Review article — Oorsigartikel

Epidemiology and control of trematode infections in cattle in Zimbabwe: a review

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ABSTRACT

In this paper the main epidemiological aspects of the major domestic ruminant trematode infections in Zimbabwe are reviewed and discussed with regard to the available options for control. Seasonal occurrence of amphistomes, *Fasciola gigantica* and *Schistosoma mattheei* are considered both in the definitive and intermediate hosts. The regional distribution of the trematodes is reviewed in relation to the distribution patterns of their snail intermediate hosts. Based on the epidemiological features of the trematodes, practical control measures are suggested.

Key words: amphistomes, cattle, epidemiology, *Fasciola gigantica*, review, *Schistosoma mattheei*, trematodes, Zimbabwe.

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INTRODUCTION

The digenetic trematodes differ from other groups of parasites in that the first larval stages develop in intermediate hosts from the same phylum, namely, the Mollusca[68]. The biotopes of the different trematodes have much in common, particularly as the snail intermediate hosts occupy identical niches in a food chain[45]. In addition, 2 or more species of snails may be found commonly together in the same habitat[4]. Thus, the prevalence of snail-borne diseases is not so much influenced by the mere abundance of infected animals as it is by the abundance and efficiency of the snail intermediate hosts[25]. Hence, availability of the snail intermediate hosts and the grazing habits of the definitive hosts to a large extent determine the epidemiology and seasonal pattern of infection with trematodes[9,19,27,62,63,69]. Therefore, to be effective, control measures depend upon a sound understanding of the epidemiology of the disease in both the definitive and intermediate hosts.

In this paper, we review the information on the epidemiology of trematodes of cattle available to date and discuss the different options with regard to their control.

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SPECIES OF TREMATODES AFFECTING CATTLE IN ZIMBABWE

Fasciolosis is caused by several species of flukes belonging to the genera *Fasciola* Linnaeus, 1758, and *Fascioloides* Ward 1917[9]. The 2 main known species of *Fasciola*, which are parasitic in domesticated animals, and sometimes in man in the tropics, are *F. gigantica* and *F. hepatica*[9]. However, only 1 species of liver fluke, *F. gigantica*, has been reported in cattle in Zimbabwe[7,14,43,73]. The fluke infects a large number of domesticated and game animals[16].

The Paramphistomidae and related trematode families comprise numerous genera and species, of which a limited number sometimes produce massive infections in domestic livestock and wild grazing animals in Africa[21–24]. Thirty-two species of amphistomes belonging to 3 families, Paramphistomidae, Gastrothylacidae, and Stephanopharyngidae, have been recorded from ruminants in Africa[21]. However, only 10 species belonging to the genera *Paramphistomum*, *Calicophoron*, *Cotylophoron* and *Carmyerius* are regarded as common amphistomes of domestic ruminants[21–24]. Although numerous species of amphistomes exist, outbreaks of amphistomosis are confined to massive infections by certain species only[27]. Out of the many species of the family Paramphistomidae recorded in Africa, *Calicophoron microbothrium* is one which causes acute amphistomosis, resulting in heavy losses amongst infected animals[21]. *Calicophoron calicophorum, C. microbothrium, C. phillerouxi, C. raja, C. sukari, C. sukumum, Cotylophoron cotylophorum, Cot. jacksoni, Carmyerius bubalis*, and *Ca. spatiosus* have been reported to occur in cattle in Zimbabwe[65].

As many as 10 different species of schistosomes infect cattle, the most important being *Schistosoma bovis, S. mattheei* and *S. curassoni* in Africa and *S. spindale, S. indicum, S. nasale* and *S. japonicum* in Asia[19]. Except for *S. nasale*, all species are parasitic in the mesenteric and rectal veins and are responsible for intestinal schistosomosis[19]. *Schistosoma margrebowiei* and *S. leiperi*, which infect antelopes, are found in Africa and *S. incognitum*, which infects pigs and occasionally cattle, is found in Asia[19]. In Zimbabwe, the only schistosome known to occur in cattle is *S. mattheei*[31].

Distribution of cattle trematodes in Zimbabwe

The occurrence of trematode infections in Zimbabwe has been recorded in various published and unpublished records. Prevalence studies based on coprological examinations (Table 1) and abattoir surveys for *F. gigantica* (Table 2) and *S. mattheei*[13] have been reported. However, limited prevalence studies have been carried out in the lowveld[50–52] and there are no data on amphistome abattoir surveys in the country. Depending on location, the prevalence of animals infected ranged from 11 % to 100 % for amphistomes, 0 % to 90 % for *F. gigantica* and 0 % to 21.5 % for *S. mattheei* (Table 1). Abattoir surveys have shown a national prevalence of 12 % to 46.3 % of cattle infected with *F. gigantica* (Table 2). The available data on *S. mattheei* abattoir surveys indicated a national prevalence of 69 % with the prevalence varying from 35 % to 92 % from one abattoir to another[13].

These data reveal that amphistomes and *F. gigantica* are the predominant trematode parasites in cattle while *S. mattheei* is less prevalent. The high prevalence of amphistomes in Zimbabwe compared with the other 2 trematodes

Table 1: **Summary of prevalence of trematode infections in cattle from various parts of Zimbabwe.**

Region	Province	Communal lands surveyed	Dip tanks surveyed	Duration of survey (months)	Prevalence (%) (mean or range)			
					Amphistomes	*F. gigantica*	*S. mattheei*	References
Highveld	Mashonaland Central	6	12	12	50.1–100	16.9–90.0	1.4–20.7	5
Highveld	Mashonaland East	15	24	12	30.0–86.2	0.0–64.9	0.0–21.5	5
Highveld	Mashonaland West	14	–	12	13.2–95.5	0.0–38.3	0.0–11.7	6
Highveld	–	–	56	28	–	26.3	–	72
Highveld	Mashonaland Central	1	–	12	65.2	61.5	3.1	73
Highveld	Mashonaland East	9	64	26	32.3	53.7	4.9	74
Highveld	Mashonaland East, West and Central	4	12	24	21.3–56.2	14.0–26.9	3.2–9.6	50,51,52
Lowveld	Matabeleland South and Midlands	3	9	24	11.0–32.0	3.9–15.4	1.1–6.8	50,51,52

– = No data provided.

can probably be attributed to the wide range of intermediate hosts and several species of the trematode affecting cattle.

