Amphistome infections in domestic and wild ruminants in East and Southern Africa: A review

In this article, the main amphistome species infecting domestic and wild ruminants in East and Southern Africa, their snail intermediate hosts and epidemiological features are reviewed and discussed. Twenty-six amphistome species belonging to nine genera from three families occur in domestic and wild ruminants in the region under review and over 70% of them belong to the genera Calicophoron, Carmyerius and Cotylophoron. Of the amphistome species, 76.9% are shared between domestic and wild ruminant hosts – an important observation when considering the different options for control. Seven freshwater snail species belonging to four genera from two families act as intermediate hosts of the identified amphistome species, with the genus Bulinus contributing 57% of the snail species. Some of the snails are intermediate hosts of amphistome species belonging to the same genus or to different genera; a phenomenon not yet fully elucidated as some snails are reported to be naturally infected with amphistome cercariae of unidentified species. Only nine (34.6%, 9/26) of the amphistome species have known snail intermediate hosts, while most (65.4%, 17/26) have unknown hosts. Species of intermediate hosts and the potential of the flukes to infect these hosts, the biological potential of the snail hosts, the definitive hosts management systems and their grazing habits are considered to be the main factors influencing the epidemiology of amphistomosis. Based on the epidemiological features of amphistome infections, various practical control options are discussed. Further research is necessary to determine amphistome–snail associations, develop diagnostic tests that can detect prepatent infections in the definitive host, determine the burden and economic importance of amphistomosis in domestic and wild ruminants and the efficacy of different anthelmintics in the treatment of patent infections.

Introduction

Amphistomosis is a disease of domestic and wild ruminants caused by digenetic trematodes of the superfamily Paramphistomoidea Fischaeder, 1901 (Lotfy et al. 2010). The superfamily has a cosmopolitan distribution and is composed of hundreds of species belonging to 12 families (Jones 2005). Given their ubiquity and their abundance within hosts, it seems likely that the importance of these flukes is underestimated globally (Lotfy et al. 2010). Various species of the different paramphistomaid families, especially members of Paramphistomidae and Gastrothylacidae, cause amphistomosis among ruminants. The disease is caused by a severe infection with immature flukes in the small intestines of immunologically incompetent hosts. The amphistomes are responsible for lower nutrition conversion and result in a loss of weight and/or a decrease in milk production, causing great economic losses (Horak 1971). However, most reports on the disease do not quote the responsible amphistome species as they are difficult to identify from a systematic point of view (Horak 1971). Calicophoron microbothrium is probably the biggest cause of this disease in Africa (Dinnik 1964a). Knowledge of the different amphistome species infecting domestic and wild ruminants facilitates a better understanding of the amphistome–host associations and the epidemiology of the disease.

A wide range of gastropods belonging to the genera Bulinus Müller 1781, Biomphalaria Preston 1910, Ceratophallus Brown and Mandal-Barth 1973 and Galba Müller 1774 act as the intermediate hosts of amphistomes in Africa (Dinnik 1961, 1965; Dinnik & Dinnik 1954; Southgate et al. 1989; Wright, Southgate & Howard 1979). The prevalence of snail-borne diseases such as amphistomosis is influenced by both the abundance of infected definitive hosts and the abundance and efficiency of the snail intermediate hosts. Hence, the epidemiology and seasonal patterns of infection with amphistomes is determined to a large extent by the availability of the snail intermediate hosts and the grazing habits of the definitive hosts (Horak 1971; Rolfe et al. 1991). Information on the snail hosts of different amphistome species is essential as knowledge of the amphistome–snail associations has an influence on amphistomosis epidemiology and control.
In this review, to avoid confusion, genera of parasites and snail hosts have been abbreviated using the first three letters of the genus name and these include: for amphistomes – Bilatorchis (Bil.), Calicophoron (Cal.), Carmyerius (Car.), Choerocotyloides (Cho.), Cotyphophoron (Cot.), Gastrothylax (Gas.), Gigantocotyle (Gig.), Orthocoelium (Ort.) and Stephanopharynx (Ste.) and for snail hosts – Biomphalaria (Bio.), Bulinus (Bul.) and Ceratophallus (Cer.). The authorities of the digenean families and species and that of the snail species referred to in this review can be found in Table 1.

In this paper, we review the information available to date on amphistome species infecting domestic and wild ruminants in east and southern African countries, the snail intermediate hosts, as well as the epidemiology of amphistomosis and available control options.

**Amphistome species infecting ruminants in East and Southern Africa**

Reported amphistome species and their respective domestic and wild ruminant hosts in east and southern African countries are shown in Table 1. Data show that the documented species belong to four families: Choerocotyloides, Gastrothylacidae, Paramphistomidae and Stephanopharyngidae, and nine genera: one from Choerocotyloides (Choerocotyloides Prudhoe, Yeh & Khalil 1964), two from Gastrothylacidae (Carmyerius Stiles & Goldberger 1910 and Gastrothylax Poirier 1887), five from Paramphistomidae (Bilatorchis Fischoeder, 1901, Calicophoron Näsmark 1937, Cotyphophoron Stiles & Goldberger 1910, Gigantocotyle Näsmark 1937 and Orthocoelium Stiles &

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**TABLE 1:** Checklist of amphistome species and their ruminant and snail intermediate hosts reported in east and southern African countries.

