refuse dump provided a rich source of food. The mean intensity of infection increased markedly in the free-ranging guineafowls after the good rains in February 1989, but not to the same extent in the birds frequenting the refuse dump. It therefore appears that the good quality diet that is continuously available for these “scavenging” guineafowls buffers the effects that environmental changes have on the free-ranging guineafowls in the rest of the study area, and that they are not as reliant on arthropods to supply their diet and hence are less likely to ingest the possible intermediate hosts of the acanthocephalans. Unfortunately, little is known about the intermediate hosts in the life cycle of Mediorhynchus. Mediorhynchus grandis develops to the infective stage in a variety of grasshoppers in the USA (Van Cleave 1947b) and it would certainly be interesting to investigate potential intermediate hosts for M. gallinarum.

Talbot (1971) reports that even in heavy infections of domestic fowls in Papua and New Guinea with M. gallinarum little evidence of severe pathology was seen during the histological examination and he concluded that M. gallinarum is not a parasite of major economic importance.

Louw, Horak, Meyer & Price (1993) when determining the lice burdens of the guineafowls examined in this study found no overt signs of distress when observing the birds prior to collection, and Penzhorn et al. (1991) found no indication of emaciation during their morphometric studies of the same birds. Crowe (1977) did not see any signs of gross pathological conditions in 206 Helmeted guineafowls, which amongst other helminth parasites, carried acanthocephalans. It would thus appear that guineafowls, at least under natural conditions, tolerate infections with Mediorhynchus well. One must, however, bear in mind, that, although not primary pathogens, these parasites compete with their host for nutrients and in the case of heavy infections might well be detrimental to the host’s condition.

ACKNOWLEDGEMENTS

The authors thank the Board of Trustees, South African National Parks, for making the guineafowls available and Prof Ivan Horak for collecting their viscera. Mr Ryno Watermeyer has provided technical assistance and the scanning electron micrographs were taken by Mr John Putterill. Mr Dean Reynecke did the statistical analyses. This study was supported by a Claude Leon Foundation Postdoctoral Fellowship grant to the senior author.

REFERENCES


**INTRODUCTION**

The genus *Tetrameres* Creplin, 1846 are cosmopolitan parasites, infecting a variety of aquatic and terrestrial avian hosts. Females are usually located in the proventricular glands, and the males are found free in the lumen of the proventriculus (Anderson 1992).

Several *Tetrameres* species have been recorded from the African continent, of which *Tetrameres fissionispira* (Diesing, 1861) Travassos, 1914 that parasitises ducks, pigeons and domestic chickens and *Tetrameres americana* Cram, 1927 that parasitises domestic chickens, turkeys and quails are the most commonly reported ones (Permin, Magwisha, Kassuku, Nansen, Bisgaard, Frandsen & Gibbons 1997; Poulsen, Permin, Hindsbo, Yelifari, Nansen & Bloch 2000).

*Tetrameres coccinea* (Seurat, 1914) Travassos, 1914 from the Greater flamingo, *Phoenicopterus ruber* Linnaeus, 1758, Cattle egret, *Bubulcus ibis* Linnaeus, 1758 and Eurasian spoonbill, *Platalea leucorodia* Linnaeus, 1758, as well as *Tetrameres ihuilleri* (Seurat, 1918) from the Rock partridge, *Alectoris graeca* (Meisner, 1804) and the Stock pigeon, *Columba oenas* Linnaeus, 1758 were recorded from Algeria (Yamaguti 1961). *Tetrameres nouveli* (Seurat, 1914) Travassos, 1914 was present in the Black-winged stilt, *Himantopus himantopus* Linnaeus, 1758 in Algeria (Yamaguti 1961), and in Nigeria *Tetrameres plectropteri* Thwaites 1926 was found in the Spur-winged goose, *Plectropterus gambensis* (Linnaeus, 1766) (Yamaguti 1961).

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**Tetrameres numida** n. sp. (Nematoda: Tetrameridae) from Helmeted guineafowls, *Numida meleagris* (Linnaeus, 1758), in South Africa

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**ABSTRACT**


*Tetrameres numida* n. sp. from the proventriculus of Helmeted guineafowls, *Numida meleagris*, in South Africa is described from eight male and four female specimens. The new species shares some characteristics with other *Tetrameres* species, but can be differentiated by a unique combination of characters. It bears two rows of cuticular spines extending over the whole length of the body and possesses two spicules. The left spicule measures 1699–2304 μm and the right one 106–170 μm. Caudal spines are arranged in three ventral and three lateral pairs and the tail is 257–297 μm long. Diagnostic criteria of some of the previously described species of the genus *Tetrameres* from Africa and other parts of the world have been compiled from the literature and are included here.

**Keywords**: Helmeted guineafowls, nematodes, *Tetrameres numida*

**INTRODUCTION**

The genus *Tetrameres* Creplin, 1846 are cosmopolitan parasites, infecting a variety of aquatic and terrestrial avian hosts. Females are usually located in the proventricular glands, and the males are found free in the lumen of the proventriculus (Anderson 1992).

Several *Tetrameres* species have been recorded from the African continent, of which *Tetrameres fissionispira* (Diesing, 1861) Travassos, 1914 that parasitises ducks, pigeons and domestic chickens and *Tetrameres americana* Cram, 1927 that parasitises domestic chickens, turkeys and quails are the most commonly reported ones (Permin, Magwisha, Kassuku, Nansen, Bisgaard, Frandsen & Gibbons 1997; Poulsen, Permin, Hindsbo, Yelifari, Nansen & Bloch 2000).

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Both *Tetrameres paradisea* Ortlepp, 1932 and *Tetrameres prozeskyi* (Ortlepp, 1964) were described from South African hosts. *Tetrameres paradisea* was recovered from a Stanley’s crane, *Anthropoides paradisea* (Lichtenstein, 1793) (Ortlepp 1932), and *T. prozeskyi* occurred in Red-billed hornbills, *Tockus erythrorhynchus* (Temminck, 1823) and Southern Yellow-billed hornbills, *Tockus leucomelas* (Lichtenstein, 1842) (= *Tockus flavirostris leucomelas*), respectively (Ortlepp 1964).

Previous records of *Tetrameres* spp. from guineafowls pertain mostly to studies in North and West Africa, *Tetrameres fissispina* being recorded from Helmeted guineafowls in these countries (Fabiyi 1972; Vercruysse, Harris, Bray, Nagalo, Pangui & Gibson 1985). Appleton (1983) found *Tetrameres* sp. females in Crested guineafowls, *Guttera edouardi* (Hartlaub, 1867) (= *Guttera pucherani*), in Natal (now KwaZulu-Natal Province), South Africa, but because males were not present, the species could not be determined.

We here describe a new species of the genus *Tetrameres* from Helmeted guineafowls in South Africa for which we propose the name *Tetrameres numida* n. sp.

With regards to the classification of the genus *Tetrameres* we have followed that of Chabaud (1975), placing the genus into the subfamily Tetramerinae Railliet, 1915 within the family Tetrameridae Travassos, 1914, which is one of four families comprising the superfamily Habronematoidea. At the time the genus had been divided into the subgenera *Tetrameres* s. str., *Gynaecophila* Gubanov, 1950, *Petrowimeres* Chertkova, 1953 and *Gubernacules* Rasheed, 1960. Chabaud (1975), arguing that this division could lead to errors and bore little phylogenetic significance, chose not to retain these, but divided the genus *Tetrameres* into the two subgenera *Tetrameres* (Tetrameres) Crepin, 1846 and *Tetrameres* (*Microtetrameres*) Travassos, 1915. In the light of new findings, especially concerning the morphology of adults and larval stages of these two subspecies, Anderson (1992), while retaining their position within the subfamily, recognized *Tetrameres* Crepin, 1846 and *Microtetrameres* Travassos, 1915 as two distinct genera, a generic classification that had been suggested by Skrjabin (1969). We adopt his view in the present paper.

**MATERIAL AND METHODS**

Fifteen Helmeted guineafowls, *Numida meleagris* (Linnaeus, 1758), were collected on a farm 60 km to the west of Musina (Messina), Limpopo Province, South Africa (22°22.139' S, 29°30.399' E) between July 2005 and November 2006. Ten of these were mature guineafowls and five were young birds, about 6–10 months old (Siegfried 1966).

Eight male *Tetrameres* sp. were recovered from the proventriculus, where they occurred free in the lumen and four females were dissected from the proventricular glands. Two guineafowls harboured a single male each, two hosts harboured two and three males respectively, and from a single host one male and four females were recovered. All hosts were mature guineafowls. The worms were fixed in 70% ethanol and cleared in lactophenol for identification. All measurements, unless otherwise indicated, are in micrometres.

**DESCRIPTION**

*Tetrameres numida* n. sp. (Fig. 1–3; Tables 1, 2)

With characters of the genus. Sexual dimorphism marked.

**MALE:** Body elongated, tapering towards both ends, posteriorly to a tail with a short, pointed tip. Cuticle with fine transverse striation and longitudinal cuticular grooves. Total length 4.3–4.5 mm; maximum width 0.16–0.17 mm. Inconspicuous lateral alae extending down the length of the body; parallel to these run two lateral rows of cuticular spines (Fig. 2F). One row of spines is situated dorsally, the second row ventrally to the lateral alae (Fig. 1B). A pair of deirids with apical spines is situated at approximately the height of the second pair of cuticular spines at a distance of 139–204 from the apex (Fig. 1B). Cuticular spines start at 93–154 from the apex, numbering approximately 42–47 per row. The nerve ring and excretory pore are 215–284 and 236–331 from the anterior extremity, respectively. The excretory pore is slightly posterior to the nerve ring. The triangular mouth is bounded by a pair of trilobed pseudolabia. The inner surface of each lobe carries two to four tooth-like processes. The precise number is difficult to assess in our specimens (Fig. 1A, 2A). Depth of buccal capsule 16–28, inner diameter 8–11. Oesophagus divided into muscular and glandular portion, 232–401 and 734–984, respectively. Total length of oesophagus 1023–1318. Spicules unequal and dissimilar. Right spicule tubular, with slight bend and spatulate tip, 106–170 long (Fig. 1C, 2D). Left spicule long and thin, trough-shaped, with spatulate tip. Shaft slightly twisted at 100–120 from proximal end. Total length 1699–2304 (Fig. 1D–F, 2C, 2E). A gubernaculum is absent. Tail
FIG. 1  *Tetrameris numida* n. sp. Male. A. Apical view of the trilobed pseudolabia surrounding the triangular mouth. Note the tooth-like processes (scale bar = 10 μm). B. Ventro-lateral view of the anterior end (scale bar = 100 μm). C. Ventral aspect of the posterior end (scale bar = 100 μm). D. Lateral view of the proximal end of the left spicule showing the slight twist (scale bar = 100 μm). E. Ventral view of the proximal end of the left spicule (scale bar = 100 μm). F. Distal end of the left spicule, ventral view (scale bar = 100 μm). Female. G. Complete female (scale bar = 1 mm). H. Anterior extremity (scale bar = 100 μm)
**FEMALE:** Specimens *in situ* red. A minute head and tail of regular nematode shape, but often twisted or bent, emerge at opposite sides from the central part of the body which is distinctly globular and slightly bent along the axis (Fig. 1G–H, 3A, 3C). The cuticle bears marked transverse striation and four longitudinal cuticular grooves. The latter divide the body into four segments of which the two segments following the outer curve are slightly longer (Fig. 1G). Much of the internal detail is obscured by the egg-
filled uterus coils surrounding a large sacular intestine. Body length 4.2–5.3 mm, maximum width 2.6–3.5 mm. The following measurements were derived from a single specimen: The deirids are at 179 and 190 and the nerve ring at 215 from the apex, respectively. The excretory pore could not be located. Depth of buccal capsule 23, inner diameter 7. Muscular part of oesophagus 333, the distal part of the glandular oesophagus obscured by the uterus. Eggs are elongate with near parallel sides, polar filaments were not seen (Fig. 3D). Eggs containing fully developed larvae are 56–59 long and 31–34 wide. Anus and vulva appeared to be confined in body folds. Emerging from the last body fold is a tail approximately 336 long with a simple tip.

SPECIFIC DIAGNOSIS: *Tetrameres numida* is differentiated from other members of the genus, by the possession of two rows of somatic spines and the arrangement of its caudal spines in two ventral and two lateral rows with usually three pairs of spines each, although deviation might occur. A short right and a long left spicule are present, ranging from 106–131 and from 1699–2 304 in length, respectively.

HOST: *Numida meleagris* (Linnaeus, 1758), Helmeted guineafowl.

SITE: Males occur free in the lumen of the proventriculus, females are situated in the proventricular glands.

LOCALITY: Musina (Messina), Limpopo Province, South Africa (22°22.139' S, 29°30.399' E).

ETYMOLOGY: The specific epithet *numida* refers to the host.

TABLE 1 The morphological characteristics of *Tetrameres numida* sp. n. males from Helmeted guineafowls, compared to *Tetrameres paradisea* Ortlepp, 1932 and to *Tetrameres prozeskyi* (Ortlepp, 1964), all described from South African hosts. All measurements in micrometres unless otherwise indicated.

<table>
<thead>
<tr>
<th>Morphological criteria</th>
<th>GFM1/N4</th>
<th>T.2191</th>
<th>T.2193</th>
<th>T.2194</th>
<th>T.2195</th>
<th>GFM11/1</th>
<th>GFM12/1</th>
<th>Tetrameres paradisea</th>
<th>Tetrameres prozeskyi</th>
</tr>
</thead>
<tbody>
<tr>
<td>Source</td>
<td>This paper</td>
<td>This paper</td>
<td>This paper</td>
<td>This paper</td>
<td>This paper</td>
<td>This paper</td>
<td>This paper</td>
<td>Ortlepp (1932)</td>
<td>Ortlepp (1964)</td>
</tr>
<tr>
<td>Body length (mm)</td>
<td>4.3</td>
<td>4.4</td>
<td>4.4</td>
<td>4.3</td>
<td>4.3</td>
<td>n</td>
<td>4.5</td>
<td>5.8</td>
<td>1.3–2.4</td>
</tr>
<tr>
<td>Body width maximum</td>
<td>n</td>
<td>160</td>
<td>160</td>
<td>164</td>
<td>170</td>
<td>162</td>
<td>140</td>
<td>60–70</td>
<td></td>
</tr>
<tr>
<td>Distance apex to first somatic spine</td>
<td>n</td>
<td>126 &amp; 117</td>
<td>96 &amp; 100</td>
<td>102 &amp; 93</td>
<td>105 &amp; 94</td>
<td>131 &amp; 154</td>
<td>96 &amp; 113</td>
<td>n</td>
<td>n</td>
</tr>
<tr>
<td>Distance apex to deirids</td>
<td>n</td>
<td>174 &amp; 180</td>
<td>139 &amp; 149</td>
<td>179 &amp; 172</td>
<td>165 &amp; 177</td>
<td>174 &amp; 181</td>
<td>175 &amp; 204</td>
<td>85</td>
<td>~ 50–60</td>
</tr>
<tr>
<td>Distance apex to nerve ring</td>
<td>n</td>
<td>256</td>
<td>215</td>
<td>234</td>
<td>244</td>
<td>264</td>
<td>n</td>
<td>~ 150–160</td>
<td></td>
</tr>
<tr>
<td>Distance apex to excretory pore</td>
<td>268</td>
<td>307</td>
<td>236</td>
<td>287</td>
<td>296</td>
<td>331</td>
<td>316</td>
<td>n</td>
<td>n</td>
</tr>
<tr>
<td>Depth of buccal capsule</td>
<td>22</td>
<td>25</td>
<td>28</td>
<td>23</td>
<td>21</td>
<td>22</td>
<td>16</td>
<td>25</td>
<td>5.0–7.0</td>
</tr>
<tr>
<td>Width of buccal capsule (inner)</td>
<td>n</td>
<td>10</td>
<td>10</td>
<td>8</td>
<td>8</td>
<td>11</td>
<td>8</td>
<td>12</td>
<td>11.0–13.0</td>
</tr>
<tr>
<td>Muscular oesophagus</td>
<td>n</td>
<td>351</td>
<td>304</td>
<td>232</td>
<td>260</td>
<td>401</td>
<td>400</td>
<td>310</td>
<td>160–210</td>
</tr>
<tr>
<td>Glandular oesophagus</td>
<td>n</td>
<td>734</td>
<td>769</td>
<td>984</td>
<td>781</td>
<td>812</td>
<td>918</td>
<td>900</td>
<td>300–400</td>
</tr>
<tr>
<td>Oesophagus total length</td>
<td>n</td>
<td>1085</td>
<td>1073</td>
<td>1216</td>
<td>1023</td>
<td>1213</td>
<td>1318</td>
<td>1210</td>
<td>n</td>
</tr>
<tr>
<td>Length of tail</td>
<td>284</td>
<td>297</td>
<td>287</td>
<td>257</td>
<td>296</td>
<td>n</td>
<td>290</td>
<td>115</td>
<td>140–160</td>
</tr>
<tr>
<td>Length of right spicule</td>
<td>131</td>
<td>130</td>
<td>106</td>
<td>110</td>
<td>131</td>
<td>120</td>
<td>170</td>
<td>Absent</td>
<td>Usually absent b</td>
</tr>
<tr>
<td>Length of left spicule</td>
<td>1 988</td>
<td>2 103</td>
<td>2 304</td>
<td>2 169</td>
<td>1 699</td>
<td>n</td>
<td>2 204</td>
<td>690; 504–626 a</td>
<td>230–260</td>
</tr>
</tbody>
</table>

n Data not available

a Range given by Mollhagen (1976) in Cremonte et al. (2001)

b A right spicule was present in three of more than 30 males
**TABLE 2** A comparison of morphological characteristics of some species of the genus *Tetrameres* Creplin, 1846