Differences in the prevalence of infections in cattle from different geographical areas have been recorded, which apart from seasonal changes, are thought to be associated with the presence or absence of intermediate snail habitats in the grazing areas of the animals. From faecal egg counts (Table 1) and abattoir studies (Table 2) the prevalence of *F. gigantica* infection is high in the high rainfall districts of the highveld compared with the relatively drier districts of the lowveld. Similar patterns have been observed for amphistomes (Table 1) and *S. mattheei*[13,28,29] (Table 1).

The greatest risk of fasciolosis in East Africa, for instance, has been reported to occur in areas of extended annual rainfall, with risk diminishing in areas of shorter wet season and/or lower temperatures[33]. Metacercariae survival is reduced in hot conditions and the duration of their viability is directly related to relative humidity and inversely to temperature and exposure to sunlight[69]. High rainfall areas favour development and survival of both the intermediate host snail and the developmental stages of the parasite and hence arid areas were found to be generally unsuitable for occurrence of fasciolosis[33].

Besides high rainfall and suitable temperatures for the survival of both the intermediate hosts and the developmental stages of the parasites, the highveld is characterised by wet/swampy grazing areas where distribution of snail habitats is widespread. On the other hand, in the lowveld rainfall is low and temperatures are high and it is characterised by dry land grazing with focal distribution of snail habitats. Thus, in the lowveld stock have little access to natural water bodies and reservoirs. The exposure of a larger population of cattle to infected herbage surrounding watering points is most likely to be higher on the highveld than the lowveld and this probably accounts for the differences in trematode prevalence noted.

Age and seasonal variation in faecal egg counts

The prevalence of *F. gigantica* and amphistomes as observed through faecal egg counts is high in adult cattle compared to calves[50,51,73,74]. By contrast, based on faecal egg counts, *S. mattheei* is more prevalent in calves than in adult cattle[52,73,74]. The high prevalence of amphistomes observed in adults is attributed to their long exposure time leading to development of immunity against the pathogenic effects of the immature flukes but still having the mature amphistomes maintaining their high capacity of egg production[27]. Similarly, the high prevalence of *F. gigantica* in older animals is related to the longer exposure time and accumulation of flukes in the liver compared to young animals[74]. Worm burdens of schistosomes in naturally infected animals increase with the age of the animal[19]. In contrast to worm burdens, faecal egg counts decrease with age of the host[17,56]. The decline in egg output of schistosomes is due to development of immunity, which acts mainly through suppression of worm fecundity[30].

The above-mentioned observations lead to the conclusion that adult cattle act as a constant source of *F. gigantica* and amphistome infection for the more susceptible young animals. In contrast to the 2 other trematodes, young animals below 2 years of age play a major role in the environmental contamination with *S. mattheei* eggs. The potential importance of older age groups in the spread of schistosomosis is further reduced by an age-related decrease in egg hatchability[18].

In Zimbabwe, *F. gigantica,* amphistome and *S. mattheei* faecal egg counts are reported to follow a seasonal pattern,

Table 2: **Summary of information on liver condemnations** (%) **due to *Fasciola gigantica* from various studies at various abattoirs in Zimbabwe.**

Years of study	Duration (months)	Abattoir*									References
		HRE	BYO	MSV	MTR	KDA	MRA	CHY	WN	Overall	
1956–1961	60	17.6	4.8	3.5	22.1	–	–	–	–	12.0	14
1972–1974	24	42.0	19.0	21.0	–	41.0	–	–	–	30.8	43
1977–1981	60	43.8	29.1	21.8	47.4	40.2	57.8	59.4	–	42.8	16
1984–1986	24	–	–	–	–	–	–	–	–	46.3	7
1988–1990	27	–	44.8	28.1	–	39.1	49.3	52.2	35.3	41.5	72
1989–1994	63	–	35.9	25.9	–	23.1	62.9	47.4	–	39.6	48
1990–1999	120	–	33.9	28.8	–	33.6	44.9	42.1	–	37.1	53

HRE = Harare, KDA = Kadoma, BYO = Bulawayo, MRA = Marondera, MSV = Masvingo, CHY = Chinhoyi, MTR = Mutare, WN = West Nicholson.

with an increase in the counts towards the end of the dry season and during the wet months of the year (October to March)[50,51,73,74]. In support of this observation, peak liver condemnations due to chronic fasciolosis have been reported during the rainy season between December and April[53]. Similar seasonal patterns for fasciolosis[1,41,46,63] and amphistomosis[2,44] have been reported elsewhere.

The above-mentioned observations combined with the data on intermediate snail population dynamics and trematode infections in the intermediate hosts, which are common during the dry season, lead to the conclusion that the definitive hosts acquire infection during the beginning and/or middle of the dry season. This results in patent infections at the end of the dry season and during the wet months of the year. However, the timing may vary depending on location, length of the rainy season and the grazing habits of the cattle.

INTERMEDIATE HOSTS OF TREMATODES OF CATTLE IN ZIMBABWE

The snail host of *F. gigantica* in tropical Africa is *Lymnaea natalensis*, the only intermediate host of *F. gigantica* so far reported in Zimbabwe[14,16,43].

