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Full Length Research Paper

Seasonality of parasitism in free range chickens from a selected ward of a rural district in Zimbabwe

Jinga Percy*, Munosiyei Pias, Bobo Desberia Enetia and Tambura Lucia

Biological Sciences Department, Bindura University of Science Education, Bag 1020, Bindura, Zimbabwe.

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A study to investigate the intensity of ectoparasites and gastro-intestinal tract worms of chickens in winter and summer was conducted in Ward 28 of Murehwa District in Zimbabwe. Sixty chickens given to local farmers to rear under the free-range system were examined for parasites; 30 in summer of 2009 and the other 30 in winter of 2010. In both seasons, ectoparasites collected were *Argas persicus*, *Echidnophaga gallinacea*, *Dermanyssus gallinae* and *Cnemidocoptes mutans*. The intensities of *A. persicus* (t= 2.54, p= 0.012) and *E. gallinacea* (t= 4.146, p= 0.000) were significantly higher in summer. There was no significant difference in seasonal intensity of *D. gallinae* (t= 0.631, p= 0.532) and *C. mutans* (t= 0.024, p= 0.978). The intensity of the nematode, *Ascaridia galli* (t= 3.889, p= 0.001) and the cestode, *Choataenia infundibulum* (t= 3.286, p= 0.004) were significantly higher in summer. There were no significant differences in the intensities of *Allodapa suctoria* (t= 0.031, p= 0.971), *Heterakis gallinarum* (t= 1.176, p= 0.248), *Capillaria obsignata* (t= 0.141, p= 0.890), *Tetrameres americana* (t= 0.514, p= 0.603), *Hymenolepis* spp. (t= 0.770, p= 0.464) and *Amoebotaenia cuneata* (t= 0.569, p= 0.579). Chickens were generally parasitised in Murehwa District. There is need to intensify parasite prevention and control, but more specifically, the control of *A. persicus*, *E. gallinacea*, *A. galli* and *C. infundibulum* in summer.

Key words: Ectoparasite, gastro-intestinal tract worm, free range system, poultry, prevalence.

INTRODUCTION

In Zimbabwe, chicken production is mostly subsistence, with almost all rural households practising it (Shumba and Whingwiri, 1988). As in many other developing tropical countries, most chickens are in backyard, free-range production systems with low output (Permin et al., 2002). Many poultry owners have marginal incomes, and poultry is therefore an important source of food and petty cash (Kelly et al., 1994). Free range chickens provide meat and eggs, both cheap sources of protein, to rural dwellers. Poultry also produces several tonnes of manure that has over the years, together with other types of farmyard manure, been used for crop growing hence fostering a healthy interaction between livestock and crop production. Rural households in Zimbabwe have relied on this livestock-crop interaction for years. However, parasitism significantly lowers productivity of free-range chickens in rural households (Okitoi et al., 1997; Mukaratirwa et al., 2001; Dube et al., 2010) mainly because of inappropriate housing and lack of pest control and preventive measures.

Several studies have shown that Zimbabwe's domestic fowl (*Gallus gallus domesticus*), like other domestic livestock species, suffers from endo- and ecto-parasite infestation (Jansen and Pandey, 1989; Kelly et al., 1994). Gastro-intestinal tract (GIT) worms in particular are known to cause poor feed conversion and utilisation. Past investigations have shown that these GIT worms are a problem to chickens in feed-scarce rural scavenging production systems (Eshetu et al., 2001; Irungu et al., 2004) as they rob nutrients and some transmit pathogens resulting in stunted growth and reduced reproductive capacity.

Common ectoparasites in chickens include lice, fleas, mites and fowl ticks. Ectoparasites may constitute a clinical problem in themselves, but may also transmit a

*Corresponding author. E-mail: percyjinga@yahoo.com. Tel: +263 712 783 913. Fax: +263 271 7552.

Abbreviation: GIT, Gastro-intestinal tract.

number of infectious pathogens and diseases to poultry, such as *Pasteurella multocida*, *Aegyptinella* spp., *Borrelia anserine*, *Plasmodium* spp., Newcastle and fowl pox. They may also act as intermediate hosts of a range of helminth infections, such as *Heterakis gallinarum, Choanotaenia infundibulum* and *Hymenolepis* spp., among others (Permin and Hansen, 1999). Ectoparasites are a clinical problem by sucking blood which can result in anaemia and death of birds. Others, like the red mite, cause irritation and egg abandonment in brooding birds.

