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RESEARCH ARTICLE

Evidence for circulation of Rift Valley fever virus in wildlife and domestic animals in a forest environment in Gabon, Central Africa

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Abstract

Rift Valley fever (RVF) is a mosquito-borne viral zoonosis caused by the Rift Valley fever virus (RVFV) that can infect domestic and wild animals. Although the RVFV transmission cycle has been well documented across Africa in savanna ecosystems, little is known about its transmission in tropical rainforest settings, particularly in Central Africa. We therefore conducted a survey in northeastern Gabon to assess RVFV circulation among wild and domestic animals. Among 163 wildlife samples tested using RVFV-specific RT-qPCR, four ruminants belonging to subfamily Cephalophinae were detected positive. The phylogenetic analysis revealed that the four RVFV sequences clustered together with a virus isolated in Namibia within the well-structured Egyptian clade. A cross-sectional survey conducted on sheep, goats and dogs living in villages within the same area determined the IgG RVFV-specific antibody prevalence using cELISA. Out of the 306 small ruminants tested (214 goats, 92 sheep), an overall antibody prevalence of 15.4% (95% CI [11.5–19.9]) was observed with a higher rate in goats than in sheep (20.1% *versus* 3.3%). RVFV-specific antibodies were detected in a single dog out of the 26 tested. Neither age, sex of domestic animals nor season was found to be significant risk factors of RVFV occurrence. Our findings highlight sylvatic circulation of RVFV for the first time in Gabon. These results stress the need to develop adequate surveillance plan measures to better control the public health threat of RVFV.

analysis, decision to publish, or preparation of the manuscript.

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Author summary

Rift Valley fever (RVF) is a mosquito-borne viral zoonosis caused by the Rift Valley fever virus (RVFV) that can affect wild and domestic animals. Although the RVFV transmission cycle has been well documented across Africa in savanna ecosystems, little is known about its transmission in tropical rainforests, especially in Central Africa. We thus conducted a survey in northeastern Gabon to assess RVFV circulation among wild and domestic animals. In this study, we demonstrated for the first time in Gabon the presence of the RVFV in two wildlife species (Peter's duiker *Cephalophus callipygus* and the blue duiker *Philantomba monticola*). In addition, we detected RVFV-specific antibodies in small domestic ruminants (sheep and goats) at an overall antibody prevalence of 15.4%, with a much higher seroprevalence rate in goats than sheep (20.1% versus 3.3%). Furthermore, RVFV-specific antibodies were also observed in a single (hunting) dog out of the 26 tested. These results stress the need to develop adequate surveillance plan measures to better control the public health threat of RVFV.

Introduction

Among the 175 species of infectious organisms that are known to be pathogenic to humans and considered to be associated with emerging infectious diseases (EIDs), 75% are zoonotic—with many emerging over the past two decades in wildlife sources [1]—making zoonotic EIDs a growing major threat to global health.

Although the emergence and re-emergence of diseases caused by arboviruses (viruses transmitted by arthropod vectors) is a constant concern in many African countries, their prevalence remains poorly documented due to the lack of efficient surveillance systems [2]. In addition, a significant number of vector-borne viruses are zoonotic, and there are gaps in the understanding of their ecology in natural wildlife niches and the factors that lead to their transmission to humans.

In Gabon, a total of 51 endemic or potentially endemic infectious viral diseases have been reported. Among them, 22 are of zoonotic origin and involve 12 families of viruses [3,4] with the most impactful arboviruses causing chikungunya, dengue, Rift Valley fever (RVF), yellow fever, West Nile fever, Crimean-Congo hemorrhagic fever and Zika virus disease. RVF is a World Organization for Animal Health (WOAH)-listed disease and a World Health Organization (WHO) priority disease for research and development due to its potential to cause major epidemics in humans [5]. RVF is a mosquito-borne, infectious disease caused by a negative single-stranded RNA virus named Rift Valley fever virus (RVFV), a member of the *Phlebovirus* genus (family *Phenuiviridae*). In humans, RVFV infection is mostly pauci-symptomatic, but the illness can progress to hemorrhagic fever syndrome in some cases [6]. In animals, abortions and stillbirths in ruminants—domestic (cattle, sheep, goats and camels) or wild (buffaloes, antelopes, wildebeest)—lead to livestock deaths and considerable economic losses in Africa, the Arabian Peninsula and the southwestern Indian Ocean region [7,8]. Epizootics (*i.e.* disease outbreaks that affect animals) of RVF are sporadic and often linked to persistent and heavy rainfall and flooding, which are in turn correlated with an abundance of mosquitoes of the *Aedes*, *Culex* and *Anopheles* genera, which are known to be involved in RVFV transmission [9,10]. Humans usually contract RVF through direct contact *via* body fluid secretions of infected livestock and, to a lesser extent, may develop the disease through bites from infected mosquitoes [11].