A wide range of species of *Bulinus* and *Biomphalaria* has been reported to act as intermediate hosts of amphistomes in Africa[81]. Field studies published on the natural intermediate hosts of amphistomes have shown *B. tropicus* to be the most common intermediate host in Zimbabwe[11,38,50]. Natural infections of *B. globosus*[11], *B. forskalii,* and *Biomphalaria pfeifferi*[11,50] with amphistome cercariae have been recently recorded in the country. The susceptibility of *B. tropicus, B. globosus, Biom. pfeifferi, L. natalensis* and *Melanoides tuberculata* to *C. microbothrium* has been experimentally examined[10]. The results showed that *B. tropicus, B. globosus, Biom. pfeifferi* and *M. tuberculata* were susceptible to *C. microbothrium* infection with varying degrees of susceptibility[10]. *Bulinus tropicus* had a prevalence of 65 %, followed by *Biom. pfeifferi* (37.5 %), *B. globosus* (6.8 %) and *M. tuberculata* (5.9 %)[10]. *Lymnaea natalensis* was refractory to infection[10].

In southern Africa, natural infections with *S. mattheei* have only been recorded in *B. globosus* and *B. africanus*[4]. In Zimbabwe, natural infections with *S. mattheei* have only been recorded in *B. globosus*[8,31,67,77–79]. *Bulinus tropicus* snails experimentally infected with *C. microbothrium* or co-infected with either *S. haematobium* or *S. mattheei* after exposure to *C. microbothrium* produced *C. microbothrium* cercaria only[10]. These findings seem to indicate that *B. tropicus* infected with *C. microbothrium* does not play any role in the transmission of *S. haematobium* or *S. mattheei,* the 2 common schistosome species of medical and veterinary importance in Zimbabwe[10].

Therefore, information gained from the field and experimental studies show that *B. tropicus, B. globosus, B. forskalii, Biom. pfeifferi* and *M. tuberculata* can serve as intermediate hosts of amphistomes in Zimbabwe. However, the actual contribution of the snail species to the transmission of amphistomes depends on a complex set of factors, which include susceptibility, distribution and relative abundance of the different snail species, habitat preferences and availability of the definitive hosts[10]. *Bulinus tropicus* is widely distributed and abundant in Zimbabwe[11,32,50]. This information, combined with relatively high natural infections in the field[11,50] and high experimental degree of susceptibility[10] makes *B. tropicus* to be the most important intermediate host in the transmission of amphistomes in Zimbabwe. *Lymnaea natalensis* and *B. globosus* serve as the intermediate hosts of *F. gigantica* and *S. mattheei,* respectively.

Distribution of intermediate hosts

Studies on ecology and population dynamics of snails in Zimbabwe have concentrated mainly on *B. globosus* and *Biom. pfeifferi*[8,66,77–80] and these studies have been mainly restricted to the highveld of Zimbabwe. Work has also been done on the population genetics of *B. globosus*[36,37], *B. tropicus*[38] and *Biom. pfeifferi*[76].

Recent snail surveys[12,32,49] provided information on the geographic distribution of snails in Zimbabwe.

Lymnaea natalensis is reported to be the most common and widely distributed freshwater snail found in the country and can tolerate a wide range of conditions[14,16,32]. It is found in a variety of habitats that include natural water bodies, reservoirs, ornamental ponds and cattle drinking troughs[14,16]. However, 13 789 freshwater snails recently collected over a 2-year study period in the highveld and lowveld regions of the country showed *B. tropicus* to be the most abundant snails species[12], contributing 31.4 % of the snails species, followed by *L. natalensis* (25.5 %), *B. globosus* (22.6 %) and *Biom. pfeifferi* (19.5 %).

Distribution of the snail intermediate hosts of trematodes showed variation between the highveld and lowveld regions. Earlier studies on the highveld showed schistosome hosts to be the most abundant snail species[77]. However, recent ecological studies showed *L. natalensis* to be the most abundant snail species on the highveld (34 %) followed by *B. globosus,* which contributed 30.5 % of the snails recorded on the highveld[12]. *Bulinus tropicus* is the most abundant snail species in the lowveld[12,49] and is significantly more abundant in the lowveld than on the highveld[50]. There seems to be no variation in the occurrence of *Biom. pfeifferi* between the 2 regions[50]. *Bulinus forskalii* is relatively more rare than the other snail species and relatively common on the highveld than in the lowveld[50].

Variations in the distribution of the snail species according to habitat have been observed[12,50–52]. The relative density of *B. globosus* and *L. natalensis* is significantly higher in highveld dams compared to lowveld dams. However, no significant difference in *L. natalensis* and *B. globosus* was noted between highveld dams and streams. By contrast, *B. tropicus* is more common in lowveld dams than highveld dams. On the highveld, *B. tropicus* has been recovered mainly from dams and *Biom. pfeifferi* mainly from streams.

The distribution of the snail species has also been observed to be associated with aquatic vegetation[12,32,50–52,77]. *Bulinus tropicus* was found to be associated with *Polygonum, Cyperus* and *Nymphaea* species and negatively associated with *Phragmites mauritianus* and *Typha* species. *Biomphalaria* is associated with *Phragmites mauritianus, Potamogeton* and *Scirpus* species and negatively associated with *Cyperus* species. *Nymphaea, Potamogeton* and *Typha* species were correlated with *B. globosus* while *L. natalensis* was correlated with *Potamogeton* and negatively associated with *Cyperus.* Although the relationships need not be causative, these plant species are useful predictors of the snail abundance in a habitat.