| Family              | Species                          | Country reported | Domestic ruminant hosts | Wild ruminant hosts | Intermediate snail hosts | References                  |
|---------------------|----------------------------------|------------------|-------------------------|---------------------|--------------------------|-----------------------------|
| Paramphistomidae    | Bilatorchis papillogenitalis     | Zambia           | Cattle                  | Blue wildebeest     | Not yet known            | Eduardo 1980; Eduardo 1986; |
|                     | Eduardo 1980                     |                  |                         |                     |                          |                             |
|                     | Calicophoron bothriophoron      | Kenya, South Africa and Tanzania | Cattle (Bos primigenius Bojanus 1827), goats (Capra hircus Linnaeus 1758) and sheep (Ovis aries Linnaeus 1758) | Buffalo (Synurus caffer Sparrman 1779) and Waterbuck (Kobus ellipispyrum Ogilby 1833) | Not yet known | Dinik 1964a; Sey & Gruber 1979a; Eduardo 1983 |
|                     | (Braun 1892)                     |                  |                         |                     |                          |                             |
|                     | Eduardo 1983                     |                  |                         |                     |                          |                             |
|                     | Calicophoron calicophororum      | Angola, Kenya, Mozambique, South Africa, Zambia and Zimbabwe | Cattle, goats and sheep | Buffalo, Blue wildebeest | Buffalo tropicus Kraus 1848 | 6, Porter 1921, 7, Porter 1938; B Swart 1954; 9, Caeiro 1961; 10, Ortlip 1961; 11, Von Roth & Dalchow 1967; 12, Cruz e Silva 1971; 13, Anderson 1983; 14, Jooste 1987; 15, Jooste 1989; 16, Dube et al. 2004; 17, Dube et al. 2004; 18, Kock et al. 2002; 19, Dube & Tizauone 2014; 20, Sibula et al. 2014; |
|                     | Näsmark 1937                     |                  |                         |                     |                          |                             |
|                     | Eduardo 1983                     |                  |                         |                     |                          |                             |
|                     | Calicophoron clivialis           | Kenya, Tanzania, Uganda and Zimbabwe | Cattle, goats and sheep | Buffalo, Hartebeest, Impala and Sable antelope | Not yet known | Eduardo 1983; Jooste 1989; Dube et al. 2002; Dube et al. 2004; Dube & Tizauone 2014; Eduardo 1987; Madzingira et al. 2002; Dube et al. 2010; Laidemitt et al. 2017 |
|                     | (Näsmark 1937)                   |                  |                         |                     |                          |                             |
|                     | Eduardo 1983                     |                  |                         |                     |                          |                             |
|                     | Calicophoron douneymi            | Kenya             | Cattle                  | Bohor reedbuck (Redunca redunca Pallas 1767), Buffalo, Eland, Hartebeest, Impala, Kafue lechwe, Kudu, Mountain gazelle (Gazella gazella Pallas 1776), Nyala, Reedbuck (Redunca redunca Pallas 1767), Roan antelope, Sable antelope, Thomson's gazelle (Gazella thomsonii Günther 1864), Topi (Damaliscus lunatus jimela Matschie 1892), Tsessebe (Damaliscus lunatus Burchell 1824), Ugandan kob (Kobus kob thomsonii Schlett 1896), Waterbuck | Bulinus forskali Ehrenberg 1831 Bulinus globosus Morelet 1866 Bulinus nasutus von Martens 1879 Bul. tropicus | Dinkin 1964a; Eduardo 1983; Swart 1954; Caeiro 1961; Ortlip 1961; Von Roth & Dalchow 1967; Cruz e Silva 1971; Jooste 1989; Dube et al. 2002; Dube et al. 2004; Kock et al. 2002; Dube & Tizauone 2014; Eduardo 1987; Madzingira et al. 2002; Dube et al. 2010; Laidemitt et al. 2017; Dinkin & Dinkin 1954; Dinkin & Dinkin 1955; Dinkin & Dinkin 1962; Dinkin et al. 1962; Dinkin 1965; Roach & Lopes 1966; Horak 1967; Wandaera 1969; Fitzsimmons 1971; Keyyu et al. 2006; Loffy et al. 2010; Dube et al. 2015; |
|                     | (Dinnik 1962)                    |                  |                         |                     |                          |                             |
|                     | Eduardo 1983                     |                  |                         |                     |                          |                             |
|                     | Calicophoron microboreuthium     | Angola, Botswana, Kenya, Lesotho, Mozambique, South Africa, Tanzania, Uganda, Zambia and Zimbabwe | Cattle, goats and sheep | Bohor reedbuck (Redunca redunca Pallas 1767), Buffalo, Eland, Hartebeest, Impala, Kafue lechwe, Kudu, Mountain gazelle (Gazella gazella Pallas 1776), Nyala, Reedbuck (Redunca redunca Pallas 1767), Roan antelope, Sable antelope, Thomson's gazelle (Gazella thomsonii Günther 1864), Topi (Damaliscus lunatus jimela Matschie 1892), Tsessebe (Damaliscus lunatus Burchell 1824), Ugandan kob (Kobus kob thomsonii Schlett 1896), Waterbuck | Bulinus forskali Ehrenberg 1831 Bulinus globosus Morelet 1866 Bulinus nasutus von Martens 1879 Bul. tropicus | Dinkin 1964a; Eduardo 1983; Swart 1954; Caeiro 1961; Ortlip 1961; Von Roth & Dalchow 1967; Cruz e Silva 1971; Jooste 1989; Dube et al. 2002; Dube et al. 2004; Kock et al. 2002; Dube & Tizauone 2014; Eduardo 1987; Madzingira et al. 2002; Dube et al. 2010; Laidemitt et al. 2017; Dinkin & Dinkin 1954; Dinkin & Dinkin 1955; Dinkin & Dinkin 1962; Dinkin et al. 1962; Dinkin 1965; Roach & Lopes 1966; Horak 1967; Wandaera 1969; Fitzsimmons 1971; Keyyu et al. 2006; Loffy et al. 2010; Dube et al. 2015; |
|                     | (Fischoeder 1901)                |                  |                         |                     |                          |                             |
|                     | Eduardo 1983                     |                  |                         |                     |                          |                             |
|                     | Calicophoron phillipouei         | Kenya, South Africa, Tanzania, Uganda and Zimbabwe | Cattle, goats and sheep | Bohor reedbuck, Buffalo, Kudu, Impala, Puku (Kobus vardonii Livingstone 1857), Reeddubuck, Roan antelope, Sable antelope, Topi, Ugandan kob | Bul. forskali | Eduardo 1983; Von Roth & Dalchow 1967; Jooste 1987; 15, Jooste 1989; Dube et al. 2004; Eduardo 1987; Laidemitt et al. 2017; Dinkin et al. 1962; Dinkin 1961; Dinkin & Hammond 1968 |
### TABLE 1 (Continues...): Checklist of amphistome species and their ruminant and snail intermediate hosts reported in east and southern African countries.