Most studies in Zimbabwe have mostly sought to investigate only the presence of parasites in free-range chickens (Kelly et al., 1994; Mukaratirwa et al., 2001; Permin et al., 2002; Dube et al., 2010) and other threats (Muchadeyi et al., 2005). Not much has been recorded in literature on the intensity of parasites by season in Zimbabwe. There is need to direct scarce resources at the right time to improve free-range chicken productivity, a source of livelihoods for millions in the rural areas. Thus, the objective of this study was to investigate seasonality of parasitism in free-range chickens, with particular emphasis on ectoparasites and GIT worms, so as to inform, more appropriately and effectively, on allocation of merger resources for control and preventive measures.

MATERIALS AND METHODS

Study area

Free-range chickens were reared in Ward 28 of Murehwa District (31°38′ E, 17°48′ S) in north-eastern Zimbabwe. The area receives a mean annual rainfall of 750 to 1000 mm (USDA, 2004). Summers are warm and wet, from September to March, with temperatures ranging from 16 to 25°C. The area experiences cold and dry winters, from April to August. Temperatures in winter range from 10 to 18°C. The ward is typically rural, with subsistence crop cultivation and small-scale animal husbandry being the major sources of livelihood for most families. Local chicken rearing is a major activity, with almost all households practising it.

Sampling

Subsistence local farmers in Ward 28 of Murehwa District were recruited as participants. They fulfilled a basic selection requirement of having a fowl run. Fifteen households who met the inclusion criterion were randomly selected from a list obtained from the district office. Free-range chicks were purchased in March 2009 and distributed to participants. Each participant was given six chicks to rear. The participants were given instructions not to administer any parasite prophylaxis. Two chickens were collected from each participant, between December 2009 and March 2010, constituting a summer collection, for parasitological examination. A second collection session, constituting a winter collection, was done between July and September 2010. Sixty of the 90 distributed chickens were finally examined. Parasitological examinations were done at the Department of Biological Sciences Laboratory of Bindura University of Science Education.

Collection and identification of ectoparasites

Ectoparasites were collected by dust-ruffling using a combination of pyrethrin and piperonyl butoxide as the insecticidal powder. Ectoparasites were collected over a coloured surface illuminated with a strong light source and stored in 70% alcohol. Paper bags used for transportation of chickens were also dusted. The ectoparasites were later dehydrated first in 80%, then 90% and finally 99% alcohol before being cleared in xylene and mounted on slides for identification with a light microscope. Ectoparasites were identified according to keys described by Soulsby (1982), Walker (1994), Kaufmann (1996), Wall and Shearer (1997), and Permin and Hansen (1999).

Collection and identification of GIT worms

Following slaughter of chickens and evisceration, the separated viscera were detached into five sections, the oesophagus, crop, gizzard with proventriculus, ceacum, and the rest of the intestines. Each section was incised longitudinally; visible worms to the naked eye were picked up using thumb forceps and preserved in 70% alcohol. The mucosae of the intestine were washed with tap water to remove any adhering worms. The contents were sieved through a 100 µm aperture test sieve to recover all smaller worms (Magwisha et al., 2002). For identification, nematodes were mounted in drops of lactic acid, whereas cestodes were stained with aceto-alum-carmine. All worms were later examined under a light microscope. Identification was based on keys described by Soulsby (1982), Anderson (1992), Ruff and Norton (1997), Reid and Mc Dougald (1997), and Permin and Hansen (1999).

Statistical method

The difference in parasite intensity between seasons was tested by the bootstrap t-test (Rozsa et al., 2000) using the software package Quantitative Parasitology 3.0.

RESULTS

Ectoparasites

Four ectoparasites were identified in the two seasons and these were *Argas persicus* (Acari: Ixodida), *Echidnophaga gallinacea* (Insecta: Siphonaptera), *Dermanyssus gallinae* (Acari: Mesostigmata) and *Cnemidocoptes mutans* (Acari: Astigmata). *E. gallinacea* was observed in 100% of sampled chickens, while for *A. persicus*, *D. gallinae* and *C. mutans*, the prevalence was 81.67, 75.00 and 56.67%, respectively. Figure 1 shows the mean intensity of ectoparasites by season.