Information gained from the above-mentioned studies indicates that *L. natalensis* and *B. globosus* are abundant in the highveld region while *B. tropicus* is more abundant in the lowveld region. *Biomphalaria pfeifferi* is common in both regions and *B. forskalii* is relatively rare but more common in the highveld region. Therefore, in Zimbabwe, both *B. globosus* and *L. natalensis* appear to thrive under 2 sets of conditions; large water bodies, which may hold water continuously for several years, and small water bodies which dry out during the annual seasonal cycle[65]. By contrast, *B. tropicus* appears to thrive well in large water bodies compared with small water bodies whereas *Biom. pfeifferi* does well in small water bodies[47]. The variation in the abundance of the snails between habitats is a reflection of vegetation type, presence or absence of other mollusc species, variation in local rainfall, seasonal water flow and water temperature.

Seasonal variations of the intermediate hosts

The population of *L. natalensis* has been reported to increase at the end of the wet season, reaching a peak during the mid-dry season and decreasing towards the end of the dry season[12,51]. The population density is negatively correlated to rainfall and positively correlated to temperature[51]. Similar trends have been reported elsewhere[40,64]. The low numbers of snails during the rainy season has been attributed to lack of suitable vegetation and rapid movement of water[40,64].

Hatchlings of *L. natalensis* were found almost throughout the year with, peaks between April and August[12,51]. The size prevalence structures seem to suggest that *L. natalensis* is a continuous breeder.

The population of *Biom. pfeifferi* showed a peak at the end of the dry season and a decline over the rainy season[77]. However, high densities of *Biom. pfeifferi* have been recorded during the rainy season and low densities during the dry season[12].

Bulinus tropicus peaked during the rainy season in highveld dams and towards the end of the rainy season and early dry season in lowveld dams[12].

Conditions favourable for natural increases in the population of *B. globosus* occur at the beginning (November/December) and end (March/April) of the rainy season[31,36,66,77,79]. However, no clear-cut seasonal patterns for this species have been observed[12,52,77] although a decline was observed during the rainy season[77]. Egg production and abundance of the juvenile snails was high during the beginning and end of the summer rainy season[66,77].

Information from the above studies indicates that the snail intermediate host populations undergo marked seasonal variations in density with generally low densities during the rainy period and high densities in the post-rainy periods. The numerical size of the population is dependent on several climatic factors, such as flooding, desiccation and temperature and on the natural rate of increase of the snail species following catastrophies[66,77].

Trematode larval infections in intermediate hosts

A variety of trematode larval infections have been reported in *L. natalensis* in Zimbabwe, which include echinostome cercariae, strigea cercariae, xiphidiocercariae and brevifurcate apharyngeate monostome cercariae[11]. However, in the available literature no quantitative data have been reported on redia and cercaria of *F. gigantica* in *L. natalensis* in Zimbabwe. None of the snails collected over a period of 24 months were found shedding *Fasciola* cercariae[51]. Shedding occurred during the day and all collected snails were returned to their respective habitats after the shedding process. Timing and method of shedding could probably have contributed to the negative results since about 80 % of *F. gigantica* cercariae are shed at night[15].

However, it is during the high peak of the snail population that the highest number of snails is liable to infection by *F. gigantica* miracidia[16]. Towards the end of winter and the beginning of spring, the fluke life cycle within the snail progresses to the cercarial stage[16] as has been observed elsewhere[40,64]. Although there is probably a seasonal variation in cercarial output of *F. gigantica,* there is no season of the year when emission of cercariae does not occur in Zimbabwe[14]. Also encysted metacercariae have been found to survive for at least 4 months in water in Zimbabwe[14] and it can be presumed that a body of water inhabited by *L. natalensis* can be a permanent source of infection. Young snails are more susceptible than adults and peaks of snail breeding are probably more important than actual snail numbers as precursors of fasciolosis outbreaks[14]. Hatchlings of *L. natalensis* were found almost throughout the year, with peaks between April and August[12,51]. Hence, transmission is most likely to be high during the month of April through to August. In addition, metacercariae were found on herbage from the fringes of the snail habitats between February and August, with most of the metacercariae concentrated on herbage 0 to 1 m from the edges of the habitats[51].

Natural patent amphistome cercarial infections were recorded from *B. tropicus, B. globosus, B. forskalii* and *Biom. Pfeifferi*[12]. *Bulinus tropicus* contributed the majority of amphistome infections (96.5 %), followed by *Biom. pfeifferi* (2 %), *B. globosus* (1.2 %) and *B. forskalii* (0.3 %). The prevalence of patent amphistome infections shows spatial heterogeneity, varying from 0 to 19.5 % on the highveld and 0 to 20.5 % in the lowveld[50]. Both on the highveld and in the lowveld, transmission by the main intermediate host, *B. tropicus,* occurred between February and September, with peak transmission between April and May and during August and September[12,50]. Metacercariae were found on herbage from the fringes of the snail habitats between February and August[50]. Hence, transmission is high during the month of April to September.

The prevalence of patent *Schistosoma* infections in *B. globosus* shows spatial heterogeneity, ranging from 0 % to 18.8 % at individual sites on the highveld and from 0 % to 4.5 % in the lowveld[52] and, exceeding 50 % in some sections of a river[77] and it varied from 0 % to 70 % over distances less than 100 m[78]. These differences in patent infected snails in space is attributed to recent river conditions, i.e. floods and drought; and patchy contamination of the water by excreta[77,79]. As observed elsewhere[17,54,55], the seasonal patterns in the transmission of *Schistosoma* cercariae have been reported in Zimbabwe[8,52,67,77]. The transmission exhibited a marked seasonal pattern, being more intensive during the hot, dry season (September–November) and markedly reduced during the cold, dry season (June–August). During the rainy (December–February) and warm, post-rainy (March–May) seasons transmission was moderate and variable.

