Review

Natural and experimental studies on domestic animal infections with visceral and cutaneous leishmaniases in Kenya.

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SUMMARY

The control of visceral and cutaneous leishmaniases in Kenya has been difficult due to poor knowledge of animal reservoirs of these diseases. Little is known about the feeding preferences of phlebotomine sand flies that transmit some of the leishmaniases that are endemic in some areas in Kenya, which makes it difficult to control the diseases by targeting the animal reservoirs. This article reviews the efforts that have been made to identify reservoirs of known *Leishmania* parasites that cause leishmaniases in Kenya. The account includes studies that have been carried out on Kenyan canids, felines, ungulates and murines.

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Introduction

Visceral leishmaniasis (VL) caused by *Leishmania (Leishmania donovani)* Lavaran and Mesnil has been known in Kenya since the 1940s [1]. The major known foci are in Turkana, Baringo, and West Pokot Districts [2], in Rift Valley Province and in Machakos [3], Kitui [4], and Meru [5] in Eastern Province. In these areas, the parasite is transmitted through the bite of an infected female sand fly, *Phlebotomus (Synphlebotomus martini)* Parrot [5-7]. Several epidemiological studies in endemic areas have been conducted on various domestic and peri-domestic animals to incriminate them as reservoirs of various for this parasite have only yielded limited data. Failure to satisfactorily incriminate wild and synanthropic animals have led to the conclusion that the disease is anthroponotic in Kenya with a man-sand fly-man cycle [6].

In the search for the *L. donovani* reservoir in 1977, Ngoka and Mutinga [7] reported from their studies on domestic dogs, *Canis familiaris L.* in West Pokot, Rift Valley province that the dog was a possible reservoir for the parasite. Later, Mutinga et al [8] isolated the parasite from 2/288 emaciated dogs trapped in homesteads of VL human cases in Machakos District, Eastern province. The small number of animals found infected made it doubtful that dogs could be reservoirs for the parasite in Kenya. Later, laboratory infection of dogs with $1x10^6$ culture derived *L. donovani* stationary phase promastigoes in four puppies did not lead to

development of the disease, suggesting that the healthy puppies were not susceptible to the disease [9].

Apart from dogs, domestic cats, *Felis domesticus L.* are also commonly found in homestead in most VL endemic areas. Many reports from the Americas have shown that the domestic cat is susceptible to New World leishmaniases. *L. (L.) mexicana* Biagi and has been isolated from a cat in Texas [10]. *L. (L) venezuelensis* has been isolated from 3 cats in Venezuela [11]. In Kenya, *L. donovani* has not been isolated from cats. To the best of our knowledge, there are only 2 reports on the isolation of *L. donovani* s.l. from a wild-caught genet cat, *Genetta genetta senegalensis* Fischer and 1 report on isolation of the same parasite from the serval, *Felis serval phillipsi* Schreber from Eastern Africa [12].

Few laboratory experiments to determine susceptibility of domestic cats to visceralizing *L. donovani* have been conducted. Kirkpatrick et al [13] demonstrated that cats inoculated intravenously with *L. chagasi* Cuhna and Chagas and *L. infantum* Nicolle (reported as *L. donovani*) were able to retain parasites in the viscera for 16 weeks without developing visceral leishmaniasis. Later, Anjili and Githure [14] experimentally inoculated 12 stray domestic cats each intravenously with $1x10^6$ culture derived *L. donovani* stationary phase promastigoes. When the cats were sacrificed opportunistically at 30, 60, 90, 120 and 150 days post-inoculation (DPI), no parasites were seen in stained smears and in cultures of liver, spleen and blood. All cats were active at necropsy and appeared healthy. The authors concluded that cats are refractory to infections with the Kenyan strain of *L. donovani* (Strain MHOM/KE/82/LRC-L445=NLB-065). It is therefore unlikely that the domestic cat plays any role in VL epidemiology in Kenya.

A parasitological and serological survey of domestic goats, *Capra hircus* L was conducted in Baringo District, R. Valley Province to detect parasites in culture and *L. donovani*-specific IgG. A total of 102 goats were sampled. No flagellates were recovered in culture. However IgG antibodies were detected from 2 goats. The authors concluded that even though goats are exposed to the parasites, the data did not support the implication that they are potential reservoirs for human leishmaniasis [15]. It has been suggested that the blackhead sheep (*Ovis aries* L.) may act as a reservoir for human *Leishmania* [16]. However, parasites collected were not identified as *Leishmania* or biochemically characterized. The determination of parasites as *Leishmania* sp. was based on the morphological characteristics of the basophilic bodies seen in tissue sections.

Experimental infection of domestic sheep with $1x10^6$ culture derived *L. donovani* stationary phase promastigoes was carried out in 4 sheep to determine susceptibility to this parasite strain. Apart from 28 DPI when a saline aspirate was taken from one out of six sheep revealed the presence of *Leishmania* promastigotes after 12 days of incubation in culture aspirate had parasites. At no time were amastigotes seen in Giemsa-stained smears throughout the 244 days sampling period. At necropsy, cultures of liver, spleen, blood, nasal skin and mandibular lymph nodes did not contain any parasites. It was concluded that this ungulate cannot support the infection with *L. donovani* [17]. However, serology using enzyme-linked immunoorbent assay (ELISA) showed that the sheep that had parasites at 28 DPI developed *L. donovani*-specific IgG antibodies. The rest of the sheep had no detectable humoral response to leishmanial antigens.