Following the first description of RVFV in 1930 in Kenya [12], epizootics were recorded in East and South Africa until 1977; evidence from serological surveys (Angola, 1960; Cameroon, 1968; Chad, 1969) and virus isolations (Democratic Republic of Congo (DRC), 1936–1954; Central African Republic (CAR), 1969) have revealed contemporaneous circulation of RVFV in central Africa. Thereafter, the disease began to spread north to Sudan and Egypt, leading to the first massive epizootic/epidemic in Egypt in 1977–78, which affected 200,000 people and led to at least 600 deaths [13]. The disease was later recorded in Madagascar in 1979 and then West Africa (Senegal and Mauritania) in 1987 [14]. The epidemic potential and human health impact of this disease have been acutely felt on the African continent. RVF is enzootic/endemic in East and South Africa causing epizootics/epidemics in Egypt (2003), Kenya (2018), Somalia, Sudan, Madagascar (2008–2009, 2019–2021), South Africa (2009–2011), Uganda (2016, 2023) [15] and various parts of West Africa, with inter-epizootic RVFV circulation. In 2000–2001, the virus left the African continent for the first time, reaching the Arabian Peninsula (Saudi Arabia, Yemen) [8].

In Central Africa, at the crossroads of major African geographical regions experiencing RVF epizootics/epidemics, several studies have demonstrated the circulation of the virus in domestic ungulates as well as in humans in a savanna-type ecosystem in Cameroon, Gabon, Equatorial Guinea and the DRC [16–25], but no major epidemics or epizootics have been reported there, in contrast to East and South Africa, West Africa and Egypt. Nevertheless, little is known about RVFV in the tropical forests of Central Africa, with only a few serological surveys suggesting RVFV circulation. These surveys revealed the presence of RVF antibodies in antelopes, wild buffaloes, warthogs and elephants in CAR [26] and in the rural human population in Gabon [27]. Moreover, in southern Cameroon, the sylvatic circulation of RVFV was suggested to explain the presence of antibodies in locally bred goats [28]. Nonetheless, the sylvatic cycle of RVFV remains poorly documented in Central African rainforests. Several wild-life vertebrate hosts, particularly wild ungulates, are possibly involved in RVFV circulation involving forest mosquito species (belonging to genera *Aedes*, *Anopheles* and *Culex*) that are involved in, or are closely related to, domestic cycles. To date, little is known about RVFV sylvatic vectors in the forests of Central Africa, and the virus has only been isolated once in *Aedes* mosquitoes belonging to the *Neomelaniconion* subgenus and the *palpalis* species group collected in the CAR [29]. Moreover, isolation of RVFV from humans [30] together with serological RVFV evidence from Pygmy populations [31] suggest the existence of an RVFV forest cycle in the CAR and probably throughout Central Africa.

The study conducted here in Gabon was therefore intended to extend our knowledge of the sylvatic circulation of the RVFV in rainforests of Central Africa by investigating wildlife and domestic animals at the edge of rainforest. We demonstrated for the first time in Gabon the presence of the RVFV in two wildlife species (Peter's duiker *Cephalophus callipygus* and blue duiker *Philantomba monticola*), along with RVFV-specific antibodies in livestock small ruminants and dogs.

Materials and methods

Ethics statement

To carry out the sampling campaign, we obtained authorization to capture and collect animals from the Ministry of Water and Forests, in charge of the environment and sustainable development (Authorization No. 0247 MEFCEDD/SG/DGFAP).

Study area

The study was carried out in 19 villages located in the Zadié Department, located in the Ogooué-Ivindo province, northeastern Gabon. This area is mainly composed of primary

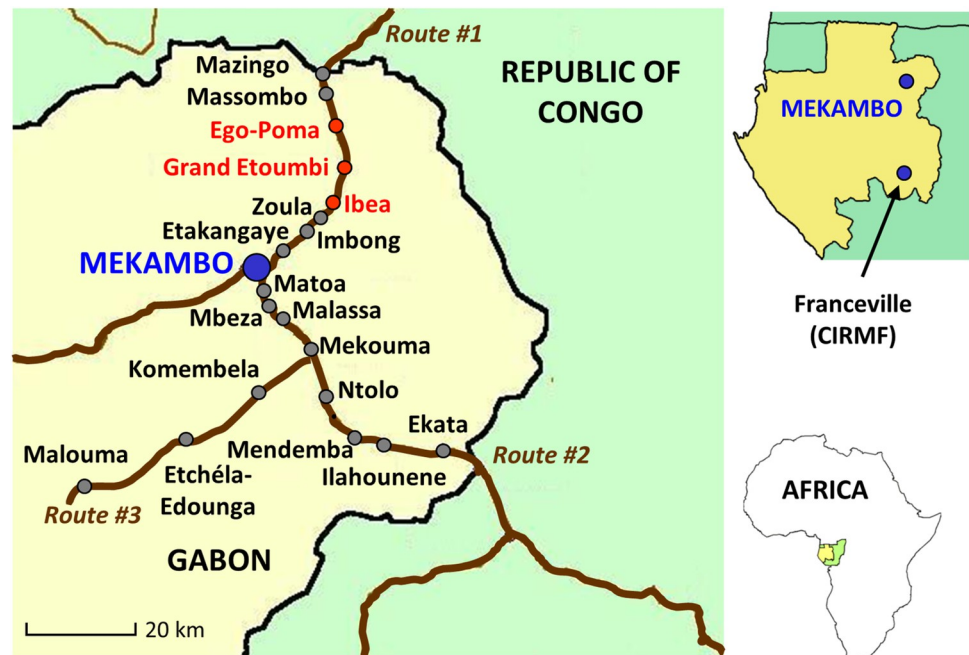


Fig 1. Map of the study area, Zadié Department, Gabon. Gray dots indicate the sampled villages and red dots, the villages where hunters brought back *Cephalophinae* detected positive for RVFV.