<table>
<thead>
<tr>
<th>Species</th>
<th>Bodylength of male (mm)</th>
<th>Number of rows of somatic spines</th>
<th>Length of rows of somatic spines</th>
<th>Number of spicules</th>
<th>Spicule length (mm)</th>
<th>Arrangement of caudal spines or papillae</th>
<th>Polar filaments on eggs</th>
<th>Source</th>
</tr>
</thead>
<tbody>
<tr>
<td>Tetrameres americana</td>
<td>5–5.5</td>
<td>4</td>
<td>n</td>
<td>2</td>
<td>Left: 0.29–0.31; right: 0.1–0.13</td>
<td>5 ventral pairs, no lateral pairs</td>
<td>n</td>
<td>Schmidt (1962); Gibbons et al. (1996)</td>
</tr>
<tr>
<td>Tetrameres araliensis</td>
<td>2.55</td>
<td>4</td>
<td>Whole body length</td>
<td>2</td>
<td>Long: 0.913; short: 0.22</td>
<td>2 ventral pairs and 2 sublateral rows with 6 and 7 spines, respectively. Two lateral tail papillae also present</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td>Tetrameres australis</td>
<td>7.8–9.0</td>
<td>2</td>
<td>Whole body length</td>
<td>2</td>
<td>Long: 5.8–6.3; short: 0.8</td>
<td>5 to 6 small spines</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td>Tetrameres biziureae</td>
<td>4.2–4.4</td>
<td>4</td>
<td>Whole body length</td>
<td>2</td>
<td>Long: 0.25–0.26; short: 0.07</td>
<td>n</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td>Tetrameres calidris</td>
<td>2.2–2.5</td>
<td>4/2</td>
<td>4 rows anteriorly, from glandular oesophagus onwards only 2</td>
<td>2</td>
<td>Left: 0.75–1.0; right: 0.08–0.09</td>
<td>5 ventral pairs, 2 lateral pairs</td>
<td>Only males known</td>
<td>Mawson (1968)</td>
</tr>
<tr>
<td>Tetrameres cardinalis</td>
<td>4.2–4.95</td>
<td>2</td>
<td>Whole body length</td>
<td>2</td>
<td>Left: 0.365–0.400; right: 0.065–0.085</td>
<td>4–5 pairs of postcloacal spines</td>
<td>Present</td>
<td>Quentin &amp; Barre (1976)</td>
</tr>
<tr>
<td>Tetrameres cladorhynchi</td>
<td>2.0–2.9</td>
<td>4</td>
<td>Whole body length</td>
<td>1</td>
<td>Left: 1.0–1.37</td>
<td>3 subventral pairs, 3 sublateral pairs</td>
<td>Present</td>
<td>Mawson (1968); Pence et al. (1975); Cremonte et al. (2001)</td>
</tr>
<tr>
<td>Tetrameres coloradensis</td>
<td>2.05</td>
<td>4</td>
<td>Whole body length</td>
<td>2</td>
<td>Left: 0.777; right: 0.067</td>
<td>4 ventral pairs, 3 lateral pairs</td>
<td>Present</td>
<td>Schmidt (1962)</td>
</tr>
<tr>
<td>Tetrameres confusa</td>
<td>4.0–5.0</td>
<td>4</td>
<td>n</td>
<td>2</td>
<td>Long: 0.291; short: 0.068</td>
<td>3 ventral pairs, 3 lateral pairs</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td>Tetrameres cordoniferens</td>
<td>n</td>
<td>4</td>
<td>n</td>
<td>n</td>
<td>Left spicule: 0.40</td>
<td>n</td>
<td>n</td>
<td>Pence et al. (1975)</td>
</tr>
<tr>
<td>Tetrameres crami</td>
<td>2.9–4</td>
<td>4</td>
<td>n</td>
<td>2</td>
<td>Left: 0.27–0.35; right: 0.136–0.185</td>
<td>n</td>
<td>n</td>
<td>Schmidt (1962); Gibbons et al. (1996)</td>
</tr>
<tr>
<td>Tetrameres crami asiatica</td>
<td>3.25–3.6</td>
<td>4</td>
<td>Whole body length</td>
<td>2</td>
<td>Long: 0.238–0.254; short: 0.099–0.106</td>
<td>5 ventral pairs, 3 lateral pairs</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td>Species</td>
<td>Bodylength of male (mm)</td>
<td>Number of rows of somatic spines</td>
<td>Length of rows of somatic spines</td>
<td>Number of spicules</td>
<td>Spicule length (mm)</td>
<td>Arrangement of caudal spines or papillae</td>
<td>Polar filaments on eggs</td>
<td>Source</td>
</tr>
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<tr>
<td>Tetrameres cygni Ryjikov &amp; Kozlov, 1960</td>
<td>n</td>
<td>4</td>
<td>n</td>
<td>2</td>
<td>Left: about one half the length of that of <em>T. finamicola</em></td>
<td>3 rows of 5 caudal papillae</td>
<td>n</td>
<td>Pence et al. (1975)</td>
</tr>
<tr>
<td>Tetrameres dubia Travassos, 1917&lt;sup&gt;6&lt;/sup&gt;</td>
<td>1.35–2.28</td>
<td>4/2</td>
<td>Dorsolateral rows reach only the level of the posterior end of the glandular oesophagus</td>
<td>2</td>
<td>Long: 0.71–0.77; short: 0.06–0.08</td>
<td>4 ventral pairs, 3 lateral pairs</td>
<td>Present</td>
<td>Mamaev (1959) cited by Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td>Tetrameres termini Vigueras, 1935</td>
<td>2.5</td>
<td>n</td>
<td>n</td>
<td>2</td>
<td>Long: 0.073; short: 0.023</td>
<td>3 pairs of postcloacal spines</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td>Tetrameres fissispina (Diesing, 1861) Travassos, 1914</td>
<td>3.0–6.0</td>
<td>n</td>
<td>n</td>
<td>2</td>
<td>Left: 0.82–1.5; right: 0.28–0.49</td>
<td>8 pairs of postanal spines</td>
<td>n</td>
<td>Gibbons et al. (1996)</td>
</tr>
<tr>
<td></td>
<td>3.2–3.9</td>
<td>4</td>
<td>n</td>
<td>2</td>
<td>Long: 0.37–0.49; short: 0.165–0.198</td>
<td>3 ventral pairs, 5 lateral pairs</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td>Tetrameres galeniculatus Oschmarin, 1956</td>
<td>3.4</td>
<td>4</td>
<td>Whole body length</td>
<td>2</td>
<td>Longer: 0.450; short: 0.086</td>
<td>Present</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td>Tetrameres gigas Travassos, 1919</td>
<td>7.5</td>
<td>4</td>
<td>Whole body length</td>
<td>2</td>
<td>Long: 0.74; short: 0.016</td>
<td>Tail papillae have not been found</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td>Tetrameres globosa (Von Linstow, 1879)</td>
<td>3.6–3.75</td>
<td>4</td>
<td>Whole body length, sparser in posterior half</td>
<td>2/1</td>
<td>Long: 0.3; short spicule rudimentary</td>
<td>Small spines posterior to cloaca</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td>Tetrameres grusi Shumakovitch, 1946</td>
<td>3.45–4.40</td>
<td>2</td>
<td>2 distinct rows, but spines scattered anterior to nerve ring and posterior to anus</td>
<td>1</td>
<td>0.638–0.783</td>
<td>Several irregular rows of spines</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963); Bush et al. (1973); Pence et al. (1975)</td>
</tr>
<tr>
<td>Tetrameres gubanovi Shigin, 1957</td>
<td>6.67</td>
<td>2</td>
<td>Whole body length, starting at transition from muscular to glandular oesophagus</td>
<td>2</td>
<td>Long: 3.996; short: 0.131</td>
<td>4 ventral pairs of conical papillae, 3 lateral pairs of stalked papillae</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td>Tetrameres hagenbecki Travassos &amp; Vogelsang, 1930</td>
<td>3.1–3.4</td>
<td>2?</td>
<td>Rows of cuticular spines along lateral fields (2 rows illustrated)</td>
<td>2</td>
<td>Long spicule: thin and ending as a spur, proximal 0.07–0.08 twisted. Short spicule 0.032–0.04</td>
<td>4 ventral pairs, 2 lateral pairs</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td>Tetrameres ihuillieri (Seurat, 1918)</td>
<td>n</td>
<td>4</td>
<td>n</td>
<td>1</td>
<td>0.48</td>
<td>n</td>
<td>Present</td>
<td>Ortlepp (1964)</td>
</tr>
<tr>
<td>Species</td>
<td>Bodylength of male (mm)</td>
<td>Number of rows of somatic spines</td>
<td>Length of rows of somatic spines</td>
<td>Number of spicules</td>
<td>Spicule length (mm)</td>
<td>Arrangement of caudal spines or papillae</td>
<td>Polar filaments on eggs</td>
<td>Source</td>
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<tr>
<td><em>Tetrameres lobibycis</em> Mawson, 1968</td>
<td>1.5</td>
<td>4/2</td>
<td>4 rows anteriorly, from nerve ring onwards only 2</td>
<td>1</td>
<td>Left: 0.73</td>
<td>6 subventral pairs</td>
<td>Only male known</td>
<td>Mawson (1968)</td>
</tr>
<tr>
<td><em>Tetrameres megaphasmidiata</em> Cremonte, Digiani, Bala &amp; Navone (2001)</td>
<td>1.94–2.03</td>
<td>4</td>
<td>Whole body length</td>
<td>1</td>
<td>Left: 0.96–1.22</td>
<td>6 subventral pairs, 2 lateral pairs</td>
<td>n</td>
<td>Cremonte et al. (2001)</td>
</tr>
<tr>
<td><em>Tetrameres micropenis</em> Travassos, 1915</td>
<td>4.0–5.0</td>
<td>2</td>
<td>Whole body length</td>
<td>2</td>
<td>Long: 0.355; short: 0.056</td>
<td>2 ventral pairs</td>
<td>n</td>
<td>Ortlepp (1932); Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td><em>Tetrameres microspinosa</em> Vigueras, 1935</td>
<td>3.0</td>
<td>2</td>
<td>Whole body length</td>
<td>2</td>
<td>Long: 1.135; short: 0.065</td>
<td>5 ventral pairs</td>
<td>Absent</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td><em>Tetrameres mohtedai</em> Bhalariao and Rao, 1944</td>
<td>4.27–5.8</td>
<td>4/2</td>
<td>Submedian spines end posterior to middle of glandular oesophagus</td>
<td>2</td>
<td>Long: 0.397–0.430; short: 0.142–0.160</td>
<td>5 subventral pairs</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td><em>Tetrameres nouveli</em> (Seurat, 1914)</td>
<td>1.0–2.4</td>
<td>4</td>
<td>Whole body length</td>
<td>1</td>
<td>Left: 350–580c</td>
<td>3 or 4 subventral pairs, 2 or 3 sublateral pairs</td>
<td>Present</td>
<td>Ortlepp (1932); Mawson (1968); Cremonte et al. (2001)</td>
</tr>
<tr>
<td></td>
<td>2.16</td>
<td>4</td>
<td>Whole body length</td>
<td>1</td>
<td>0.480; second spicule rudimentary (Seurat 1914, cited by Skrjabin &amp; Sobolev 1963)</td>
<td>4 ventral and 3 lateral pairs illustrated; according to text 2 papillae in posterior third of tail</td>
<td>Present</td>
<td>Ortlepp (1932); Mawson (1968); Cremonte et al. (2001)</td>
</tr>
<tr>
<td><em>Tetrameres numinii</em> Mamaev, 1959</td>
<td>1.64–2.4</td>
<td>4/2</td>
<td>Dorsolateral rows reach only the level of the posterior part of the oesophagus</td>
<td>2</td>
<td>Long: 1.08–1.24; short: 0.08–0.10</td>
<td>4 ventral pairs, 3 lateral pairs</td>
<td>Absent</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td><em>Tetrameres numida</em> n. sp.</td>
<td>4.3–4.4</td>
<td>2</td>
<td>Whole body length</td>
<td>2</td>
<td>Left: 1.699–2.304; right: 0.106–0.131</td>
<td>3 ventral pairs, 3 lateral pairs</td>
<td>Absent</td>
<td>This paper</td>
</tr>
<tr>
<td><em>Tetrameres oxylabiatus</em> Oschmarin, 1956</td>
<td>5.0</td>
<td>n</td>
<td>Whole body length</td>
<td>2</td>
<td>Long: 0.940; short: 0.125</td>
<td>Extend posteriorly to middle of tail, getting very small</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td><em>Tetrameres paraaraliensis</em> Oschmarin, 1956</td>
<td>1.71</td>
<td>4</td>
<td>Whole body length</td>
<td>1</td>
<td>0.405–0.420</td>
<td>n</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963); Mawson (1968); Mollhaugen (1976) in Cremonte et al. (2001)</td>
</tr>
<tr>
<td>Species</td>
<td>Bodylength of male (mm)</td>
<td>Number of rows of somatic spines</td>
<td>Length of rows of somatic spines</td>
<td>Number of spicules</td>
<td>Spicule length (mm)</td>
<td>Arrangement of caudal spines or papillae</td>
<td>Polar filaments on eggs</td>
<td>Source</td>
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<tr>
<td>Tetrameres paradisea</td>
<td>5.8</td>
<td>2</td>
<td>Whole body length</td>
<td>1</td>
<td>0.69</td>
<td>3 ventral pairs, 3 dorso-external pairs</td>
<td>Absent</td>
<td>Ortlepp (1932)</td>
</tr>
<tr>
<td>(Ortlepp, 1932)</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
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<tr>
<td>Tetrameres paradoxa</td>
<td>12–15</td>
<td>2</td>
<td>n</td>
<td>2</td>
<td>Long: 3.0 or longer; short: 0.480</td>
<td>Drashe (1884) illustrated a very small pair of ventral papillae and 3 and 4 lateral papillae respectively</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963), Drashe (1884) cited by Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td>(Diesing, 1835)</td>
<td></td>
<td></td>
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<tr>
<td>Tetrameres pattersoni</td>
<td>4.2–4.6</td>
<td>2</td>
<td>Whole body length</td>
<td>1</td>
<td>1.2–1.5</td>
<td>n</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td>Cram, 1933</td>
<td></td>
<td></td>
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<td></td>
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<tr>
<td>Tetrameres paucispina</td>
<td>n</td>
<td>2</td>
<td>Few, only in posterior 2/3</td>
<td>2</td>
<td>Left: 0.328–0.371; right: 0.012–0.154</td>
<td>3 caudal papillae</td>
<td>n</td>
<td>Bush et al. (1973); Quentin &amp; Barre (1976)</td>
</tr>
<tr>
<td>Sandground, 1928</td>
<td></td>
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<tr>
<td></td>
<td>3.1–4.5</td>
<td>1</td>
<td>1 row in median ventral field, not more than 25 spines, only in post 2/3</td>
<td>2</td>
<td>Long: 0.328–0.371; short: 0.154</td>
<td>3 caudal papillae</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td>Tetrameres pavlovskii</td>
<td>n</td>
<td>4</td>
<td>n</td>
<td>1</td>
<td>n</td>
<td>4 ventral pairs, 4 lateral pairs</td>
<td>n</td>
<td>Pence et al. (1975)</td>
</tr>
<tr>
<td>lygis, 1965</td>
<td></td>
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<tr>
<td>Tetrameres pavonis</td>
<td>4.7</td>
<td>n</td>
<td>Irregular and dense anteriorly, in middle and posterior part almost invisible</td>
<td>2</td>
<td>Long: 0.43; short: 0.105</td>
<td>4 rows of spines, and 3 papillae: 1 lateral pair, 1 unpair median papilla</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
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<tr>
<td>Tschenkova, 1953</td>
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<tr>
<td>Tetrameres phaenicopterus</td>
<td>n</td>
<td>4</td>
<td>n</td>
<td>2</td>
<td>n</td>
<td>n</td>
<td>n</td>
<td>Pence et al. (1975)</td>
</tr>
<tr>
<td>All, 1970</td>
<td></td>
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<tr>
<td>Tetrameres plectropteri</td>
<td>n</td>
<td>n</td>
<td>n</td>
<td>n</td>
<td>Left: 0.85</td>
<td>n</td>
<td>n</td>
<td>Ortlepp (1964)</td>
</tr>
<tr>
<td>Thwaite, 1926</td>
<td></td>
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<tr>
<td>Tetrameres prozeskyi</td>
<td>1.3–2.4</td>
<td>4</td>
<td>Whole body length</td>
<td>1</td>
<td>Left: 0.23–0.26</td>
<td>3 ventral pairs, 3 lateral pairs</td>
<td>n</td>
<td>Ortlepp (1964)</td>
</tr>
<tr>
<td>(Ortlepp, 1964)</td>
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<tr>
<td>Tetrameres puchovi</td>
<td>3.86–4.339</td>
<td>2</td>
<td>Whole body length</td>
<td>1</td>
<td>0.307–0.309; second spicule rudimentary: 0.008</td>
<td>n</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
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<td>Gushanskaia, 1949</td>
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<tr>
<td>Tetrameres ryjikovi</td>
<td>4.5</td>
<td>4</td>
<td>Whole body length</td>
<td>2</td>
<td>Long: 0.208; short: 0.062</td>
<td>4 ventral pairs, 3 lateral pairs</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td>Chuan, 1961</td>
<td></td>
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<tr>
<td>Tetrameres sakharowi</td>
<td>9.47</td>
<td>4</td>
<td>n</td>
<td>2</td>
<td>Left: 0.195; right: 1.021</td>
<td>n</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td>Petrov, 1926</td>
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<tr>
<td>Species</td>
<td>Bodylength of male (mm)</td>
<td>Number of rows of somatic spines</td>
<td>Length of rows of somatic spines</td>
<td>Number of spicules</td>
<td>Spicule length (mm)</td>
<td>Arrangement of caudal spines or papillae</td>
<td>Polar filaments on eggs</td>
<td>Source</td>
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</tr>
<tr>
<td><em>Tetrameres scolopacidis</em></td>
<td>1.06–1.8</td>
<td>4/2</td>
<td>4 rows anteriorly, from end of oesophagus only 2 rows</td>
<td>2</td>
<td>Left: 0.70–0.85; right: 0.07–0.105</td>
<td>4 subventral pairs, 3 sublateral pairs</td>
<td>Present</td>
<td>Mawson (1968)</td>
</tr>
<tr>
<td><em>Tetrameres somateriae</em></td>
<td>4.8</td>
<td>4</td>
<td>No spines in the middle part of the body</td>
<td>2</td>
<td>Long: 0.576; short: 0.086</td>
<td>5 ventral pairs, 4 lateral pairs</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td><em>Tetrameres spiruospiculum</em></td>
<td>2.52–4.06</td>
<td>n</td>
<td>Thinly dispersed and poorly developed</td>
<td>2</td>
<td>Left: 0.82–1.08; right: n</td>
<td>n</td>
<td>n</td>
<td>Pinto &amp; Vicente (1995)</td>
</tr>
<tr>
<td><em>Tetrameres skrjabini</em></td>
<td>2.6</td>
<td>4</td>
<td>Whole body length</td>
<td>2</td>
<td>Long: 1.543; short: 0.103</td>
<td>Not found</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td><em>Tetrameres tetrica</em></td>
<td>2.6</td>
<td>4</td>
<td>Dissapear near last quarter of body length</td>
<td>2</td>
<td>Long: 0.2; short: 0.022</td>
<td>4 lateral pairs, 4 sublateral pairs</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td><em>Tetrameres timopheewoi</em></td>
<td>4.7</td>
<td>n</td>
<td>Whole body length</td>
<td>2</td>
<td>Long: 0.421; short: 0.189</td>
<td>n</td>
<td>n</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td><em>Tetrameres tinamicola</em></td>
<td>6.52</td>
<td>4</td>
<td>Ventral rows whole body length, dorsal rows end 1.02 mm from apex</td>
<td>2</td>
<td>Left: 2.26; right: 0.207</td>
<td>5 subventral pairs, 3 ventro-lateral pairs</td>
<td>Absent</td>
<td>Pence et al. (1975)</td>
</tr>
<tr>
<td><em>Tetrameres uxorius</em></td>
<td>n</td>
<td>4</td>
<td>n</td>
<td>2</td>
<td>Left: 2.1–2.3; right: 0.088</td>
<td>4 ventrolateral pairs, 2 subdorsal pairs</td>
<td>Absent</td>
<td>Mamaev (1959); Pence et al. (1975)</td>
</tr>
<tr>
<td></td>
<td>4.76–5.0</td>
<td>4/2</td>
<td>Dorsolateral rows reach only the beginning of the glandular oesophagus</td>
<td>2</td>
<td>Long: 2.1–2.24; short: 0.086–0.088</td>
<td>4 ventrolateral pairs, 2 subdorsal pairs</td>
<td>Absent</td>
<td>Skrjabin &amp; Sobolev (1963)</td>
</tr>
<tr>
<td><em>Tetrameres vietnamensis</em></td>
<td>n</td>
<td>4</td>
<td>n</td>
<td>2</td>
<td>Left: 1.28; right: 0.148</td>
<td>5 ventral pairs (lateral absent)</td>
<td>n</td>
<td>Fan the Viet (1968) in Helminthological Abstracts (1970), Pence et al. (1975)</td>
</tr>
</tbody>
</table>

n No information at our disposal
a The original reads 65–350 μm. We consider this a typing error and include the range of single measurements provided by Quentin & Barre (1976)
b Skrjabin & Sobolev (1963) also include a description after Cram (1927), which differs slightly from that of Mamaev (1959)
c Cremonte et al. (2001) give a range of 0.312–0.587 mm
d Cremonte et al. (2001) quote Mollhagen (1976) giving a range of 0.504–0.626 mm
e The length provided by Quentin & Barre (1976) is 12–154 μm. We consider this an error. Skrjabin & Sobolev give the width of the right spicule as 12 μm
f According to Ortlepp (1964) in three of about 30 males a right spicule was present
g Cremonte et al. (2001) quote Mollhagen (1975) as *T. prozeskyi* having varying caudal papillae (3/0, 3/3, 4/1, 4/2)
h Calculated from a 1:24 to 1:26 ratio between right and left spicule
DISCUSSION

Some of the main morphological characteristics of many of the species belonging to the genus *Tetrameres* are listed in Table 2.

Of the *Tetrameres* species with two rows of cuticular spines, *Tetrameres pattersoni* Cram, 1933, *T. paradisea* and *Tetrameres grusi* Shumakovitsch, 1946 have only one spicule and the spicule measurements of the latter two species differ distinctly from those in our specimens (Ortlepp 1932; Schmidt 1962; Bush, Pence & Forrester 1973).

*Tetrameres gubanovi* Shigin, 1957 bears two rows of body spines, but has seven pairs of caudal papillae (Pence et al. 1975), as opposed to six pairs of caudal spines in *T. numida* n. sp.

The use of the term caudal spines or caudal papillae is not always clear. Pence et al. (1975) use the term caudal papillae for several species in their publication. They list *T. paradisea* as well as *T. prozeskyi* as having caudal papillae, but in the original descriptions Ortlepp (1932, 1964) clearly refers to cuticular spines. Thus, Pence et al. (1975) seem to use the term indiscriminately. Mawson (1968), however, describes *T. nouveli* as having caudal spines, but points out that in *Tetrameres lobiybicus* Mawson, 1968 the spines are more like elongate papillae, and refers to *Tetrameres calidis* Mawson, 1968 and *Tetrameres scolopacidis* Mawson, 1968 as having papillae.

The left spicules of *Tetrameres cardinalis* Quentin & Barre, 1976 and *Tetrameres paucispina* Sandground, 1928 are much shorter than those measured in our specimens (Quentin & Barre 1976). *Tetrameres micropenis* Travassos, 1915 has been recovered from ciconiform hosts, *Nyctanassa violacea* (Linnaeus, 1758) and *Cochlearius cochlearia* (Linnaeus, 1766) (Yamaguti 1961), whose geographic distribution is restricted to North and South America (Lepage 2006).

*Tetrameres fissispina* has been recorded from guineafowls in Africa (Fabiyi 1972; Vercruysse et al. 1985) and, like *T. americana*, has a high prevalence in domestic chickens, whose nematode fauna is similar to that of guineafowls (Mukaratirwa, Hove, Esmann, Hoj, Permin & Nansen 2001; Magwisha, Kassuku, Kyvsgaard & Permin 2002). *Tetrameres fissispina* distinguishes itself from the new species by its shorter spicules and the larger number of caudal spines. *Tetrameres americana* differs not only in the spicule size and the number and arrangement of caudal spines, but also in its four rows of somatic spines (Schmidt 1962; Gibbons, Jones & Khalil 1996).

The head of the female and the apical view of the head of the male of *T. numida* n. sp. most closely resemble *Tetrameres tinamicola* Pence, Mollhagen & Prestwood, 1975. The authors of the latter species describe the male head as possessing a triangular mouth surrounded by a pair of trilobed structures originating from the inner surface of the pseudolabia. Each lobe bears a pair of tooth-like processes in *T. tinamicola*. Similar processes can be seen in our specimens, but it is difficult to determine their exact number. However, there seem to be three or four per lobe. Pronounced lateral alae, as illustrated by Pence et al. (1975), were not found in our specimens. Moreover, *T. tinamicola* has a total of four rows of cuticular spines and the deirids are without apical spines. While the length of the left spicule of both species is similar, the right spicule of *T. numida* is only approximately half the length of *T. tinamicola*.

Ortlepp (1932) described the buccal capsule of *T. paradisea* as having trilobed structures showing two to three bright refringent markings towards its posterior border. This, as well as other features of our specimens such as the transverse grooves anterior to the cloaca and the size of the spines, appeared so similar to *T. paradisea* that we initially considered assigning them to *T. paradisea*, especially in view of the fact that both were recovered from South African hosts. Close examination has nevertheless revealed distinct differences between the two. *Tetrameres paradisea* possesses a single spicule, whereas in our males two spicules are consistently present. While the arrangement of caudal spines is nearly identical and both carry three pairs of ventral and three pairs of extero-dorsal or lateral spines, the tail of *T. paradisea* is considerably shorter than that of our specimens (see Table 1).

Ortlepp (1932) described and illustrated two rows of body spines found in *T. paradisea* and he uses this criterion to distinguish his species from *Tetrameres nouveli* which he lists as possessing four rows of spines. Cremonte, Digiani, Bala & Navone (2001) record *T. paradisea* as having four rows of spines, but cite Mollhagen (1976) as describing the dorsal rows of spines as very short, ending at 94–155 from the anterior end.

When comparing *T. paradisea* to *T. prozeskyi*, Ortlepp (1964) lists the length of the left spicule of the former species as 0.48 mm, but his original description of *T. paradisea* (Ortlepp, 1932) clearly states the length of the spicule as 0.69 mm. We list *T. pro-
zeskyi as monospicular, which differentiates it from our bispicular specimens. As regards *T. prozeskyi* it should be borne in mind that Ortlepp (1964) found a well-chitinized right spicule in three of the more than 30 males he examined.

In the summary of the description of *Tetrameres cardinalis* Quentin & Barre, 1976, the range of the length of the right spicule is given as 65–350 μm (Quentin & Barre 1976). As this seems erroneous, we decided to include the range provided in the same paper, namely 365–400, in Table 2. Similarly, we consider the first measurement these authors provide for the range of the right spicule is given as 65–350 μm (Quentin & Barre 1975). Relative to body length, however, there are other species with long spicules, such as *T. lobycis* where the single spicule reaches about half of the body length (1.5 mm) and *T. scolopacidis* where the spicule length reaches almost two thirds of the body length (1.06–1.8 mm) (Mawson 1968).

To our knowledge, *Tetrameres phaenicopterus* All, 1970 is the only member of the genus *Tetrameres* possessing a gubernaculum (Pence et al. 1975) and *Tetrameres greeni* Mawson, 1979 is unique in the genus *Tetrameres* in that it has caudal alae (Mawson 1979). *Tetrameres spiroscillum* Pinto & Vicente, 1995 is distinguished from our specimens and all the other species of *Tetrameres* by the spiral shaped distal end of the longer of its two spicules (Pinto & Vicente 1995).

The numbers of *T. numida* n. sp. recovered from the guineafowl hosts from Musina (Messina) were low, and the parasite was only found in the older birds, being absent in young adults. While it is possible that guineafowls are not the main host for this parasite, we attribute the low intensity of infection to the fact that the area had been experiencing a severe drought during the past years. This would decrease the survival rates of nematode eggs while at the same time causing the numbers of possible intermediate hosts necessary for the completion of the life-cycle to decline. While differences in the immune status between guineafowls of different age might play a role in the intensity of infection, we believe that the presence of *T. numida* n. sp. in older hosts simply reflects the increased possibility of prior exposure to the parasite as a function of time.

**ACKNOWLEDGEMENTS**

The authors are indebted to Dr S. Sokolov of the Institute of Parasitology, Russian Academy of Science, Moscow, for obtaining the extensive chapter on the genus *Tetrameres* in "Principles of nematodology XI" (Skrijabin & Sobolev 1963) and to Dr D.A. Apanaskevich, Georgia Southern University, for the translation from the original Russian into English. The authors thank Mr K. Meyer and Mr M. Storm, the previous and current owners of the farm Sandown, Musina (Messina), respectively, for placing the guineafowls at our disposal and Mr H.E. Hattingh, University of Limpopo, for collecting them. Dr W.J. Luus-Powell, University of Limpopo, has kindly facilitated the co-operation. Ms D.T. Durand, University of Pretoria, photographed the three complete females of *T. numida*. This research was made possible through a Claude Leon Foundation Post-doctoral Fellowship grant to the first author.

**REFERENCES**


Tetrameres numida n. sp. (Nematoda: Tetrameridae) from Helmeted guineafowls in South Africa


Nematodes from Swainson’s spurfowl 
*Pternistis swainsonii* and an Orange River francolin *Scleroptila levaillantoides* in Free State Province, South Africa, with a description of *Tetrameres swainsonii* n. sp. 
(Nematoda: Tetrameridae)

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¹Department of Veterinary Tropical Diseases, University of Pretoria, Private Bag X04, Onderstepoort, 0110 South Africa: ²Department of Zoology, DST/NRF Centre of Excellence in Birds at the Percy FitzPatrick Institute, University of Cape Town, Private Bag, Rondebosch, 7701 South Africa: ³Department of Environmental, Water and Earth Sciences, Tshwane University of Technology, Private Bag X680, Pretoria, 0001 South Africa

Abstract

Five Swainson’s spurfowl collected in Free State Province, South Africa, were examined for helminth parasites, and the nematodes *Acuaria gruveli, Cyrnea parroti, Gongylonema congoense, Subulura dentigera, Subulura suctoria* and a new *Tetrameres* species were recovered. Their respective prevalence was 100, 20, 80, 20, 20 and 20%. These nematodes are all new parasite records for Swainson’s spurfowl, and *Acuaria gruveli* constitutes a new geographical record as well. A single specimen of *Cyrnea eurycerca* was found in an Orange River francolin, representing a new host and geographical record for this parasite. The new species, for which the name *Tetrameres swainsonii* is proposed, can be differentiated from its congeners by a combination of the following characters of males: two rows of body spines, a single spicule which is 1152–1392 μm long, and eight pairs of caudal spines arranged in two ventral and two lateral rows of four spines each. The single female has the globular shape typical of the genus.

Introduction

Swainson’s spurfowl *Pternistis swainsonii* (Smith, 1836) (Phasianidae: spurfowls) is endemic to southern Africa. In South Africa it has undergone a major southward range expansion and can now be found east of approximately 23°E and south as far as 30°S in the Eastern Cape, Free State, Gauteng, Limpopo, Mpumalanga, Northern Cape and North West Provinces. It is absent from the coastal lowlands of KwaZulu-Natal Province (Little, 2005). Its preferred habitat in South Africa is dense grassland in proximity to cultivated lands, where it exploits crops and associated insects. While some authors refer to Swainson’s spurfowl as one of the most water-dependent perdicine birds in Africa (del Hoyo *et al.*, 1994; Little, 2005), a study in Limpopo Province, South Africa, revealed no or little reliance on easily accessible drinking water and birds seldom drank (Jansen & Crowe, 2002).

The Orange River francolin *Scleroptila levaillantoides* (Smith, 1836) (Phasianidae: francolins) is found in two distinct geographical areas on the African continent.
(del Hoyo et al., 1994). While it is a frequent to common bird in Ethiopia and Somalia, numbers appear to have declined in its southern population, especially in South Africa and Namibia. This is thought to be mainly due to habitat pressure following the conversion of natural grass- and woodland habitats into farmland, despite the fact that, like Swainson’s spurfowl, it will forage at the edges of cultivated land (Little et al., 2000). The natural range of Orange River francolin in South Africa used to be restricted to north-western Northern Cape Province (del Hoyo et al., 1994), but it has expanded to include north-eastern Eastern Cape Province, and Free State and North West Provinces, as well as the region east of the highveld of Mpumalanga and Gauteng Provinces (Little et al., 2000; Little, 2005).

Only incidental findings on helminth parasites of both these gamebirds in South Africa have been published. Oosthuizen & Markus (1967) collected Subulura sp. from a single Swainson’s spurfowl, while the only record pertaining to helminths of S. levaillantoides is that of Bennett et al. (1992) who reported Microfilaria sp. when cataloguing haematozoa of sub-Saharan birds.

This paper reports on helminths collected from the gastrointestinal tract (GIT) of five Swainson’s spurfowl and a single Orange River francolin in Free State Province, South Africa and describes a new nematode, Tetrameres swainsonii, from the proventriculus of the former.

**Materials and methods**

Five Swainson’s spurfowl, a single second-year male and four adult females (at least third-year), and a single adult male Orange River francolin were collected during a gamebird hunt in the vicinity of Petrus Steyn (27°39’S; 28°’E), Free State Province, in August 2007. The habitat in the survey area was made up primarily of cereal plantings (maize) and sunflower, in a mosaic of grazing land.

Within 4 hours of being shot, the entire GIT was removed from the birds and placed in a plastic tray. The GITs of the birds were ligated at the entrance of the oesophagus and removed from the birds and placed in a plastic tray. The GITs of the various birds were placed in individual containers, stored at 2°C overnight and then fixed in 70% ethanol.

Subsequently, the crop, proventriculus, gizzard, small intestine and caeca were washed separately over a 150 μm sieve and, together with the residue, examined under a stereoscopic microscope. Helminths in the gizzard usually only became visible after removal of the lining.

All helminths were stored in 70% ethanol. For identification purposes, nematodes were cleared in lactophenol and studied under a standard microscope. Intensity of infection, mean intensity of infection, mean abundance and prevalence are used in accordance with Margolis et al. (1982).

**Results**

All five Swainson’s spurfowl harboured nematodes and a total of six species, Acuaria gruveli (Gendre, 1913), Cyrnea parroti Seurat, 1917, Gongylomena congolense Fain, 1955, Subulura dentigera Orlepp, 1937, S. suctoria (Molin, 1860) and T. swainsonii n. sp., was recovered. Their habitat, prevalence, mean intensity of infection and mean abundance are listed in table 1. A single host harboured a total of four species, a second three, and three birds had two nematode species each. The mean species richness was 2.6 (SD = 0.9). The intensity of infection ranged from 3 to 68, with a mean intensity of 19 (SD = 27.7). The second-year male had the highest species diversity as well as highest intensity of infection.

Two nematode species were recovered from both the gizzard and caeca, and a single nematode species from the proventriculus and crop, respectively. No helminths were found in the small intestine.

With the exception of a single C. eurycerca Seurat, 1914 in its gizzard, the Orange River francolin harboured no helminth parasites.

The presence of A. gruveli in Swainson’s spurfowl constitutes both a new host record and a new geographical record for this parasite, while C. parroti, G. congolense and S. suctoria are new parasite records for this host. This is the first report of S. dentigera from a host other than helmeted guineafowl Numida meleagris (Linnaeus, 1758) (Phasianidae: guineafowls). Cyrnea eurycerca is recorded from Orange River francolin as well as from South Africa for the first time.

**Tetrameres swainsonii n. sp**

**Description.** Tetrameres swainsonii is described from four males and one female from a single Swainson’s spurfowl. Males were found free in the lumen of the proventriculus, while the female was dissected from the proventricular glands. All measurements are in micrometres unless otherwise stated (fig. 1).