| Family | Species | Country reported | Domestic ruminant hosts | Wild ruminant hosts | Intermediate snail hosts | References |
|--------|---------|------------------|--------------------------|---------------------|--------------------------|------------|
| Calicophoron raje | Näsmark 1937 | Botswana, Kenya, Namibia, South Africa, Tanzania, Zambia and Zimbabwe | Cattle, goats and sheep | Blue wildebeest, Buffalo, Bushbuck, Eland, Gemsbok, Kudu, Hartebeest, Impala, Kafue lechwe, Puku, Reddeback, Roan antelope, Sable antelope, Thomson’s gazelle, Topi, Tsessebe, Waterbuck | Bul. globosus | Dinnik 1964a; Eduardo 1983; Von Roth & Dalchow 1967; Jooste 1989; Dube et al. 2002; Dube et al. 2004; Dube & Tizauone 2014; Eduardo 1987; Madsingera et al. 2002; Dinnik & Dinnik 1954; Dinnik & Dinnik 1955; Dinnik & Hammond 1968; Mettrick 1962 |
| Calicophoron sukuri | (Dinnik 1954) Eduardo 1983 | Angola, Kenya, Tanzania, Uganda, Zambia and Zimbabwe | Cattle, sheep and goats | Buffalo | Biomphalaria pfefferi Krauss 1848 | Dinnik 1964a; Eduardo 1983; Von Roth & Dalchow 1967; Dube et al. 2004.; Eduardo 1987;Dinnik & Dinnik 1965; Dinnik & Hammond; Dinnik 1954; Dinnik & Dinnik 1957; Sachs & Sachs 1968 |
| Calicophoron sukumum | (Dinik 1964a) Eduardo 1983 | Tanzania, Zambia and Zimbabwe | Cattle | Buffalo, Blue wildebeest, Eland, Kafue lechwe, Reddeback, Roan antelope, Topi, Waterbuck | Not yet known | Eduardo 1983; Von Roth & Dalchow 1967; Jooste 1989; Sachs & Sachs; 1968; Dinnik 1964b |
| Cotylophoron cinctum | (Fisch. 1901) Stiles and Goldberger 1910 | Kenya, Malawi, Mozambique, South Africa, Tanzania, Uganda, Zambia and Zimbabwe | Cattle, goats and sheep | Blue wildebeest, Bohor reeddeback, Buffalo, Bushbuck, Common duiker (Sylvicapra grimmia Linnaeus 1758), Eland, Kudu, Hartebeest, Kafue lechwe, Nyala, Puku, Reddeback, Roan antelope, Sable antelope, Sittatunga (Trachelaphus speki Speke 1863), Tsessebe, Topi, Ugandan kob, Waterbuck | Not yet known | Ortlepp 1961; Von Roth & Dalchow 1967; Cruz e Silva 1971; Anderson 1983; Laidemitt et al. 2017; Dinnik et al. 1962; Mettrick 1962; Le Roux 1930b; Le Roux 1930b; Le Roux 1932; Mettman 1932; Fitzsimmons 1964; Bwangamoi 1968; Eduardo 1985a; |
| Cotylophoron jacksoni | Näsmark 1937 | Kenya, Tanzania, Uganda, Zambia and Zimbabwe | Cattle | Kudu, Hartebeest, Impala and Sable antelope | Not yet known | Anderson 1983;Dinik et al. 1962, Eduardo 1985a |
| Cotylophoron macrospynctris | Sey and Graber 1979b | Uganda – | Buffalo, Hartebeest and Oribi (Ourebia ourebi Zimmermann 1783) | Not yet known | Eduardo 1985a; Sey & Graber 1979b; Sey 1982 |
| Gigantocotyle simmenni | Näsmark 1937 | Botswana, South Africa, Zambia and Zimbabwe | Cattle | Buffalo, Hartebeest, Kafue lechwe, Kudu and Roan antelope | Not yet known | Sey & Graber 1979b Dube et al. 2002; Dube & Tizauone 2014; Sibula et al. 2014; Yeh 1957; Eduardo 1984; |
| Orthoecocelium scolicocelium | (Fischoder 1904) Yamaguti 1971 | Kenya | Cattle | – | Ceratophillus natalensis Krauss 1848 | Dinnik 1951; Dinnik 1956; Eduardo 1985b; |
| Gastrothylacidae | | | | | | |
| Stiles and Goldberger 1910 | | | | | | |
| Carminurus babalis | (Linnes 1812) Sturkard 1925 | Zimbabwe – | – | Hartebeest and Bongo (Tragelaphus eurycerus Ogilby 1837) | Not yet known | Sey 1983 |
| Carminurus dolius | Golvan Chabaud and Grelitiat 1957 | Botswana | Cattle | – | Not yet known | Dube et al. 2015 |
| Carminurus exopus | Mapleton 1923 | Kenya, Malawi, Tanzania and Zimbabwe | Cattle and sheep | Buffalo, Reddeback, Roan antelope, Sittatunga, Topi and Waterbuck | Cer. natalensis | Dinnik 1964a; Von Roth & Dalchow 1967; Jooste 1989; Laidemitt et al. 1977; Dinnik 1965; Sibula 1963; Prudhoe 1957; Dinnik & Dinnik 1960 |
| Carminurus gregarius | (Looss 1896) Stiles and Goldberger 1910 | Kenya and South Africa | Cattle | Buffalo, Bushbuck and Imababala (Trachelaphus scriptus sylvaticus (Pallas 1766) Sparman 1780) | Bulinus species | Laidemitt et al. 2017; Sey 2000 |
| Carminurus mancatus | (Fischoder, 1901) Stiles and Goldberger 1910 | Kenya and Tanzania | Cattle, goats and sheep | Bohor reeddeback, Buffalo, Bushbuck, Eland, Kafue lechwe, Roan antelope and Waterbuck | Cer. natalensis | Dinnik 1964a; Laidemitt et al. 2017;Dinnik 1965; Prudhoe 1957 |
| Carminurus porpypogillatus | Grelitiat 1962 | Kenya and Zambia | Cattle | Bushbuck, Sable antelope, Topi and Waterbuck | Bul. globosus | Dinnik 1965; Sey 1983 |
| Carminurus spatiosus | (Brandes 1898) Stiles and Goldberger 1910 | Kenya, Mozambique, South Africa, Tanzania, Zambia and Zimbabwe | Cattle, goats and sheep | Bohor reeddeback, Buffalo, Bushbuck, Kafue lechwe, Hartebeest, Reddeback, Roan antelope, Sable antelope, Topi and Waterbuck | Not yet known | Dinnik 1964a; Ortlepp 1961; Cruz e Silva 1971; Le Roux 1934; 63; Pike & Condy 1966 |
| Gastrothylacidae | | | | | | |
| | | | | | |
Goldberger 1910] Price & McIntosh 1953) and one from Stephanopharyngidae (Stephanopharynx Fisschoeder 1901).

Twenty-six species occur in domestic and wild ruminants in the area under review. Seventy-seven per cent of them (20/26) belong to Calicophoron, Carmyerius and Cotylyphoron with the genus Calicophoron accounting for approximately 35% of the species, followed by Carmyerius (27%) and Cotylyphoron (15%). Seventy-five per cent (9/12) of the known Calicophoron species and more than 40% of Carmyerius (43.8%, 7/16) and Cotylyphoron (57.1%, 4/7) species occur in the area under review. However, less than 40% of Gastrolyphax (33.3%, 1/3), Gigantocotyle (25%, 1/4) and Orthocoelium (9.1%, 1/11) known species occur in ruminants in east and southern Africa. Most of the Calicophoron species have a wider distribution with respect to countries where reported compared with species of the other genera. Calicophoron microbothrium has the widest distribution followed by Cot. cotylyphorum and Cal. raja. Calicophoron calcicophorum, Cal. phillerouxi, Cal. sukari, Cal. fuelleborni and Cal. spatiosus also have a wider distribution compared with the rest of the other species. Seven species; Bil. papillogenitalis, Cal. daubnegyi, Car. bubalis, Car. dollfusi, Cho. Onotragi, Ort. macrospinhinctris and Ort. scoliocoeleum had the narrowest distribution, being reported in only one country each.

The majority of the species (76.9%) are shared between domestic and wild ruminant hosts and approximately 12% of them have not been documented in domestic ruminants as yet (Bil. papillogenitalis, Car. bubalis and Cot. macrospinhinctris), while another 12% (3/26) have not been reported in wild ruminants (Cal. daubnegyi, Car. dollfusi and Ort. scoliocoeleum). Approximately 85% (22/26) are found in domestic and 88% (23/26) in wild ruminants. All the species recorded in domestic ruminants occur in cattle with Cal. daubnegyi, Car. dollfusi and Ort. scoliocoeleum documented in this ruminant host only. Sheep are hosts to 55% (12/22), while goats are hosts to 50% of the species reported in domestic ruminants. Half (11/22) of the species have been reported in all the domestic ruminants with most (63.6%, 7/11) of them being Calicophoron species. The range of wild ruminant hosts varies for the different species. Cotylyphoron cotylyphorum has the highest wild ruminant host range, 19 host species belonging to 10 genera followed by Cal. raja, 18 host species belonging to 12 genera, Cal. microbothrium, 17 host species belonging to 11 genera and Cal. spatiosus, 10 host species belonging to 10 genera. Calicophoron sukari has the lowest wild ruminant host range, with only one wild ruminant host, that is, the buffalo. Of the wild ruminant hosts, buffaloes are hosts to 78% (18/23) of the amphistome species recorded in wild ruminants followed by the waterbuck (52.2%, 12/23), the Kafue lechwe (47.8%, 11/23), the roan antelope (47.8%, 11/23) and the hartebeest (43.4%, 10/23). The blue wildebeest, bushbuck, eland, impala, kudu and sable antelope are also hosts to more than 25% of the amphistome species documented in wild ruminant hosts.