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tropical rainforests along three main routes radiating from Mekambo, the main city in Zadié: Mekambo-Mazingo (Route #1), Mekambo-Ekata (Route #2) and Mekambo-Malouma (Route #3) (Fig 1).

Sampling and data collection

Wild animals were sampled along the three routes described above (Fig 1) in July 2019 during the long dry season (June to September) and legal hunting season (March to September). Organ samples (liver and spleen) were collected from animals hunted during both campaigns in the surrounding forest and displayed roadside and sold for consumption (Table 1). Samples were kept in coolers during sample collection. Samples were then temporarily stored in liquid

Table 1. List of wild animal species screened for the RVFV genome.

Wild animal species	<i>n</i>
<i>Atherurus</i> sp.	25
<i>Cephalophus agilbigi</i>	1
<i>Cephalophus callipygus</i>	4
<i>Cephalophus dorsalis</i>	13
<i>Philantomba monticola</i>	92
<i>Cephalophus</i> sp.	22
<i>Cercopithecus cephus</i>	1
<i>Genetta abyssinica</i>	2
<i>Genetta</i> sp.	2
<i>Potamochoerus porcus</i>	1
Total	163

<https://doi.org/10.1371/journal.pntd.0011756.t001>

nitrogen at the Mekambo health center, before being transferred to CIRMF (*Centre Interdisciplinaire de Recherches Médicales de Franceville*) laboratory for storage at -80°C .

Additionally, livestock (goats and sheep) and dogs were sampled at two time points, once in November 2018 (short rainy season) and once in July 2019 (long dry season), along Route #1 and Route #2 (Fig 1). There is no census of livestock in this region. Domestic animals were selected based on the willingness of the livestock owners to participate in the study. The number of sheep and goats sampled thus depended on the livestock owners' availability and their ability to restrain their animals for sampling. Ultimately, most owners in the villages agreed to have their animals sampled and participated in the two sampling campaigns.

Data on species, sex, period of sampling and age (or sexual maturity stage) were collected using a standard questionnaire submitted to each animal owner. Sheep and goats were classified as young or adult according to the criterion of sexual maturity: young (under 3 years old) and adult (aged >3 years) using both morphological characters observed by the veterinarians and information provided by animal owners. For each domestic animal, a blood sample was collected in EDTA tubes upon jugular venipuncture and preserved in a cooler box until transport to the laboratory.

RVFV genome detection in wildlife

A total of 163 wild animals, comprising mostly *Cephalophus* spp. ruminants and *Atherurus* spp. rodents, were sampled (Table 1) and tested for the presence of RVFV genome using a RT-qPCR method. Briefly, after grinding the organs (liver and spleen) in RA1 lysis buffer supplemented with a 1% Triton X-100 solution (Sigma, France), RNA was extracted using the Nucleospin RNA kit (Macherey-Nagel, Germany) followed by a RVFV-specific RT-qPCR amplification [32] in a Lightcycler L96 (Roche). When RVFV was detected in wildlife samples, DNA was extracted using the Qiagen DNeasy Blood & Tissue Kit (Qiagen, Courtaboeuf, France) to amplify a 710 bp long fragment of the mitochondrial cytochrome oxidase I (*COX1*) gene using PCR to identify/confirm the vertebrate species [33]. *COX1* sequences generated were then aligned and compared with Cephalophinae sequences from Central Africa.

Sequencing and phylogenetic analysis of RVFV

After PCR amplification of the RVFV-S segment, all positive samples were sequenced using a previously described method [34]. The phylogenetic analyses were done after multiple alignments of the obtained sequences, along with GenBank reference sequences using ClustalW (v1.8.1 in BioEdit v.7.0.9.0. software). Indeed, before phylogenetic analysis, datasets and multiple sequence alignments were thoroughly checked to eliminate misalignments and ensure the correct framing of the coding sequences. Maximum likelihood (ML) methods were used for tree construction using full-length sequences of the S segment (1690 nucleotides). Sequence evolution was modeled using the general time reversible (GTR) + Gamma model, as determined using Model Test [35]. The best-fitting ML model according to Akaike's information criterion was the general time-reversible + γ distribution for nucleotides, as identified by Model Test [35]. The ML trees and corresponding bootstrap support values were obtained using the online software PhyML, based on nearest neighbor exchange and subtree pruning, regrafting, branch swapping and 100 bootstrap replicates [36] (available at the ATGC bioinformatics facility: <http://www.atgc-montpellier.fr/>).