Female. Bright red in situ as typical for the genus, damaged; only buccal capsule, 24 deep and 16 wide, maximum body width (3 mm) and length (4 mm) as

<table>
<thead>
<tr>
<th>Nematode</th>
<th>Habitat</th>
<th>Prevalence (%)</th>
<th>Mean intensity (± SD)</th>
<th>Range</th>
<th>Mean abundance (± SD)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Acuaria gruveli</td>
<td>Gizzard</td>
<td>100</td>
<td>2.4 (0.9)</td>
<td>1–3</td>
<td>2.4 (0.8)</td>
</tr>
<tr>
<td>Cyrnea parroti</td>
<td>Gizzard</td>
<td>20</td>
<td>2.0</td>
<td>2</td>
<td>0.4 (0.8)</td>
</tr>
<tr>
<td>Gongylomena congolense</td>
<td>Crop</td>
<td>80</td>
<td>4.25 (5.9)</td>
<td>1–13</td>
<td>3.4 (4.8)</td>
</tr>
<tr>
<td>Subulura dentigera</td>
<td>Caeca</td>
<td>20</td>
<td>12.0</td>
<td>12</td>
<td>2.4 (4.8)</td>
</tr>
<tr>
<td>Subulura suctoria</td>
<td>Caeca</td>
<td>20</td>
<td>47.0</td>
<td>47</td>
<td>9.4 (18.8)</td>
</tr>
<tr>
<td>Tetrameres swainsonii n. sp</td>
<td>Proventriculus</td>
<td>20</td>
<td>5.0</td>
<td>5</td>
<td>1.0 (2.0)</td>
</tr>
</tbody>
</table>
Fig. 1. *Tetrameres swainsonii* n. sp. male. (A) Ventral view of anterior extremity illustrating the position of the deirids, nerve ring, excretory pore and first pair of body spines. (B) Ventral view of posterior extremity showing the arrangement of the caudal spines. (C) Proximal end of the single spicule, lateral view. (D) Distal end of the spicule, lateral view. Scale bars = 100 µm.
well as egg length and width could be measured. Eggs 

\( n = 10 \), length 49 (SD = 2.98), between 43 and 52, width 

32 (SD = 1.47), between 30 and 34; polar filaments 

not seen. Body globular with anterior and posterior 

extremities forming short protuberances; surface divided 

into four segments by four conspicuous longitudinal 

cuticular grooves; each segment with numerous trans-

verse striations.

Male. Measurements of holotype male given in text, 
those of two paratypes and a further specimen in table 2. 
Body elongated, tapered at both ends, 5.1 mm long and 
188 wide. Cuticle striated transversely as well as 
longitudinally. Cuticular spines arranged in two lateral 
rows, one dorsal and one ventral to inconspicuous lateral 
ala; 41 spines per row in holotype, 40 to 43 in paratypes; 
first pair of spines at 269 and 285 from anterior extremity. 
Deirids with apical spines at 261 and 251 from anterior 
extremity. Nerve ring and excretory pore at 252 and 
265 from apex, respectively. Deirids at approximately 
centre of nerve ring with first pair of cuticular spines 
in close proximity, but posterior to deirids. Excretory pore in 
same vicinity, sometimes slightly anterior, slightly 
posterior or on same level as first pair of cuticular spines 
(fig. 1A). Depth of buccal capsule 19, inner diameter 6. 
Oesophagus divided into muscular and glandular parts, 
(fig. 1A). Depth of buccal capsule 19, inner diameter 6. 
Oesophagus total length 1377 1285 1451.

<table>
<thead>
<tr>
<th>Morphological criteria</th>
<th>Specimen A</th>
<th>Paratype 1</th>
<th>Paratype 2</th>
</tr>
</thead>
<tbody>
<tr>
<td>Body length (mm)</td>
<td>4.7</td>
<td>4.8</td>
<td>5.1</td>
</tr>
<tr>
<td>Body width max.</td>
<td>203</td>
<td>200</td>
<td>216</td>
</tr>
<tr>
<td>Distance from apex to first pair of somatic spines</td>
<td>276; 260</td>
<td>250; 272</td>
<td>340; 340</td>
</tr>
<tr>
<td>Distance from apex to nerve ring</td>
<td>244</td>
<td>245</td>
<td>263</td>
</tr>
<tr>
<td>Distance from apex to deirids</td>
<td>243; 235</td>
<td>237; 242</td>
<td>268; 286</td>
</tr>
<tr>
<td>Distance from apex to excretory pore</td>
<td>282</td>
<td>275</td>
<td>310</td>
</tr>
<tr>
<td>Depth of buccal capsule (inner)</td>
<td>21</td>
<td>23</td>
<td>23</td>
</tr>
<tr>
<td>Width of buccal capsule (inner)</td>
<td>5</td>
<td>6</td>
<td>5</td>
</tr>
<tr>
<td>Muscular oesophagus</td>
<td>368</td>
<td>418</td>
<td>428</td>
</tr>
<tr>
<td>Glandular oesophagus</td>
<td>1005</td>
<td>914</td>
<td>1031</td>
</tr>
<tr>
<td>Oesophagus total length</td>
<td>1377</td>
<td>1285</td>
<td>1451</td>
</tr>
<tr>
<td>Length of tail</td>
<td>291</td>
<td>306</td>
<td>309</td>
</tr>
<tr>
<td>Length of single spicule</td>
<td>1152</td>
<td>1392</td>
<td>1183</td>
</tr>
</tbody>
</table>
distinctly different from that seen in the present specimens (Junker & Boomker, 2007a). The first pair of cuticular spines of *T. numida* is situated anterior to the deirids, which are approximately at the level of the second pair of cuticular spines, and the nerve ring is distinctly posterior to the deirids. Only 12 caudal spines are described for *T. numida* and, although additional ventral spines may occasionally be present, the two lateral rows consistently carried three spines each. In addition, *T. numida* possesses a right and a left spicule, ranging from 106 to 170 and from 1699 to 2304, respectively (Junker & Boomker, 2007a).

Of the 54 species of *Tetrameres* listed by Junker & Boomker (2007a), only *T. paradisea*, *Tetrameres grusi* Shumakovitch, 1946, *Tetrameres puchovi* Cram, 1933 and *Tetrameres puchovi* Gushanskaja, 1949 share the combination of two rows of cuticular spines and a single spicule with the present specimens. However, the spicules of *T. grusi* (638–783) and of *T. puchovi* (307–309) (Skrjabin & Sobolev, 1963) are distinctly shorter than those of *T. swainsonii* n. sp. (1152–1392). Moreover, the caudal spines of *T. grusi* are arranged in several irregular rows, and several pairs of cuticular spines originate anterior to the nerve ring (Skrjabin & Sobolev, 1963), whereas in *T. swainsonii* n. sp. the first pair of cuticular spines emerges posterior to the nerve ring. The distance from the apex to the deirids is 160 in *T. puchovi* (Skrjabin & Sobolev, 1963), which is considerably shorter than that observed in the new taxon, namely 235–286.

*Tetrameres puchovi* is closest to *T. swainsonii* n. sp. in spicule length, with a single, strongly chitinized spicule of length 1200–1500; but it differs in the arrangement of caudal spines in three lateral and four subventral pairs, as opposed to four pairs each in the new taxon. The distance of the deirids from the apex, which is less than half that seen in *T. swainsonii* n. sp., namely 83–112 (Skrjabin & Sobolev, 1963), clearly separates *T. pattersoni* from *T. swainsonii* n. sp.

**Discussion**

The single second-year male Swainson’s spurfowl yielded the largest number of helminth species as well as individuals. Phasianid chicks are reported to rely heavily on insect food in the early stages of their lives (del Hoyo *et al.*, 1994). Chicks of grey partridge *Perdix perdix* Linnaeus, 1758 (Phasianidae: partridges) in Europe, for example, consume a diet consisting of 80% insect matter for the first 2 weeks after hatching (del Hoyo *et al.*, 1994). Arthropods only make up approximately 7% of the crop weight of adult *P. swainsonii*, reaching a maximum of up to 20% in summer (del Hoyo *et al.*, 1994). Higher intake of live food by juvenile versus adult birds is likely to increase exposure to infected intermediate hosts, which, in turn, would result in higher worm burdens. However, because of the small sample size it is not possible to establish whether our findings are due to chance or reflect a true pattern in the helminth community of Swainson’s spurfowl.

Only nematodes were collected from Swainson’s spurfowl and the single Orange River francolin. This is noteworthy, especially taking into account that all nematodes collected from these two hosts are heteroxenous; that is, their life cycles include various arthropod intermediate hosts, such as orthopterans and coleopterans (Anderson, 1992), which in addition serve as intermediate hosts for cestodes and acanthocephalans (Moore, 1962; Reid, 1962). Moreover, helmeted guineafowl collected at the same locality during the course of this study harboured nematodes and cestodes as well as acanthocephalans (Davies *et al.*, in review), thereby confirming their presence in the environment.

While Swainson’s spurfowl had a markedly less diverse helminth fauna than helmeted guineafowl at the study site, the former seem to be more suitable hosts of the gizzard nematode *A. gruveli*, since it was collected from all five spurfowl, but was absent in more than 40 helmeted guineafowl (Davies *et al.*, in review). Other galliform birds recorded as final hosts of *A. gruveli* include double-spurred spurfowl *Pternistis bicalcaratus* (Linnaeus, 1766) (= *Francolinus bicalcaratus*) (Phasianidae: spurfowls) in Togo (Quentin & Seureau, 1983), common quail *Coturnix coturnix* (Linnaeus, 1758) (Phasianidae: quails) in the Palearctic region (Baruš & Sonin, 1983) and red-legged partridge *Alectoris rufa* (Linnaeus, 1758) (Phasianidae: partridges) in Spain (Tarazona *et al.*, 1979), suggesting that perdicine birds feature more prominently in the life cycle of this parasite than do guineafowls.

A possible explanation for the presence/absence of helminths in Swainson’s spurfowl versus helmeted guineafowl at the same locality might be a difference in their dietary preferences, which in turn would influence the probability of exposure to intermediate hosts of certain parasites. Moreover, differences in the immune competence of the two bird species might result in a higher resistance in guineafowl. Similarly, morphological differences between hosts, such as the nature of the gizzard lining, could prevent establishment of, for example, *A. gruveli* in guineafowl, but allow colonization of spurfowl.

*Cynnea parroti*, *G. congolense* and *S. suctoria* collected from Swainson’s spurfowl are also commonly found in other galliform birds (Junker & Boomker, 2007b). Contrary to this, *S. dentigera* had hitherto been recorded from helmeted guineafowl only.

*Cynnea eurycerca*, which was present in the single Orange River francolin, seems a relatively common parasite in francolins and spurfowls, and has previously been collected from black francolin *Francolinus francolinus* (Linnaeus, 1766) (Phasianidae: francolins) in Italy, grey francolin *Francolinus pondicerianus* (Gmelin, 1789) (Phasianidae: francolins) in India and double-spurred spurfowl in Togo (Marconcini & Triantafilis, 1970; Jehan, 1974; Seureau & Quentin, 1983).

The low prevalence and intensity of infection of *T. swainsonii* n. sp. in Swainson’s spurfowl is in keeping with data obtained for *T. numida* from helmeted guineafowls in Limpopo Province, as well as in the present study area (Junker & Boomker, 2007a; Davies *et al.*, in review). Similarly, only two of 158 bobwhite quail *Colinus virginianus* (Linnaeus, 1758) (Phasianidae: quails) examined in northern Florida harboured *T. pattersoni*, and intensity of infection ranged from 0 to 1 (Moore & Simberloff, 1990).
The overall low helminth diversity and intensity of infection seen in Swainson’s spurfowl at the study site might be attributable to several factors. First, they occur in pairs or small family groups rather than in large flocks (Little, 2005; Jansen & Crowe, 2006), which would facilitate parasite transmission (Moore et al., 1988). Jansen & Crowe (2002) reported a covey size ranging from 1 to 4. Second, the birds were collected in winter, when the volume of their diet consists mainly of grass seeds, weed seeds and agricultural seeds, while invertebrates play a minor role (Jansen & Crowe, 2006). In terms of crop volume, 5.74% is made up of invertebrates during the summer months and 3.64% during the winter months (Jansen & Crowe, 2006). Third, much of their habitat consisted of cultivated lands, the insect fauna of which might be depauperate because of low habitat diversity and the use of pesticides. In addition, while Swainson’s spurfowl from a cereal-crop habitat, similar to that found in the current study area, ingested the greatest number of invertebrates, when compared to savanna spurfowl from a cereal-crop habitat, similar to that found in the current study area, ingested the greatest number of invertebrates, when compared to savanna paints or small family groups rather than in large flocks (Jansen & Crowe, 2002) reported a covey size ranging from 1 to 4.

Acknowledgements
We thank Mr J.P. Wales for access to ‘The Jimmy Wales Shoot’ for collection of gastrointestinal tracts. The research was supported financially by the Centre of Excellence in Birds at the Percy FitzPatrick Institute (funded by the South African Department of Science and Technology and the National Research Foundation), the University of Cape Town’s Research Committee, and the Department of Veterinary Tropical Diseases, University of Pretoria. K.J. was supported by a University of Pretoria Postdoctoral Fellowship Grant.

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Junker, K. & Boomker, J. (2007b) A check list of the helminths of guineafowls (Numididae) and a host list of these parasites. Onderstepoort Journal of Veterinary Research 74, 315–337.


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CHAPTER 2

Population dynamics

of

parasites of guineafowls
INTRODUCTION

Helmeted guineafowls, *Numida meleagris* (Linnaeus, 1758), are distributed throughout most of South Africa and almost the entire African continent (Del Hoyo, Elliot & Sargatal 1994). Studies to elucidate the helminth fauna of these hosts in South Africa have been undertaken by Saayman (1966), Crowe (1977) and Verster & Ptasinska-Kloryga (1987), but were restricted to the Eastern Cape, the Northern Cape and Gauteng Provinces.

Although relatively wide-spread in Africa, Crested guineafowls, *Guttera edouardi* (Hartlaub, 1867), are scarce and have a limited distribution within South Africa. They occur in the Limpopo, North West, Mpumalanga and KwaZulu-Natal Provinces and are listed as rare or accidental in Gauteng Province (Hockey, Dean & Ryan 2005; Lepage 2007). To date our knowledge concerning their helminth fauna is virtually non-existent.

Ortlepp (1937, 1938a,b, 1963) reported on the cestode and nematode parasites of guineafowls present in the National Collection of Animal Helminths, formerly known as the Onderstepoort Helminthological Collection, or material made...
available to him by various collectors. He described several new species of cestodes and nematodes and added numerous parasites to the host-parasite list of guineafowls in South Africa. His research, however, was of a taxonomic nature and the material at his disposal represented incidental findings rather than complete collections.

In this paper we present data obtained from 16 birds, including a single Crested guineafowl, at Musina, Limpopo Province, and from five Helmeted guineafowls at Mokopane, Limpopo Province, South Africa.

**MATERIAL AND METHODS**

In July and August 2005 a total of five Helmeted guineafowls were sampled in the vicinity of Mokopane (Potgietersrus), Limpopo Province. A complete helminth recovery was not possible, but some of the worms present in the small intestine of three of the birds, the complete caeca of one of them and part of the intestinal and caecal contents of another were collected and fixed separately in 70% ethanol.

In July and May, July and November 2006, three, five, three and four Helmeted guineafowls (eight males and seven females) were collected on a farm approximately 60 km west of Musina (Mes sina), Limpopo Province (22°22’ S, 29°30’ E, Altitude 700–800 m). The vegetation-type in the study area is classified as Mopani veld (Acocks 1988).

The birds were aged according to the criteria established by Siegfried (1966) and in total ten adults and five juveniles were collected. The juveniles were between six and ten months old (Siegfried 1966). In November 2006 a single adult female Crested guineafowl, found moribund in a wire snare, was made available to us for examination.

The carcasses of the birds were opened according to standard techniques for necropsies of chickens, and the viscera removed. The trachea was opened and macroscopically examined for helminths.

The crop, proventriculus, gizzard, small intestine and caecum/colon were separated and individually washed over a 150 μm sieve. The livers of nine Helmeted guineafowls and the single Crested guineafowl were sliced into 5 mm wide sections and incubated in phosphate-buffered saline at 40°C for 30 min. Subsequently, the slices together with the saline were washed over a 150 μm sieve. The gastrointestinal and liver residues left on the sieves, as well as the organs themselves were fixed separately in 70% ethanol and transported to the laboratory at Onderstepoort. Each sample was examined under a stereoscopic microscope and the helminths removed.

Cestodes were stained in haematoxylin and mounted in Canada balsam or mounted and cleared in Hoyer’s medium. Acanthocephalans were cleared in Hoyer’s medium and studied as temporary mounts in the same medium. All nematodes were cleared in lactophenol for identification.

The ecological terms are used in accordance with the definitions of Margolis, Esch, Holmes, Kuris & Schad (1982).

**RESULTS**

All the guineafowls were infected and all were concurrently parasitized by acanthocephalans, cestodes and nematodes.

Data on the prevalence, intensity and habitat preference of the parasites from the Helmeted guineafowls in Musina are presented in Tables 1 and 2. Five of the nine hosts (55.6%), whose livers were examined, harboured *Dicrocoelium macrostomum*, the intensity of infection ranging from 8 to 182 flukes. In addition, the livers of three of the nine birds yielded five, 11 and five young specimens of *Porogynia paronai*. These had the typical three circles of large hammer-shaped rostellar hooks and small, unarmed suckers. No differential development could be seen in any of the proglottids of the short strobilae which ranged from 2.3 to 3.8 mm \((n = 5)\) in length. The scolecis were 689–746 μm wide and the rostella were 261–329 μm wide.

Birds from Mokopane yielded the nematodes *Subulura suctoria*, *Subulura dentigera* and *Ascaridia numidae* and seven cestodes, namely *Hispaniolepis multiuncinata*, *Porogynia paronai*, *Raillietina steinhartii*, *Raillietina pintneri*, *Raillietina sp.*, *Numidella numida* and *Octopetalum numida*.

*Subulura dentigera* and *S. suctoria* were co-specific in the two hosts from Mokopane. One of these harboured a total of 579 nematodes consisting of 142 male and 159 female *S. suctoria*, 134 male and 126 female *S. dentigera* and 18 immature *Suctoria spp.*. These nematodes were suspended freely in the contents of the posterior saccate part of the caeca, virtually occupying the entire lumen (Fig. 2D).

Eight of the 15 helmeted guineafowls from Musina harboured *S. dentigera* and *S. suctoria* concurrently, and in all these hosts *S. suctoria* by far outnumbered...
TABLE 1 The site preference, prevalence and intensity of infection of acanthocephalans and cestodes collected from 15 Helmeted guineafowls in Limpopo Province, South Africa. Additional data on guineafowl helminths in southern Africa from various authors are included for comparison.

<table>
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<tbody>
<tr>
<td></td>
<td>Site</td>
<td>Prevalence (%)</td>
<td>Intensity Mean (± SD) Range</td>
<td>Prevalence (%)</td>
<td>Intensity Mean Range</td>
</tr>
<tr>
<td>Acanthocephalans</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Mediorhynchus gallinarum</td>
<td>SI</td>
<td>100</td>
<td>55.7 (± 78.3 ) 2–231</td>
<td>–</td>
<td>–</td>
</tr>
<tr>
<td>Mediorhynchus numidae</td>
<td>SI</td>
<td>–</td>
<td>–</td>
<td>–</td>
<td>–</td>
</tr>
<tr>
<td>Mediorhynchus taeniatus</td>
<td>SI</td>
<td>–</td>
<td>–</td>
<td>27</td>
<td>1.7</td>
</tr>
<tr>
<td>Cestodes</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Abuladzugnia gutterae</td>
<td>SI</td>
<td>80</td>
<td>11.7 (± 8.2) 1–28</td>
<td>–</td>
<td>–</td>
</tr>
<tr>
<td>Abuladzugnia transvaalensis</td>
<td>SI</td>
<td>–</td>
<td>–</td>
<td>–</td>
<td>–</td>
</tr>
<tr>
<td>Davainea nana</td>
<td>SI</td>
<td>33</td>
<td>5.8 (± 4.4) 1–10</td>
<td>–</td>
<td>–</td>
</tr>
<tr>
<td>Hispaniolepis multiuncinata</td>
<td>SI</td>
<td>87</td>
<td>9.3 (± 5.2) 2–14</td>
<td>–</td>
<td>–</td>
</tr>
<tr>
<td>Hispaniolepis cantaniana</td>
<td>SI</td>
<td>40</td>
<td>42.7 (± 70.4) 1–124</td>
<td>–</td>
<td>–</td>
</tr>
<tr>
<td>Numidella numida</td>
<td>SI</td>
<td>67</td>
<td>55.9 (± 72.7) 1–144</td>
<td>29</td>
<td>1.8</td>
</tr>
<tr>
<td>Octopetalum numida</td>
<td>SI</td>
<td>67</td>
<td>91.9 (± 110.7) 1–360</td>
<td>48</td>
<td>8</td>
</tr>
<tr>
<td>Paroniella sp. a</td>
<td>SI</td>
<td>–</td>
<td>–</td>
<td>25</td>
<td>1.5</td>
</tr>
<tr>
<td>Porogynia paronai</td>
<td>SI</td>
<td>47</td>
<td>12.3 (± 13.3) 5–39</td>
<td>–</td>
<td>–</td>
</tr>
<tr>
<td>Raillietina angusta</td>
<td>SI</td>
<td>53</td>
<td>10.3 (± 7.9) 1–25</td>
<td>8</td>
<td>&lt; 1.0</td>
</tr>
<tr>
<td>Raillietina pintneri</td>
<td>SI</td>
<td>80</td>
<td>5.3 (± 3.9) 2–12</td>
<td>44</td>
<td>3.9</td>
</tr>
<tr>
<td>Raillietina steinhardti</td>
<td>SI</td>
<td>53</td>
<td>49.0 (± 60.2) 4–137</td>
<td>31</td>
<td>1.9</td>
</tr>
<tr>
<td>Raillietina sp.</td>
<td>SI</td>
<td>73</td>
<td>15.8 (± 8.8) 6–28</td>
<td>–</td>
<td>–</td>
</tr>
<tr>
<td>Raillietina sp. a</td>
<td>SI</td>
<td>–</td>
<td>–</td>
<td>35</td>
<td>2.7</td>
</tr>
<tr>
<td>Skrijabinia deweti</td>
<td>SI</td>
<td>–</td>
<td>–</td>
<td>–</td>
<td>–</td>
</tr>
</tbody>
</table>

a Listed by Verster & Ptasinska-Kloryga (1987) as a new species, but were not subsequently described
SI = small intestine
TABLE 2 The site preference, prevalence and intensity of infection of nematodes collected from 15 Helmeted guineafowls in Limpopo Province, South Africa. Additional data on guineafowl nematodes in southern Africa from various authors are included for comparison.

<table>
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<tbody>
<tr>
<td></td>
<td>Site</td>
<td>Prevalence (%)</td>
<td>Intensity Mean (± SD)</td>
<td>Prevalence (%)</td>
<td>Intensity Mean Range</td>
</tr>
<tr>
<td>Ascaridia galli</td>
<td>SI</td>
<td>–</td>
<td>–</td>
<td>2</td>
<td>&lt; 1 0–2</td>
</tr>
<tr>
<td>Ascaridia numidae</td>
<td>SI</td>
<td>6</td>
<td>4.0± 4±</td>
<td>13</td>
<td>&lt; 1 0–19</td>
</tr>
<tr>
<td>Cyrena parroti</td>
<td>Giz</td>
<td>100</td>
<td>13.8± 2–75</td>
<td>13</td>
<td>&lt; 1 0–16</td>
</tr>
<tr>
<td>Dispharynx nasuta</td>
<td>Prov</td>
<td>–</td>
<td>–</td>
<td>10</td>
<td>1.8 0–59</td>
</tr>
<tr>
<td>Gongylonema congolense</td>
<td>Crop</td>
<td>40</td>
<td>23.0± 2–61</td>
<td>–</td>
<td>–</td>
</tr>
<tr>
<td>Gongylonema ingluvicola</td>
<td>Crop</td>
<td>–</td>
<td>–</td>
<td>–</td>
<td>–</td>
</tr>
<tr>
<td>Hadjelia inermis</td>
<td>Giz</td>
<td>–</td>
<td>–</td>
<td>–</td>
<td>–</td>
</tr>
<tr>
<td>Hadjelia truncata</td>
<td>Giz</td>
<td>53</td>
<td>1.6± 1–2</td>
<td>–</td>
<td>–</td>
</tr>
<tr>
<td>Heterakis gallinarum</td>
<td>Caeca</td>
<td>–</td>
<td>–</td>
<td>4</td>
<td>&lt; 1 0–2</td>
</tr>
<tr>
<td>Sicarius caudatus</td>
<td>Giz, SI</td>
<td>53</td>
<td>2.1± 1–6</td>
<td>–</td>
<td>–</td>
</tr>
<tr>
<td>Subulura dentigera</td>
<td>Caeca</td>
<td>53</td>
<td>15.9± 1–31</td>
<td>6</td>
<td>1.3 0–54</td>
</tr>
<tr>
<td>Subulura suertoria</td>
<td>Caeca</td>
<td>100</td>
<td>536.3± 9–214</td>
<td>23</td>
<td>&lt; 1 0–40</td>
</tr>
<tr>
<td>Subulura sp.</td>
<td>Caeca</td>
<td>40</td>
<td>44.0± 1–170</td>
<td>10</td>
<td>&lt; 1 0–4</td>
</tr>
<tr>
<td>Unidentified subulurid</td>
<td>SI</td>
<td>13</td>
<td>2.5± 2–3</td>
<td>–</td>
<td>–</td>
</tr>
<tr>
<td>Tetrameres numida</td>
<td>Prov</td>
<td>33</td>
<td>2.4± 1–5</td>
<td>–</td>
<td>–</td>
</tr>
</tbody>
</table>

a Only a single host harboured this parasite

SI = small intestine
Giz = gizzard
Prov = proventriculus
S. dentigera, the ratio ranging from 4.5:1 to 53:1. In the remaining hosts only S. suctoria was present (Fig. 2E, F).

The Crested guineafowl harboured a single acanthocephalan species, Mediorhynchus gallinarum \((n = 48)\), five species of cestodes, namely Abuladzugnia gutterae \((n = 1)\), H. multiuncinata \((n = 1)\), N. numida \((n = 114)\), O. numida \((n = 57)\) and P. paronai \((n = 52)\), as well as three species of nematodes, S. suctoria \((n = 260)\), Gongylonema congolense \((n = 56)\) and Hadjelia truncata \((n = 2)\), representing a total of 591 helminths.

Our finding of *M. gallinarum*, *A. gutterae*, *H. multiunci- nata*, *H. truncata* and *Sicarius caudatus* in Helmeted guineafowls in South Africa constitutes new host associations, as well as new geographic records for these parasites. *Dicrocoelium macrostomum*, *G. congolense* and *Davainea nana* are recorded in South Africa for the first time, and the Crested guineafowl is a new host for the nematodes *S. suctoria*, *G. congolense* and *H. truncata*.

Despite the generally high helminth burdens, the Helmeted guineafowls were in good physical condi-

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**FIG. 2**  
A, B, C. *Sicarius caudatus*. A. Anterior extremity of male. The deirids are marked by arrows. B. Posterior extremity of female. Note the finger-like protruberances (arrow) at the tip of the tail. C. Posterior extremity of male. D. Distal part of guineafowl caecum filled with *Subulura* spp. E. *Subulura dentigera* female, anterior part. The arrow indicates the cuticular denticles as described by Ortlepp (1937); x 400. F. *Subulura suctoria* female, anterior part; x 400.
tion and no obvious lesions were associated with the presence of helminths. The crop mucosa of a single bird from Musina had an inflamed appearance. This, however, did not seem to be related to *G. congolense* living in shallow tunnels under the crop lining, but rather to the presence of numerous thorny seeds of *Tribulus terrestris*.