Information gained from the above observations indicates that the transmission of trematodes by the intermediate hosts is high during the dry season. The increase in transmission during the dry season is attributed to decreased water volume observed in the habitats during the dry season leading to high focal concentration of the intermediate hosts. This is accompanied by increased contact of the habitats by livestock due to scarcity of pasture and increased grazing around water bodies, thereby favouring accumulation of trematode eggs in close proximity to snail habitats. These factors result in increased frequency of contact between miracidia and snail intermediate hosts thereby increasing the prevalence of infection in the latter.

EPIDEMIOLOGICAL CYCLE OF TREMATODE INFECTIONS IN CATTLE IN ZIMBABWE

Extrapolation of observations mentioned above give a picture of the epidemiology of amphistomosis, fasciolosis and schistosomosis in Zimbabwe as shown in Figs 1, 2 and 3, respectively. During the wet season there is abundant grazing and alternative sources of drinking water. During an average year, from November to July, grazing is usually adequate in most areas of the country[14]. Therefore, this reduces the need for animals to graze near, and to drink, particularly from permanent water holes. Cattle and sheep prefer higher, drier ground in the summer months[14] and thereby avoid fluky parts of the pasture as long as this choice of grazing is available to them.

Also during the wet season animals are deliberately pastured away from wet, low-lying marshy areas and permanent pools especially in the communal areas

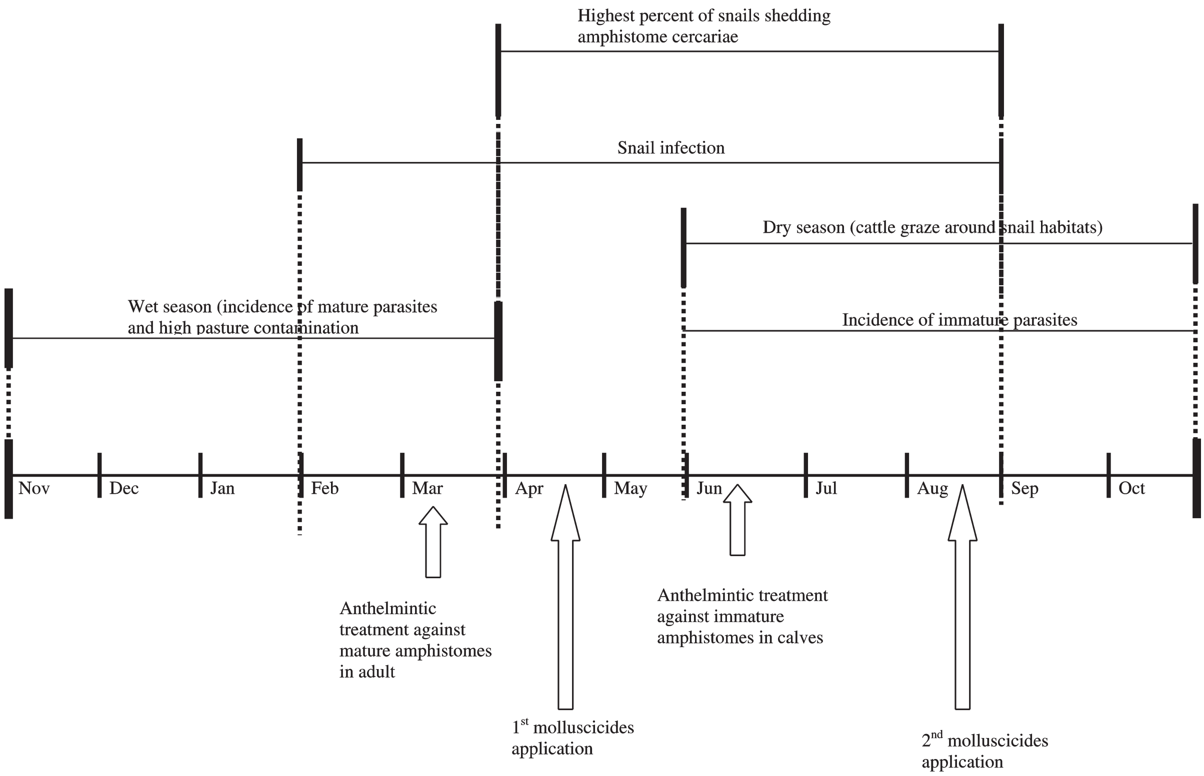

Fig. 1: **Seasonal occurrences related to the epidemiology and treatment of amphistomes in cattle in Zimbabwe.**

where most of the land will be under crop production. In addition, snail habitats and pastures are constantly flooded and, therefore, snails and the free-living stages of the parasites are regularly flushed out and disseminated over a large area[9]. As a result, only light infections are likely to be acquired during the wet season.

Towards the end of the rainy season (March–April) eggs of *Fasciola*, amphistomes and S*chistosoma* dropped on pasture survive to infect the new generation of snails, which start to grow at the end of the rainy and beginning of the dry seasons. During this period the miracidia would be fairly abundant, as the egg-excretion by cattle has been observed to be high during this period[64]. In addition, the egg survival and miracidium/snail contact would be better during this period[64].

During the dry season, animals are free-ranging, especially in communal areas. Therefore, the observed peak cercarial shedding during the dry season coincides with a reduction of the available grazing areas and sources of drinking water for livestock. This therefore increases the need for animals to graze near, and drink from permanent water sources. The increased animal concentration at the few permanent watering points would lead to increased contamination of these areas with fluke eggs, giving rise to heavy infections in the snails. A significant increase in herbage metacercarial and water cercarial density would lead to acute infections of the animals with flukes.