Within the VL endemic focus of Baringo District, R.Valley Province, cutaneous leishmaniasis (CL) caused by *L. (L.) major* Yarkimoff and Schokhor is also endemic. The parasite in this area is transmitted by *P. (P.) duboscqi* Neveu-Lemaire [18], which gets infected after feeding the known peri-domestic rodent reservoirs mainly *Tatera robusta* Cretzschmar, *Aethomys kaiseri* Noack, *Taterillus emini* Thomas, *Mastomys natalensis* Smith and *Arvicantis niloticus* Geoffrey [19,20]. Apart from these rodents, the parasite has never been isolated from any other animal except infected human patients [21]. It was suggested that goats may act as reservoirs for *Leishmania* after 4/457 goats from West Pokot, Kenya yielded leishmanial parasites in culture [22] these parasites were not identified as *L. major* or *L. donovani* by characterization. Parasitological and serological studies were only able to detect *L. major*-specific IgG antibodies from 2/102 goats that were sampled by Robert et al [15]. No parasites were seen in cultures. Experimental studies have shown that goats can become experimentally infected with *L. major* through needle inoculation of culture-derived promastigotes (Strain IDUB/KE/83=NLB-144), but not by bites of infected *P. duboscqi* [23]. This study showed that goats do not seem particularly susceptible to the disease and that his disease is a true zoonosis in Kenya.

Around the slopes of Mt. Elgon in Western Kenya, *L. (L.). aethiopica* Bray, Ashford and Bray that causes CL is endemic. In this area, parasites have been isolated from rock hyrax, *Provavia johnstoni* Thomas, the tree hyrax, *Dendrohyrax arboreus* Smith and the giant rat, *Cricetomys gambianus* Waterhouse [24, 25]. In this area, the parasite is a zoonosis and is transmitted by *P. (Larroussius) pedifer* Lewis, Mutinga and Ashford which readily feeds on these murines. No other animal reservoir hosts have been identified. Isoenzyme characterization of one isolate from a goat in Kenya showed that *L. aethiopica* was responsible for the infection [26]. Unfortunately, the authors did not indicate what geographical area of Kenya the goat came from.

Since the discovery of autochthonous human cases of CL caused due to *L. tropica* Wright in R. Valley, Kenya [27], considerable effort was made to search for the vector and reservoir of the disease. The active foci for the disease were found to be Muruku Sublocation, Laikipia District [28], where the vector was identified as *P. (Larroussius) guggisbergi* Kirk and Lewis [29], and Utut Reserve, Njoro, Nakuru District [30] , where the probable vector was identified as *P. (Larroussius) aculeatus* Lewis , Minter and Ashford [31]. Within the Muruku focus, *P. guggisbergi* is mainly found in caves thus strongly suggesting the presence of an animal reservoir even though no domestic animal has been found infected with the parasite [29].

Considering that no rural sylvatic reservoirs for *L. tropica* have been found in the Laikipia focus Johnson et al [32] conducted a study to determine the feeding preferences of *P. guggisbergi.* They set up baits in the caves containing a sheep, goat, dog, rat, cat, hamster (*Mesocricetus auratus* Waterhouse), rabbit (*Oryctolagus cuniculus* L.), giant rat, crested rat (*Lophiomys imhausi* Milne-Edwards) and the rock hyrax, all of which (except hamsters) are normally found in the vicinity of the study site. Sand fly collections from traps baited with goat, sheep, cat, dog, rabbit or wild rodent species were significantly higher than the control SSAM miniature light traps (John W. Hock, C., Gainsville, FL, U.S.A) baited with solid CO_2 whereas trap collections with

hamster and rock hyrax were not significantly greater than from the rabbit and rodents. The authors concluded that emphasis should be placed on greater surveying larger animals to assess their status as reservoir hosts for *L. tropica* in Kenya. In the Utut focus a rock hyrax was found infected with *Leishmania* parasites leading to the suggestion that it could be a reservoir and hence the disease a zoonosis [33]. The parasites were not characterized biochemically. There is therefore no proof that they were *L. tropica*.

Conclusion

Apart from chemotherapy, use of insecticidal nets and repellants, control of animal reservoirs of the leishmaniases also contributes significantly in the control of the leishmaniases. More knowledge about animal reservoirs of anthropophilic sand fly species can be useful when deciding how a certain form of leishmaniasis can be controlled. Considering that in Kenya the only well known animal reservoirs are for *L. major* and *L. aethiopica,* more efforts should be made to survey in detail whether AVL caused by *L. donovani* is a zoonosis or a purely anthroponotic disease. The sources of infections in sand fly vectors in the *L. tropica* foci of Muruku and Utut still need further investigations.

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