**TAXONOMIC REMARKS**

*Cyrnea parroti* Seurat, 1917 (Table 3; Fig. 1A, B)

Ortlepp (1938b) described *Habronema numidae* from Helmeted guineafowls in Malawi, South Africa and Swaziland. This nematode has subsequently been included in the genus *Cyrnea* Seurat, 1914, but it is still listed under its original name in Yamaguti (1961) as well as in Verster & Ptasinska-Kloryga (1987).

In his work on the Habronematinae, Chabaud (1958) divided the genus *Cyrnea* into two subgenera, *Procyrnea* Chabaud, 1958 and *Cyrnea* Chabaud, 1958, which he later raised to genus level (Chabaud 1975). Following an in-depth study of the cephalic structures, he synonymized *Cyrnea (Cyrnea) numidae* (Ortlepp, 1938) with *Cyrnea (Cyrnea) parroti* Seurat, 1917 (Chabaud 1958).

Specimens from our hosts mounted *en face* show the same arrangement of submedian lobes and simple lips as illustrated for *C. parroti* by Chabaud (1958) and otherwise conform well with the description and measurements supplied by Ortlepp (1938b) for *C. numidae*. The range of measurements in our specimens was, however, generally wider than that provided by the latter author (Table 3). Ortlepp (1938b) himself stated that his new species most closely resembled *C. eurycerca* and *C. parroti* and that the arrangement of the caudal papillae in the males as well as the spicules were very similar.

*Gongylonema congolense* Fain, 1955 (Table 4; Fig. 1C, D)

This parasite was first described by Fain (1955a) from domestic chickens, a single duck, *Cairina moschata domestica* and from *N. melagris* from the Democratic Republic of the Congo and Rwanda. Subsequently it has been recorded from *N. melagris* in Burundi, Nigeria, Ethiopia, Ghana and Burkina Faso (Fain & Thienpont 1958; Fabiyi 1972; Graber 1976; Hodasi 1976; Vercruysse, Harris, Bray, Nagalo, Pangu & Gibson 1985).

One of the main morphological characteristics of this species is the hook situated at a distance of about 50 µm from the distal tip of the left spicule (Fain 1955a) (Fig. 1D). The hook itself carries three fine barbs. In our specimens the barbed hook of the tip of the left spicule was often difficult to see, but in specimens where the distance could be determined it varied from 31 to 46 µm.

It is not always easy to judge whether the left spicule is intact or damaged, which could lead to measuring errors. There are, however, sufficient other characteristics, such as the gubernaculum, the extent and arrangement of the cuticular plaques (Fig. 1C), as well as the length of the right spicule to differentiate *G. congolense* from other species utilizing avian hosts (Fain 1955a).

While our specimens fit in well with Fain’s (1955a, b) description of *G. congolense*, we have not been able to confirm that the excretory pore opens on a transversally elongated plaque as was described by him. In our specimens it would seem that the two median ventral longitudinal rows of plaques are interrupted, leaving a plaque-free zone immediately anterior and posterior to the excretory pore (Fig. 1C).

Measurements of our specimens and those of Fain (1955a) taken from guineafowl hosts are presented in Table 4. These indicate that there is little geographic variation in the morphology of *G. congolense* from the same host species.

*Hadjelia truncata* (Creplin, 1825) (Table 5; Fig. 1E, F)

The most obvious differences between *H. truncata* and sympatric specimens of *C. parroti* are the position of the vulva and the winged appearance of the lips of *H. truncata* in ventral view (Fig. 1E, F). In *H. truncata* the vulva is distinctly anterior and positioned in front of the posterior end of the oesophagus. These characteristics are in accordance with the generic diagnosis of *Hadjelia* provided by Yamaguti (1961).

Measurements of the specimens from the guineafowls fall well within the range of measurements provided by Ortlepp (1964) for *Hadjelia inermis* (Gedoelst, 1919) (Table 5). *Hadjelia inermis* had been synonymized with *H. truncata* by Chabaud & Campana (1950), and Ortlepp (1964) commented on this, but chose to retain the former species. He lists his own measurements for *H. inermis* collected from Red- and Yellow-billed hornbills from South Africa, together with measurements for *H. inermis* taken from Gedoelst (1919) and for *H. inermis* and *H. truncata* as provided by Cram (1927, cited by Ortlepp...
### TABLE 3
The main morphological criteria of *Cyrnea parroti* Seurat, 1917 from Helmeted guineafowls. The range of measurements is provided. All measurements in micrometres unless otherwise stated.

<table>
<thead>
<tr>
<th>Source</th>
<th>Present study</th>
<th>Ortlepp (1938b)</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>Males (n = 6)</td>
<td>Females (n = 4)</td>
</tr>
<tr>
<td>Body length (mm)</td>
<td>9–11</td>
<td>11–16</td>
</tr>
<tr>
<td>Distance apex to nerve ring</td>
<td>187–262</td>
<td>237–263</td>
</tr>
<tr>
<td>Distance apex to deirids</td>
<td>220–370</td>
<td>309–364</td>
</tr>
<tr>
<td>Distance apex to excretory pore</td>
<td>220–362</td>
<td>311–357</td>
</tr>
<tr>
<td>Depth of buccal capsule</td>
<td>29–36</td>
<td>34–45</td>
</tr>
<tr>
<td>Width of buccal capsule (inner)</td>
<td>10–15</td>
<td>12–16</td>
</tr>
<tr>
<td>Muscular oesophagus</td>
<td>304–393</td>
<td>–</td>
</tr>
<tr>
<td>Glandular oesophagus</td>
<td>2 234–2 526</td>
<td>–</td>
</tr>
<tr>
<td>Oesophagus total length</td>
<td>2 284–2 830</td>
<td>2 039–3 056</td>
</tr>
<tr>
<td>Length of tail</td>
<td>120–193</td>
<td>128–150</td>
</tr>
<tr>
<td>Distance vulva to posterior end</td>
<td>–</td>
<td>661–897</td>
</tr>
<tr>
<td>Egg length x egg width</td>
<td>–</td>
<td>45–46 x 25–27</td>
</tr>
<tr>
<td>Length of right spicule</td>
<td>410–510a</td>
<td>–</td>
</tr>
<tr>
<td>Length of left spicule</td>
<td>834–1 354a</td>
<td>–</td>
</tr>
<tr>
<td>Length of gubernaculum</td>
<td>63–84a</td>
<td>–</td>
</tr>
<tr>
<td>Length of caudal alae</td>
<td>437–618</td>
<td>–</td>
</tr>
</tbody>
</table>

\* Measurements of the spicules and the gubernaculum are derived from ten males.
TABLE 4 The main morphological criteria of *Gongylonema congolense* Fain, 1955 males from Helmeted guineafowls from South Africa (present study, GFM/N represents our specimen number) and from the Democratic Republic of the Congo and Rwanda (Fain 1955a). All measurements in micrometres unless otherwise stated.

<table>
<thead>
<tr>
<th>Source</th>
<th>Present study</th>
<th>Fain (1955b)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Morphological criteria</td>
<td>GFM3/N1</td>
<td>GFM1/N16</td>
</tr>
<tr>
<td>Body length (mm)</td>
<td>17</td>
<td>15</td>
</tr>
<tr>
<td>Maximum width</td>
<td>266</td>
<td>228</td>
</tr>
<tr>
<td>Distance apex to deirids</td>
<td>84</td>
<td>70; 86</td>
</tr>
<tr>
<td>Distance apex to nerve ring</td>
<td>210</td>
<td>190</td>
</tr>
<tr>
<td>Distance apex to excretory pore</td>
<td>305</td>
<td>303</td>
</tr>
<tr>
<td>Distance apex to end of plaques</td>
<td>440</td>
<td>385</td>
</tr>
<tr>
<td>Distance apex to cervical ailes</td>
<td>102; 110</td>
<td>118</td>
</tr>
<tr>
<td>Depth of buccal capsule</td>
<td>31</td>
<td>–</td>
</tr>
<tr>
<td>Muscular oesophagus</td>
<td>–</td>
<td>362</td>
</tr>
<tr>
<td>Glandular oesophagus</td>
<td>–</td>
<td>2 967</td>
</tr>
<tr>
<td>Oesophagus total length</td>
<td>4 125</td>
<td>3 407</td>
</tr>
<tr>
<td>Length of tail</td>
<td>207</td>
<td>173</td>
</tr>
<tr>
<td>Caudal alae (left; right)</td>
<td>600 (left)</td>
<td>–</td>
</tr>
<tr>
<td>Length of gubernaculum</td>
<td>87</td>
<td>73</td>
</tr>
<tr>
<td>Length of right spicule</td>
<td>98</td>
<td>101</td>
</tr>
<tr>
<td>Length of left spicule (mm)</td>
<td>8.7</td>
<td>–</td>
</tr>
</tbody>
</table>
TABLE 5 The main morphological criteria of *Hadjelia truncata* (Creplin, 1825) from Helmeted guineafowls. The range of measurements is provided. All measurements in micrometres unless otherwise stated.

<table>
<thead>
<tr>
<th>Source</th>
<th>Present study</th>
<th>Gedoelst (1919)</th>
<th>Ortlepp (1964)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Morphological criteria</td>
<td>Males</td>
<td>Females</td>
<td>Males</td>
</tr>
<tr>
<td>Body length (mm)</td>
<td>GFM9/1</td>
<td>7</td>
<td>6.1–6.45</td>
</tr>
<tr>
<td></td>
<td>GFM11</td>
<td>8</td>
<td>–</td>
</tr>
<tr>
<td></td>
<td>GFM1/10</td>
<td>10</td>
<td>11</td>
</tr>
<tr>
<td></td>
<td>GFM1/14</td>
<td>–</td>
<td>–</td>
</tr>
<tr>
<td></td>
<td>GFM6/1</td>
<td>–</td>
<td>–</td>
</tr>
<tr>
<td>Maximum width</td>
<td>160</td>
<td>145</td>
<td>209</td>
</tr>
<tr>
<td></td>
<td>208</td>
<td>212</td>
<td>185</td>
</tr>
<tr>
<td>Distance apex to nerve ring</td>
<td>237; 239</td>
<td>238; 231</td>
<td>206; 204</td>
</tr>
<tr>
<td>Distance apex to deirids</td>
<td>275</td>
<td>259</td>
<td>234</td>
</tr>
<tr>
<td>Distance apex to excret. pore</td>
<td>44</td>
<td>42</td>
<td>39</td>
</tr>
<tr>
<td>Depth of buccal capsule</td>
<td>5</td>
<td>7</td>
<td>5</td>
</tr>
<tr>
<td>Width of buccal capsule (inner)</td>
<td>369</td>
<td>397</td>
<td>358</td>
</tr>
<tr>
<td>Muscular oesophagus</td>
<td>1 750</td>
<td>1 927</td>
<td>1 948</td>
</tr>
<tr>
<td>Glandular oesophagus</td>
<td>2 119</td>
<td>2 324</td>
<td>2 306</td>
</tr>
<tr>
<td>Oesophagus total length</td>
<td>–</td>
<td>–</td>
<td>1 698</td>
</tr>
<tr>
<td>Distance apex to vulva</td>
<td>–</td>
<td>–</td>
<td>–</td>
</tr>
<tr>
<td>Length of tail</td>
<td>–</td>
<td>–</td>
<td>50 x 32</td>
</tr>
<tr>
<td>Egg length x egg width</td>
<td>–</td>
<td>–</td>
<td>1 346</td>
</tr>
<tr>
<td>Length of left spicule</td>
<td>271</td>
<td>254</td>
<td>–</td>
</tr>
<tr>
<td>Length of right spicule</td>
<td>–</td>
<td>–</td>
<td>–</td>
</tr>
</tbody>
</table>

<table>
<thead>
<tr>
<th></th>
<th></th>
<th></th>
<th></th>
<th></th>
<th></th>
</tr>
</thead>
<tbody>
<tr>
<td>Morphological criteria</td>
<td><em>Sicarius dipterum</em></td>
<td><em>Sicarius hoopoe</em></td>
<td><em>Sicarius caudatus</em></td>
<td><em>Sicarius caudatus</em></td>
<td><em>Sicarius renatae</em></td>
</tr>
<tr>
<td>Male</td>
<td>Female</td>
<td>Male</td>
<td>Female</td>
<td>Male</td>
<td>Female</td>
</tr>
<tr>
<td>Body length (mm)</td>
<td>10.2–11.9</td>
<td>12.9–16.1</td>
<td>6.7–9.4</td>
<td>11.2–18.2</td>
<td>7.3</td>
</tr>
<tr>
<td>Distance apex to deirids</td>
<td>70–74&lt;sup&gt;a&lt;/sup&gt;</td>
<td>80–90&lt;sup&gt;a&lt;/sup&gt;</td>
<td>60–70</td>
<td>50–60</td>
<td>85</td>
</tr>
<tr>
<td>Depth of buccal capsule</td>
<td>43–45</td>
<td>52–58</td>
<td>14–17</td>
<td>25</td>
<td>28</td>
</tr>
<tr>
<td>Glandular oesophagus</td>
<td>2 880–3 920</td>
<td>3 160–3 910</td>
<td>2 800–3 040</td>
<td>3 200–3 600</td>
<td>2 170</td>
</tr>
<tr>
<td>Oesophagus total length</td>
<td>3 400–3 500</td>
<td>3 700–4 500</td>
<td>–</td>
<td>–</td>
<td>2 550</td>
</tr>
<tr>
<td>Length of tail</td>
<td>210</td>
<td>185–210</td>
<td>176–208</td>
<td>167–256</td>
<td>190</td>
</tr>
<tr>
<td>Length of left spicule</td>
<td>620–690</td>
<td>–</td>
<td>470–600</td>
<td>–</td>
<td>400–450</td>
</tr>
<tr>
<td>Distance vulva to tip of tail</td>
<td>–</td>
<td>5 000</td>
<td>–</td>
<td>–</td>
<td>–</td>
</tr>
<tr>
<td>Max. width of alae</td>
<td>–</td>
<td>–</td>
<td>–</td>
<td>–</td>
<td>45</td>
</tr>
<tr>
<td>Extension alae</td>
<td>Whole body</td>
<td>Whole body</td>
<td>Whole body</td>
<td>Whole body</td>
<td>Whole body</td>
</tr>
</tbody>
</table>

<sup>a</sup> Cervical papillae in Ali (1961) interpreted here as deirids.
According to Quentin & Wertheim (1975) the deirids (Cancrini et al. 1991) in length (Cancrini, Balbo & Iori 1991). Our measurements best fit the description of S. caudatus as atrophied, the tail consisting merely of a smooth stump, which at best has rugged edges. Our specimens possess about seven distinct, albeit short, cuticular extensions similar to those illustrated by Cancrini et al. (1991) for S. renatae (Fig. 2B). Despite these differences we have allocated our specimens to S. caudatus. Apart from the original description and their inclusion in some taxonomic reviews (Chabaud 1958; Ali 1961), we have not found any other references to S. caudatus in the literature. The measurements of the specimens collected during this study are included in Table 6.

**DISCUSSION**

Despite the fact that various studies on the helminths of guineafowls in South Africa have been conducted, direct comparisons between the results of these studies are not always possible, as they had different objectives. Ortlepp (1937, 1938a, b, 1963) studied the helminths of all the organs and the entire alimentary canal, but his work was of a taxonomic nature, based on incidental findings, and presented no epidemiological data. Crowe (1977) listed the helminth species recovered from the small intestine, caeca and rectum of guineafowls, but in his subsequent analysis grouped them as acanthocephalans, cestodes and nematodes respectively. The two studies providing data on the prevalence and intensity of the helminths are those of Saayman (1966) and Verster & Ptasinska-Kloryga (1987). However, Saayman (1966) only examined the intestinal tract and Verster & Ptasinska-Kloryga (1987) collected helminths from the gizzard, intestine and caeca. Thus, their data on species richness would not reflect worms located in e.g. the crop or proventriculus.

The study conducted on guineafowls in Burkina Faso by Vercruysse et al. (1985) lends itself best to comparison with ours, as they examined the complete alimentary tract, including the crop and proventriculus. Of the total of 13 helminth species collected by these authors, eight species coincide with species recovered from our hosts. If the single acanthocephalan present in the birds from Burkina Faso
is taken into account, this number will increase by one. Vercruysse et al. (1985) record the acanthocephalan *Mediorhynchus selengensis*, which has been synonymized with *M. gallinarum* by Schmidt & Kuntz (1977), and the nematodes *Cyrena parroti*, *S. suctoria*, *G. congolense* and *A. numidae*, which are also recorded in this study. In addition to these species, Vercruysse et al. (1985) recorded the cestode *Cotugnia digonopora* and the nematodes *Eucoleus fissispina* and *Dispharynx spiralis*.

**Nematodes**

*Cyrena*

With the exception of *C. parroti*, helminths were recovered from their usual predilection sites. According to Anderson (1992) members of the genus *Cyrena* occur in the proventriculus of birds and he records *Cyrena colini* in the wall of the proventriculus near the gizzard of Bobwhite quails. We did not recover *C. parroti* from the proventriculus, but in all infected guineafowls the parasites were situated under the gizzard lining and could only be seen after the horny layer had been removed. There seemed, however, to be a preference for the proventricular-gizzard isthmus as described for *Cyrena neeli* from wild turkeys in the south-eastern United States (Davidson, Hon & Forrester 1977). Similarly, *C. parroti* recovered from Helmeted guineafowls in Burkina Faso were also present in the gizzard (Vercruysse et al. 1985).

*Subulura*

The genus *Subulura* has a wide distribution in gallinaceous birds on the African continent and records exist from Zimbabwe, Tanzania, Ghana, Nigeria and Somalia (Nicholls, Bailey, Gibbons, Jones & Samour 1995; Nior, Ajanus, Agbede & Esievo 1999; Poulsen, Permin, Hindsbo, Yelifar, Nansen & Bloch 2000; Permin, Esmann, Hoj, Hove & Mukaratiwa 2002; Magwisha, Kassuku, Kyvsgaard & Permin 2002). However, the genus is not restricted to the African continent and, according to Yamaguti (1961) is a cosmopolitan species.

Ortlepp (1937) recovered *S. suchoria* in association with *S. dentigera* from guineafowls from various regions in South Africa and Swaziland and concluded that the two species had a wide distribution. Contrary to our findings, he found *S. dentigera* to be far more abundant than *S. suchoria*.

Verster & Ptasinska-Kloryga (1987) collected helminths from 48 guineafowls in the vicinity of Pretoria. *Subulura suchoria* was present in 11 and *S. dentigera* in three of the hosts examined. From these and our own results it is apparent that the two species, *S. suchoria* and *S. dentigera* often share the same habitat. It is difficult to judge from our data whether these two species are interactive and compete for the same resources. If so, *S. suchoria* would seem the stronger competitor as it consistently occurred in higher numbers than *S. dentigera*. However, the numbers of *S. dentigera* were not greater in hosts with relatively low burdens of *S. suchoria*, but rather the numbers of *S. dentigera* were low in these hosts as well. It is possible, that this association is similar to the major-minor species concept, as seen with *Theladorsagia circumcincta* and *Theladorsagia daviani* in sheep and goats.

A literature study confirms the dominance of *S. suchoria* in guineafowls and Vercruysse et al. (1985) recorded a 100% prevalence of *S. suchoria* from 103 Helmeted guineafowls in Burkina Faso. In addition to being the most prevalent nematode, these authors also found *S. suchoria* to be one of the most numerous parasites (26–1071 worms per host). *Subulura dentigera* was not reported from these hosts.

*Ascaridia numidae*

*Ascaridia numidae* is another nematode commonly encountered in Helmeted guineafowls and has been recorded from various geographic localities in Africa. The prevalence and intensity of this parasite varies greatly from 98.1% with a range of intensity from 1 to 1452 in hosts in Burkina Faso (Vercruysse et al. 1985) and 86.7% with intensities ranging from 1 to 504 in birds in Ghana (Hodasi 1976) to a low prevalence of 13% with a maximum of 19 worms per host in South Africa (Verster & Ptasinska-Kloryga 1987). In the present study *A. numidae* was present in a single host only.

*Gongylonema*

Both Hodasi (1976) and Vercruysse et al. (1985), record *G. congolense* from hosts they examined, with a prevalence of 48.9 and 73.8%, respectively. This indicates that *G. congolense* not only forms a regular part of the helminth community of guineafowls in South Africa, but throughout the African continent. With the exception of *Gongylonema ingluviolica* allegedly recorded by Ortlepp ("1937, 1938, unpublished records" cited by Verster & Ptasinska-Kloryga 1987), the absence of this genus in previous reports on helminths of guineafowls in South
Africa, is most likely due to the fact that earlier authors did not examine the crop of the hosts in their studies.

*Tetrameres*

While *Tetrameres numida* was recovered in low numbers from the Musina guineafowls, none of the more commonly reported species of this genus was present in our material. A second species, which has been recorded from guineafowls and is also a common parasite of domestic chickens, is *Tetrameres fissispina* Diesing, 1861. Verbrugse et al. (1985) report a 48.5% prevalence and an intensity of infection ranging from 1 to 146 worms per host from Helmeted guineafowls in Burkina Faso, and 23.3% of 126 Helmeted guineafowls in Nigeria were infected with *T. fissispina* (Fabiyi 1972). In Ghana the prevalence of infection in the same host was 8.9% with a mean worm burden of 2.8, ranging from one to eight. Young scavenging chickens in Ghana had a prevalence of *T. fissispina* of 58% (Poulsen et al. 2000).

We are aware of a single record of three females of *T. fissispina* from a single Helmeted guineafowl in South Africa (Le Roux 1926), and the same author reports a high percentage of infection (78%) in 60 domestic chickens in the same country. The proveentriculus of a single, heavily infected host contained a minimum of 150 females (Le Roux 1926).

A third species commonly infecting domestic chickens, namely *Tetrameres americana*, which had a 60 and 62% prevalence in adult chickens in Tanzania and Zimbabwe, respectively (Permin, Magwisha, Kassuku, Nansen, Bisgaard, Frandsen & Gibbons 1997; Permin et al. 2002), has not yet been recorded from guineafowls.

From the literature cited above it would appear that the prevalence of the genus *Tetrameres* is slightly higher in domestic chickens than in Helmeted guineafowls. Since the data above concern the domestic chickens above pertain to free-ranging or scavenging chickens, guineafowls and domestic fowls probably had an equal chance of exposure to the parasite. Whether the higher infection rates in chickens are a result of higher host densities or whether guineafowls are generally more resistant towards helminth infections remain speculation.

**Trematodes**

The presence of young stages of *Porogynia paronai* in the liver of infected hosts is unusual. Hodasi (1976), however, recovered adult *Cotugnia meleagridis* from the small intestine of Helmeted guineafowls in Ghana, and recorded numerous young forms of this parasite from the host’s gallbladder. Since the life cycle of *Porogynia* is not known, one can only speculate on the presence of immatures in the liver.

During the normal course of cestode development in avian hosts, the cysticercoid is freed from the arthropod intermediate host in the intestine as a result of mechanical and chemical actions. Subsequently, the scolex evaginates and the cysticercoid attaches itself to the gut wall (Reid 1962). The fact that young *P. paronai* were recovered from the liver of three birds and in relatively high numbers, in addition to
their uniform stage of development, suggests that their presence is not a result of post-mortem migration. Whether the newly freed cysticercoid, assuming that an arthropod is the intermediate host, migrates up the common bile duct to mature to a certain stage, before leaving the liver to resume its final maturation in the small intestine, or whether we have observed aberrant migration of juvenile stages will remain speculation until the development of *P. paronai* can be studied in more detail.

**Abuladzugnia**

Interestingly, the cestode *A. gutterae*, which was common in the guineafowls examined by us was not found in any of the previous surveys. Ortlepp (1963) originally described this species as *Cotugnia gutterae* from three specimens that had been collected from Crested guineafowls in Mozambique. Since then there seem to have been no further records of this parasite. Spasskii (1973) created the genus *Abuladzugnia* to accommodate *A. gutterae* and another of Ortlepp’s (1963) species formerly described as *Cotugnia transvaalensis*.

**Conclusion**

The above findings suggest, that despite geographical variation in the prevalence and intensity of individual helminth species, probably caused by environmental conditions, such as temperature, rainfall and soil conditions, the helminth community of guineafowls in Africa is composed of a relatively stable body of core and secondary species enriched by satellite species. The latter probably depend on local conditions and can be influenced by abiotic conditions, but also the presence or absence of certain intermediate hosts and other terrestrial birds which may serve as reservoir hosts for certain parasites. We interpret the relative uniformity in the helminth community of Helmeted guineafowls in Africa as flowing from a long host/parasite association during which parasites have spread in conjunction with their hosts.

**ACKNOWLEDGEMENTS**

We thank Dr W.J. Luus-Powell, University of Limpopo, who enabled the collection of Helmeted guineafowls in Musina and Mr H.E. Hattingh, University of Limpopo, for collecting and placing the Mokopane material at our disposal as well as for collecting the birds at Musina. Mr K. Meyer and Mr M. Storm kindly made the guineafowls available to us. The technical assistance of Mr. Ryno Watermeyer, University of Pretoria, especially with the processing of the cestodes, is greatly appreciated. This study was made possible by a Claude Leon Foundation Postdoctoral Fellowship grant to the first author.

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INTRODUCTION

Despite the remarkable diversity of South African birdlife, knowledge concerning their helminth parasites is scant (Ortlepp 1937, 1938a, b, 1963; Verster-Patsinska-Kloryga 1987) and even sparser on the structure of their helminth communities.

A first step was taken by Crowe (1977), who compared the influence of sex, age and habitat on the intestinal helminths of Helmeted Guineafowls, *Numida meleagris* (Linnaeus, 1758), at Kimberley, Northern Cape Province, South Africa. Thereafter, Alexander & McLaughlin (1997) provided a comprehensive analysis of the helminth communities of four species of ducks at Barberspan, South Africa. It is also apparent from Bush’s (1990) chapter on helminth communities in avian hosts, that considerably more information on helminth community dynamics in birds from aquatic environments than those from terrestrial habitats is available.

This paper analyses the composition and structure of the helminth community of 15 Helmeted Guineafowls in the Limpopo Province, even though small numbers of hosts were available and a larger sample might have a different outcome. Data on the various
species with a prevalence of 70% and higher, were Holmes (1986a) and Alexander & McLaughlin (1997), by Holmes & Price (1986). As suggested by Bush & component parasite community is used as defined metapopulation follows Riggs & Esch (1987) and infracomunity follow Bush & Holmes (1986a, b), fertility, Lotz & Shostak (1997). Infrapopulation and Esch, Holmes, Kuris & Schad (1982) and Bush, Lazard and caeca respectively were calculated. To detect species association with presence-abundance, as well as their feeding guilds, feeding active on semi-solid food materials such as blood, bile, mucus and intestinal debris as well as directly absorbing nutrients through their tegumental surface, was restricted to the liver and represented by a single species. The nematode, being mucosal and engulfing tissue and/or luminal contents, occupied the largest number of sites along the alimentary canal, namely the crop, proventriculus, gizzard as well as the small and large intestine. The females of *Tetrameres numida* are an exception in so far as they inhabit the glands of the proventriculus, where they suck blood.