Anti-RVFV antibody detection in domestic ruminants and dogs

In domestic animals, RVFV-specific IgM and IgG antibodies were detected using ELISA with respectively the ID Screen Rift Valley fever IgM Capture and ID Screen Rift Valley fever

competition multispecies kits (Innovative Diagnostics, Grabels, France) according to the manufacturer's instructions. Diagnostic sensitivity of the IgG kit is 98% and specificity 100% [37]. Anti-RVFPV antibody detection could not be carried out in wildlife because blood samples were not available.

Because the circulation of phleboviruses other than RVFPV cannot be excluded in Gabon, a subset of randomly selected positive and negative samples was tested using the virus neutralization test (VNT), considered as the gold standard method by WOAHP [38]. Briefly, duplicates of two-fold serial dilutions of sera starting from 1:5 were added to 100 TCID₅₀ (50% tissue culture infectious dose) of Smithburn RVFPV in 96-well microtiter plates and incubated for 1 h at 37°C. Next, 100,000 Vero cells were added to each well and the plates were incubated under 5% CO₂ for 5–6 days at 37°C. Titers were expressed as the inverse highest dilutions giving 50% of cytopathic effect. A positive control serum was included. A serum sample with a titer of 1:10 or higher was considered seropositive.

Statistical analysis

We analyzed small ruminant serological data from ELISA using GLM (generalized linear models), with the individual serological status as the response, and potential risk factors (species, age, gender, period of sampling) as explanatory variables. Multicollinearity among explanatory variables was assessed using variance inflation factors (VIFs). The selection of the best models was based on the Akaike information criterion (AIC). A multi-model inference approach was used for the set of models with an AIC within 2 units difference of the best model [39]. Data analyses were performed using R software version 4.3.0 [40].

Results

RVFPV genome detection and genetic diversity

Of the 163 wildlife animals sampled along three main routes in northeastern Gabon, the RVFPV-specific genome was detected in four of them, *i.e.* two duiker species: one sample from *Cephalophus callipygus* and three from *Philantomba monticola* (Table 2 and Fig 1). After sequencing the entire S segment, phylogenetic analyses were carried out to explore their genetic relatedness with all previously published RVFPV S segment nucleotide sequences. All four sequences detected in duikers clustered with a human strain of RVFPV isolated in Namibia in 2004, with nucleotide identity between our sequences and the Namibian sequence ranging from 99.0 to 99.8%. This cluster is included in Group A following Grobbelaar's classification [41] that now includes viruses from Egypt, Zimbabwe (1978) and Madagascar (1979) (Fig 2). Viral isolation was attempted on Vero cells without success.

RVFPV-specific antibody prevalence

Following the detection of RVFPV in wild duikers, a cross-sectional serological study was conducted in populations of small domestic ruminants and dogs living in villages where hunted animals were sampled. Power calculations indicated that a sample size of 400 goats and 400 sheep is necessary to attain 2.5% precision, given an expected prevalence of 6.5%, according to the only estimation of RVFPV seroprevalence in small ruminants in Gabon [23]. Based on an estimated 1200 goats and 600 sheep living in our study area, according to data collected during campaigns, our sample sizes represent approximately 18% of goats and 15% of sheep. As mentioned above, the main difficulty was that small ruminants are not held in enclosures, but roam freely in the villages. Therefore, we could only sample animals that villagers could capture. The difficulties of carrying out this type of study in a region composed mainly of primary

Table 2. RVFV genome detection in wildlife using RT-qPCR according to sampled route and village.

Route	Village	Number of positive samples/Total number of samples
Route #1	Etakangaye	-
	Imbong	-
	Zoula	1/20 ¹
	Ibea	-
	Grand Etoumbi	2/53 ^{1,2}
	Ego Pouma	1/11 ¹
	Massombo	-
	Mazingo	-
Route #2	Matoa	0/2
	Mbeza	-
	Malassa	0/5
	Mekouma	0/11
	Ntolo	0/3
	Mendemba	-
	Ilahounéné	-
	Ekata	0/13
Route #3	Komenbela	0/23
	Etchela-Edounga	0/19
	Malouma	0/3
	Total	4/163

¹*Philantomba monticola*²*Cephalophus callipygus*<https://doi.org/10.1371/journal.pntd.0011756.t002>

tropical rainforests, which are difficult to access, also explains our “relatively modest” sample sizes. Overall, a total of 306 small ruminants (214 goats and 92 sheep) and 26 dogs (including 3 hunting dogs) were sampled and screened for RVFV specific antibodies (IgM and IgG) using ELISA. RVFV-specific IgM was not detected in any of the samples. RVFV-specific IgG antibody prevalence in livestock was 15.4% (47/306; 95% CI [11.5–19.9]) (Table 3). VNT was used to confirm RVFV specificity in 19 samples with a strong positive signal and in 78 samples found negative using cELISA. A total of 15 samples out of the 19 tested were confirmed positive and all the negative samples were confirmed negative by both methods. RVFV-specific antibodies were found in goats along both routes with similar prevalence rates (Route #1: 24.5% (23/94); Route #2: 17.5% (21/120)). Unlike goats with a seroprevalence of 20.6% (95% CI [15.4–26.6]), RVFV-specific antibodies were detected only in three sheep with a seroprevalence of 3.3% (95% CI [0.7–9.2]): Route #1: 2.6% (2/76); Route #2: 6.2% (1/16). Finally, RVFV-specific antibodies were detected in only one dog (3.8% (1/26), 95% CI [0.0–19.6]), which was a hunting dog (Table 3).