Mixed farming systems of cattle and game, particularly antelope, have become an important agricultural activity in most east and southern African countries. In addition, there has been the creation of Transfrontier Conservation Areas (TFCAs) involving many African countries, particularly in Southern Africa, resulting in increased livestock–wildlife interface areas. Therefore, domestic and wild animals are coming into ever more intimate contact in many interface areas, particularly in rural areas at the edges of the TFCAs and in farms practising mixed cattle and game farming, thus promoting the possibility of parasite exchange. These observations are important when considering the different options for their control. For instance, Phiri et al. (2011) observed that the host range of many helminths found in the Kafue lechwe is broad and they could serve as a potentially stable source of infection to domestic animals such as goats and cattle. Hence, issues concerning livestock management and conservation may arise.

### Snail intermediate hosts

Table 1 shows the reported intermediate snail hosts of different amphistome species recorded in the study areas under review. Data show that seven snail species – Bio. pfeifferi, Bul. forskalii, Bul. globosus, Bul. nasatus, Bul. tropicus and Cer. natalensis all Planorbidae Rafinesque 1815 and Gaiba truncatula belonging to Lymnaeidae Rafinesque 1815 – are so far confirmed intermediate hosts of identified amphistome species. The genus Bulinus contributes 57% (4/7) of the confirmed snail intermediate hosts, while the remaining genera contribute one species each. The data also show that some snail species are intermediate hosts of amphistome species belonging to the same genus, for example, Bul. forskalii (Cal. microbothrium and Cal. phillerouxi), Bul. tropicus (Cal. calicophorum and Cal. microbothrium) and Cer. natalensis (Car. exoporus and Car. mancupatus) or amphistome species belonging to different genera, for example, Bul. globosus (Cal. microbothrium and Car. parvippapillatus) and Cer. natalensis (Car. exoporus, Car. mancupatus and Ort. scoliocoeleum). The capacity of the various snail species to act as intermediate hosts for paramphistomoids has not been fully elucidated yet, as some of the snail hosts have been reported to be naturally infected with amphistome cercariae of unidentified species (Chingwena et al. 2002a; Dinnik 1961; Loker, Mayo & Gardner 1981; Lotfy et al. 2010; Mukaratirwa et al. 1998; Pfuenyji et al. 2005a; Wright et al. 1979). Adult amphistomes are difficult to identify using their anatomical and morphological features as they have thick robust bodies in which the internal organs are difficult to characterise (Jones 1990). As amphistomes in snail hosts have not been fully elucidated yet, as some of the snail hosts have been reported to be naturally infected with amphistome cercariae of unidentified species (Chingwena et al. 2002a; Dinnik 1961; Loker, Mayo & Gardner 1981; Lotfy et al. 2010; Mukaratirwa et al. 1998; Pfuenyji et al. 2005a; Wright et al. 1979). Adult amphistomes are difficult to identify using their anatomical and morphological features as they have thick robust bodies in which the internal organs are difficult to characterise (Jones 1990). As amphistomes in snail hosts have not been fully elucidated yet, as some of the snail hosts have been reported to be naturally infected with amphistome cercariae of unidentified species (Chingwena et al. 2002a; Dinnik 1961; Loker, Mayo & Gardner 1981; Lotfy et al. 2010; Mukaratirwa et al. 1998; Pfuenyji et al. 2005a; Wright et al. 1979).
Car. mancupatus, Car. parvipapillatus and Ort. scoliocoleium) have known snail hosts. Except for Cal. microbothrium, presently with four known snail hosts (Bul. forskalii, Bul. globosus, Bul. nasutus and Bul. tropicus) in East and Southern Africa, all the other species have one known snail host each. In addition, Bio. pfeifferi and Melanoides tuberculata are known experimentally to serve as snail hosts of this parasite (Chingwena et al. 2002b). Calicophoron microbothrium is widely distributed in the areas under review and the wide range of its snail hosts probably supports its reported broad geographical distribution.

Data under review show that most of the known amphistome species have unknown snail hosts (65.4%, 17/26). The snail hosts of four Calicophoron species (Cal.bothriophoron, Cal. clavula, Cal. raja and Cal. sukumum) are currently not known. In Tanzania, amphistome cercariae of unidentified Calicophoron species were recorded in Bul. forskalii (Lotfy et al. 2010). Lotfy et al. (2010) suggested that besides Cal. phillerouxi, which is known from Bul. forskalii, two other Calicophoron species known in Tanzania, Cal. bothriophoron and Cal. sukumum with unknown snail hosts, cannot be ruled out. In the East African region, Bul. abyssinicus is reported as the intermediate host of Cal. clavula in Somalia (Sobrero 1962). Dinnik and Hammond (1968) suggested Bul. globosus as a likely snail host of Cal. raja as it is experimentally proven to be susceptible to infection. Four Carmyerius species with presently unknown snail hosts are Car. bubalis, Car. dolphis, Car. gregarious and Car. spatiatus. Amphistome cercariae belonging to an unidentified species of the family Gastrothylacidae were recorded from Cer. natalensis in Kenya (Lotfy et al. 2010). The genus Carmyerius is one of four genera belonging to the family Gastrothylacidae. Hence, besides Car. Mancupatus and Car. exoporus already known from this snail host, one other Carmyerius species known in Kenya, Car. spatiatus with unknown snail hosts, cannot be ruled out (Lotfy et al. 2010). Besides Cer. natalensis, Wright et al. (1979) also suggested Bul. forskalii as likely snail hosts of Car. spatiatus in Zambia. To date, all four Cotylophoron species have unknown snail hosts. The other species with unknown snail hosts are Bul. papillogenitalis, Cho. onotragi, Gas. crumenifer, Gig. symmeri and Ste. compactus. However, even though not yet confirmed, Bul. forskalii has been speculated to act as the intermediate host of Ste. compactus (Dinnik 1965).

Epidemiological features of amphistome infections in ruminants in East and Southern Africa

The epidemiology and prevalence of amphistomosis depend on several factors. These include the species of definitive and intermediate hosts (Rolfe et al. 1991), the potential of the flukes to infect these hosts (Dinnik 1964a; Dinnik & Dinnik 1954; Horak 1967), the topography and biological potential of the snail hosts (Dinnik 1964a; Horak 1971; Rolfe et al. 1991; Swart & Reinecke 1962a, 1962b), the definitive hosts’ management systems and their grazing habits as well as climate (Rolfe et al. 1991).