The cestodes and acanthocephalans occurred in the small intestine only. *Mediorhynchus gallinarum* has a short neck and its attachment to the mucosa remains superficial. The larger part of its abdomen is suspended freely in the intestinal lumen, absorbing nutrients via the body surface (Junker & Boomker 2007a). The terms prevalence, intensity and abundance are used in accordance with the proposals of Margolis, Esch, Holmes, Kuris & Schad (1982) and Bush, Lafferty, Lotz & Shostak (1997). Infrapopulation and infracomunity follow Bush & Holmes (1986a, b), metapopulation follows Riggs & Esch (1987) and component parasite community is used as defined by Holmes & Price (1986). As suggested by Bush & Holmes (1986a) and Alexander & McLaughlin (1997), species with a prevalence of 70% and higher, were categorized as core species, those with prevalences of <40% as satellite species and those with prevalences ≥40% but <70% as secondary species. A summary of these definitions is to be found in Esch, Shostak, Marcogliese & Goater (1990).

A Wilcoxon rank sum test was performed to determine differences in species richness, as well as in the abundance of the various species between male and female hosts, juveniles and adults and between birds shot in winter and spring. A variance ratio test of Schluter (1984) and McCulloch (1985) was used to detect species association with presence-absence data for all parasites, parasites in the small intestine (SI) and parasites in the caeca. 

Pairwise Spearman’s rank correlation for every possible species combination in the small intestine, gizzard and caeca respectively were calculated. To avoid possible distortions inherent to this form of analysis, double zero matches, i.e. absence of both species from a host, were eliminated. Of the 14 helminth species present in the small intestine only the single acanthocephalan and the 12 cestodes were included in the analysis, because the occurrence of two nematodes, *Ascaridia numidae* and an unidentified subulurid, was restricted to one and two hosts respectively, while a third nematode, *Sicarius caudatus*, utilized the SI as well as the gizzard. We tested for a correlation between *Subulura dentigera* and *Subulura suatoria* from the caeca only, as *Subulura* sp. most probably represents either of the former two nematodes.

Significance was set at the 95% level throughout. In the absence of scoleces, counts were not always possible for all the cestodes of a particular host. While these hosts were included in analyses based on presence/absence data, they were excluded from the sample pool in the Wilcoxon rank sum tests pertaining to the abundance of helminths at species level.

**RESULTS**

A total of 11 951 helminths representing ten nematode, 11 cestode and a single acanthocephalan species were recovered from the alimentary canal of the 15 guineafowls. Data on their prevalence, intensity of infection and abundance, as well as their feeding guild classification and status as core, secondary or satellite species are summarized in Table 1. In all likelihood, *Raillietina* sp. and *Subulura* sp. are representatives of the remaining species of these two genera listed in Table 1. A single trematode species, *Dicrocoelium macrostomum*, was present in five of nine guineafowls examined for this parasite. Although included in the general results and discussion, these trematodes do not form part of the community study outlined below.

Following the classification of Bush (1990) four feeding guilds, i.e. organisms using the same feeding-mode, without regard to their taxonomic affinity, were present in the helminth community. The trematode guild, feeding actively on semi-solid food materials such as blood, bile, mucus and intestinal debris as well as directly absorbing nutrients through their tegumental surface, was restricted to the liver and represented by a single species. The nematode guild, being mucosal and engulfing tissue and/or lumen contents, occupied the largest number of sites along the alimentary canal, namely the crop, proventriculus, gizzard as well as the small and large intestine. The females of *Tetrameres numida* are an exception in so far as they inhabit the glands of the proventriculus, where they suck blood.

helminth species collected have been presented in a companion publication (Junker & Boomker 2007a).

**MATERIAL AND METHODS**

During July 2005 to November 2006 the gastrointestinal helminths of 15 Helmeted Guineafowls on a farm about 60 km west of Musina (Messina), Limpopo Province (22°22′ S, 29°30′ E), were examined as detailed in Junker & Boomker (2007a). Three of the birds shot in May 2006 and two collected in July 2006 were young birds, between 6 and 10 months old (Siegfried 1966), the remainder were adults. Three of the juveniles were females and two males, and the adults comprised four females and six males.

The birds shot in winter and spring. A variance ratio test of infection and abundance, as well as their feeding guild classification and status as core, secondary or satellite species were summarized in Table 1. In all likelihood, *Raillietina* sp. and *Subulura* sp. are representatives of the remaining species of these two genera listed in Table 1. A single trematode species, *Dicrocoelium macrostomum*, was present in five of nine guineafowls examined for this parasite.

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The cestodes and acanthocephalans occurred in the small intestine only. *Mediorhynchus gallinarum* has a short neck and its attachment to the mucosa remains superficial. The larger part of its abdomen is suspended freely in the intestinal lumen, absorbing nutrients via the body surface (Junker & Boomker 2007a).
TABLE 1  Helminths recovered from 15 Helmeted Guineafowls in Musina, Limpopo Province, South Africa

<table>
<thead>
<tr>
<th>Parasite</th>
<th>Guild</th>
<th>Site</th>
<th>Status</th>
<th>No. of inf. hosts</th>
<th>Prevalence (%)</th>
<th>Intensity</th>
<th>Abundance</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td>Median</td>
<td>Mean</td>
</tr>
<tr>
<td><strong>Acanthocephala</strong></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Mediorhynchus gallinarum</td>
<td>L</td>
<td>SI</td>
<td>C</td>
<td>15</td>
<td>100</td>
<td>23</td>
<td>55.7</td>
</tr>
<tr>
<td><strong>Cestoda</strong></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Abuladzugnia guttata</td>
<td>L</td>
<td>SI</td>
<td>C</td>
<td>12</td>
<td>80</td>
<td>13</td>
<td>11.7</td>
</tr>
<tr>
<td>Davainea nana</td>
<td>M</td>
<td>SI</td>
<td>Sat</td>
<td>5(^a)</td>
<td>36</td>
<td>6</td>
<td>5.8</td>
</tr>
<tr>
<td>Hymenolepis cantaniana</td>
<td>M</td>
<td>SI</td>
<td>Sec</td>
<td>6</td>
<td>40</td>
<td>3</td>
<td>42.7</td>
</tr>
<tr>
<td>Numidella numida</td>
<td>L</td>
<td>SI</td>
<td>C</td>
<td>10</td>
<td>67</td>
<td>14</td>
<td>55.9</td>
</tr>
<tr>
<td>Octopetalum numida</td>
<td>L</td>
<td>SI</td>
<td>C</td>
<td>10</td>
<td>67</td>
<td>79</td>
<td>91.9</td>
</tr>
<tr>
<td>Ortleppolepis multucincata</td>
<td>M</td>
<td>SI</td>
<td>C</td>
<td>13</td>
<td>87</td>
<td>11</td>
<td>9.3</td>
</tr>
<tr>
<td>Porogynia paronai</td>
<td>L</td>
<td>SI</td>
<td>Sec</td>
<td>7</td>
<td>47</td>
<td>6.5</td>
<td>12.3</td>
</tr>
<tr>
<td>Raillietina angusta</td>
<td>L</td>
<td>SI</td>
<td>Sec</td>
<td>8</td>
<td>53</td>
<td>10</td>
<td>10.3</td>
</tr>
<tr>
<td>Raillietina pintneri</td>
<td>L</td>
<td>SI</td>
<td>C</td>
<td>12</td>
<td>80</td>
<td>4</td>
<td>5.3</td>
</tr>
<tr>
<td>Raillietina steinhardtii</td>
<td>L</td>
<td>SI</td>
<td>Sec</td>
<td>8</td>
<td>53</td>
<td>27.5</td>
<td>49.0</td>
</tr>
<tr>
<td>Raillietina sp.</td>
<td>L</td>
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<td>C</td>
<td>11</td>
<td>73</td>
<td>13</td>
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<tr>
<td><strong>Nematoda</strong></td>
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<td></td>
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<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Ascaridia numidae</td>
<td>N</td>
<td>SI</td>
<td>Sat</td>
<td>1</td>
<td>6</td>
<td>4</td>
<td>4.0</td>
</tr>
<tr>
<td>Cynnea parroti</td>
<td>N</td>
<td>Gizz</td>
<td>C</td>
<td>15</td>
<td>100</td>
<td>7</td>
<td>13.8</td>
</tr>
<tr>
<td>Gongylonema congolense</td>
<td>N</td>
<td>Crop</td>
<td>Sec</td>
<td>6</td>
<td>40</td>
<td>19</td>
<td>23.0</td>
</tr>
<tr>
<td>Hadjelia truncata</td>
<td>N</td>
<td>Gizz</td>
<td>Sec</td>
<td>8</td>
<td>53</td>
<td>2</td>
<td>1.6</td>
</tr>
<tr>
<td>Sicarius caudatus</td>
<td>N</td>
<td>Gizz, SI</td>
<td>Sec</td>
<td>8</td>
<td>53</td>
<td>1.5</td>
<td>2.1</td>
</tr>
<tr>
<td>Subulura dentigeri</td>
<td>N</td>
<td>Caeca</td>
<td>Sec</td>
<td>8</td>
<td>53</td>
<td>15</td>
<td>15.9</td>
</tr>
<tr>
<td>Subulura succoria</td>
<td>N</td>
<td>Caeca</td>
<td>C</td>
<td>15</td>
<td>100</td>
<td>345</td>
<td>536.3</td>
</tr>
<tr>
<td>Subulura sp.</td>
<td>N</td>
<td>Caeca</td>
<td>Sec</td>
<td>6</td>
<td>40</td>
<td>15</td>
<td>44.0</td>
</tr>
<tr>
<td>Unidentified subulurid</td>
<td>N</td>
<td>SI</td>
<td>Sat</td>
<td>2</td>
<td>13</td>
<td>2.5</td>
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<tr>
<td>Tetrameres numida</td>
<td>N</td>
<td>Prov</td>
<td>Sat</td>
<td>5</td>
<td>33</td>
<td>2</td>
<td>2.4</td>
</tr>
</tbody>
</table>

\(^a\) Data from 14 hosts only

L = lumenal absorber; M = mucosal absorber; N = nematode
SI = small intestine, Gizz = gizzard, Prov = proventriculus
C = core species; Sat = satellite species; Sec = secondary species
2006). It is therefore included in Bush’s (1990) category of lumenal absorbers, together with the majority of the larger cestodes. Small and delicate cestodes such as Davainea nana, Hymenolepis cantaniana and Ortiepope lis multiuncinata, whose entire body is virtually buried amongst the mucosal villi, constitute the fourth guild, namely that of mucosal absorbers.

Except for the monoxenous nematode Ascaridia numidae, all members of the component community have indirect life-cycles. The component community comprised nine core species as well as nine secondary species, each representing 40.9% of the total number of species, and four satellite species accounting for 18.2% of the species present (Table 1). Despite their prevalence of 67% being slightly below the 70% threshold, we have arbitrarily included Numidella numida and Octopetalum numida with the core species, as they were two of the most numerous helminths recovered in this study. The core species accounted for 91% of all individuals, the secondary species for

<table>
<thead>
<tr>
<th>Host</th>
<th>Number of species</th>
<th></th>
<th></th>
<th>Total number of species</th>
</tr>
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<tr>
<td></td>
<td>Acanthocephalans</td>
<td>Cestodes</td>
<td>Nematodes</td>
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</tr>
<tr>
<td>GFM1</td>
<td>1</td>
<td>9</td>
<td>6</td>
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<td>12</td>
</tr>
<tr>
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<td>1</td>
<td>7</td>
<td>5</td>
<td>13</td>
</tr>
<tr>
<td>GFM4</td>
<td>1</td>
<td>7</td>
<td>4</td>
<td>12</td>
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</tr>
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<td>6</td>
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<td>12</td>
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<td>Average</td>
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<td>11.6</td>
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<tr>
<td>SD</td>
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<td>1.2</td>
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</tr>
<tr>
<td>Range</td>
<td>1</td>
<td>5–9</td>
<td>2–6</td>
<td>8–16</td>
</tr>
</tbody>
</table>

FIG. 1 Frequency distribution of the total number of helminth species found in Helmeted Guineafowls in Musina, Limpopo Province, South Africa. The number of individuals hosts infected by a certain number of helminth species is indicated by the vertical bars.

FIG. 2 The frequency distribution of the total number of helminths in individual Helmeted Guineafowls in Musina. Helminth burdens were grouped into size classes (0–400, 401–800, etc. to 2 000+ helminths per guineafowl) represented on the x-axis. The y-axis displays the number of guineafowls infected with a certain size class of helminth burdens.
TABLE 3  Pairwise Spearman’s rank correlations for helminths in the small intestine of Helmeted guineafowls. Only species significantly correlated with at least one other species have been included. The correlation coefficients are displayed in the upper right corner of the matrix. The lower left half includes the respective p-values for each pair of species.

<table>
<thead>
<tr>
<th></th>
<th>Core species</th>
<th>Secondary species</th>
<th>Satellite species</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>1</td>
<td>2</td>
<td>3</td>
</tr>
<tr>
<td>1. Mediorhynchus gallinarum</td>
<td>0.576*</td>
<td>–0.464</td>
<td>0.096</td>
</tr>
<tr>
<td>2. Numidella numida</td>
<td>0.031*</td>
<td>–0.662*</td>
<td>–0.570</td>
</tr>
<tr>
<td>3. Octopetalum numida</td>
<td>0.082</td>
<td>0.010*</td>
<td>0.303</td>
</tr>
<tr>
<td>4. Ortleppolepis multiuncinata</td>
<td>0.820</td>
<td>0.140</td>
<td>0.467</td>
</tr>
<tr>
<td>5. Raillietina pintneri</td>
<td>0.860</td>
<td>0.228</td>
<td>0.347</td>
</tr>
<tr>
<td>6. Hymenolepis cantaniana</td>
<td>0.528</td>
<td>0.463</td>
<td>0.005*</td>
</tr>
<tr>
<td>7. Porogynia paronai</td>
<td>0.498</td>
<td>0.879</td>
<td>&lt;0.0001*</td>
</tr>
<tr>
<td>8. Raillietina angusta</td>
<td>0.574</td>
<td>0.375</td>
<td>0.915</td>
</tr>
<tr>
<td>9. Raillietina steinhardti</td>
<td>0.092</td>
<td>0.113</td>
<td>0.474</td>
</tr>
<tr>
<td>10. Davainea nana</td>
<td>0.146</td>
<td>0.255</td>
<td>0.010</td>
</tr>
</tbody>
</table>

* Data pertaining to significantly correlated pairs (P < 0.05) are in bold and marked with an asterisk.
8.6% and satellite species made up 0.4% of the total worm count.

Infracommunities in the Helmeted Guineafowls from Musina were moderately species rich, ranging from 8 to 16 species, with a mean number of 11.6 ± 2. Sixty percent of the hosts were infected with 12 or more species (Table 2, Fig. 1). The total number of helminths in individual guineafowls was highly aggregated and ranged from 66 to 2,724. In ten of the 15 hosts the intensity of infection was below 800 (Fig. 2), but three guineafowls had worm burdens of 1,457, 1,496 and 2,724 worms and when combined, the abundance of this parasite was thus significantly higher in juvenile birds than in adults (P = 0.0156).

The pairwise Spearman’s rank correlation test yielded 11 significantly correlated species pairs in the small intestine, of which one was positive and 10 negative. The results are presented in Table 3. The gizzard nematodes, C. parroti and H. truncata, were positively correlated, whereas S. dentigera and S. suctoria from the caeca were negatively correlated. Both results were, however, not significant.

DISCUSSION

Helmeted Guineafowls are non-selective omnivores feeding on a large variety of dietary items that, among others, include arthropods. Saayman (1966) recovered a wide variety of prey taxa, namely Orthoptera (four families), Coleoptera (five families), Isopota, Hemiptera, Lepidoptera, Hymenoptera, Hymenoptera, Diptera, Myriapoda and Araneida, from the crops of 36 Helmeted Guineafowls in the Eastern Cape Province.

Notwithstanding their being a sedentary species, the birds can cover a considerable distance during their daily forays (Del Hoyo, Elliot & Sargatal 1994). These characteristics and a well structured, complex alimentary canal are among the major host factors contributing to parasite community richness (Kennedy, Bush & Aho 1986). This might explain why, despite the harsh climatic conditions and the largely undiversified mopani (Colophospermum mopane) veld habitat of the study area (Acocks 1988), the helminth community of Helmeted Guineafowls from Musina is diverse. The inclusion of live food in their diet, up to 12% of the annual total, but higher during the summer months when insects are abundant (Mentis, Poggenpoel & Maguire 1975), may also account for the dominance of helminths with an indirect life cycle in the guineafowls.

We attribute the low prevalence and intensity of D. nana and especially of A. numidae to the arid environment characteristic of the study area. Ascaridia spp. were also the only nematodes with a direct life cycle recovered from guineafowls by Verster & Pta-sinska-Kloryga (1987). Their eggs are resistant and can survive for several months in suitable moist soil conditions (Anderson 1992), but these were certainly not met in the present study area. Furthermore, earthworms can harbour eggs and larvae, thus serv-
ing as paratenic hosts (Anderson 1992), but environmental conditions were not conducive for this route of transmission either. No information is available as to the intermediate hosts of *D. nana*, but we assume that they are similar to those used by the congeneric Davainea progolltina, namely snails and slugs (Anderson 1992). Little evidence of these invertebrates was seen in or around water troughs and one would not expect them to occur in large number under the prevailing conditions. Thus, despite the high numbers of available final hosts, the scarcity of intermediate hosts seems to limit these parasites.

Similarly, the absence of the trematode guild from the small intestine of the guineafowls appears to be related to the availability of intermediate hosts. Most digeneans are dependant on molluscs or very rarely an annelid intermediate host for completion of their life cycles (Gibbons, Jones & Khalil 1996). Hence, they are more frequently associated with an aquatic habitat. Hodasi (1969, 1976) concluded that trematodes were rare parasites in gallinaceous birds.

It is difficult to determine the host specificity of helminths, and whether a certain parasite is regarded as a specialist or a generalist is often subjective, especially as helminths which are specialists in a certain host can nevertheless occur in other, often related hosts (Bush 1990). Based on the host-parasite check list of guineafowls of Junker & Boomker (2007b), we consider the following helminths as generalists: *S. suertoria*, *G. congolense*, *H. truncata* and possibly *A. numidae* as well as the cestode *H. cantianiana* and the eacanthocephalan *M. gallinarum*. Each of these has been reported from a variety of hosts.

Many of the remaining helminths collected during this study are currently recorded from the guineafowl genera *Numida* and/or *Guttera* only, such as the nematodes *S. dentigera* and *T. numida* or the cestodes *O. numida*, *Railletina angusta*, *Railletina pinnleri* and *Railletina steinhardtii*. *Numidella numida* which is equally common in guineafowls in the USA was also found in turkeys and domestic chickens in that country. However, failure to experimentally infect the latter hosts with the parasite (Jones 1933) led Reid (1962) to believe that chickens and turkeys were not natural hosts. While *S. dentigera*, *T. numida*, *N. numida*, *O. numida*, *R. angusta*, *R. pinnleri* and *R. steinhardtii* would therefore seem to be specialists in guineafowls, this, at best tentative, classification might simply reflect a general lack of data and could well change as more information on other gamebirds, such as korhaans, bustards, francolins, spurfowls and quails, becomes available. In an environment where high temperatures combined with low rainfall jeopardize successful completion of helminth life cycles, spreading the risk of transmission between various final hosts would appear a more reliable way to assure high parasite survival rates than a specialist approach. We would therefore expect the generalists to outweigh the specialists.

Similarly, helminths collected in this study use a wide range of intermediate hosts and are often not limited to a specific host or even host taxon. *Numidella numida*, for example, is reported to use ground and dung beetles as well as grasshoppers (Reid 1962), and the common nematode, *S. suertoria* makes use of coleopterans, dermapterans and orthopterans (Anderson 1992). This strategy of spreading the risk of transmission between several intermediate hosts, all serving as prey to the final host, might well explain the aforementioned helminths’ success in colonizing their final hosts, resulting in a prevalence of 100% in *S. suertoria*, even under adverse environmental conditions. However, intermediate host data are usually very generalized in respect of the taxonomic status of the hosts. Hence, as more life cycle data become available especially elucidating parasite-intermediate host associations at species level, this picture of lack of specificity might change.

The aggregated pattern of dispersion seen in our data is common in parasite communities (Pielou 1974; Bush & Holmes 1986a, b; Alexander & McLaughlin 1997) and is a result of a number of factors, such as differences in the individual host’s immune competence, feeding preferences and species specific host behaviour (Petney, Van Ark & Spickett 1990; Horak & Boomker 2000). Saayman (1966) demonstrated a pronounced difference in feeding-preferences between different members of the same guineafowl flock both in the amount of food consumed as well as the composition of crop contents. The higher the food intake and the higher the percentage of insect matter in the individual’s diet, the higher the probability of ingesting an infected intermediate host and becoming infected.

A further reason for the aggregation of helminths in certain host individuals is the fact that a single infected guineafowl can excrete hundreds of nematode eggs in its faeces and a single tapeworm proglottid can contain hundreds of hexacanth larvae. Consequently, dung beetles, or other insects, feed-
ing on contaminated faeces or around contaminated patches can be exposed to large numbers of parasite eggs during a single meal. Reid (1962) records up to 50 cysticercoids of *N. numida* in infected intermediate hosts, up to 930 cysticercoids of *Skrjabinia cesticillus* were present in a single beetle, while dung beetles have been found to contain 100 or more cysticercoids of *H. cantiana*. Thus the ingestion of a single infected intermediate host can lead to the presence of a large number of helminths in individual final hosts.

Nine core species were identified within the helminth community of Helmeted Guineafowls at Musina. The helminth infracomunity of a single Crested Guineafowl, *Guttera edouardi*, from a nearby locality examined by Junker & Boomker (2007a) suggests a considerable overlap between the two parasite communities. Nine helminth species were present in the Crested Guineafowl, of which seven are core species and two are secondary species in Helmeted Guineafowls. This can probably be attributed much the same feeding habits, exposing them to a similar pool of intermediate arthropod hosts.

Core species are usually the first to appear in juvenile birds (Hair 1975) and our data reflect the high colonization ability of these species, in that their proportional density in juvenile birds was distinctly higher than that seen in the overall host population (60.6% vs 40.9%). In contrast, the percentage of secondary species in juvenile birds was 34.8% compared to 40.9% in the overall population, and satellite species averaged 4.6% in comparison with an overall average of 18.2%.

Pairwise Spearman’s rank correlation detected 11 significant correlations between helminth species in the small intestine. Of these, the only significant positive correlation occurred between the acanthocephalan *M. gallinarum* and the cestode *N. numida*, in that their intensities increased or decreased in unison. Positive associations between species can be due to several factors, amongst others the use of a common intermediate host. In this case a positive association in the source community would merely be transferred to the target community and would not necessarily reflect an interaction of the two species in the final host (Lotz & Font 1994). As is the case with many of the other parasite species collected in our study, there is no data on the intermediate hosts used by *M. gallinarum* in South Africa. Its North American counterpart, *Mediorhynchus grandis*, however, has been reported to use several species of grasshoppers as intermediate hosts (Moore 1962), and grasshoppers also form part of the life cycle of *N. numida* (Mohler 1936; Reid 1962). Whether a source community is the origin of the positive correlation between these two species, or if one parasite indeed changes the habitat in the final host in such a way as to facilitate the colonization by the other, would necessitate experimental studies. Conversely, *N. numida* had a significant negative correlation with *O. numida*, which also uses orthopterans as intermediate hosts (Gwyun & Hamilton 1935), and *O. numida* was negatively, albeit not significantly so, correlated with *M. gallinarum*.

Another positive correlation, although not significant (*P = 0.0819*), was found between *C. parroti* and *H. truncata* in the gizzard. The few data available on their intermediate hosts suggest that these do not overlap. *Cyreena parroti* has been reported from orthopteran intermediate hosts and *H. truncata* from beetles (Anderson 1992). Their positive correlation might be a result of the fact that both seem to make use of a window period during the development of their host in which the latter is more susceptible to infection (see below).

Negative correlations between species, where an increased intensity of the one leads to a decreased intensity of the other, may result from competition for resources such as carbohydrates or attachment sites (Smyth & McManus 1989). Or it could indicate that the presence of one species alters the habitat to such an extent that it is less suitable for the other. Smyth & McManus (1989) report a number of substances that are produced by *Hymenolepis diminuta* and which might act as inhibitory factors, producing a crowding effect. Moreover, the host’s immune response triggered by a certain species could well make this host less susceptible to subsequent colonization by other parasites.

Some of the factors influencing parasite community patterns in other hosts seem to be of little importance in structuring the helminth communities of Helmeted Guineafowls. One of these is age. Moore, Freehling, Horton & Simberloff (1987) concluded that age can occasionally have an important influence on the prevalence and intensity of helminth infections of Bobwhite Quail, *Colinus virginianus* (Linnaeus, 1758), and Pence (1990) reported changes in host age over seasons to be one of the factors most frequently cited when discussing prevalence and intensity. However, in the present study neither overall abundance nor species richness in juvenile guineafowls differed significantly from those in adults.

In contrast, Crowe (1977) reported that juvenile Helmeted Guineafowls, i.e. birds younger than 10
months, from the Kimberley district, South Africa had significantly higher burdens of cestodes and acanthocephalans than adults, and Davies, Junker, Jansen, Crowe & Boomker (in preparation) found higher burdens of *S. suatoria*, *O. numida* and *M. gallinarum* in juveniles during a study on Helmeted Guineafowls in the Free State Province. Forrester, Conti, Bush, Campbell & Frohlich (1984) found no significance in the differences between the prevalence of helminth species in chick and adult bobwhites, but the intensity of infection of a single helminth species was higher in chicks than in adults. When studying the helminth communities in willets, *Tringa semipalmatus* (Gmelin, 1789) (= *Catoptrophorus semipalmatus*), both on their breeding grounds (freshwater) and in their wintering habitat (saltwater), Bush (1990) found young birds to be depauperate, but within the course of 2 weeks the diversity of their helminth communities increased considerably and, in the case of helminths with freshwater life cycles, at 3 months of age no longer differed from those of adult birds.

Several factors could influence the prevalence and intensity of helminths in guineafowls of different ages. Young birds might well be more susceptible to helminth infections when compared to adults, as has been suggested by a number of authors (Ackert & Reid 1937; Biester & Schwarte 1959; Soulsby, 1969). However, this would be counterbalanced by a time-dependant higher probability of previous exposure to infected intermediate hosts, and thus to the various parasites, in older birds, therefore evening out differences between different ages on component community level. On the other hand, it is well documented that the diet of juvenile Helmeted Guineafowls and other gamebirds consists of a larger percentage of arthropods than that of adults (Del Hoyo et al. 1994; Crowe 2000), increasing their exposure to possible intermediate hosts.