There was a higher intensity of *A. persicus* ($t= 2.54$, $p= 0.012$) and *E. gallinacea* ($t= 4.146$, $p= 0.000$) in summer. There was no significant difference in the intensity of *D. gallinae* ($t= 0.631$, $p= 0.532$) and *C. mutans* ($t= 0.024$, $p= 0.978$) between summer and winter. The most prevalent ectoparasite in both seasons was *E. gallinacea* (100%) while the least was *C. mutans* (56.67%).

GIT worms

Six nematode species were identified, which were *Ascaridia galli* (Secernentea: Ascaridida), *Allodapa*

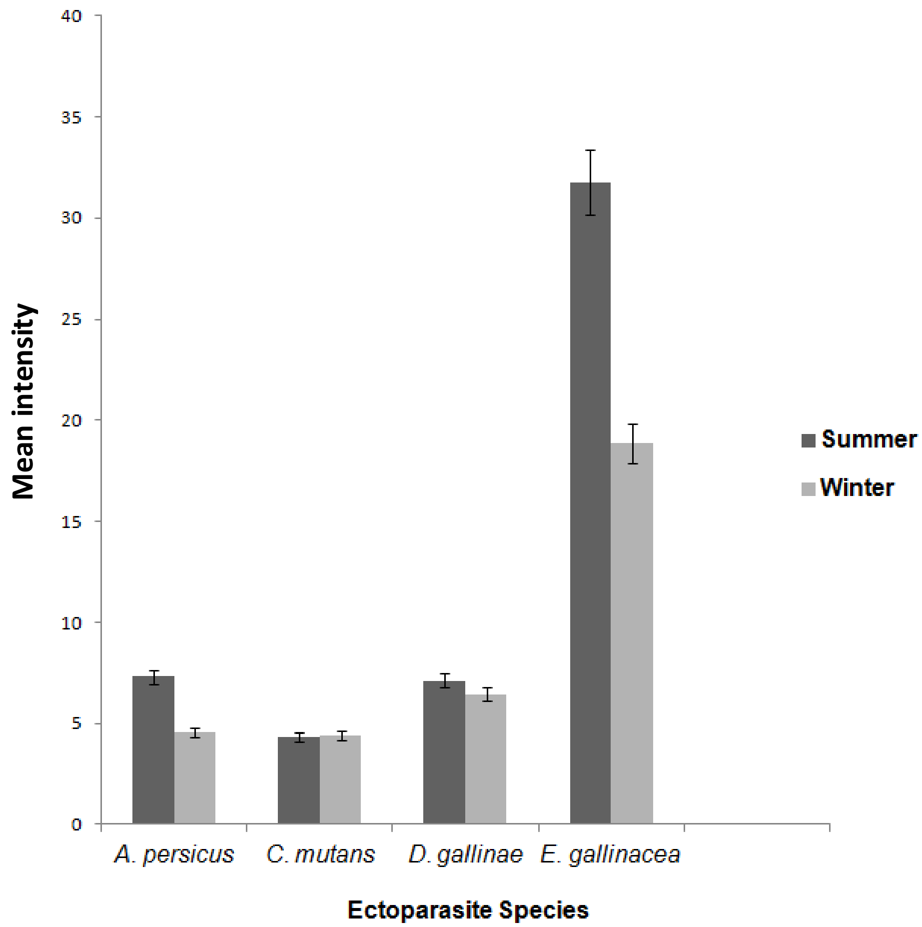

Figure 1. Seasonal mean intensities of ectoparasites.

suctoria (Secernentea: Ascaridida), *Heterakis gallinarum* (Rhabditia: Ascaridida), *Capillaria obsignata* (Adenophorea: Trichocephalida) and *Tetrameres americana* (Secernentea: Spirurida), infesting 93.33, 96.67, 95.00, 100, and 56.67% of the chickens, respectively. Cestodes identified were *Choanotaenia infundibulum* (Neoophora: Cyclophyllidae), *Amoebotaenia cuneata* (Neoophora: Cyclophyllidea) and *Hymnolepis* spp. (Cestoda: Cyclophyllidea), infesting 100, 30.00 and 33.33% of the chickens, respectively. Figure 2 shows the average seasonal mean intensity of the GIT worms. The most prevalent GIT worms were *C. obsignata* and *C. infundibulum*, which were collected from all chickens examined. The least prevalent were *Hymenolepis* spp. (33.33%) and *A. cuneata* (30.00%).