Data on amphistome infection prevalence are scarce for the reviewed countries and are currently only available from six countries (Table 2). The prevalence data are based on coprology and fluke counts with most studies having conducted in cattle. Because of difficulties in amphistome species identification, specific species prevalence data are lacking. Prevalence studies are limited for goats, sheep and wild ruminants. The available data show a high prevalence in the Kafue lechwe, but low prevalence rate in goats and sheep. In cattle, the coprological prevalence varies from 23.7% to 86.5%, while it varies from 25.5% to 96% on fluke counts, the high prevalence perhaps being explained by the fact that amphistome infection in ruminants is commonly because of several species. In the highlands of Kenya, Cal. microbothrium, Cal. daubneyi and Cal. jacksoni were recovered from a single animal and in few cases Cal. sukari, Car. exoporus and Car. mancupatus were present as well (Dinnik 1964a). Another combination of six species (Cal. microbothrium, Cal. phillerouxi, Cal. raja, Cot. cotylophorum, Cal. parvipapillatus and Ste. compactus) was found in an ox in Zambia (Dinnik 1964a). Amphistomes recovered in slaughtered cattle were a combination of Cal. microbothrium and Cot. jacksoni in Tanzania (Keyyu et al. 2006). Infections with different amphistome species are also reported in Zimbabwe in cattle (Dube et al. 2004; Dube & Tizauone 2014) and in sheep and goats (Dube, Masangani & Dube 2010). In addition, most amphistome species (85%) are shared between domestic and wild ruminant hosts (Table 1), providing a potentially stable source of infection among the ruminant animals. Furthermore, the availability of a wide range of the snail hosts (Table 1) with high biological potential also increases the successful propagation of amphistomes in the environment leading to increased infection exposure in ruminants. Limited routine anthelmintic treatment, particularly in rural communities who practice communal grazing, and the lack of effective drugs against amphistomes are also possible explanations for the high prevalence in domestic ruminants. An increase in the prevalence of amphistome infections has been reported in Western Europe (Foster et al. 2008; Mage et al. 2002; Murphy et al. 2008; Toolan et al. 2015). Besides an improvement in quality of diagnosis, the increase has also been attributed to the absence of an effective anthelmintic against amphistome infections (Mage et al. 2002).

Studies on animal-breed predisposition to amphistome infection are limited. Indigenous cattle breeds were observed to have a significantly higher prevalence and intensity than the exotic breeds and crosses in Kenya and Uganda (Howell 2011; Kanyari, Kagira & Mhoma 2010). In Tanzania, the Maasai Zebu cattle had a significantly higher prevalence than the Iringa Red cattle; however, the numbers of animals involved were too small for any meaningful interpretations to be made (Nzalawae et al. 2015). Literature reports a variable effect of sex in domestic ruminants. Despite females tending to record higher prevalences than males, the associations were not significant (Kanyari et al. 2009, 2010; Keyyu et al. 2006; Phiri, Chota & Phiri 2007a; Phiri, Phiri & Monrad 2006). However, Pfukenyi et al. (2005a) observed significantly higher prevalences in pregnant and lactating
TABLE 2: Prevalence of amphistomes in ruminants in east and southern African countries based on faecal egg and fluke counts.

| Host         | Location     | Total examined | Positive | Prevalence (%) | 95% CI | Publication year | References               |
|--------------|--------------|----------------|----------|---------------|--------|------------------|--------------------------|
| Cattle       | Tanzania     | 450            | 283      | 62.9          | 58.2–67.3 | 2015            | Nzalawahe et al. 2015  |
| Cattle       | Tanzania     | 241            | 90       | 37.3          | 31.3–43.8 | 2014            | Nzalawahe et al. 2014  |
| Cattle       | Uganda       | 233            | 158      | 67.8          | 61.3–73.7 | 2011            | Howell 2011             |
| Cattle       | Kenya        | 344            | 108      | 31.4          | 26.6–36.6 | 2010            | Kanyari et al. 2010    |
| Cattle       | Zambia       | 50             | 38       | 76.0          | 61.5–85.6 | 2008            | Yabe et al. 2008       |
| Cattle       | Zambia       | 268            | 96       | 35.8          | 30.1–41.9 | 2007            | Phiri et al. 2007a     |
| Cattle       | Zambia       | 101            | 33       | 32.7          | 23.9–42.8 | 2007            | Phiri et al. 2007b     |
| Cattle       | Tanzania     | 482            | 302      | 62.7          | 58.2–67.0 | 2006            | Keyyu et al. 2006      |
| Cattle       | Zambia       | 709            | 366      | 51.6          | 47.9–55.4 | 2006            | Phiri et al. 2006      |
| Cattle       | Tanzania     | 301            | 168      | 55.8          | 50.0–61.5 | 2005            | Keyyu et al. 2005      |
| Cattle       | Zimbabwe     | 16 264         | 4790     | 29.5          | 28.8–30.2 | 2005            | Pfukenyi et al. 2005a  |
| Cattle       | Zimbabwe     | 12 472         | 6697     | 53.7          | 52.8–54.6 | 1999            | Vassilev 1999          |
| Cattle       | Zimbabwe     | 796            | 490      | 61.6          | 58.1–64.9 | 1994            | Vassilev 1994          |
| Cattle       | Kenya        | 1878           | 481      | 25.6          | 23.7–27.7 | 1993            | Waruiru et al. 1993    |

**Total**

|            |              | **34 589**     | **14 100** | **40.8**      | **40.3–41.3** |

| Host         | Location     | Total examined | Positive | Prevalence (%) | 95% CI | Publication year | References               |
|--------------|--------------|----------------|----------|---------------|--------|------------------|--------------------------|
| Cattle       | Uganda       | 32             | 27       | 84.4          | 66.5–94.1 | 2011            | Howell 2011             |
| Cattle       | Zambia       | 50             | 48       | 96.0          | 85.1–99.3 | 2008            | Yabe et al. 2008        |
| Cattle       | Zimbabwe     | 3225           | 822      | 25.5          | 24.0–27.0 | 2004            | Dube et al. 2004        |
| Cattle       | Zimbabwe     | 1377           | 429      | 31.2          | 28.7–33.7 | 2002            | Dube et al. 2002        |

**Total**

|            |              | **4684**       | **1326**  | **28.3**      | **27.0–29.6** |

| Host         | Location     | Total examined | Positive | Prevalence (%) | 95% CI | Publication year | References               |
|--------------|--------------|----------------|----------|---------------|--------|------------------|--------------------------|
| Goats        | Kenya        | 33             | 4        | 12.1          | 4.0–29.1 | 2009            | Kanyari et al. 2009     |
| Sheep        | Kenya        | 54             | 16       | 29.6          | 18.4–43.8 | 2009            | Kanyari et al. 2009     |
| Buffalo      | Uganda       | 10             | 6        | 60.0          | 27.4–86.3 | 2011            | Howell 2011             |
| Kafue lechwe | Zambia       | 22             | 11       | 50.0          | 28.8–71.2 | 2002            | Kock et al. 2002        |
| Cattle       | Uganda       | 32             | 27       | 84.4          | 66.5–94.1 | 2011            | Howell 2011             |
| Cattle       | Zambia       | 50             | 48       | 96.0          | 85.1–99.3 | 2008            | Yabe et al. 2008        |
| Cattle       | Zimbabwe     | 3225           | 822      | 25.5          | 24.0–27.0 | 2004            | Dube et al. 2004        |
| Cattle       | Zimbabwe     | 1377           | 429      | 31.2          | 28.7–33.7 | 2002            | Dube et al. 2002        |

**Total**

|            |              | **4684**       | **1326**  | **28.3**      | **27.0–29.6** |

| Host         | Location     | Total examined | Positive | Prevalence (%) | 95% CI | Publication year | References               |
|--------------|--------------|----------------|----------|---------------|--------|------------------|--------------------------|
| Goats        | Zimbabwe     | 3000           | 60       | 2.0           | 1.5–2.6  | 2010            | Dube et al. 2010        |
| Sheep        | Zimbabwe     | 1000           | 60       | 6.0           | 4.7–7.7  | 2010            | Dube et al. 2010        |
| Kafue lechwe | Zambia       | 8              | 7        | 87.5          | 46.7–99.3 | 2012            | Munang’andu et al. 2012 |
| Wildebeest   | Zambia       | 6              | 4        | 66.7          | 24.1–94.0 | 2012            | Munang’andu et al. 2012 |
| Kafue lechwe | Zambia       | 65             | 65       | 100.0         | 93.1–99.9 | 2011            | Phiri et al. 2011      |
| Kafue lechwe | Zambia       | 40             | 40       | 100.0         | 89.1–99.8 | 2010            | Munyeme et al. 2010    |
| Impala       | South Africa | 46             | 41       | 89.1          | 75.6–95.9 | 1983            | Anderson 1983          |

Cows compared with bulls, oxen and dry cows. Similarly, Howell (2011) reported a significantly higher prevalence in female cattle compared with males. The differences between sexes could probably be related to grazing patterns, sex hormones and treatment regimes.