Some age-related differences on metapopulation level, i.e. when singling out certain parasite species from the Musina hosts, were observed. *Cyrnea parroti*, whose predilection site is under the lining of the gizzard, was significantly more abundant in juvenile birds. We observed a distinct hardening of the gizzard lining in adult guineafowls which was not nearly as pronounced in the younger birds and which could easily impede establishment of this parasite in older hosts. This phenomenon might also explain why the prevalence of *H. truncata*, using the same site, decreased from 80% in juveniles to only 40% in older guineafowls. Dogiel (1964) suggested that the normal development of a host, such as a thickening of skin, could result in a habitat being no longer suitable for the parasite, hence leading to resistance against the latter.

The same mechanism is obviously not in play with *G. congolense*, which lives in tunnels under the crop mucosa. While the observed thickening of the mucosa should make colonization with *G. congolense* more difficult with increasing host age, this parasite was not found in any of the younger birds, but was present in 60% of the older hosts. A possible explanation might be that *G. congolense* is only a secondary species indicating that its prevalence in the entire ecosystem is lower than that of a core species such as *C. parroti*. Consequently, age, if seen as an increase of the probability of prior exposure to a certain parasite with time, might have a more pronounced influence on the distribution pattern of this particular parasite. Using the same reasoning, one could expect the prevalence of *H. truncata*, also a secondary species, to increase in adult birds. As has been discussed this is not the case. However, the hardening of the crop mucosa never seems as pronounced as that of the gizzard’s and, while the latter would seem likely to form a suitable barrier against the establishment of parasites, this is not necessarily so in the former.

Similarly to *C. congolense*, the cestode *P. paronai* had a significantly higher abundance in adult guineafowls, being absent in young birds. Little is known about the life cycle of this parasite except that it is one of the cestodes making use of sites other than the small intestine, in this case the bile ducts of guineafowls (Smyth & McManus 1989). Junker & Boomker (2007a) have reported immature stages of this parasite from the liver/bile ducts and adults from the small intestine of Helmeted Guineafowls. Whether morphological changes, such as the size of the bile ducts, or biochemical changes, such as the bile composition, during the ontogenesis of the guineafowl hosts in some way facilitate the migration and establishment of developing *P. paronai* has to remain speculation. On the other hand, a change in the prey preference in growing birds, possibly taking larger prey items not formerly included in the diet, may expose older guineafowls to a wider range of parasites.

The significantly higher abundance of *Subulura* sp. in juvenile birds can be attributed to the fact that the population of *Subulura* spp. in these hosts was mainly represented by infective larvae that do not yet display sufficient diagnostic characters to distinguish between the two species *S. dentigera* and *S.
Helminth community of Helmeted Guineafowls in Limpopo Province, South Africa

suctoria. We consider this a result of the fact that infections in the juvenile hosts had been recently acquired, thus comprising a high number of immature, as opposed to the more mature infections found in older guineafowls.

Host gender was another determinant that had no significant influence on the distribution of worm burdens and species richness within the guineafowl population from Musina. Similar results were obtained by Crowe (1977), who attributed the absence of sexual variation in helminth infections to the fact that there is little behavioural or dietary difference between sexes outside the breeding season. All hosts in our and also Crowe’s (1977) study were collected during the non-breeding season, extending from March to October in South Africa (Del Hoyo et al. 1994). Helmeted Guineafowls collected in the Free State Province during August 2007, however, showed sex related differences regarding the intensities of some helminths (Davies et al. in preparation). Possible reasons for this given by the latter authors are a difference in the length of the small intestine and caeca between males and females, as demonstrated by Prinsloo (2003), resulting in a larger habitat in the females. Moreover, females have a relatively higher intake of insects prior to breeding, which is often aided by the male’s foraging for its mate (Hockey, Dean & Ryan 2005).

When discussing helminth communities in avian hosts, Bush (1990) concluded that host age and sex played a minor role, whereas the overall environment and habitat diversity therein exercised a major influence on the patterns of helminth communities. He argued that the latter would directly influence the “supply” of helminths available in the system. Keeping in mind that the current set of data was based on a limited number of hosts, and that a larger sample size might change the emerging picture, it nevertheless suggests that Helmeted Guineafowls are no exception to this general pattern.

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A check list of the helminths of guineafowls (Numididae) and a host list of these parasites

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ABSTRACT

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Published and personal records have been compiled into a reference list of the helminth parasites of guineafowls. Where data on other avian hosts was available these have been included for completeness’ sake and to give an indication of host range. The parasite list for the Helmeted guineafowls, *Numida meleagris*, includes five species of acanthocephalans, all belonging to a single genus, three trematodes belonging to three different genera, 34 cestodes representing 15 genera, and 35 nematodes belonging to 17 genera. The list for the Crested guineafowls, *Guttera edouardi*, contains a single acanthocephalan together with 10 cestode species belonging to seven genera, and three nematode species belonging to three different genera. Records for two cestode species from genera and two nematode species belonging to a single genus have been found for the guineafowl genus *Acryllium*. Of the 70 helminths listed for *N. meleagris*, 29 have been recorded from domestic chickens.

Keywords: Acanthocephalans, cestodes, check list, guineafowls, host list, nematodes, trematodes

INTRODUCTION

Guineafowls (Numididae) originated on the African continent, and with the exception of an isolated population of Helmeted guineafowls in north-west Morocco, their natural distribution is restricted to sub-Saharan Africa (Del Hoyo, Elliott & Sargatal 1994). In the wake of commercial game bird farming, but also as ornamental birds in aviculture, they have been introduced to many other parts of the world, such as France, Hungary, Italy, Greece, the United Kingdom, the USA, Australia and different regions of the former USSR (Haziev & Khan 1991). According to Belshaw (1985) guineafowls were imported into the southern Mediterranean region several millennia before turkeys and hundreds of years before junglefowls from which today’s domestic chickens were derived. Currently four genera of guineafowls are recognized, namely *Acryllium* Gray, 1840, *Ageastes* Bonaparte, 1850, *Guttera* Wagler, 1832 and *Numida* Linnaeus, 1766 (Del Hoyo et al. 1994).

Many publications on the helminth fauna of guineafowls originate from northern and western Africa, where, second only to the introduced and native domestic fowls, they are farm-reared as a source of protein. The economic importance of guineafowls and domestic fowls within the poultry industry, as well as the fact that domestic fowls are kept by many private households to augment their income, necessitated a better understanding of factors, such as gastro-intestinal parasites, influencing the success-
ful rearing of these birds. Consequently studies have been conducted to assess the extent to which guineafowls and domestic fowls can serve as alternative hosts for their respective helminths and possibly be adversely affected by them (Hodasi 1969, 1976; Fabiyi 1972; Fatunmbi & Olufemi 1982; Vercruysse, Harris, Bray, Nagalo, Pangui & Gibson 1985).

In southern Africa Ortlepp (1937, 1938a, b, 1963), Saayman (1966), Crowe (1977) and Verster & Ptatsinska-Kloryga (1987) have published on the helminth fauna of guineafowls. No data on the helminths infecting species of the guineafowl genus *Agelastes* could be found, and we are of the opinion that the comparatively short parasite lists for the genera *Acyllium* and *Guttera* reflect a lack of data rather than an absence of parasites.

The check list herein is intended as a quick reference aid and is split into two sections. The first section contains the parasites listed under their scientific names and authorities. Synonyms are provided either as generic synonyms in the case where whole genera have been synonymized or specific synonyms. The second section lists the hosts and their synonyms alphabetically, together with their respective parasites, also in alphabetical order.

The synonymy of the acanthocephalan genus *Mediorhynchus* Van Cleave 1916 is as given by Van Cleave (1947) and Schmidt & Kuntz (1977) and specific synonymy is according to Yamaguti (1963). For an in-depth review of the involved history of this genus’s nomenclature the reader is referred to Van Cleave (1947).

The taxonomy of digenean trematodes follows Yamaguti (1958), but since the application of molecular techniques to this group has recently led to many changes, the reader is encouraged to consult the latest literature.

The classification of cestodes is based on the works of Khalil, Jones & Bray (1994). Information on generic synonyms and type species follows Khalil *et al.* (1994), while that on other species as well as the hosts and geographic distribution has mainly been derived from Yamaguti (1959), Schmidt (1986) and additional published records.

As regards nematode taxonomy, the authors have followed the CIH Keys to the nematode parasites of vertebrates (Anderson, Chabaud & Willmott, 1974–1983) and, where differences have occurred, have accepted the validity of genera and species as listed by Gibson (2005). With regard to generic synonyms, only synonyms listed in the CIH keys and by Gibson (2005) have been included in the check list. Specific synonyms, Type species and other species, as well as much of the data on hosts and geographic distribution are according to Yamaguti (1961) and Gibson (2005). Host and geographic data have been supplemented by including additional literature references.

The families and subfamilies of cestodes and nematodes are listed according to the system of Khalil *et al.* (1994) and the CIH Keys, respectively, but genera within these families are presented in alphabetical order. Synonyms have been arranged chronologically. The hosts and geographic localities per author are listed alphabetically. If several authors made reference to the same host, the authors are listed in chronological order.

The nomenclature and taxonomy of the avian hosts mainly follows Peterson (1999) and has been supplemented by Lepage (2007). Avian orders and families, as well as the nomenclature of southern African hosts follow Hockey, Dean & Ryan (2005).

In order to avoid excessive duplication, Helmeted guineafowls are listed below as *N. meleagris* only without regards to the subspecies. A total of nine subspecies of *N. meleagris* are currently recognized (Del Hoyo *et al.* 1994, Peterson 1999). These are: *N. m. coronatus* Gurney, 1868, *N. m. galeatus* Pallas, 1767, *N. m. marungensis* Schalow, 1884, *N. m. meleagris* (Linnaeus, 1758), *N. m. mitratus* Pallas, 1767, *N. m. papillosus* Reichenow, 1894, *N. m. reichenowi* Ogilvie-Grant, 1894, *N. m. sablyi* Hartert, 1919 and *N. m. somaliensis* Neumann, 1899. Del Hoyo *et al.* (1994) give a detailed list of the geographic range of the various subspecies of Helmeted guineafowls.


Hosts listed in the literature as *Gallus domesticus* or *Gallus gallus domesticus* are referred to below as domestic chicken. Lepage (2007) lists domestic chicken as unconfirmed subspecies, *G. g. domesticus* (no authority given), of the Red Junglefowl, *Gallus gallus* (Linnaeus, 1758). However, this subspecies is not included in the five subspecies listed by Peterson (1999).
PARASITE/HOST CHECK LIST

PHYLUM ACANTHOCEPHALA

Class Archiacanthocephala
Order Gigantorhynchidea
Family GIGANTORHYNCHIDAE Hamann, 1892

GENUS MEDIORHYNCHUS Van Cleave 1916

Echinorhynchus Zoega in Müller, 1776, in part; Gigantorhynchus Hamann, 1892; Empodius Travassos, 1916; Leiperacanthus Bhalariao, 1937; Disteganus Lehmann, 1953, nomen nudum; Empodisma Yamaguti, 1963

Type species: Mediorhynchus papillosus Van Cleave, 1918

1. Mediorhynchus empodius (Skrjabin, 1913) Meyer, 1933
   Ardea, Ardeotis arabs, Numida meleagris
   Yamaguti (1963), Belgium, Russia

2. Mediorhynchus gallinarum (Bhalerao, 1937) Van Cleave, 1947
   Domestic chicken
   Yamaguti (1958), North Africa
   Gallinaceous birds
   Schmidt & Kuntz (1977), (East-) Africa, India, Papua and New Guinea
   Numida meleagris
   Junker & Boomker (2006), South Africa

3. Mediorhynchus numidae (Baer, 1925) Meyer, 1933
   Numida meleagris
   Meyer (1932), Namibia
   Oosthuizen & Markus (1967), South Africa

4. Mediorhynchus selengensis Harris, 1973
   Numida meleagris
   Vercryusse et al. (1985), Burkina Faso
   Schmidt & Kuntz (1977) synonymized this species with M. gallinarum.

5. Mediorhynchus taeniatus (Von Linstow, 1901) Dollfus, 1936
   Empodius segmentatus De Marval, 1902
   Ardeotis arabs
   Dollfus (1951) in Yamaguti (1963), Mauritania
   Chlamydotis undulata
   Dollfus (1951) in Yamaguti (1963), Morocco
   Guttera edouardi
   Southwell & Lake (1939), Democratic Republic of the Congo
   Numida meleagris
   Graber (1959), Chad
   Von Linstow (1901), Kenya
   Meyer (1932), Africa, Malawi
   Southwell & Lake (1939), Democratic Republic of the Congo
   Graber (1959), Chad

PHYLUM PLATHYHELMINTHES

Class Trematoda
Order Digenea
Family BRACHYLAEMIDAE Joyeaux & Foley, 1930

GENUS POSTHARMOSTOMUM WITENBERG, 1923

Type species: Postharmostomum gallinum (Witenberg, 1923)

1. Postharmostomum gallinum (Witenberg, 1923)
   Crested guineafowl
   Yamaguti (1958), Russia
   Domestic chicken
   Lamaguti (1958), Hawaii, Japan, Russian Turkestan
   Numida meleagris
   Yamaguti (1958), North Africa

Family DICROCOELIIDAE Odhner, 1911

GENUS DICROCOELIUM DUDARDIN, 1845

Type species: Dicrocoelium lanceatum Stiles & Hassal, 1898

1. Dicrocoelium macrostomum Odhner, 1911
   Coturnix coturnix
   Yamaguti (1958), Russia
   Numida meleagris
   Lesbouyries (1941), Egypt
   Junker & Boomker (2007b), South Africa

GENUS LUTZTREMA TRAVASSOS, 1941

Type species: Lutztrema olliquum (Travassos, 1917)

Numida meleagris
Hodasi (1976), Ghana

Class Cestoda
Subclass Eucestoda
Order Cyclophyllidea
Family DAVAINEIDAE Braun, 1900
Subfamily Davainaeinae Braun, 1900

GENUS ABULADZUGNIA SPASSKII, 1973

Type species: Abuladzugnia gutterae (Ortlepp, 1963)

1. Abuladzugnia gutterae (Ortlepp, 1963)
   Cotugnia gutterae Ortlepp, 1963
   Guttera edouardi
   Ortlepp (1963), Mozambique
   Numida meleagris
   Junker & Boomker (2007b), South Africa
Check list of helminths of guineafowls (Numididae) and host list of parasites

2. **Abuladzugnia transvaalensis** (Ortlepp, 1963)
   - *Cotugnia transvaalensis* Ortlepp, 1963
   - *Numida meleagris*
     - Ortlepp (1963), South Africa

**GENUS Cotugnia, Di Mare, 1893**

*Ershovitugnia*, Spasskii, 1973; *Pavugnia* Spasskii, 1984; *Rosteu-lugnia* Spasskii, 1984

Type species: *Cotugnia digonopora* (Pasquale, 1890) Diamare, 1893

1. **Cotugnia crassa** Fuhrmann, 1909
   - Guineafowl
     - Hudson (1934), East Africa
     - Bwangamoi (1968), Uganda
   - *Numida meleagris*
     - Fuhrmann (1909) in Ortlepp (1963), the White Nile
     - Baer (1925), Namibia
     - Baer (1926), East Africa, West Africa
     - Ortlepp (1963), Tanzania
   - The White Nile rises from Lake Victoria in Uganda and enters the Sudan where it joins the Blue Nile in Khartoum to form the Nile. White Nile is one of the states of Sudan.

2. **Cotugnia digonopora** (Pasquale, 1890) Diamare, 1893
   - *Taenia digonopora* Pasquale, 1890
   - *Anser*, *Columba livia*, *Gallus gallus*, *Numida meleagris*
     - Schmidt (1986), Africa, Burma, India, Indonesia, Philippines
   - Guineafowl
     - Baylis (1934), Uganda

3. **Cotugnia meleagridis** Joyeux, Baer & Martin, 1936
   - *Numida meleagris*
     - Joyeux, Baer & Martin (1936), Northern Somalia
     - Graber (1959), Chad
     - Fabiyi (1972), Nigeria
     - Hodasi (1976), Ghana

4. **Cotugnia shohoi** Sawada, 1971
   - *Acryllium vulturinum*
     - Schmidt (1986), Somalia

5. **Cotugnia tuliensis** Mettrick, 1963
   - *Numida meleagris*
     - Schmidt (1986), Zimbabwe

**GENUS Davainea Blanchard, 1891**

Type species: *Davainea proglottina* (Davaine, 1860) Blanchard, 1891

1. **Davainea nana** Fuhrmann, 1912
   - *Guttera edouardi*
     - Ortlepp (1963), Zambia
   - *Numida meleagris*
     - Fuhrmann (1912), Northern Africa
     - Junker & Boomker (2007b), South Africa
   - *Vanellus cinereus*
     - Schmidt (1986), Africa, Japan

2. **Davainea paucisegmentata** Fuhrmann, 1909
   - *Numida meleagris*
     - Baer (1926), Sudan, West Africa
     - Schmidt (1986), Africa, Europe

3. **Davainea paucisegmentata var. dahomeensis** Joyeux & Baer, 1928
   - *Numida meleagris*
     - Schmidt (1986), France

4. **Davainea proglottina** (Davaine, 1860) Blanchard, 1891
   - *Taenia proglottina*
   - *Davainea variaiensis* Sweet, 1910; *Davainea dubious* Meggitt, 1916
   - *Alectoris graeca*, *Bonasa umbellus*, *Gallus gallus*, *Perduce perdivis* Schmidt (1986), Cosmopolitan
   - Domestic chicken
     - Baer (1926), South Africa
     - Magwisha, Kassuku, Kyvggaard & Permin (2002), Tanzania
   - *Numida meleagris*
     - Nfor, Ajianusi, Agbede & Esievo (1999), Nigeria

**GENUS Numidella Spasskay & Spasskii, 1971**

Type species: *Numidella numida* (Fuhrmann, 1912) Spasskay & Spasskii, 1971

1. **Numidella numida** (Fuhrmann, 1912) Spasskay & Spasskii, 1971
   - *Davainea numida* Fuhrmann, 1912; *Raillietina (Paronella) numida* (Fuhrmann, 1912)
   - *Davainea (Paronella) magnumunda* Jones, 1930
   - Guineafowl
     - Baylis (1934), Uganda
   - *Guttera, Numida meleagris*
     - Schmidt (1986), Africa, Cuba, North America
   - *Guttera*
     - Baer (1933), Zimbabwe
     - Baer (1933) lists *Guttera edouardi* Elliot as host. None of the subspecies of *Guttera edouardi* listed in Lepage (2007) has been described by Elliot, but Lepage (2007) lists *Guttera pucherani verreauxii* (Elliot, 1870).
   - *Meleagris gallopavo, Numida meleagris*
     - Jones (1930), North America
   - *Numida meleagris*
     - Baer (1925), Namibia
     - Ortlepp (1963), South Africa
     - Fabiyi (1972), Nigeria

**GENUS Porogynia Ralliet & Henry, 1909**

*Polycoelia* Fuhrmann, 1907, preoccupied

Type species: *Porogynia paronai* (Moniez, 1892) Ralliet & Henry, 1909

1. **Porogynia paronai** (Moniez, 1892) Ralliet & Henry, 1909
   - *Taenia paronai*
   - *Linstowia lata* Fuhrmann, 1901; *Polycoelia lata* (Fuhrmann, 1901)
   - *Malika numida* Woodland, 1929; *Raillietina (Paronella) woodlandii*
     - Baylis, 1934
**GENUS Kotlania**

Movsesyan, 1966; *Kotlanotaurus mania*

Type species: *Kotlania pintneri* (Klaptocz, 1906) Fuhrmann, 1924

**GENUS Raillietina**

Fuhrmann, 1920


Type species: *Raillietina tetragona* (Molin, 1858)

1. *Raillietina angusta* Ortlepp, 1963

2. *Raillietina cohnii* (Baczynska, 1914) Fuhrmann, 1924

3. *Raillietina echinobothrida* (Megin, 1880) Fuhrmann, 1924

4. *Raillietina pintneri* (Klaptocz, 1906) Fuhrmann, 1924

5. *Raillietina somalensis* Sawada, 1971

6. *Raillietina steinhardti* Baer, 1925

7. *Raillietina tetragona* (Molin, 1858) Fuhrmann, 1924

**GENUS Numida meleagris**

Baer (1925), Namibia

Baer (1926), East Africa, West Africa

Woodland (1928), Sudan

Ortlepp (1963), South Africa

Cruz e Silva (1971), Mozambique

Sawada (1955) Gutierrez (1931), Tanzania

Ayeni, Dipeolu & Okaeme (1983), Nigeria

71. *Guttera edouardi*, *Numida meleagris* Schmidt (1986), Africa

72. *Guttera edouardi* Ortlepp (1963), Mozambique, Zambia

73. *Numida meleagris* Schmidt (1986), Africa

74. *Numida meleagris* Persids (1986), Mozambique, Zambia

75. *Numida meleagris* Verster & Plasinska-Kloryga (1987), South Africa

76. *Numida meleagris* Verster & Plasinska-Kloryga (1987), South Africa

77. *Numida meleagris* Schmidt (1986), Cosmopolitan

**GENUS Gallus**

Baer (1923), Zimbabwe

Baer (1933) lists *Guttera edouardi* Elliot as host. None of the subspecies of *Guttera edouardi* listed in Lepage (2007) have been described by Elliot, but Lepage (2007) lists *Guttera pucheraneri* verreauxi (Elliot, 1870).

**GENUS Gallus**

Baer (1923), Zimbabwe

Baer (1933) lists *Guttera edouardi* as host. None of the subspecies of *Guttera edouardi* listed in Lepage (2007) have been described by Elliot, but Lepage (2007) lists *Guttera pucheraneri* verreauxi (Elliot, 1870).

**GENUS Gallus**

Baer (1923), Zimbabwe

Baer (1933) lists *Guttera edouardi* as host. None of the subspecies of *Guttera edouardi* listed in Lepage (2007) have been described by Elliot, but Lepage (2007) lists *Guttera pucheraneri* verreauxi (Elliot, 1870).
Check list of helminths of guineafowls (Numididae) and host list of parasites

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**Family DILEPIDIDAE** Railliet & Henry, 1909

**GENUS Choanotaenia** Railliet, 1896

Type species: *Choanotaenia infundibulum* (Bloch, 1779) Railliet, 1896

1. *Choanotaenia infundibulum* (Bloch, 1779) Railliet, 1896
   
   Domestic chicken
   
   Poulsen *et al.* (2000), Ghana
   
   Magwisha *et al.* (2002), Tanzania

**Numida meleagris**

Haziev & Khan (1991), Republic of Bashkortostan

Nfor *et al.* (1999), Nigeria

**Family PARUTERINIDAE** Fuhrmann, 1907

**GENUS OCTOPETALIDAE** Fuhrmann, 1914

Type species: *Octopetalum guttareae* Baylis, 1914

1. *Octopetalum guttareae* Baylis, 1914
   
   Ascometra guttareae (Baylis, 1914) Baer, 1955
   
   Guttera edouardi, *Numida meleagris*
   
   Baer (1926), East Africa
   
   Baer (1955), Democratic Republic of Congo, Malawi, South Africa
   
   Schmidt (1986), Africa, France

2. *Octopetalum numida* (Fuhrmann, 1909) Baylis, 1914
   
   Rhabdometra numida numida (Fuhrmann, 1909); *Octopetalum longicirrosum* Baer, 1925; *Uncinia sudanea* Woodland, 1928; Ascometra numida (Fuhrmann, 1909) Baer, 1955
   
   Guineafowl
   
   Baylis (1934), Uganda
   
   Guttera edouardi
   
   Baer (1955), Sub-Saharan Africa
   
   Ortlepp (1963), South Africa, Zambia

**Numida meleagris**

Baer (1925), Namibia

Baer (1926), Sudan, West Africa

Woodland (1928), Sudan

Baer (1955), Sub-Saharan Africa

Ortlepp (1963), Central Africa, North Africa, South Africa, southern Africa, Swaziland

Fabiyi (1972), Nigeria

Hodasi (1976), Ghana

**GENUS METROLIASTHES** Ransom, 1900

*Hexaparuterina* Palacios & Barroeta, 1967

Type species: *Metroliasthes lucida* Ransom, 1900

1. *Metroliasthes lucida* Ransom, 1900
   
   Alectoris graeca, Alectoris rufa, Coturnix coturnix, Gallus gallus, Gallus gallus bankiva, Guttera edouardi, Meleagris gallopavo, *Numida meleagris*, Perdix perdix
   
   Schmidt (1986), Africa, Australia, Europe, India, North and South America, Russia

**Numida meleagris**

Southwell & Lake (1939), Democratic Republic of Congo

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**GENUS SKRJABINIA** Fuhrmann, 1920

*Raillietina* (Skrjabinia) Fuhrmann, 1920; *Brumptiella* López-Neyra, 1920; *Armacetabulum* Moversyan, 1966; *Markewitchella* Spasskii & Spasskaya, 1972; *Daovantienia* Spasskii & Spasskaya, 1976

Type species: *Skrjabinia cesticillus* (Molin, 1858) Fuhrmann, 1920

1. *Skrjabinia cesticillus* (Molin, 1858) Fuhrmann, 1920
   
   *Taenia cesticillus* (Molin, 1858) Fuhrmann, 1920
   
   *Davainea cesticillus* (Molin, 1858) Baer, 1955
   
   *Ransomia deweti* (Molin, 1858) Fuhrmann, 1920
   
   *Colinus virginianus*, *Coturnix coturnix*, *Gallus gallus*, *Lagopus lagopus*, *Lagopus lagopus scotica*, *Lyurus tetrix*, *Meleagris gallopavo*, *Numida meleagris*, *Perdix perdix*, *Phasianus colchicus*, *Tetrao urogallus*, *Tetraastes bonasia*
   
   Schmidt (1986), Cosmopolitan
   
   Domestic chicken
   
   Baer (1926), West Africa
   
   Le Roux (1926), South Africa
   
   Joyeux *et al.* (1936), Northern Somalia
   
   Poulsen *et al.* (2000), Ghana
   
   Magwisha *et al.* (2002), Tanzania
   
   Permin *et al.* (2002), Zimbabwe

**Numida meleagris**

Nfor *et al.* (1999), Nigeria

2. *Skrjabinia deweti* Ortlepp, 1938

**Numida meleagris**

Ortlepp (1938a), South Africa

**Subfamily Idiogeninae** Fuhrmann, 1907

**GENUS IDIOGENES** Krabe, 1867

*Erisogenes* Spasskaya, 1961; *Paraiidiogenes* Moversyan, 1971

Type species: *Idiogenes otidis* Krabe, 1867

1. *Idiogenes sp.*
   
   **Numida meleagris**
   
   Hodasi (1976), Ghana

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Family HYMENOLEPIDIDAE Ariola, 1899

Subfamily Hymenolepidinae Perrier, 1897

GENUS ECHINOLEPIS SPASSKII & SPASSKAYA, 1954

Type species: Echinolepis carioca (Maghalaës, 1898) Spasskii & Spasskaya, 1954

1. Echinolepis carioca (Maghalaës, 1898) Spasskii & Spasskaya, 1954
   Davainea carioca Maghalaës, 1898; Taenia conardi Zürn, 1898; Hymenolepis carioca (Maghalaës, 1898) Ransom, 1902; Hymenolepis puliae Cholodkovsky, 1913; Weinlandia rustica Meggitt, 1926; Hymenolepis rustica Fuhrmann, 1932; Dicranotaenia carioca (Maghalaës, 1898) Skrjabin & Mathevossian, 1945; Dicranotaenia rustica (Meggitt, 1926) Skrjabin & Mathevossian, 1945
   Alectoris graeca, Bonasa umbellus, Colinus virginianus, Coturnix coturnix, Gallus gallus, Meleagris gallopavo
   Schmidt (1986), Cosmopolitan
   Domestic chicken
   Le Roux (1926), South Africa
   Magwisha et al. (2002), Tanzania
   Numida meleagris
   Baer (1926), West Africa

GENUS HYMENOLEPIS WEINLAND, 1858


Type species: Hymenolepis diminuta (Rudolphi, 1819) Weinland, 1858

1. Hymenolepis cantaniana (Polonio, 1860) Ransom, 1909
   Taenia cantaniana Polonio, 1860; Davainea oligophora Maghalaës, 1898; Davainea cantaniana Railliet & Lucet, 1899; Hymenolepis inermis (Yoshida, 1910) Fuhrmann, 1932
   Colinus virginianus, Coturnix coturnix, Gallus gallus, Meleagris gallopavo, Numida meleagris, Pavo cristatus, Perdix perdix, Phasianus colchicus, Tetrao parvirostris, Tetrao tetrastes bonasia, Turnix suscitator
   Schmidt (1986), Cosmopolitan
   Domestic chicken
   Le Roux (1926), South Africa
   Magwisha et al. (2002), Tanzania
   Le Roux (1926) chose to retain the name H. inermis for his unarmed specimens and not to accept the synonymy of H. inermis and H. cantaniana since the latter had been described as having an armed rostellum.