Explanatory variables were not collinear (variance inflation factors (VIFs) less than 2) in the full model including all explanatory variables (species, age, gender, period of sampling). According to AIC, two models were suitable for describing small ruminant seroprevalence and thus were analyzed using a multi-model inference approach. These two best models included species and period of sampling as explanatory variables. The seroprevalence in goats was significantly higher than in sheep ($p < 0.001$; odds ratio (OR) = 8.1, 95% CI [1.4–27.1]), whereas the sampling period was not significant using multi-model inference.

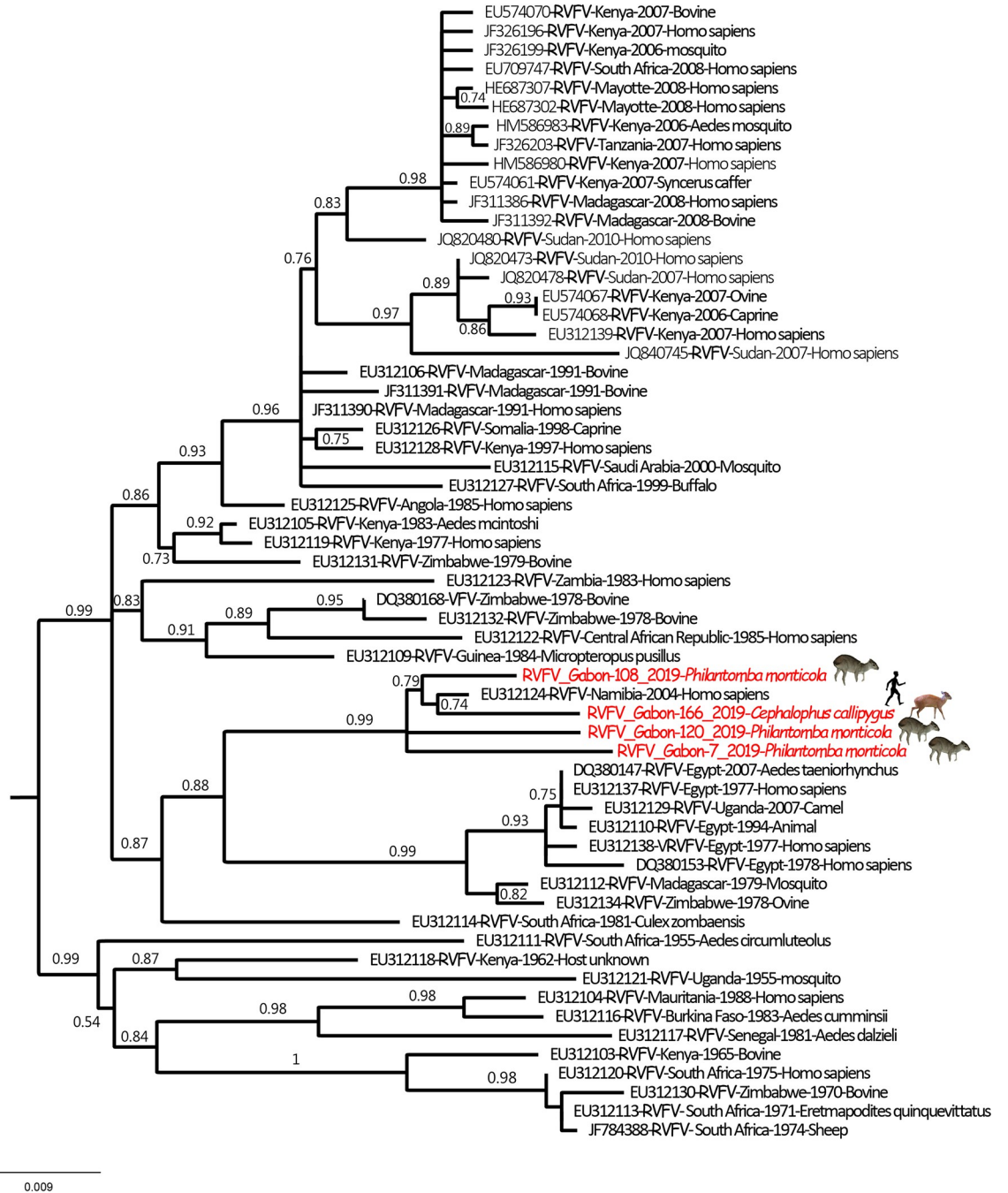


Fig 2. Phylogenetic tree derived from nucleotide sequence data of the entire S segment. The phylogenetic analyses were carried out after multiple alignments of the obtained sequences along with the GenBank reference sequences (including all published sequences). Maximum likelihood (ML) methods were used to construct trees based on full sequences of the S segment (1690 nt). The GenBank accession numbers for the S gene are OR528950, OR528951, OR528952, OR528953 for samples 7, 108, 120 and 166, respectively.