Adult domestic ruminants are reported to have a significantly higher prevalence compared with young animals (Howell 2011; Kanyari et al. 2009, 2010; Keyyu et al. 2005, 2006; Nzalawahe et al. 2014; Pfukenyi et al. 2005a; Phiri et al. 2007a; Vassilev 1999). This is attributed to a long exposure time in adults leading to immunity against the pathogenic effects of immature amphistomes but still having the mature ones maintaining their high egg production capacity (Horak 1971). The resistance to amphistome re-infection in cattle was demonstrated clinically (Horak 1967) with no simultaneous studies on the cellular effector systems that characterise the acquired resistance. Mavenyengwa et al. (2008) showed that the resistance to Cal. microbothrium re-infection in cattle involves eosinophils and mast cells that are targeted at immature flukes. Epidemiologically, adults act as a constant source of infection for successive generations of snail hosts. In rural areas, the grazing management of young and adult animals may differ, where young animals graze around farms or homesteads while adults are trekked long distances to valleys, flood plains or swampy areas where they are exposed to high metacercariae-contaminated pastures (Keyyu et al. 2005, 2006).

Animal grazing area and/or habitat is significantly associated with prevalence and intensity of amphistomes in domestic ruminants (Howell 2011; Kanyari et al. 2009, 2010; Keyyu et al. 2005, 2006; Nzalawahe et al. 2014, 2015; Pfukenyi et al. 2005a; Phiri et al. 2006). The prevalence is highest in animals grazing in areas characterised by wetlands or swampy or marshy grazing areas where the distribution of suitable snail habitats is widespread. For instance, the highveld region in Zimbabwe, characterised by wet/swampy grazing areas where distribution of snail habitats is widespread, is associated with a higher prevalence compared with the lowveld which is characterised by dry land grazing with a focal distribution of snail habitats (Pfukenyi et al. 2005a, 2005b).
Similarly, the presence of wetlands and high livestock density in the cattle grazing areas of the Zambian western and southern provinces is associated with an increased risk of acquiring amphistome infections (Phiri et al. 2006) and the same observations have been reported in Kenya (Kanyari et al. 2009, 2010), Tanzania (Keyyu et al. 2005, 2006; Nzalawahe et al. 2015) and Uganda (Howell 2011). In Tanzania, traditional communal grazing areas exhibited the highest prevalence of amphistomes compared with other sectors and this is attributed to heavy contamination of the habitats with eggs where intermediate host snails breed, because of high stocking densities with subsequent heavy metacercarial density on vegetation grazed by the animals especially during the dry season (Keyyu et al. 2005, 2006). Villages practising irrigation of crops are associated with high amphistome infection rates in Tanzania (Nzalawahe et al. 2014) as this provides favourable ecological conditions for growth of snail hosts and development of trematode larval stages.

The prevalence in domestic ruminants as measured by coprology follows a seasonal pattern with an increase towards the end of the dry season and a peak during the wet months of the year (Keyyu et al. 2005; Pfukenyi et al. 2005a; Phiri et al. 2007a; Reinecke 1983; Vatta & Krecke 2002). Outbreaks of acute clinical amphistomosis because of immature flukes are usually confined to the drier months of the year (Boray 1969; Butler & Yeoman 1962; Dinnik 1964a; Horak 1967, 1971; Rolfe et al. 1991; Vassilev 1999). Towards the end of the rainy season and onset of the dry season, conditions in permanent water sources become favourable for an increase in the number of the snail intermediate hosts, reaching their peak during the mid-to-end of the dry season (Chingwena et al. 2002a; Pfukenyi et al. 2005a, 2005b; Phiri et al. 2007b). As the snail hosts are extremely adaptable and prolific breeders, this ensures their widespread availability as well as heavy shedding of cercariae which encyst on vegetation surrounding the habitats (Dinnik 1964a). The proportion of infected snails increases from the end of the rainy season into the dry season (Chingwena et al. 2002a; Pfukenyi et al. 2005a). A combination of high snail numbers, asexual multiplication of the fluke in infected snails and survival of snails in suitable environments for several months may result in shedding of large numbers of cercariae. During this period, the infective metacercariae are spread over pastures surrounding permanent water sources where they can survive for several months. This coincides with pasture areas being narrowed around permanent water sources or wetland environments where animal concentration becomes high (Pfukenyi et al. 2005b). Contamination rates of these areas are increased, resulting in more snails being infected and high numbers of metacercariae on surrounding herbage, leading in turn to acute infections of animals with amphistomes. Thus, a build-up of immature flukes occurs, accounting for clinical amphistomosis outbreaks and low amphistome prevalence as measured by coprology. The outbreaks are common in ruminants that graze in marshy or swampy areas and are usually confined to the dry season.

However, on irrigated pastures, moisture is often adequate for the survival of snail hosts and metacercariae, and hence outbreaks can occur throughout the year. In Tanzania, some villages practising year-round zero-grazing had high levels of amphistome infections attributed to the acquisition of cattle fodder from irrigation canals and swamps contaminated with metacercariae (Nzalawahe et al. 2014).

Development of amphistomes into adults takes 5–9 months (Dinnik & Dinnik 1962) and the prepatent period is 56–89 days (Dinnik & Dinnik 1962; Horak 1971). Five to 9 months after infection, the immature flukes become fully mature and this would lead to high faecal egg production and thus, account for the high prevalence during the rainy season as measured by coprology. During this period, abundant grazing and alternative water sources are available. Hence, drinking from and grazing around infected permanent water sources is greatly reduced. Furthermore, snail habitats and pastures are constantly flooded, and thus snails and the parasitic free-living stages are regularly flushed (Pfukenyi et al. 2005b). In summary, the intermediate and definitive hosts acquire most of the infection during the beginning and/or middle of the dry season. This results in immature fluke infections and clinical amphistomosis during the dry season and patent (mature fluke) infections during the wet months and at the end of the dry season. However, the timing may vary depending on location, length of the rainy season and the grazing habits of the animals (Pfukenyi et al. 2005b).