Numida meleagris
   Hodasi (1976), Ghana
   Junker & Boomker (2007b), South Africa

GENUS HISPANOLEPIS LÓPEZ-NEYRA, 1942

Satyolepis Spasskii, 1965

Type species: Hispanolepis villosa (Bloch, 1782) López-Neyra, 1942

1. Hispanolepis falsata (Meggitt, 1927) López-Neyra, 1942
   Hymenolepis falsata Meggitt, 1927.
   Numida meleagris
   Myers, Wolfgang & Kuntz (1960), Sudan
   Chlamydotis undulata
   Schmidt (1986), Egypt

2. Hispanolepis fedtschenkoi (Solowiow, 1911) López-Neyra, 1942
   Hymenolepis fedtschenkoi/Solowiow, 1911; Hymenolepis gwilletica Dinnik, 1938
   Gallus gallus, Lyrurus tetrax, Numida meleagris, Tetraogallus illyricus, Tetraogallus caucasicus, Tetrao tetrastes bonasia
   Schmidt (1986), Russia, Europe, Asia, Africa

3. Hispanolepis hilmyi (Skrjabin & Mathevossian, 1942) López-Neyra, 1942
   Numida meleagris
   Schmidt (1986), Liberia

4. Hispanolepis villosa (Bloch, 1782) López-Neyra, 1942
   Numida meleagris
   Baer (1926), East Africa

GENUS ORTLEPPOLEPIS SPASSKII, 1965

Type species: Ortleppolepis multiuncinata (Ortlepp, 1963) Spasskii, 1965

1. Ortleppolepis multiuncinata (Ortlepp, 1963) Spasskii, 1965
   Hispanolepis multiuncinata Ortlepp, 1963
   Guttera edouardi
   Ortlepp (1963), Zambia
   Numida meleagris
   Junker & Boomker (2007b), South Africa

PHYLUM NEMATHELMINTHES

Class Nematoda
Subclass Adenophorea
Order Enoplida
Superfamily Trichinelloidea Hall, 1916
Family TRICHURIDAE (Ransom, 1911) Railliet, 1915
Subfamily Capillariinae Railliet, 1915

GENUS AONCHOTHECA LÓPEZ-NEYRA, 1947

Avesaonchotheca auct.; Baruscapillaria auct.; Capillaria auct.; Pterothonyx auct.; Skrabinocapillaria Skarbiliovich, 1946

1. Aonchotheca caudinflata (Molin, 1858)
Check list of helminths of guineafowls (Numididae) and host list of parasites

1. **Capillaria anatis** (Schrank, 1790) Travassos, 1915

   *Trichosoma* Rudolphi, 1819; *Trichosomum* Creplin, 1829; *Thominx* Dujardin, 1845; *Tridentocapillaria* Barus & Sergeeva, 1990; *Aonchotheca* Dujardin, 1845; *Ptherominx* auct.; *Trichocelphalus* auct.

   **Type species**: *Capillaria anatis* (Schrank, 1790) Travassos, 1915

   **Domestic chicken**

   **Magwisha et al.** (2002), Tanzania

   **Numida meleagris**

   **Ayeni et al.** (1983), Nigeria

2. **Eucoleus dujardini** Molin, 1858

   *Capillaria* auct.; *Thominx* auct.; *Trichocelphalus* auct.

   1. **Eucoleus annulatus** (Molin, 1858)

      *Trichosoma* annulatum Molin, 1858

      *Bonasa*, *Chrysolophus*, *Colinus*, *Gallus*, *Lyrurus*, *Meleagris*, *Numida meleagris*, *Perdix*, *Phasianus*, *Syrmaticus*, *Tetrao*

      **Yamaguti** (1961), *Asia*, *Europe*, *North and South America*

      **Domestic chicken**

      **Magwisha et al.** (2002), Tanzania

      **Numida meleagris**

      **Nfor et al.** (1999), Nigeria

**Order Strongyliida**

**Superfamily Strongyloidea**

**Family SYNGAMIDAE Leiper, 1912**

**Subfamily Syngaminae Baylis & Daubney, 1926**

**GENUS SYNGAMUS SIEBOLD, 1836**

*Cyathostoma* auct.; *Ornithogamus* Ryjikov, 1948

**Type species**: *Syngamus trachea* (Montagu, 1811) Siebold, 1836

1. **Syngamus trachea** (Montagu, 1811) Siebold, 1836

   *Fasciola trachea* Montagu, 1811; *Syngamus trachealis* Siebold, 1836; *Strongylus trachealis* Nathusius, 1937 in Ortlepp (1923); *Strongylus pictus* Creplin, 1849; *Sclerostomum* *Syngamus* Diesing, 1951 in Ortlepp (1923); *Syngamus furcatus* Theob., 1896; *Syngamus primitivus* Molin, 1861; *Syngamus sclerostomum* Molin, 1861

   **Galliformes**, *Passeriformes*; rarely *Anseriformes*, "Ardeiformes", "Pelecaniformes", *Piciformes*, *Otidiformes*

   **Yamaguti** (1961), *Africa*, *Australia*, *Europe*, *India*, *North and South America*

   **Domestic chicken**

   **Magwisha et al.** (2002), Tanzania

   **Numida meleagris**

   **Hodasi** (1976), *Ghana*

   **Nfor et al.** (1999), *Nigeria*

**Order Ascaridida**

**Superfamily Heterakoidea**

**Family HETERAKIDAE Railliet & Henry, 1912**

**Subfamily Heterakinae Railliet & Henry, 1912**

**GENUS HETERAKIS SIEBOLD, 1836**

*Heterakis* auct.; *Sclerostoma* Siebold, 1836

1. **Heterakis vesicularis** (Frölich, 1791)

   *Ascaris* vesicularis Frölich, 1791; *Ascaris papillosa* Bloch, 1782, in part

   **Anas**, *Colinus*, *Coturnix*, *Cygnus*, *Gallus*, *Lagopus*, *Meleagris*, *Numida meleagris*, *Oreortyx pictus*, *Otis*, *Pavo*, *Perdix*, *Phasianus colchicus*, *Polyleptodon*, *Tetrao*

   **Yamaguti** (1961), *Africa*, *Europe*, *North America*

   **Lophophorus**, *Lopha*ura

   **Yamaguti** (1961), *Nepal*

   **Yamaguti** (1961) lists *Heterakis vesicularis* from *Otis* without giving the host’s species name. It is therefore not clear whether *Otis* refers to the current genus *Otis*, *Ardeotis*, *Neotis* or *Chlamydotis*.

2. **Heterakis brevispiculum** Gendre, 1911

   **Domestic chicken**, *Numida meleagris*, *Pterinistis bicalcaratus*

**Suborder ASCARIDOIDEA**

**Superfamily ASCARIDAE Cope, 1866**

**Family ASCARIDAE Cope, 1866**

**Subfamily ASCARIDINAe Railliet & Henry, 1912**

**GENUS ASCARIS LINNAEUS, 1758**

*Ascaris* auct.; *Syngamus* Siebold, 1836

1. **Ascaris lumbricoides** (Line, 1758)

   *Ascaris lumbricoides* Linnaeus, 1758

   **Anas**, *Colinus*, *Coturnix*, *Cygnus*, *Gallus*, *Lagopus*, *Meleagris*, *Numida meleagris*, *Oreortyx pictus*, *Otis*, *Pavo*, *Perdix*, *Phasianus colchicus*, *Polyleptodon*, *Tetrao*

   **Yamaguti** (1961), *Africa*, *Europe*, *North America*

   **Lophophorus**, *Lopha*ura

   **Yamaguti** (1961), *Nepal*

   **Yamaguti** (1961) lists *Heterakis vesicularis* from *Otis* without giving the host’s species name. It is therefore not clear whether *Otis* refers to the current genus *Otis*, *Ardeotis*, *Neotis* or *Chlamydotis*.

2. **Heterakis brevispiculum** Gendre, 1911

   **Domestic chicken**, *Numida meleagris*, *Pterinistis bicalcaratus*
Family ASCARIIDIDAE Travassos, 1919

GENUS ASCARIDIA DUJARDIN, 1845

_Cotylyascaris_ Sprent, 1971

Type species: _Ascaridia hermaphrodita_ (Frölich, 1789) Railliet & Henry, 1914

1. _Ascaridia calcarata_ (Gendre, 1909)
   _Numida meleagris_
   Yamaguti (1961), Africa
   Junior synonym of _Ascaridia numidae_ (Leiper, 1908) according to Sprehn (1932) in Yamaguti (1961).

2. _Ascaridia compar_ (Schrank, 1790) Travassos, 1913
   _Ascaris compar_ Schrank, 1790
   _Alectoris, Coturnix, Gallus, Lyurus, Numida meleagris, Oreortyx pictus, Perdix, Tetrao, Tretastes_
   Yamaguti (1961), America, Europe, India, Philippines

3. _Ascaridia galli_ (Schrank, 1788) Freeborn, 1932
   _Ascaris galli_ Schrank, 1788; _Fusaria inflexa_ Zeder, 1800 (Baylis 1932, cited in Yamaguti 1961); _Fusaria reflexa_ Zeder, 1800, in part; _Fusaria stromosa_ Zeder, 1800, in part (López-Neyra 1946, cited in Yamaguti 1961); _Heterakis brasiliensis_ Magalhães, 1892 (Pinto & Lins de Almeida 1935, cited in Yamaguti 1961); _Heterakis granulosa_ Von Linstow, 1906 (Baylis 1932, cited in Yamaguti 1961); _Ascaridia hamia_ Lane, 1914
   Domestic chicken, guineafowl
   Yamaguti (1961), Europe, Japan
   _Alectoris, Bonasa, Cairina, Colinus, duck, Ithaginis, Lyurus, Meleagris, Numida meleagris, Perdix, Phasianus, Streptopelia, Tetrao, Tympanuchus_
   Yamaguti (1961), Cosmopolitan
   Domestic chicken
   Poulser et al. (2000), Ghana
   Magwisha et al. (2002), Tanzania
   Permin et al. (2002), Zimbabwe
   _Numida meleagris_
   Ayeni et al. (1983), Nigeria
   Verster & Ptasinska-Kloryga (1987), South Africa
   Haziev & Khan (1991), Republic of Bashkortostan

4. _Ascaridia lineata_ (Schrank, 1866)
   _Ascaris lineata_ Schrank, 1866
   _Alectoris, Anas, Anser, Bonasa, duck, Francolinus, Gallus, goose, Meleagris, Meleagris ocellata, Numida, partridge, Phasianus, pigeon, Tympanuchus_
   Yamaguti (1961), Africa, Brazil, China, Cuba, Europe, Formosa, India, Malaya, North America, Philippines, Puerto Rico, Turkestan
   Domestic chicken
   Le Roux (1926), South Africa

5. _Ascaridia numidae_ (Leiper, 1908) Travassos, 1913
   _Heterakis numidae_ Leiper, 1908
   _Alectoris, Guttera_
   Yamaguti (1961), Africa
   Guinea fowl
   Yamaguti (1961), Puerto Rico
   Bwagamoi (1968), Uganda
   _Numida meleagris_
   Graber (1959), Chad
   Yamaguti (1961), Africa, the White Nile
   Myers et al. (1960), Sudan
   Fabiyi (1972), Nigeria
   Hodasi (1976), Ghana
   Verstruyse et al. (1985), Burkina Faso
   Verster & Ptasinska-Kloryga (1987), South Africa
   The White Nile rises from Lake Victoria in Uganda and enters the Sudan where it joins the Blue Nile in Karthoum to form the Nile. White Nile is one of the states of Sudan.
6. *Ascaridia perspicillum* (Rudolphi, 1803)
   *Ascaris perspicillum* Rudolphi, 1803
   *Anas acuta*, domestic chicken, *Meleagris gallopavo*, *Numida meleagris*, *Pavo cristatus*, *Tetrao urogallus*, *Tetrastes bonasia rupestris*, *Turdus viscivorus* 
   Yamaguti (1961), Europe, Hawaii, India, Indonesia, Japan, Malaya

**Superfamily Subuluroidea**

**Family SUBULURIDAE** (Travassos, 1914) Yorke & Maplestone, 1926

**Subfamily Subulurinae Travassos, 1914**

**GENUS**

*Subulura* Molin, 1860

Type species: *Subulura acutissima* Molin, 1860

   *Oxystrongylus acuticauda* Von Linstow, 1901; *Heterakis acuticauda* (Von Linstow, 1901) Von Linstow, 1909
   *Numida meleagris*
   Von Linstow (1901), Kenya
   Yamaguti (1961), Africa

2. *Subulura brumpti* (Lopez-Neyra, 1922)
   *Alodapa brumpti* Lopez-Neyra, 1922
   *Alectoris graeca*, *Anas*, *Colinus virginianus texanus*, *domestic chicken*, *Meleagris gallopavo*, *Numida, Perdix perditia*, *Streptopelia orientalis* 
   Yamaguti (1961), Europe, Palestine, Cyprus, Cuba, Puerto Rico, Panama, North America, Africa, China
   Domestic chicken
   Hodasi (1969), Ghana
   Mukaratiwia, Hove, Esmann, Hoj, Permin & Nansen (2001), Zimbabwe
   *Numida meleagris*
   Graber (1959), Chad
   Hodasi (1976), Ghana
   Nfor et al. (1999), Nigeria

3. *Subulura dentigera* Ortlepp, 1937
   *Numida meleagris*
   Ortlepp (1937), South Africa

4. *Subulura differens* (Sonsino, 1890)
   *Heterakis differens* Sonsino, 1890
   *Alectoris graeca*, *Centropus phasianus*, *domestic chicken*, *Euplectes orix*, *Numida meleagris*, *Perdix perditia canescens*, *Pternistis bicalcaratus* 
   Yamaguti (1961), Cosmopolitan

5. *Subulura victoria* (Molin, 1860)
   *Heterakis victoria* Molin, 1860; *Ascaris forcipata* Rudolphi, 1819, in part
   *Caprimulgus, Podager, Nyctibius* 
   Yamaguti (1961), Brazil
   *Burhinus, Coturnix, Numida, Pternistis* 
   Yamaguti (1961), South Africa (Transvaal)

**Superfamily Thelazioidea**

**Family THELAZIIDAE** Skrjabin, 1915

**GENUS OXYSPIRURA** Skrjabin, 1931

Type species: *Oxyspirura cephaloptera* (Molin, 1860)

1. *Oxyspirura mansoni* (Cobbold, 1879)
   *Filaria mansoni* Cobbold, 1879; *Spirooptera emmerezii* Emmez & Mégnin, 1901 (Marotel & Carougeau 1902, cited in Yamaguti 1961)
   *Domestic chicken, Gallus gallus, Meleagris gallopavo, Pavo cristatus* 
   Yamaguti (1961), Atlantic and Pacific islands, Australia, Democratic Republic of Congo, Formosa, India, Japan, North America
   *Numida meleagris*
   Hodasi (1976), Ghana

**Superfamily Spiruroidea**

**Family GONGYLONEMATIDAE** (Hall, 1916, subfam.) Sobolev, 1949

**GENUS GONGYLONEMA** Molin, 1857

Type species: *Gongylonema musculi* (Rudolphi, 1819) Neumann, 1894

1. *Gongylonema ingluvicola* Ransom, 1904
   *Gongylonema sumani* Bahlerao, 1933 (Baylis 1939, cited in Yamaguti 1961)
Gallus, Meleagris, pheasants
Yamaguti (1961), Cosmopolitan

Domestic chicken
Poulsen et al. (2000), Ghana
Magwisha et al. (2002), Tanzania
Permin et al. (2002), Zimbabwe

Numida meleagris
Nfö et al. (1999), Nigeria

2. Gongylonema congolense Fain, 1955

Cairina moschata
Fain (1955), Democratic Republic of the Congo

Gallus
Fain (1955), Burundi, Democratic Republic of the Congo, Rwanda

Guttera edouardi
Junker & Boomker (2007b), South Africa

Numida meleagris
Fain (1955), Burundi, Democratic Republic of the Congo, Rwanda

Fabiyi (1972), Nigeria
Graber (1976), Ethiopia
Hodasi (1976), Ghana
Vercruysse et al. (1985), Burkina Faso
Junker & Boomker (2007b), South Africa

Scleroptila levaillantii
Fain (1955), Rwanda

3. Gongylonema sumani Bhalerao, 1933

Gallus gallus
Bhalerao (1933), India

Numida meleagris
Fain (1955), Burundi, Democratic Republic of the Congo, Rwanda

Fabiyi (1972), Nigeria
Vercruysse et al. (1985), Burkina Faso
Chabaud (1958) divided the genus Cyrene into the two subgenera Procyrnea Chabaud, 1958 and Cyrene Chabaud, 1958, subsequently raising them to genus level (Chabaud, 1975). He also synonymized Cyrene (Cyrene) numidae Ortlepp, 1938 and Cyrene (Cyrene) seurati Lopéz-Neyra, 1918 with Cyrene (Cyrene) parroti Seurat, 1917. Our specimens of Cyrene parroti collected from Numida meleagris in South Africa comply with Ortlepp’s (1938b) description of C. numidae, but the arrangement of cephalic structures in apical view is that of C. parroti. Not having examined Ortlepp’s (1938b) specimens we adopt the classification of Chabaud (1958) and list Ortlepp’s specimens as C. parroti.

GENUS Sicarius Li, 1934

Type species: Sicarius dipterum (Popova, 1927) Li, 1934

1. Sicarius caudatus Quentin & Wertheim, 1975

Numida meleagris
Junker & Boomker (2007b), South Africa

Pycnonotus capensis
Quentin & Wertheim (1975), Israel

Quentin & Wertheim (1975) described S. caudatus from P. capensis present in the collection of the “Helmhthological Laboratory Jerusalem” and list Jerusalem as locality. It should be noted that P. capensis is endemic to South Africa (Lepage 2007). We therefore conclude that the authors were either looking at birds kept in captivity in Israel, making it difficult to determine the geographic origin of the parasites or did not have any information on the original locality if the birds had been collected in South Africa.

2. Sicarius renatae Cancrini, Balbo & Iori, 1991

Acryllium vulturinum
Cancrini, Balbo & Iori (1991), Somalia

Subfamily Histrocephalinae Gendre, 1922

GENUS Hadjelia Seurat, 1916

Gilsonia Gedoelst, 1919; Stellabronema Guschanskaja, 1937; Sobolevicephalus Parukhin, 1964

1. Hadjelia truncata (Creplin, 1825)

Spiroptera truncata Creplin, 1825; Hadjelia inermis (Gedoelst, 1919)

Aceros corrigatus
Ortlepp (1964), Malucca Islands, Indonesia

Columba livia
Tadros & Iskander (1975), Egypt

Guttera edouardi, Numida meleagris
Junker & Boomker (2007b), South Africa

Tockus erythrorhynchus, Tockus leucomelas
Ortlepp (1964), South Africa

Tockus fasciatus semifasciatus
Cram (1927, cited in Ortlepp 1964), Africa
Coracias benghalensis, Halcyon smyrnensis, Upupa epops

Singh (1949), India

Chabaud & Campana (1950) synonymized H. inermis with H. truncata. Orlepp (1964) did not follow this and recorded his specimens as H. inermis. Tadros & Iskander (1975) synonymized H. inermis, H. parva and H. thullieri with H. truncata, designating H. truncata as the new type species of the genus.

Family TETRAMERIDAE Travassos, 1914

Subfamily Tetramerinae Railliet, 1915

GENUS Tropisurus Travassos, 1915

Tropisurus Diesing, 1835; Tropidurus Wiegmann, 1835, preoccupied; Gynaeophila Gubanov, 1950; Petrovimeres Tschertkova, 1953; Microtetrameres auct.

Type species: Tetrameres paradoxa (Diesing, 1835)

1. Tetrameres fissispina Diesing, 1861

Acantophorus horridus Von Linstow, 1876; Acanthophorus tenuis Von Linstow, 1876; Filaria pulcis Von Linstow, 1894

Alectoris, Anas acuta, Anas clypeata, Anas platyrhynchos, Anas querquedula, Aythya ferina, Bucephala clangula, Columba livia, Cygnus melanocoryphus, Fulica atra, Gallus, Melanitta fusca, Meleagris, Meleagris gallopavo, Mergus merganser, Nycticorax nycticorax, Perdix, Somateria molissima, Tachybaptus fluviatilis

Yamaguti (1961), Africa, Canton, Europe, Formosa, Guam, India, Malaya, North and South America, Philippines, Russian Turkestan, Siberia, Turkey

Domestic chicken

Le Roux (1926), South Africa

Poulse et al. (2000), Ghana

Magwisha et al. (2002), Tanzania

Guineafowl

Le Roux (1926), South Africa

Numida meleagris

Fabiyi (1972), Nigeria

Hodasi (1976), Ghana

Vercruysse et al. (1985), Burkina Faso

1. Tetrameres numida Junker & Boomker 2007

Numida meleagris

Junker & Boomker (2007a), South Africa

Chabaud (1975) divided the genus Tetrameres into the two subgenera Tetrameris (Tetrameres) Creplin, 1846 and Tetrameris (Microtetrameres) Travassos, 1915. We adopt the view of Anderson (1992) and consider the two as valid genera.

Superfamily Acuarioidae

Family Acuariidae (Railliet, Henry & Sisoff, 1912, subfam.)

Subfamily Acuariinae Railliet, Henry & Sisoff, 1912

GENUS ACUARIA BREMSE,R, 1811

Chelospirura auct.