There was a higher intensity of *A. galli* (t= 3.889, p= 0.001) and *C. infundibulum* (t= 3.286, p= 0.004) in summer. There was no difference in the intensity of the other nematodes, *A. suctoria* (t= 0.031, p= 0.971), *H. gallinarum* (t= 1.176, p= 0.248), *C. obsignata* (t= 0.141, p= 0.890), and *T. americana* (t= 0.514, p= 0.603), and the cestodes, *Hymnolepis* spp. (t= 0.770, p= 0.464) and *A. cuneata* (t= 0.569, p= 0.579).

DISCUSSION

Ectoparasites

In summer, there was a higher intensity of *A. persicus* and *E. gallinacea.* The presence of *A. persicus* has been reported in Zimbabwe (Dube et al., 2010) and elsewhere in sub-Saharan Africa (Mungube et al., 2008). Higher temperatures in summer yield faster development rates of tick larvae, nymphs, and adults, with the precise rate of development varying between stages and species (Olson and Patz, 2010). Decreasing day length reduces probability of tick questing and low temperatures may inhibit activity altogether, while increasing day length may be permissive, but only if temperatures are high enough (Randolph, 2004). The mean intensity observed could have been higher as *A. persicus* is a nocturnal feeder, moving back to cracks and crevices at dawn. The collection strategy potentially excluded many other parasites which feed during the night, such as mosquitoes, because chickens were collected during daytime.

The predilection site of *E. gallinacea* was the skin of the

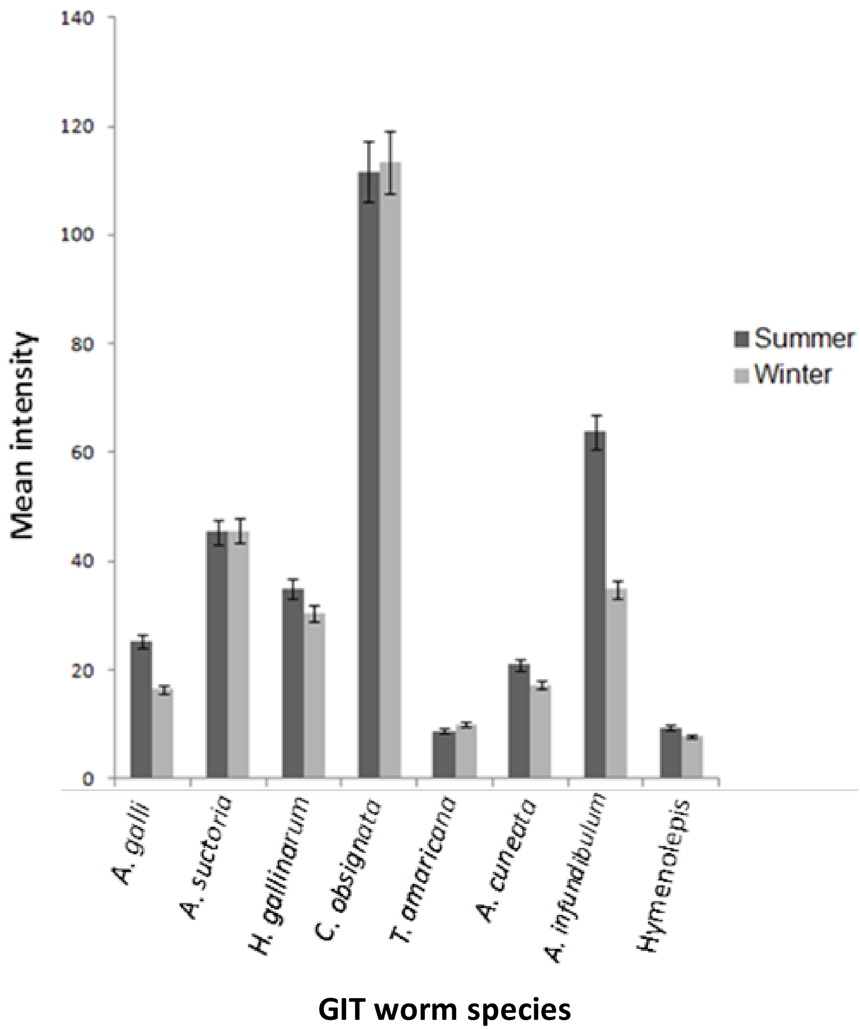

Figure 2. Seasonal mean intensities of GIT worms.