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Discussion

Although 100 years have elapsed since RVFV was first detected, outbreaks are still difficult to anticipate and control, because the drivers of RVF endemicity are not clearly understood. The multiplicity of vertebrate hosts and mosquito species involved in the RVFV transmission cycle,

Table 3. RVFV-specific IgG antibodies detection in livestock (sheep and goats) and dogs using ELISA according to sampled route and village.

Route	Village	Goats			Sheep			Dogs
		Positive samples/total	Age		Positive samples/total	Age		Positive samples/total
			Young	Adult		Young	Adult	
Route #1	Etakangaye	5/23	5/11	0/12	-	-	-	-
	Imbong	9/28	3/12	6/16	1/23	0/11	1/12	1/6
	Zoula	3/13	3/7	0/6	0/8	0/2	0/6	0/2
	Ibea	1/6	0/1	1/5	0/1	-	0/1	-
	Grand Etoumbi	0/6	0/1	0/5	0/9	-	0/9	-
	Ego Pouma	0/1	-	0/1	1/9	0/5	1/4	-
	Massombo	1/6	1/1	0/5	0/3	0/2	0/1	-
	Mazingo	4/11	1/5	3/6	0/23	0/14	0/9	0/1
Route #2	Matoa	-	-	-	-	-	-	-
	Mbeza	4/7	3/5	1/2	1/5	0/1	1/4	0/5
	Malassa	1/7	1/6	0/1	0/3	0/1	0/2	-
	Mekouma	10/34	7/12	3/22	0/1	-	0/1	0/8
	Ntolo	3/20	2/8	1/12	0/2	-	0/2	0/4
	Mendemba	2/26	0/7	2/19	-	-	-	-
	Ilahounéné	1/8	0/5	1/3	-	-	-	-
	Ekata	0/18	0/11	0/7	0/5	0/1	0/4	-
	Total	44/214	26/92	18/122	3/92	0/37	3/55	1/26
	Seroprevalence (%)	20.6	28.3	14.8	3.3	0.0	5.5	3.8
	95% CI	[15.4–26.6]	[19.4–38.6]	[9.0–22.3]	[0.7–9.2]	[0.0–9.5]	[1.4–15.1]	[0.0–19.6]

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the diversity of ecosystems in which RVFV occurs and changes in human activities along with their environmental dynamics, make the entire epidemiological RVFV cycle complex and hard to determine. Although sylvatic circulation has been suspected for a long time in Central Africa, RVF rarely occurs in an epizootic form in livestock and very few clinical cases of infection have been reported in humans. The aim of this study was to investigate the circulation of RVFV in selected wildlife and livestock species living in the forest (wildlife) and villages (domestic animals) in northeastern Gabon.

Sylvatic compartment

We detected the RVFV genome for the first time in a forest environment in two wildlife ruminant species (duiker antelopes, *C. callipygus* and *P. monticola*) sampled in three neighboring villages in northeastern Gabon in a rainforest area. The sequencing of the S segment showed that RVFV detected in these duikers clustered with a sequence of RVFV from Namibia sampled in 2004, closely related to the Egyptian clade A based on the Bird et al. and Grobelaar classifications [34,41]. Unfortunately, the lack of recent RVFV sequences precludes establishing links with strains circulating in Central Africa. Our results demonstrate the circulation of the virus in wild animals. However, the mode of circulation of this virus remains unknown and the community of potential RVFV mosquito vectors in forest ecosystems has been poorly characterized, thereby requiring further investigation to model virus transmission and maintenance. Nevertheless, we know that at least 14 mosquito species incriminated as potential RVFV vectors are present in Gabon [10,42,43]. For genus *Aedes* (subgenus *Neomelanoconion*), *Ae. macintoshi* has previously been reported in the country (as *Ae. lineatopennis*), as well as *Ae. palpalis* (from which the virus was previously isolated in a forest in the CAR). Interestingly, among the potential vectors present in Gabon, *Anopheles coustani* has been shown to bite wild

ungulates in the forests (including *C. callipygus*), as well as six additional anopheline species (*An. carnevalei*, *An. marshallii*, *An. moucheti*, *An. obscurus*, *An. paludis*, *An. vinckei*), making these species putative candidates for the sylvatic transmission of RVFV in wild ungulates [43]. Further investigations must focus on mosquitoes that feed on wild ungulates and the Cephalophinae antelope species, they favor to determine vector candidates and are likely to shed light on sylvatic vector transmission of RVFV in Gabon.