Amphistome metacercariae are present on vegetation from February to October with peak concentration in April/May and August/September (Fig. 1). From April to September/October, cattle are therefore ingesting metacercariae, leading to a build up of immature parasites in the small intestine. This would account for the low faecal egg production observed during the early to the middle of the dry season and also for reports of acute outbreaks of acute amphistomosis[74] in the country during the dry months. Development of amphistomes into adults takes 5 to 9 months[20] and the prepatent period is 56 days[27] to 89 days[20]. Hence 5 to 9 months after infection, the immature amphistomes become fully mature and this would account for high faecal egg production observed during the period from August to March (Fig. 1).

The prepatent period of *F. gigantica* varies between 9 and 12 weeks in susceptible young animals but may be longer in adult or previously exposed animals[26]. High snail populations observed during the dry season and peaking of faecal egg counts from August to March (Fig. 2) suggest that the infective stages are picked up around June/July and transmission occurs until to the month of December. Therefore, the incidence of the immature flukes is high during the dry period (Fig. 2) and this would account for low egg production observed during this period. In 9 to 12 weeks post-infection the flukes mature and this explains the high faecal egg count seen during the wet season (Fig. 2).

In contrast to the other 2 trematodes, transmission of *S. mattheei* occurs throughout the year, with peak transmission during the hot-dry season (September–November) (Fig. 3). Studies have shown that the prepatent period of *S. mattheei* is usually 6 to 7 weeks[30]. The observed peaking in faecal egg counts in cattle from October/November to March/April suggests that high exposure to the infective stages occur from around August/September to March (Fig. 3). This pattern of transmission would therefore explain the high faecal egg production and prevalence of schistosomosis observed during the wet season.

CONTROL

The epidemiological information on trematode parasites of cattle gathered in Zimbabwe can be used to design appropriate control measures. In principle, control should aim at the reduction of transmission rates. Several control methods, which include cultural, chemical, biological and immunological control, have been proposed[42].

Cultural and husbandry control

Cultural and husbandry control methods include practices such as controlling stocking rates, rotational grazing, and the provision of clean grazing[42]. The best way to prevent amphistomosis, fasciolosis and schistosomosis is to keep cattle away from potentially dangerous water habitats. Drainage or fencing-off of wet areas pre-

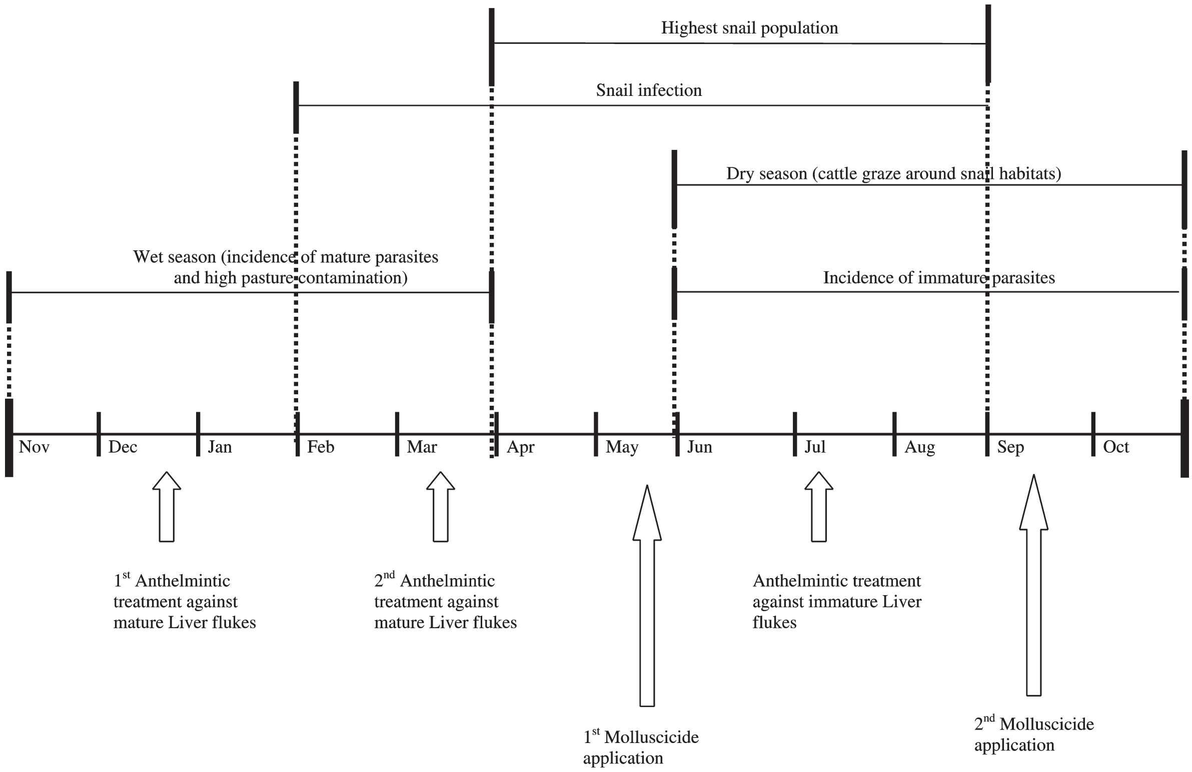

Fig. 2: **Seasonal occurrences related to the epidemiology and treatment of *Fasciola gigantica* in cattle in Zimbabwe.**

vents infection of pastures but is rarely cost effective on grazing land in developed countries and neither is it feasible in developing countries[61]. Complete separation of stock from snail-infested areas is only practical in intensive farming husbandry systems[45]. For instance, fencing snail-infested waters and supplying cercariae-free water in troughs controlled *S. mattheei* infections in cattle on a South African farm[54]. Hence cultural and husbandry control methods can be applicable in the commercial farming areas. In communal grazing areas animals are communally grazed and therefore practices such as rotational grazing and provision of clean pastures would not be feasible.