**Impact on production**

Adult flukes are not associated with clinical amphistomosis (Mavenyengwa, Mukaratirwa & Monrad 2010). However, in heavy infections they have been hypothesised to cause weakness, recurrent ruminal tympany, ruminal atony, weight loss, anaemia and production losses (Anuracpreeda, Wanichanon & Sobhon 2008). They are also reported to be associated with inflammation of the mucosa and mucoid diarrhoea (Rolfe & Boray 1993). Based on coprology, poor body condition is reported to be significantly associated with high amphistome prevalence in cattle (Kanyari et al. 2010). A similar observation was noted in small ruminants (Kanyari et al. 2009), but the association was not significant. Cattle infected with more than 500 adult amphistomes had a significant reduction in final carcass mass when compared with controls (Marchand 1984; Dube & Tizauone 2014). The concurrent infection of amphistomes with other parasites known to depress growth rate such as strongyles (Kanyari et al. 2010), *Fasciola* species (Kanyari et al. 2010; Keyyu et al. 2006; Nzalawahe et al. 2014; Phiri et al. 2006; Yabe et al. 2008) and *Moniezia* species (Kanyari et al. 2010) is a likely explanation of the significant association between poor body condition and amphistome infections. However, further studies on the effect of adult amphistomes on production are required.

Clinical amphistomosis is caused by the immature flukes that lodge in the first 3 m of the small intestine (Mavenyengwa et al. 2010). The occurrence of clinical amphistomosis and
subsequent clinical pathology in ruminants is dependent on the dose, pathogenicity of the species and the level of establishment of the metacercariae in the host’s small intestine (Horak 1967; Mavenyengwa et al. 2010). In ruminants, the disease is characterised by anorexia, anaemia, submandibular oedema, and hypoproteinaemia, foul-smelling fetid diarrhoea, general weakness, polydipsia, and a reduction in feed conversion, weight and milk production and mortality in young animals (Boray 1969; Horak 1966; Mavenyengwa et al. 2010; Mohan 2011; Pillai & Alikutty 1995; Rolfe, Boray & Collins 1994; Spencer, Fraser & Chang 1996). Together with gastrointestinal nematodes, amphistome infection in cows can reduce milk production by approximately 0.4 L/day – 3 L/day (Mohan 2011; Spencer et al. 1996). Anthelmintic treatment of dairy cows infected with gastrointestinal nematodes (oxfendazole) and amphistomes (oxyclozanide) resulted in a significant increase in milk production, averaging 0.4 L/day (Spencer et al. 1996). The reduction in milk yield during clinical amphistomosis is associated with fetid diarrhoea (Mohan 2011). Mohan (2011) also reported anoestrus during clinical amphistomosis, while a functional obstruction or paralytic ileus of the intestine because of severe amphistomosis was reported in a cow (Yogeshpriya et al. 2011). Despite their ubiquity and abundance, as well as an increase in their prevalence in domestic ruminants, the economic importance of amphistome infections is not yet fully known and is likely to be underestimated in eastern and southern Africa – an area which requires further studies.

Diagnosis

Diagnosis of amphistomes in live animals is still dependent on faecal detection of eggs (Rieu et al. 2007) and this method only detects the presence of adult rumen fluke infection (Malrait et al. 2015). The filtration technique with sieves and sedimentation is the most accurate method to identify eggs in faeces (Horak 1971). Using contrast stains such as methylene blue or methyl green to distinguish amphistome eggs from Fasciola ova is advisable. One drawback of this diagnostic method is that, in acute infections, it is highly probable not to find eggs or only very few as this is usually associated with massive infection with immature flukes (Horak 1971). The agreement between a modified McMaster method and necroscopic diagnosis of amphistome infection is reported to be high (Rieu et al. 2007) with no significant differences being observed between the two methods. The modified McMaster method showed a significant association between eggs per gram (epg) counts and parasite burden; more than 100 epg indicated the presence of more than 100 adult amphistomes in the rumen and/or reticulum (Rieu et al. 2007). Similarly, the mini-FLOTAC is a reliable method of assessing the presence of adult amphistome infection with both sensitivity and specificity being above 0.9 (Malrait et al. 2015). A good correlation was found between faecal egg count (FEC) and estimated rumen fluke burden with a FEC > 200, indicating the presence of more than 200 adult rumen flukes in the rumen and/or reticulum (Malrait et al. 2015). The adult worms are difficult to identify to species level because most have thick robust bodies in which the internal organs are difficult to see. Even by using histological techniques, species identification is still problematic (Lotfy et al. 2010). As the flukes responsible for disease are sexually immature, specific identification is made even more difficult and the diagnosis has to rely on the dubious procedure of identifying a few adult worms, which may be present in the rumen of the animal (Horak 1971). Because of these problems, PCR-based techniques providing rDNA ITS2 sequences have proven to be reliable tools to identify amphistome species and to determine their phylogenetic relationships (Itagaki et al. 2003; Rinaldi et al. 2005). Using cercariae and rediae from snail hosts and adult flukes obtained from slaughterhouses, Lotfy et al. (2010) confirmed ITS2 as a good molecular marker for amphistome identification that can also be used to determine phylogenetic and amphistome–snail associations.

The clinical diagnosis of amphistomosis remains challenging as immunological techniques are usually not conclusive (Horak 1967, 1971). Cropsoscopic examination cannot be used for the early diagnosis of clinical amphistomosis which is vital for prompt treatment before considerable damages and economic losses are incurred. For the identification of immature flukes, the recommended method is to mix approximately 10 g of faeces with 100 mL – 200 mL of water (Horak 1971). The mixture is allowed to stand for 5 min, followed by decanting any supernatant fluid and then repeating the procedure four to five times. Young flukes, resembling small white or pink rice grains, will be seen after pouring the sediment on a black surface for examination (Horak 1971). In dead animals, postmortem, pathological and clinical pathological findings combined with the presence of immature flukes in the affected intestines would be confirmative. The gross pathological, histopathological and clinical pathological findings are as described in the literature (Horak 1966, 1967, 1971; Horak & Clarke 1963; Mavenyengwa et al. 2005, 2008, 2010; Pillai & Alikutty 1995). An indirect ELISA performed to detect coproantigens in faecal supernatants of 100 cattle known to be infected with Gas. crumenifer had a sensitivity of 74% (Kandasamy & Devada 2011). Generally, the sensitivity of the indirect ELISA ranges from 74% to 86% and its specificity from 79% to 90% (Hassan et al. 2005; Kandasamy & Devada 2011; Salib et al. 2015; Sanchis et al. 2012; Shivjot et al. 2009). However, Shivjot et al. (2009) reported a very low specificity of 23.7%. The ELISA was shown to be more specific and accurate but less sensitive than Western blotting for the diagnosis of amphistome infections in cattle and buffaloes (Salib et al. 2015). Results indicate the feasibility of ELISA for the detection of coproantigens of amphistome infections, especially for the diagnosis of immature amphistomosis where faecal examination may not reveal eggs.

Control

The available epidemiological information on amphistomes of ruminants in the area under review can be used to design appropriate integrated control measures. Options available for the control of amphistome infections are mainly based on
chemical treatment, non-chemical management practices and immunological control.