Domestic compartment

A cross-sectional serological study on domestic animals living in villages in the Mekambo area highlighted that goats, sheep and dogs are exposed to RVFV (overall anti-RVFV antibody prevalence of 15.4% for domestic ruminants), demonstrating its circulation in an anthropogenic environment. Most of these animals are raised locally with no history of importation or vaccination, even though a few of them come from rare imports from the Zadié Department (or from villages located on the other side of the border, in the Republic of the Congo) as gifts for weddings (dowries), deaths or religious celebrations [44]. Although our results suggest RVFV transmission at the edge of the rainforest, the origin of this circulation could also be explained by possible and rare introductions (purchases, gifts) of infected small ruminants from another area in Zadié Department or from neighboring villages in the Republic of the Congo, thus leading to virus circulation in this region.

Our study also showed that the antibody prevalence of RVFV specific antibodies was higher in goats than in sheep (20.6% versus 3.3%, Table 1). To our knowledge, such a difference in seroprevalence levels observed between goats and sheep has not been reported in previous studies. However, none of them have been conducted in villages located in a forest environment, notably the recent studies carried out in the Congo Basin [18,19,23]. The variation in the observed antibody prevalence is likely to be caused by several factors including mosquito trophic behavior and the efficacy of induced immune responses linked to malnutrition or stress. RVFV can be transmitted in livestock through different routes: bites of competent mosquito vectors, contact with infected blood, body fluids and tissues of infected animals, aborted fetuses, placental membranes containing large numbers of virus particles that can either contaminate the local environment directly or infect animals [45]. In our study area, the small ruminants are not enclosed in pens and roam around the villages. In this type of environment, goats are known to venture to the outskirts of villages, and particularly to the edge of the forest [46], according to testimonies collected from owners and villagers. Goats would be more likely exposed to forest-dwelling mosquito vectors, including those that transmit RVFV among wild-life. In contrast, sheep, which are reared around houses, are likely mainly exposed to a more domestic mosquito community. Moreover, the sheep and goats in the area may not be similarly exposed to mosquito vectors, due to qualitative and/or quantitative differences in their attractiveness to mosquitoes. Although comparative studies of goats and sheep regarding mosquito attraction are rare, some of them—undertaken in West [47] and East Africa [48,49]—suggest that there are both qualitative and quantitative differences. In Nigeria, the overall exposure of goats to mosquito bites is twice as high in goats as in sheep, but at a specific level, some mosquito species, such as *Anopheles squamosus*, incriminated as a candidate vector species during RVFV epizootics in Madagascar and Kenya [10], prefer (about 4 times as much) sheep over goats [47]. Among mosquito species involved in RVFV transmission in Kenya, *Aedes ochraceus* and *Aedes mcintoshi* seem to prefer goats over sheep, but the contrary has been observed for *Mansonia uniformis* [48]. In another study from Kenya, most of the RVFV vector species, including *Ae. mcintoshi*, showed no differences in their trophic preferences between goats and sheep, although *Aedes dentatus* tended to prefer goats and *Culex pipiens* preferred

sheep [49]. Nevertheless, the community of potential RVFV mosquito vectors in villages of this study area as part of the forest ecosystems remains poorly understood. It would be helpful to better document mosquito species' blood feeding patterns in the villages of the Mekambo area, as well as to test for a possible differential host trophic preferences between goats and sheep. Excess mortality in sheep due to RVFV infection could also explain the differences in seroprevalence, but no animal owner indicated significant mortality in sheep during the sampling campaigns.

Very little data is available on RVFV circulation in dogs. To our knowledge, only one dog was reported seropositive, out of four tested using the hemagglutination-inhibition test, during the RVFV epizootic in Egypt in 1977–78 [50]. Another study reported RVFV specific antibodies using the same method in wild dogs, and none in domestic dogs, in Botswana, Kenya and South Africa [51]. However, these results could not be confirmed by VNT. Interestingly, despite our small sample size, the only seropositive dog was a hunting dog (1/3 versus 0/23 domestic dogs). Therefore, this dog may have been infected in the forest (*via* mosquito bites, or contact with an infected animal or its fluids/tissues), offering an additional opportunity for the RVFV to be introduced into the domestic compartment and subsequent transmission to domestic animals and humans and in turn be the source of further transmission: numerous anthropogenic mosquito species have opportunistic feeding habits in tropical Africa (e.g. *Culex quinquefasciatus*, *Anopheles gambiae*, *Anopheles funestus*, *Aedes albopictus*) [52–55].

Interconnections between sylvatic and domestic compartments

The interconnections between the sylvatic and domestic compartments in a forest environment may thus be a source of zoonotic disease emergence, specifically RVFV in our case. Exploration of RVFV transmission to domestic animals in an anthropogenic environment, including the identification, the role and the blood-feeding patterns of the potential vectors, need to be explored. Comparison of viral sequences obtained from wild animals, small ruminants and dogs can help confirm whether the virus circulates between the sylvatic and domestic compartments. If there are indeed interconnections, several hypotheses need to be tested: identification of the insect vectors feeding on both wild and domestic animals; evaluation of the overlapping areas/ecosystems where animals may be exposed to common vectors; assessment of the mobility of these animals and characterization of the compartment where the virus emerge.