Habitat management in the form of vegetation clearance is potentially effective both through reducing feed availability of snails and also by enhancing water flow rates during the rainy season[79]. This method of control could probably be possible especially in commercial farming areas in the lowveld region where snail habitats are not widespread. Owing to widespread distribution of the snail habitats, cultural methods are difficult or perhaps impossible in the highveld region and in communal grazing areas.

Chemical control

Where the economy will withstand the expense of the use of often-costly drug treatment, chemotherapy remains the most widely used method of control[58]. During the rainy season, mature stages of the trematodes are expected and anthelmintic treatment of cattle with drugs effective against the mature stages of amphistomes and *F. gigantica* is indicated. Because animals are often infected with a wide range of helminths the need for broad-spectrum compounds active against trematodes, cestodes and nematodes and their larval stages is obvious.

A broad-spectrum anthelmintic administered strategically in December/January will control immature and adult forms of the parasitic nematodes and, mature forms of *F. gigantica.*(Fig. 2). Albendazole is highly active against all stages of parasitic nematodes and, is also active against tapeworms but it shows variable activity against liver flukes[58]. Oxyclozanide either alone or with levamisole shows reasonable activity against mature amphistomes (73–90 % efficacy) of cattle[58]. Its use would lie in the fact that it is also an efficient compound against mature *F. gigantica*[58]. Another anthelmintic treatment against adult amphistomes and *F. gigantica* should be given at the end of the rainy season (March/April) in order to reduce the opportunity for infection in snails (Figs 1, 2).

Praziquantel is the anti-schistosomal drug of choice[19,58]and is highly effective against all stages and also most trematodes and cestodes[58]. A combined treatment of praziquantel and artemether has been suggested as a strategy for transmission control[71]. Praziquantel and artemether are safe and efficacious anti-schistosomal drugs that act against adults and developmental stages, respectively[71]. Recent laboratory experiments with rabbits and hamsters infected with *S. japonicum* and *S. mansoni*, respectively, proved that a combined treatment with praziquantel and artemether at low doses is safe and more effective than praziquantel alone[71]. The 2 drugs can be administered in February/March to control acute schistosomosis in calves and to reduce pasture contamination (Fig. 3). However, the indiscriminate treatment of clinically affected animals is not recommended as it may produce more serious consequences than the disease itself[60]. Worms paralysed or killed by treatment move towards the liver where they may cause extensive thrombosis[35]. Schistosomes are relatively large worms (>10 mm) and the sudden accumulation of considerable numbers of them in the portal veins may cause occlusion and focal hepatic infection[35].

During mid- to the end of the dry period large burdens of immature *F. gigantica* and amphistomes are expected. Oxyclozanide shows reasonable activity (60–90 %) against immature amphistomes of cattle[58] and triclabendazole has been found to be effective against both immature and mature *F. gigantica*[70,75]. Oxyclozanide or Niclosamide and triclabendazole can be administered in July/August to treat against the immature amphistomes and immature liver flukes, respectively (Figs 1, 2).

However, in communal grazing areas it is imperative that the anthelmintic treatments mentioned above should be

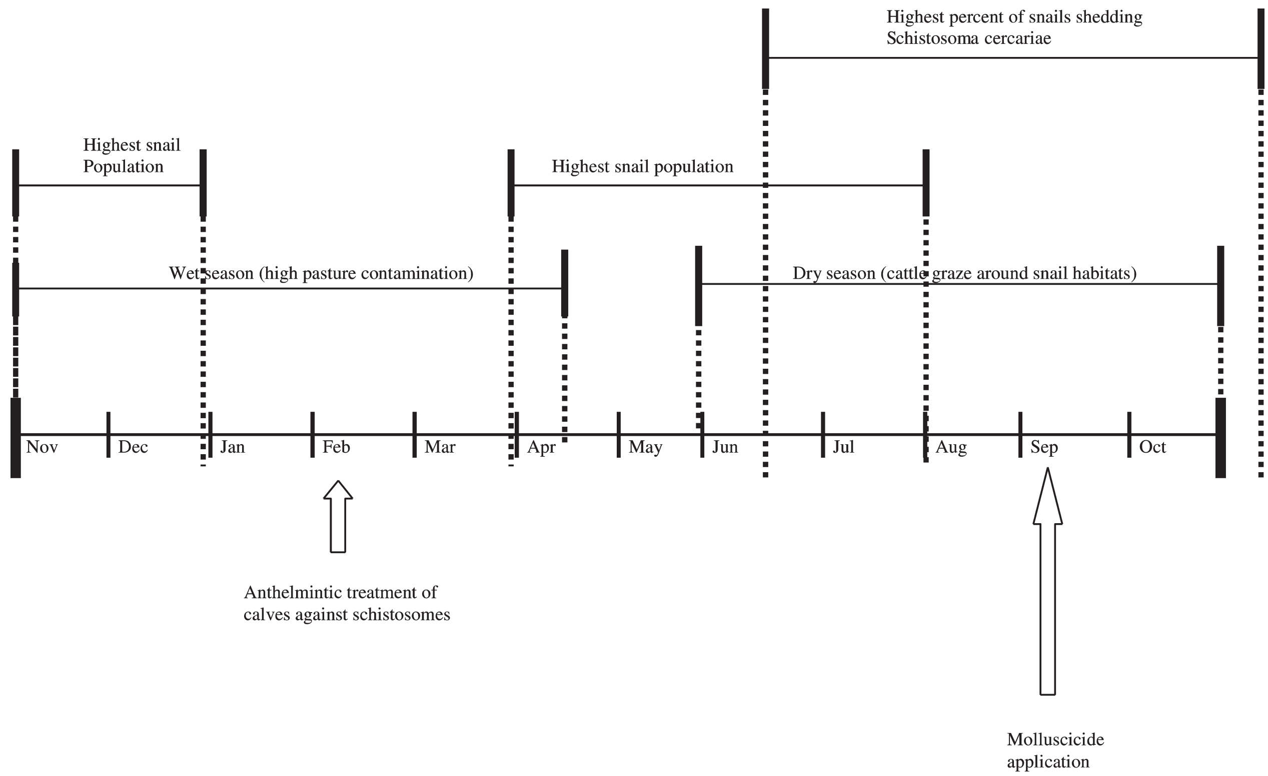

Fig. 3: **Seasonal occurrences related to the epidemiology and treatment of *Schistosoma mattheei* in cattle in Zimbabwe.**