Type species: Acuaria anthuris (Rudolphi, 1819)

1. Acuaria hamulosa (Diesing, 1851)

Spiroptera hamulosa Diesing, 1851; Chelospirura hamulosa Diesing, 1861; Spiroptera perforans Centoccd, 1911

Coturnix coturnix, Gallus gallus, Meleagris, pheasant

Yamaguti (1961), Cosmopolitan

Domestic chicken

Le Roux (1926), South Africa

Poulse et al. (2000), Ghana

Magwisha et al. (2002), Tanzania

Numida meleagris

Fabiyi (1972), Nigeria

Hodasi (1976), Ghana

Vercruysse et al. (1985), Burkina Faso

GENUS SYNTHIMANTUS RAILLIET, HENRY & SISOFF, 1912

Type species: Synthimantus laticeps (Rudolphi, 1819)

1. Synthimantus spiralis (Linstow, 1883)

Dispharagus spiralis Linstow, 1883

Accipiter, Alectoris, Bonasa, Ciconia, Colinus, Columba, Coracias, Corvus, Gallus, Meleagris, Metopidius, Numida meleagris, Passer, Perdix, Phasianus, Quiscalus, Turdus, Turdus migratorius

Yamaguti (1961), Cosmopolitan

Numida meleagris

Fabiyi (1972), Nigeria

Hodasi (1976), Ghana

Vercruysse et al. (1985), Burkina Faso

GENUS DISPHARYNX RAILLIET, HENRY & SISOFF, 1912

Type species: Dispharynx nasuta (Rudolphi, 1819)

1. Dispharynx nasuta (Rudolphi, 1819)

Spiroptera nasuta Rudolphi, 1819

Passer domesticus

Yamaguti (1961), Europe

Gallus gallus

Yamaguti (1961), Africa, America, Australia, Ceylon, Cuba, Formosa

Domestic chicken, turkeys

Gibbons, Jones & Khalil (1996), no geographic data given

Numida meleagris

Verster & Ptasinska-Kloryga (1987), South Africa

HOST/PARASITE CHECK LIST

Order Tinamiformes

Family Tinamidae (Tinamous)

GENUS TINAMUS HERMANN, 1783

Subulura strongylina

GENUS CRYPTURELLUS BRABOURNE & CHUBB, 1914

Crypturus

Subulura strongylina
Order Galliformes

Domestic chicken

Mediorhynchus gallinarum
Postharmostomum gallinum
Choanotaenia infundibulum
Davainea proglottina
Echinolepis carioca
Hymenolepis cantaniana
Raillietina cohni
Raillietina echiunobothrida
Raillietina tetragona
Skrjabinia cesticillus
Acuaria hamulosa
Aonchotheca caudinflata
Ascaridia galli
Ascaridia lineata
Ascaridia perspicillum
Capillaria anatis
Dispharynx nasuta
Eucoleus annulatus
Gongylonema ingluvicola
Heterakis brevispiculum
Heterakis dispar
Heterakis gallinarum
Oxyspirura mansoni
Subulura brumpti
Subulura differens
Subulura strongylina
Subulura suotoria
Syngamus trachea
Tetrameres fissispina

Gallinaceous birds, galliformes

Mediorhynchus gallinarum
Syngamus trachea

Family Numididae (Guineafowls)

Crested guineafowl
Postharmostomum gallinum

Guineafowl

Cotugnia crassa
Cotugnia digonopora
Numidella numida
Octopetalum numida
Porogynia paronai
Ascaridia galli
Ascaridia numidae
Tetrameres fissispina

GENUS NUMIDA LINNAEUS, 1766

1. *Numida meleagris* (Linnaeus, 1758) (Helmeted Guineafowl)
   Phasianus meleagris
   Numida meleagris galeatus Pallas, 1767
   Numida galeata
   Numida meleagris meleagris (Linnaeus, 1758)
   Numida ptilorhyncha
   Numida meleagris mitratus Pallas, 1767
   Numida mitrata
   Numida meleagris marungensis Schalow, 1884
   Numida frommi, Numida marungensis, Numida meleagris
   bodalaya, Numida meleagris frommi, Numida meleagris max-
   ima, Numida meleagris rikwe, Numida mitrata frommi, Numi-
   da mitrata maximia, Numida mitrata rikwe, Numida rikwe
   Mediorhynchus empodius
   Mediorhynchus gallinarum
   Mediorhynchus numidae
   Mediorhynchus selengensis
   Mediorhynchus taeniatus
   Dicrocoelium macrostomum
   Lutzrema sp.
   Postharmostomum gallinum
   Abuladzugnia guttare
   Abuladzugnia transvaalensis
   Choanotaenia infundibulum
   Cotugnia crassa
   Cotugnia digonopora
   Cotugnia meleagridis
   Cotugnia tuliensis
   Davainea nana
   Davainea paucisegmentata
   Davainea paucisegmentata var. dahomeensis
   Davainea progollitina
   Echinolepis carioca
   Hispanolepis falsata
   Hispanolepis fedtschenkoi
   Hispanolepis hilmyi
   Hispanolepis villosa
   Hymenolepis cantaniana
   Idiogenes sp.
   Metroliasthes lucida
   Numidella numida
   Octopetalum guttare
   Octopetalum numida
   Ortleppolepis multiuncinata
   Porogynia paronai
   Raillietina angusta
   Raillietina cohni
   Raillietina echiunobothrida
   Raillietina pintneri
   Raillietina steinhardti
   Raillietina tetragona
   Raillietina tetragonoides
   Raillietina toyohashiensis
   Skrjabinia cesticillus
   Skrjabinia dewet
   Acuaria hamulosa
   Ascaridia calcarata
   Ascaridia compar
   Ascaridia galli
   Ascaridia lineata
   Ascaridia numidae
   Ascaridia perspicillum
   Aonchotheca caudinflata
   Capillaria anatis
   Cymea eurycerca
Check list of helminths of guineafowls (Numididae) and host list of parasites

**Cyrnea parroti**
**Eucoleus annulatus**
**Gongylonema congolense**
**Gongylonema ingluvicola**
**Gongylonema sumani**
**Hadjelia truncata**
**Heterakis brevispiculum**
**Heterakis dispar**
**Heterakis gallinarum**
**Heterakis vesicularis**
**Oxyspirura mansoni**
**Sicarius caudatus**
**Sicarius renatae**

**Subulura acuticauda**
**Subulura brumpti**
**Subulura dentigera**
**Subulura differens**
**Subulura strongylina**
**Subulura suctoria**
**Strongyloides avium**
**Syngamus trachea**
**Dispharynx nasuta**
**Synhimantus spiralis**
**Tetrameris fissispina**
**Tetrameris numida**

**GENUS: GUTTERA WAGLER, 1832**

  Numidella numida  
  Raillietina pintneri  
  Ascaridia numidae  

  1. **Guttera edouardi** (Hartlaub, 1867) (Crested Guinea-fowl)  
      **Numida edouardi** (Hartlaub, 1867); **Guttera pucherani edouardi** (Hartlaub, 1867)  
      **Mediorhynchus taeniatus**  
      **Abuladzugnia gutterae**  
      **Davainea nana**  
      **Metroliasthes lucida**  
      **Octopetalum gutterae**  
      **Octopetalum numida**  
      **Ortellopolepis multiuncinata**  
      **Porogynia paranai**  
      **Raillietina pintneri**  
      **Raillietina steinhardi**  
      **Raillietina tetragona**  
      **Gongylonema congolense**  
      **Hadjelia truncata**  
      **Subulura suctoria**

**GENUS: ACRYLLIUM GRAY, 1840**

  **Heterakis gallinarum**  

  1. **Acryllium vulturinum** Gray, 1840 (Vulturine Guinea-fowl)  
      **Cotugnia shohoi**  
      **Raillietina somaiensis**  
      **Heterakis tenuicauda**  
      **Sicarius renatae**

**Family ODONTOPHORIDAE (New World quails)**

**GENUS OREORTYX BAIRD, 1858**

  **Ortyx in Yamaguti (1961)**

  1. **Oreortyx pictus** (Douglas, 1829) (Mountain Quail)  
      **Ortyx picta**  
      **Ascaridia compar**  
      **Heterakis vesicularis**

**GENUS CALLIPEPLA WAGLER, 1832**

  **Lophortyx**  
  **Subulura strongylina**

**GENUS COLINUS GOLDFUSS, 1820**

  **Ascaridia galli**  
  **Eucoleus annulatus**  
  **Heterakis vesicularis**  
  **Heterakis gallinarum**  
  **Subulura strongylina**  
  **Synhimantus spiralis**

  1. **Colinus virginianus** (Linnaeus, 1758) (Northern Bob-white)  
      **Tetrao virginianus**  
      **Echinolepis carioca**  
      **Hymenolepis cantaniana**  
      **Skrjabinia cesticillus**

  1a. **Colinus virginianus texanus** Lawrence, 1853  
      **Subulura brumpti**

**GENUS ODONTOPHORUS VIEILLOT, 1816**

  **Subulura strongylina**

  1. **Odontophorus capueira** (Spix, 1825) (Spot-winged Wood-quail)  
      **Perdix capueira**  
      **Subulura strongylina**

**Family PHASIANIDAE (Partridges, francolins, spurfowls, pheasants, etc.)**

  **Partridge**  
  **Ascaridia lineata**  
  **Pheasant**  
  **Acuaria hamulosa**  
  **Gongylonema ingluvicola**  
  **Turkey**  
  **Dispharynx nasuta**

**GENUS MELEAGRIS LINNAEUS, 1758**

  **Acuaria hamulosa**  
  **Ascaridia galli**  
  **Ascaridia lineata**  
  **Eucoleus annulatus**  
  **Gongylonema ingluvicola**  
  **Heterakis vesicularis**  
  **Heterakis gallinarum**  
  **Synhimantus spiralis**
Tetrameres fissispina

1. Meleagris gallopavo Linnaeus, 1758 (Wild Turkey, Common Turkey)
   Echinolepis carioca
   Hymenolepis cantaniana
   Metroliasthes lucida
   Numidiella numida
   Raillietina echinobothrida
   Raillietina tetragona
   Skrjabinia cesticillus
   Ascaridia perspicillum
   Oxyspirura mansoni
   Subulura brumpti
   Tetrameres fissispina

2. Meleagris ocellata Cuvier, 1820 (Ocellated Turkey)
   Agriocharis ocellata
   Ascaridia galli

GENUS BONASA Stephens, 1819

Ascaridia galli
Ascaridia lineata
Eucoleus annulatus
Heterakis gallinarum
Subulura strongylina
Synhimanus spiralis
1. Bonasa umbellus (Linnaeus, 1766) (Ruffed Grouse)
   Tetrao umbellus
   Davainea proglottina
   Echinolepis carioca

GENUS TETRASTES Keyserling & Blasius, 1840

Ascaridia compar
1. Tetrastes bonasia (Linnaeus, 1758) (Hazel Grouse)
   Bonasa bonasia, Bonasia bonasia, Tetrao bonasia
   Hispaniolepis fedtschenkoi
   Hymenolepis cantaniana
   Skrjabinia cesticillus
1a. Tetrastes bonasia rupestris (Brehm, 1831)
   Ascaridia perspicillum

GENUS TETRAO Linnaeus, 1758

Aonchotheca caudinflata
Ascaridia compar
Ascaridia galli
Eucoleus annulatus
Heterakis gallinarum
Heterakis vesicularis
Subulura strongylina
1. Tetrao urogallus Linnaeus, 1758 (Western Capercaillie)
   Tetrao major
   Skrjabinia cesticillus
   Ascaridia perspicillum
2. Tetrao pavostris Bonaparte, 1856 (Black-billed Capercaillie)

GENUS LYRURUS Swainson, 1832

Aonchotheca caudinflata
Ascaridia compar
Ascaridia galli
Capillaria anatis
Eucoleus annulatus
Heterakis gallinarum
1. Lyrurus tetrix (Linnaeus, 1758) (Black Grouse)
   Tetrao tetrix
   Hispaniolepis fedtschenkoi
   Skrjabinia cesticillus

GENUS TYMPANUCHUS Gloger, 1841

Cupidonia
Ascaridia lineata
Heterakis gallinarum
1. Tymanuchus phasianellus (Linnaeus, 1758) (Sharptailed Grouse)
   Pedioecetes phasianellus (Linnaeus, 1758), Tetrao phasianellus
   Ascaridia galli
   Ascaridia lineata
   Heterakis gallinarum
   Subulura strongylina

GENUS LAGOPUS Brisson, 1760

Aonchotheca caudinflata
Heterakis gallinarum
Heterakis vesicularis
1. Lagopus lagopus (Linnaeus, 1758) (Willow Ptarmigan)
   Tetrao lagopus
   Raillietina tetragona
   Skrjabinia cesticillus
   Subulura suctoria
1a. Lagopus lagopus scotica (Latham, 1787)
   Lagopus scotica
   Skrjabinia cesticillus
2. Lagopus muta (Montin, 1781) (Rock Ptarmigan)
   Raillietina tetragona

GENUS TETRAGALLUS Gray, 1832

1. Tetragallus caucasicus (Pallas, 1811) (Caucasian Snowcock)
   Tetrao caucasicus
   Hispaniolepis fedtschenkoi
2. Tetragallus himalayensis Gray, 1843 (Himalayan Snowcock)
   Megaloperdix nigelli
   Hispaniolepis fedtschenkoi

GENUS ALECTORIS Kaup, 1829

Caccabis in Yamaguti (1961)
Check list of helminths of guineafowls (Numididae) and host list of parasites

Ascaridia compar
Ascaridia galli
Ascaridia numidae
Cyrnea eurycerca
Heterakis dispar
Heterakis gallinarum
Synhimantus spiralis
Tetrameres fissispina

1. Alectoris barbara (Bonnaterre, 1792) (Barbary Partridge)
   Caccabis petrosa, Perdix barbara
   Cyrnea parroti

2. Alectoris graeca (Meisner, 1804) (Rock Partridge)
   Perdix graeca
   Davainea progollitina
   Echinolepis carioca
   Metroliastrae lucida
   Heterakis tenuicauda
   Subulura brumpti
   Subulura differens

2a. Alectoris graeca saxatilis (Bechstein, 1805)
   Caccabis saxatilis chukar
   Heterakis tenuicauda

   Lepage (2007) states that the original Alectoris graeca has been split into four species, namely Alectoris graeca, Alectoris chukar (Gray, 1830), Alectoris philbyi Lowe, 1934 and Alectoris magna (Prjevalski, 1876).

3. Alectoris rufa (Linnaeus, 1758) (Red-legged Partridge)
   Caccabis rufa, Coturnix rufa, Tetrao rufus
   Metroliastrae lucida
   Cyrnea eurycerca
   Cyrnea parroti

GENUS FRANCOLINUS STEPHENS, 1819
   Ascaridia lineata
   Cyrnea eurycerca
   Heterakis gallinarum

GENUS SCLEROPTILA BLYTH, 1852
1. Scleroptila levaillantii (Valenciennes, 1825) (Red-winged Francolin)
   Francolinus levaillantii, Perdix levaillantii
   Gongylonema congolense

GENUS PTERNISTIS WAGLER, 1832
   Subulura suctoria

1. Pternistis natalensis (Smith, 1834) (Natal Spurfowl)
   Francolinus natalensis, Pternistes natalensis
   Porogynia paronai

2. Pternistis bicalcaratus (Linnaeus, 1766) (Double-spurred Spurfowl)
   Francolinus bicalcaratus, Tetrao bicalcaratus
   Heterakis brevispiculum
   Subulura differens

GENUS PERDIX BRISSON, 1760
   Aonchotheca caudinflata
   Ascaridia compar
   Ascaridia galli
   Capillaria anatis
   Euculeus annulatus
   Heterakis vesicularis
   Heterakis gallinarum
   Subulura strongylina
   Synhimantus spiralis
   Tetrameres fissispina

1. Perdix perdix (Linnaeus, 1758) (Grey Partridge)
   Tetrao perdix
   Davainea progollitina
   Hymenolepis cantaniana
   Metroliastrae lucida
   Raillietina echinobothrida
   Skrjabinia cesticillus
   Subulura brumpti

1a. Perdix perdix canescens Buturlin, 1906
   Subulura differens

GENUS COTURNIX BONNATERRE, 1791
   Aonchotheca caudinflata
   Ascaridia compar
   Cyrnea eurycerca
   Heterakis gallinarum
   Heterakis vesicularis
   Subulura suctoria

1. Coturnix coturnix (Linnaeus, 1758) (Common Quail)
   Tetrao coturnix
   Dicrocoelium macrostomum
   Echinolepis carioca
   Hymenolepis cantaniana
   Metroliastrae lucida
   Skrjabinia cesticillus
   Acuaria hamulosa
   Subulura suctoria

GENUS ITHAGINIS WAGLER, 1832
   Ascaridia galli

GENUS TRAGOPAN CUVIER, 1829
   Ceriornis
   Heterakis gallinarum

GENUS LOPHOPHORUS TEMMINCK, 1813
   Heterakis gallinarum
   Heterakis vesicularis

GENUS GALLUS BRISSON, 1760
   Aonchotheca caudinflata
   Ascaridia compar
   Ascaridia lineata
   Euculeus annulatus
   Gongylonema congolense
Gongylonema ingluvicola
Heterakis vesicularis
Strongyloides avium
Subulura strongylina
Synhimantus spiralis
Tetrameres fissispina

1. Gallus gallus (Linnaeus, 1758) (Red Junglefowl)
   Gallus ferrugineus, Phasianus gallus
   Davainea proglottina
   Echinolepis carioca
   Hispanicolepis fedtschenkoi
   Hymenolepis cantaniana
   Metroliaastes lucida
   Raillietina cohni
   Raillietina echinobothrida
   Raillietina tetragona
   Skrjabinia cesticillus
   Acuaria hamulosa
   Dispharynx nasuta
   Gongylonema sumani
   Oxyspirura mansoni
1a. Gallus gallus bankiva Temminck, 1813

   Metroliaastes lucida
   Raillietina echinobothrida

GENUS LOPHURA FLEMING, 1822

   Gennaeus
   Heterakis gallinarum

1. Lophura nycthemera (Linnaeus, 1758) (Silver Pheasant)
   Euplocamus nycthemerus, Gennaeus nycthemerus, Phasianus nycthemerus
   Heterakis vesicularis

GENUS SYRMATICUS WAGLER, 1832

   Graphophasisanus
   Eucoleus annulatus

1. Syrmaticus soemmerringii (Temminck, 1830) (Copper Pheasant)
   Graphophasisanus soemmerringii, Phasianus soemmerringii
   Heterakis gallinarum

GENUS PHASIANUS LINNAEUS, 1758

   Aonchotheca caudinflata
   Ascaridia galli
   Ascaridia lineata
   Capillaria anatis
   Cymera eurycerca
   Eucoleus annulatus
   Heterakis gallinarum
   Synhimantus spiralis

1. Phasianus colchicus Linnaeus, 1758 (Common Pheasant, Ring-necked Pheasant)
   Hymenolepis cantaniana
   Raillietina echinobothrida
   Skrjabinia cesticillus
   Heterakis vesicularis
   Subulura suctoria
1a. Phasianus colchicus mongolicus Brandt, 1844
   Phasianus mongolicus turkestanicus
   Subulura suctoria
1b. Phasianus colchicus principalis Sclater, 1885
   Phasianus principalis
   Subulura suctoria

GENUS CHRYSOLOPHUS GRAY, 1834

   Thaumalea
   Aonchotheca caudinflata
   Eucoleus annulatus
   Heterakis gallinarum

GENUS POLYPELECTRON TEMMINCK, 1807

   Heterakis vesicularis

GENUS PAVO LINNAEUS, 1758

   Heterakis gallinarum
   Heterakis vesicularis

1. Pavo cristatus Linnaeus, 1758 (Indian Peafowl)
   Hymenolepis cantaniana
   Raillietina tetragona
   Ascaridia perspicillum
   Oxyspirura mansoni
2. Pavo muticus Linnaeus, 1766 (Green Peafowl)
   Raillietina tetragona

Order Anseriformes

   Syngamus trachea

Family ANATIDAE (Ducks, geese and swans)

Duck
   Ascaridia galli
   Ascaridia lineata

Goose
   Ascaridia lineata

GENUS ANSER BRISSON, 1760

   Cotugnia digonopora
   Ascaridia lineata
   Capillaria anatis
   Heterakis dispar
   Heterakis gallinarum

1. Anser cygnoides (Linnaeus, 1758) (Swan Goose)
   Cygnopsis cygnoides
   Heterakis dispar

GENUS BRANTA SCOPOLI, 1769

   Heterakis dispar

1. Branta bernicla (Linnaeus, 1758) (White-bellied Brant)
Check list of helminths of guineafowls (Numididae) and host list of parasites

**GENUS CYGNUS BECHSTEIN, 1803**
1. *Cygnus atratus* (Latham, 1790) (Australian Black Swan)
   *Chenopsis atrata*
   *Heterakis vesicularis*
2. *Cygnus melanocoryphus* (Molina, 1782)
   *Tetrameres fissispina*

**GENUS CHLOEPHAGA EYTON, 1838**

**GENUS TADORNINA BOIE, 1822**

**GENUS CAILRINA FLEMING, 1822**

**GENUS ANAS LINNAEUS, 1758**
1. *Anas platyrhynchos* Linnaeus, 1758 (Mallard)
   *Anas boschas*
   *Tetrameris fissionisina*
2. *Anas clypeata* Linnaeus, 1758 (Northern Shoveler)
   *Anas spathula, Spathula clypeata*
   *Tetrameris fissionisina*
3. *Anas acuta* Linnaeus, 1758 (Northern Pintail)
   *Dafila acuta*
   *Ascaridia perspicillum*
   *Tetrameris fissionisina*

According to Lepage (2007) *A. acuta* has been split into *A. acuta* and *Anas eatoni*, but some authors consider *A. eatoni* a subspecies of *A. acuta*. Peterson (1999) lists the two as separate species.

4. *Anas querquedula* Linnaeus, 1758 (Garganey)
   *Querquedula querquedula*
   *Capillaria anatis*
   *Tetrameris fissionisina*

**GENUS AYTHYA BOIE, 1822**
1. *Aythya ferina* (Linnaeus, 1758) (Common Pochard)
   *Anas ferina, Aristonetta ferina, Nyroca ferina*
   *Tetrameris fissionisina*

**GENUS SOMATERIA LEACH, 1819**
1. *Somateria mollissima* (Linnaeus, 1758) (Common Eider)
   *Anas mollissima*
   *Tetrameris fissionisina*

**GENUS MELANITTA BOIE, 1822**

**GENUS CLANGULA LEACH, 1819**
1. *Clangula hyemalis* (Linnaeus, 1758) (Oldsquaw, Long-tailed Duck)
   *Anas hyemalis, Harelda hyemalis, Ereunetes occidentalis*
   *Capillaria anatis*

**GENUS Bucephala Baird, 1858**
1. *Bucephala clangula* (Linnaeus, 1758) (Common Goldeneye)
   *Anas clangula, Clangula clangula, Glaucionetta clangula*
   *Tetrameris fissionisina*

**GENUS MERGUS LINNAEUS, 1758**

**Order Turniciformes**

**Family TURNICIDAE (Buttonquail)**

**GENUS TURUX BONNATERRE, 1791**
1. *Turnix suscitator* (Gmelin, 1789) (Barred Buttonquail)
   *Tetrao suscitator*
   *Hymenolepis cantaniana*

**Order Piciformes**

**Family SYNGAMIDAE (Buttonquail)**

**Order Galbuliformes**

**Family BUCCONIDAE (Puffbirds)**

**GENUS BUCO BRISON, 1760**

**GENUS MALOCOPTILA GRAY, 1841**

**Subulura strongylina**
GENUS NONNULA SCLATER, 1854  
Subulura strongylina

GENUS MONASA VIEILLOT, 1816  
Subulura strongylina

GENUS CHELIDOPTERA GOULD, 1837  
Subulura strongylina

Order Bucerotiformes  
Family BUCEROTIDAE (Hornbills)  
GENUS ACEROS HODGSON, 1844  
1. Aceros corrugatus (Temminck, 1832) (Wrinkled Hornbill)  
   Buceros corrugatus, Rhynicerus corrugatus  
   Hadjelia truncata

2. Tockus fasciatus semifasciatus (Hartlaub, 1855) (Allied Hornbill)  
   Lophoceros semifasciatus  
   Hadjelia truncata

3. Tockus leucomelas (Liechtenstein, 1842) (Southern Yellow-billed Hornbill)  
   Buceros leucomelas  
   Hadjelia truncata

Some authors consider T. leucomelas a subspecies of Tockus flavirostris (Rüppell, 1853) (Lepage 2007)

Order Upupiformes  
Family UPUPIDAE (Hoopoes)  
GENUS UPUPA LINNAEUS, 1758  
1. Upupa epops Linnaeus, 1758 (Hoopoe)  
   Hadjelia truncata

Family CORACIIDAE (Rollers)  
GENUS CORACIAS LINNAEUS, 1758  
   Synhimantus spiralis

1. Coracias benghalensis (Linnaeus, 1758) (Indian Roller)  
   Corvus benghalensis  
   Hadjelia truncata

GENUS HALCYON SWAINSON, 1821  
1. Halcyon smyrnensis (Linnaeus, 1758) (White-throated Kingfisher)  
   Hadjelia truncata

Family MEROPIDAE (Bee-eaters)  
GENUS MEROPS LINNAEUS, 1758  
   Cymea eurycerca

Order Cuculiformes  
Family CUCULIDAE (Cuckoos, coucals, anis, roadrunners, couas, etc.)  
GENUS CUCULUS LINNAEUS, 1758  
   Subulura strongylina

GENUS CENTROPUS ILLIGER, 1811  
1. Centropus phasianinus (Latham, 1802) (Pheasant Coucal)  
   Cuculus phasianinus  
   Subulura differens

Order Strigiformes  
Family STRIGIDAE (Typical owls)  
GENUS STRIX LINNAEUS, 1758  
   Heterakis dispar  
   Heterakis gallinarum

GENUS SURNIA DUMERIL, 1805  
   Heterakis dispar

GENUS GLAUCIDIUM BOIE, 1826  
   Heterakis dispar

Family NYCTIBIIDAE (Potoos)  
GENUS NYCTIBIUS VIEILLOT, 1816  
   Subulura suctoria

Order Columbiformes  
Family COLUMBIDAE (Pigeons and doves)  
GENUS COLUMBA LINNAEUS, 1758  
   Aonchotheca caudinflata  
   Synhimantus spiralis

1. Columba livia Gmelin, 1785 (Rock Pigeon)
Check list of helminths of guineafowls (Numididae) and host list of parasites

- Cotugnia digonopora
- Raillietina echinobothrida
- Hadjelia truncata
- Tetrameres fissispina

**GENUS STREPTOPELIA Bonaparte, 1855**

Spilopelia

- Ascaridia galli

1. Streptopelia orientalis (Latham, 1790) (Oriental Turtle-Dove)

**Order Gruiformes**

**Family RALLIDAE (Rails, crakes, moorhens and coots)**

**GENUS Fulica Linnaeus, 1758**

1. Fulica atra Linnaeus, 1758 (Common Coot)

**Family OTIDIDAE (Bustards and korhaans)**

Yamaguti (1961) lists Aonchotheca caudinflata, Heterakis gallinarum and Heterakis vesicularis from *Otis* without giving the host’s species name. It is therefore not clear whether *Otis* refers to the current genus *Otis*, *Ardeotis*, *Neotis* or *Chlamydotis*.

**Order Charadriiformes**

**Family PTEROCOLIDAE (Sandgrouse)**

**GENUS Pteroicles Temminck, 1815**

Calopterocles

- Heterakis gallinarum

1. *Pteroicles exustus* Temminck, 1825 (Chestnut-bellied Sandgrouse)

- *Pteroicles leucogaster* Temminck, 1815 (White-breasted Sandgrouse)

- *Pteroicles orientalis* Temminck, 1815 (Eastern Black-bellied Sandgrouse)

- *Pteroicles oedicnemus* (Linnaeus, 1758) (Grey-throated Sandgrouse)

- *Pteroicles oedicnemus* nana (Lapwings)

2. *Pteroicles orientalis* arenarius (Pallas, 1775) (Eastern Black-bellied Sandgrouse)

**Family JACANIDAE (Jacanas)**

**GENUS Metopidius Wagler, 1832**

Synhimantus spiralis

**Family BURHINIDAE (Thick-knees)**

**GENUS Burhinus Illiger, 1811**

1. Burhinus oedicnemus (Linnaeus, 1758) (Eurasian Thick-knee)

2. Burhinus oedicnemus (Linnaeus, 1758) (Eurasian Thick-knee)

**Order Falconiformes**

**Family ACCIPITRIDAE (Eagles, hawks, buzzards, kites, vulures)**

**GENUS Accipiter Brisson, 1760**

Synhimantus spiralis

**Family CICONIIFORMES**

**Family Podicipedidae (Grebes)**

**GENUS Tachybaptus Reichenbach, 1853**

1. Tachybaptus novaehollandiae (Stephens, 1826) (Australian Grebe)
Podiceps fluviatilis, Podiceps novaehollandiae
Tetrameres fissispina

Family ARDEIDAE (Herons, egrets and bitterns)
“Ardeiformes”
Syngamus trachea

GENUS Ardea Linnaeus, 1758
Mediorhynchus empodius

GENUS Nycticorax Forster, 1817
1. Nycticorax nycticorax (Linnaeus, 1758) (Black-crowned Night-Heron)
Ardea nycticorax
Tetrameres fissispina

Family PELECANIDAE (Pelicans)
“Pelicaniformes”
Syngamus trachea

Family CICONIIDAE (Storks)
GENUS Ciconia Brisson, 1760
Synhimantus spiralis

Order Passeriformes
Syngamus trachea

Family CORVIDAE (Crows and ravens)
GENUS Corvus Linnaeus, 1758
Heterakis gallinarum
Synhimantus spiralis

Family PYCNONOTIDAE
GENUS Pycnonotus Boie, 1826
1. Pycnonotus capensis (Linnaeus, 1766) (Cape Bulbul)
Sicarius caudatus

Family MUSCICAPIDAE (Old World flycatchers)
GENUS Turdus Linnaeus, 1758
Aonchotheca caudinflata
Synhimantus spiralis
1. Turdus viscivorus Linnaeus, 1758 (Mistle Thrush)
Ascaridia perspicillum
2. Turdus migratorius Linnaeus, 1766 (American Robin)
Planesticus migratorius
Synhimantus spiralis

Family STURNIDAE (Starlings)
GENUS Sturnus
Aonchotheca caudinflata

Family PASSERIDAE (Old World sparrows, sOWfinches and relatives)
GENUS Passer Brisson, 1760
Aonchotheca caudinflata
Synhimantus spiralis
1. Passer domesticus (Linnaeus, 1758) (House Sparrow)
Fringilla domestica
Dispharynx nasuta

GENUS Euplectes Swainson, 1829
1. Euplectes orix (Linnaeus, 1758) (Red Bishop)
Pyromelana oryx
Subulura differens

Family FRINGILLIDAE (Canaries and buntings)
GENUS Quiscalus Vieillot, 1816
Synhimantus spiralis

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REFERENCES
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