Chemical treatment

Chemical control involves treatment with a product that is effective against both adult and immature flukes. Oxyclozanide given twice, three days apart, has a high efficacy against both adult and juvenile amphistomes (Rolfe & Boray 1987) and a high anthelmintic performance in cattle (Arias et al. 2013; Rolfe & Boray 1987; Spencer et al. 1996) and small ruminants (Paraud et al. 2009; Rolfe & Boray 1988; Sanabria et al. 2014). Studies in Tanzania showed a reduced efficacy of levamisole–oxyclozanide combination against amphistomes in cattle (Keyyu et al. 2008) and this is of great concern as they are the commonly available drugs in the country. However, levamisole is widely used to treat nematode infections in livestock and it is not intended as treatment against trematodes. When given orally at a higher dosage (10 mg/kg), closantel has a high efficacy against mature flukes (Arias et al. 2013). However, treatment of mature flukes with intra-ruminally (Rolfe & Boray 1993) or subcutaneously administered (Malrait et al. 2015) closantel is not effective. In countries where oxyclozanide is unavailable, the use of closantel to treat against mature flukes is recommended. When administered at high doses (50 mg/kg and 100 mg/kg), niclosamide has 94% – 99% efficacy against immature amphistomes (Rolfe & Boray 1987).

Even though it is not of direct benefit to the animal, treatment against mature amphistomes will prevent egg laying and thus reduce pasture contamination (Horak 1971), while treatment against the immature flukes will reduce the impact of the disease. During the rainy season, mature amphistomes are expected and anthelmintic treatment with drugs effective against adult flukes is indicated. The strategic anthelmintic treatment against mature amphistomes should be given in adult animals at the end of the rainy season (Pfukenyi et al. 2005a, 2005b) or beginning of the dry season (Keyyu et al. 2005) to reduce the opportunity for snail infections. The timing of this treatment is dependent on local factors, length of the rainy season and the grazing habits of the animals. Where possible, adult animals targeted for treatment should have high levels of infection based on coprology. Depending on availability, oxyclozanide or closantel can be administered during this period to treat against mature amphistomes.

Disease epidemiology indicates that large burdens of immature amphistomes are expected during the dry season. As adult animals are resistant to the pathogenic effects of the migrating immature amphistomes, the target for treatment would be young animals being exposed to the infection for the first time (Pfukenyi et al. 2005a). Hence, the first anthelmintic treatment can be administered in young animals during the mid-dry season period when maximum migration of immature amphistomes starting 3–4 weeks after infection in the early dry season is expected. To remove potentially high burdens of immature amphistomes acquired later in the dry season, a second treatment could be given towards the end of the dry season (Pfukenyi et al. 2005a). Oxyclozanide or niclosamide can be administered during this period to treat against immature amphistomes. In communal areas, animals are communally grazed and for optimum benefits, the recommended anthelmintic treatments should be well organised and preferably done at the same time within a village. Where cattle are dipped for the control of ticks, dip tank facilities where all animals are gathered during dipping sessions could be used for organised fluke control (Pfukenyi et al. 2005a, 2005b).

The efficacy of medicinal plant extracts against amphistomes has recently been evaluated. The ethanol extract of Punica granatum L. (Lythraceae), commonly known as pomegranate, is highly effective against amphistomes in naturally infected sheep (Lalhmingchhuannawii, Veerakumari & Raman 2014). The authors concluded that the plant extract could be successfully used as an anthelmintic to treat amphistomes in domestic ruminants. Similarly, an aqueous extract of Acacia concinna (Willd.) DC. (Fabaceae) significantly reduced egg counts of amphistomes in naturally infected sheep and also restored the haematobiochemical profile to normal in extract-treated sheep (Priya, Veerakumari & Raman 2013). However, efficacy of the P. granatum and A. concinna extracts was not established in immature amphistomes. Other studies have also shown medicinal plants extracts to be effective against amphistomes (Elango & Rahuman 2011; Kamaraj et al. 2010).

Chemical control of the snail hosts through application of molluscicides such as niclosamide may also be done. To achieve cost-effective control, this type of control should be done during the peak transmission period to reduce numbers of infected snails and cercarial shedding. Thus, the application could be done during the mid-dry and towards the end of the dry season (Pfukenyi et al. 2005b). The application is practical and economical in areas where snail habitats are focal and not widespread, but regular application may be necessary because of the rapid recovery of the snail populations during brief periods of favourable conditions. However, molluscicide application causes environmental pollution and also kills non-targeted aquatic organisms (Roberts & Suhardono 1996).

Immunological control

Hafeez and Rao (1981) showed that the lifespan and pathogenicity of amphistomes developing from gamma irradiated (2 or 3 krad) metacercariae were greatly reduced with the higher irradiation dose resulting in the complete absence of the flukes in infected animals. Single vaccination of kids and lambs with 3000 irradiated (2 or 3 krad) metacercariae stimulated a significant degree of resistance against challenge and the resistance was more pronounced in the group vaccinated with a higher irradiation dose (Hafeez & Rao 1981). Earlier, Horak (1967) successfully immunised sheep, goats and cattle against massive artificial infections with Cal. microbothrium. The animals were given immunising infections with at least 40 000 metacercariae and later challenged with larger doses of metacercariae (Horak 1967).
Cattle were the most suitable subjects for immunisation with the immunity being effective for at least a year post-immunisation (Horak 1967, 1971). Mavenyengwa et al. (2008) demonstrated that cattle acquire resistance to amphistome infection. This resistance is targeted at immature amphistomes and it involves eosinophils and mast cells. However, despite promising immunisation results, the mass production of snail hosts and metacercariae remains a challenge and a major limiting factor (Horak 1967, 1971; Mavenyengwa et al. 2006; Swart & Reinecke 1962a, 1962b). Thus, the success of a large-scale immunisation program is dependent on a viable metacercariae mass production system.

Non-chemical control

The best preventive method against amphistome infections is to keep domestic ruminants from infected pastures (Pfukenyi et al. 2005b). Fencing-off or drainage of wetlands or marshy/swampy areas and provision of clean pastures and cercariae-free water in troughs are advised (Roberts & Suhardono 1996). Similarly, habitat management through vegetation clearance is also effective in controlling the snails (Woolhouse & Chandiwana 1990). However, habitat management and complete separation of stock from snail habitats are only practical and economical where the snail habitats are focal and not widespread (Pfukenyi et al. 2005b). These control methods are not feasible in communal grazing areas. It is also important to repair any leaks in dams and water troughs as they can create an ideal habitat for the snail hosts.

Conclusions

Twenty-six amphistome species belonging to nine genera from three families occur in domestic and wild ruminants in the area under review and seven snail species belonging to four genera from two families act as their intermediate hosts. Eighty-five per cent of the amphistome species are shared between domestic and wild ruminant hosts. Some snails are intermediate hosts of amphistome species belonging to the same genus or to different genera – a phenomenon not yet fully elucidated. Only nine (34.6%) of the amphistome species have known snail intermediate hosts, while most (65.4%) have unknown hosts. The epidemiology of amphistomosis depends on the species of definitive and intermediate hosts and the potential of the flukes to infect these hosts, the topography and biological potential of the snail hosts, the management systems of the definitive host and their grazing habits and climatic factors. Based on current epidemiological information, the strategic anthelmintic treatment against mature amphistomes should be given in adult animals at the end of the rainy or early dry season. The anthelmintic treatment in young animals against immature amphistomes should be administered during the mid-dry and towards the end of the dry season. Further research is necessary to determine the economic importance of amphistomosis, amphistome–snail associations, efficacy of different anthelmintics and to develop diagnostic tests that can detect prepatent infections in the definitive host.

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Competing interests

The authors declare that they have no financial or personal relationships that may have inappropriately influenced them in writing this article.

Authors’ contributions

Both authors contributed equally in the writing of this manuscript.

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