village-based as cattle in communal areas are grazed together and there is no benefit for only a few farmers to carry out the recommended control measures. The anthelmintic treatment should be organised and preferably done at the same time within a village.

Molluscicides have been used successfully as a short-term control method of snail intermediate hosts and can be cost effective but have gained little acceptance[61]. The main problems being environmental pollution and killing of non-targeted aquatic organisms[61]. Also due to rapid recovery of the snail populations[4,79] during brief periods of favourable conditions, recolonisation should be expected and this may necessitate regular molluscicide application.

However, as already mentioned above, snail densities and transmission are seasonal in the country and measures to control snails only need to be applied when high densities of infected snails are expected. Hence, molluscicide application can be done in May/June and September/October (Figs 1, 2, 3). However, molluscicides have mainly been used or recommended for use in dams because the more extensive habitats such as rivers make the cost prohibitive[70]. Therefore, attempting to control the snails, using molluscicides, especially in the highveld region and communal grazing areas would prove to be difficult due to the widespread distribution of the snail habitats, the great biotic potential of the snails and the recurrent labour and equipment costs. Molluscicide application is probably practical in intensive farming husbandry systems especially in the lowveld region where snail habitats are not widespread.

Biological control

Two main types of biological control agents of snails have been studied and tested against the snail host of *Schistosoma* spp. in the Caribbean region[57]. Several species of competitor snails belonging to the Ampullariidae (*Pomacea glauca, Marisa cornuarietis*) and Thiaridae ((*Tarebia granifera, Melanoides tuberculata*) families have been tested with success in several types of habitats[57]. Competitor snails have also proven to be useful in preventing the recolonisation by the snail hosts after molluscicide treatments[57]. The introduction of *M. tuberculata* into transmission sites resulted in the interruption of transmission and the near total disappearance of the snail hosts[57]. However, *M. tuberculata* has been demonstrated naturally and experimentally to be a potential intermediate host of *C. microbothrium*[10]. In addition, through experimental and natural infection, *M. tuberculata* has been confirmed to be the intermediate host of *Philophthalmus gralli*, the 'oriental eye-fluke' of ostriches in Zimbabwe[39]. Therefore, in light of these observations and the fact that *M. tuberculata* is viviparous and reproduces parthenogenetically[4] and hence colonises new areas rapidly, its use in biological control needs to be approached with caution[10].

Predators may also be used to eliminate the free larval stages of the schistosomes[19]. Some trematode species such as *Echinostoma malayanum* can be used in the control of schistosomosis. These trematodes may not only interfere with the reproductive capacity of the intermediate snail host; they may also exert an antagonistic effect against the larval stages of the schistosomes inside the snails[19].

Free-ranging ducks or geese, which feed on snails, have also been proposed as a possible means for control of *F. gigantica*[59] but the degree at which control is likely to be achieved has not been measured. Effective control would require that ducks were present in sufficient numbers to feed on snails in a habitat before they shed cercariae, and this may be achievable along limited stretches of the shore of lakes and streams where stock drink.

Immunological control

Vaccines consisting of irradiated schistosomula have been shown to significantly protect cattle against schistosome infection[82]. An economic study in Sudan indicated that the development and production of such a vaccine would yield favourable returns from livestock production[34]. However, despite the fact that great potentials of irradiated vaccines have been known for more than 15 years, the vaccines have never been used on a large scale[19]. Live attenuated vaccines are difficult to produce, especially those against *S. japonicum* because of the relatively low cercarial production by each

Oncomelania snail[19]. In addition, the vaccines require cryopreservation and are not easy to apply in the field. The current research for schistosomosis control is to identify defined protective antigens that are easier to standardise and deliver than live attenuated vaccines[19].

In cattle, using irradiated metacercariae as the immunising vaccine, a 98 % reduction in *F. gigantica* fluke burdens in vaccinated calves has been reported[3]. Using a range of immunising regimes it has been shown that vaccination of Zebu calves with irradiated metacercariae reduced fluke burdens by 45–68 %[83]. The use of Glutathione S-transferase (GST) isolated from *F. gigantica* as a vaccine alone or in combination with either aluminium hydroxide or saponin in sheep against *F. gigantica* infection has also been evaluated[47]. The highest fluke reduction was observed in the group vaccinated with GST-saponin (32 %), but the reduction was not statistically significant in comparison with the control group.

The successful immunisation of sheep, goats and cattle against massive artificial infections with *C. microbothrium* has been reported[27]. The results indicated that cattle were the most suitable subjects for immunisation. Immunity in adult cattle was attained within 4 to 6 weeks after immunisation and the immunity was effective for at least a year post-immunisation. However, to immunise 100 cattle simultaneously with 40 000 metacercariae each would require 4 million metacercariae and these need to be produced within a period of 60 days because thereafter their viability decreases with age[27]. Hence, large-scale immunisation depends entirely on whether the considerable numbers of metacercariae required for immunisation could be produced[27].

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