head, often around the eyes in clusters of dozens. Environmental factors affect the survival of pre-imaginal fleas by virtue of the large part of their life cycle that is spent off the host (Krasnov et al., 2001). Adult sticktight fleas overwinter in a cocoon and emerge in response to a sharp increase in temperature (Robinson, 2005), resulting in a higher intensity in summer. Eggs typically hatch in high humidity of around 70%. Low humidity increases egg mortality. Hatching of eggs occurs at around 25°C (Robinson, 2005), thus avoiding emergence of larvae in winter when temperatures are lower. The higher intensity in summer of *E. gallinacea* may also be attributed to an increase in population of their alternative hosts, such as rodents.

There was no significant difference in the intensity of *D. gallinae* and *C. mutans* in summer and winter. The intensity of *D. gallinae* could possibly have been higher as these are nocturnal feeders. *C. mutans* completes its entire life cycle on the host (Permin and Hansen, 1999), engorged under the scales of legs, where the microclimate is almost perennially homogeneous, thus rendering external seasonal variation in climate inconsequential. A study on developmental time on pre-oviposition, oviposition, eggs, larvae, protonymph and deutonymph of *D. gallinae* showed that the parasite invariably persists at temperatures ranging from 15 to 30°C (Tucci et al., 2008). There was, however, a life cycle duration increase at 35°C, suggesting extremely high temperature retards growth of *D. gallinae* (Tucci et al., 2008). With maximum temperatures of up to 25°C in summer in Murehwa District, the intensity of *D. gallinae* remained unaffected.

GIT worms

The intensity of *A. galli* and *C. infundibulum* was higher in summer. Results of *A. galli* are consistent with those observed by Mungube et al. (2008) and Permin et al. (1997). Wet and humid conditions are necessary for development of eggs to infective stages. During the summer season, the population of earthworms, paratenic hosts of *A. galli*, increases, and thus potentially increasing the intensity of the parasite. However, a study by Magwisha et al. (2002) found the intensity of *A. galli* to be invariable all year. *A. galli* infestation causes occlusion of the intestinal lumen leading to rupture of the intestines.

The presence of the cestode *C. infundibulum* in free range chickens of sub-Saharan Africa has been reported in several studies (Mungube et al., 2008; Permin et al., 2002). Its higher intensity in summer in this study could possibly be explained by an increase in the population of its intermediate hosts. The intermediate hosts are, among others, beetles of the genera *Tribolium*, *Geotrupes*, *Aphodius* and *Calathus*.

There was no seasonal variability in the intensity of the nematodes *A. suctoria*, *C. obsignata*, *H. gallinarum* and *T. americana*, and the cestodes *A. cuneata* and *Hymenolepis* spp. Presence of these GIT worms has been reported in Zimbabwe (Permin et al., 2002). Results of this study are different from those obtained by Magwisha et al. (2002) who reported seasonal variability in the parasite load of *Capillaria* spp, *T. americana* and *Hymenolepis* spp. in domestic fowl in Tanzania. In Ethiopia, Ashenafi and Eshetu (2004) observed variability in the intensity of *Amoebotaenia* spp., *Capillaria* spp., *H. gallinarum* and *Hymenolepis cantainana* according to agro-climatic zones, while in Kenya, Mungube et al. (2008) observed an increase in the intensity of *H. gallinarum* during the rainy season. Differences of parasitism among African countries could be a result of differences in climatic conditions and altitudinal differences of the countries.

Conclusion

The presence of ectoparasites and GIT worms suggest chickens kept under the free range system are generally parasitized in Ward 28 of Murehwa District in Zimbabwe. The higher intensity of *A. persicus*, *E. gallinacea*, *A. galli* and *C. infundibulum* in summer suggests that the occurrence of these parasites is affected by climatic conditions. There is need to increase preventive and control methods of the parasites in summer to avoid loss of income to rural poultry farmers. There is also need to step up efforts to contain the most prevalent parasites, *C. obsignata*, and *C. infundibulum*. Although this study did not attempt to investigate the presence of diseases vectored by the parasites, the presence of parasites in the production systems should guide the scope of control strategies to include the diseases.

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