Further studies need to be carried out to understand how RVFV circulates in the forest environment of Central Africa—which is at the crossroads between West and East Africa—to investigate the sylvatic circulation of RVFV in Central African rainforests. A larger sample size can help confirm our seroprevalence results in wild and domestic animals. Additional studies on mosquitoes should also be carried out. Because we did not know whether the virus was circulating in this region before the domestic animals and wildlife samples were processed, no mosquito sampling had been planned in the study area. The study should also be extended to humans to better characterize the circulation and transmission of RVFV in this region of Gabon. These studies are needed to explore the mechanisms by which the virus shifts from its sylvatic compartment to an anthropic one, *i.e.* transmission to domestic animals and humans in village.

This preliminary study emphasizes the need to develop adequate event-based surveillance and control measures to limit the threat of RVF, such as awareness campaigns for villagers to report unusual deaths or abortions in domestic and wild ruminants and on the risk of RVFV infection through the manipulation of aborted fetuses, if clinical cases occur. Limiting the movement of livestock can also be proposed as a control measure. Therefore, further

virological and serological dynamic surveys to investigate RVFV circulation (wet and dry seasons) in domestic animals, wildlife, hematophagous arthropods and in humans can lead to a better understanding of RVFV circulation in the forest ecosystem of the Congo Basin.

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References

1. Pastoret PP, Vallat B. Essential veterinary education in infectious diseases of livestock and related scientific disciplines. *Rev Sci Tech.* 2010/02/05 ed. 2009; 28: 537–44. <https://doi.org/10.20506/rst.28.2.1895> PMID: 20128461
2. Gould E, Pettersson J, Higgs S, Charrel R, de Lamballerie X. Emerging arboviruses: Why today? *One Health.* 2017/08/09 ed. 2017; 4: 1–13. <https://doi.org/10.1016/j.oneht.2017.06.001> PMID: 28785601
3. GIDEON. GIDEON The world's premier global infectious diseases database, 2009. Available from: <http://web.gideononline.com/web/epidemiology/>
4. Bourgairel M, Wauquier N, Gonzalez JP. Emerging viral threats in Gabon: health capacities and response to the risk of emerging zoonotic diseases in Central Africa. *Emerg Health Threats J.* 2010/01/01 ed. 2010; 3: e7. <https://doi.org/10.3134/ehj.10.163> PMID: 22460397

5. (WOAH) WO for AH. Rift Valley fever. Available from: <https://www.woah.org/en/disease/rift-valley-fever/>
6. Javelle E, Lesueur A, Pommier de Santi V, de Laval F, Lefebvre T, Holweck G, et al. The challenging management of Rift Valley Fever in humans: literature review of the clinical disease and algorithm proposal. *Ann Clin Microbiol Antimicrob*. 2020/01/24 ed. 2020; 19: 4. <https://doi.org/10.1186/s12941-020-0346-5> PMID: 31969141
7. Anywaine Z, Lule SA, Hansen C, Warimwe G, Elliott A. Clinical manifestations of Rift Valley fever in humans: Systematic review and meta-analysis. *PLoS Negl Trop Dis*. 2022/03/26 ed. 2022; 16: e0010233. <https://doi.org/10.1371/journal.pntd.0010233> PMID: 35333856
8. Clark MHA, Warimwe GM, Di Nardo A, Lyons NA, Gubbins S. Systematic literature review of Rift Valley fever virus seroprevalence in livestock, wildlife and humans in Africa from 1968 to 2016. *PLoS Negl Trop Dis*. 2018/07/24 ed. 2018; 12: e0006627. <https://doi.org/10.1371/journal.pntd.0006627> PMID: 30036382
9. Tchouassi DP, Bastos AD, Sole CL, Diallo M, Lutomiah J, Mutisya J, et al. Population genetics of two key mosquito vectors of Rift Valley Fever virus reveals new insights into the changing disease outbreak patterns in Kenya. *PLoS Negl Trop Dis*. 2014/12/05 ed. 2014; 8: e3364. <https://doi.org/10.1371/journal.pntd.0003364> PMID: 25474018
10. Tantely LM, Boyer S, Fontenille D. A review of mosquitoes associated with Rift Valley fever virus in Madagascar. *Am J Trop Med Hyg*. 2015/03/04 ed. 2015; 92: 722–9. <https://doi.org/10.4269/ajtmh.14-0421> PMID: 25732680
11. Metras R, Edmunds WJ, Youssouffi C, Dommergues L, Fournie G, Camacho A, et al. Estimation of Rift Valley fever virus spillover to humans during the Mayotte 2018–2019 epidemic. *Proc Natl Acad Sci U A*. 2020/09/16 ed. 2020; 117: 24567–24574. <https://doi.org/10.1073/pnas.2004468117> PMID: 32929025
12. Daubney R, Hudson JR, Graham PC. Enzootic hepatitis of Rift Valley Fever, an undescribed virus disease of sheep, cattle and man from Eastern Africa. *J Pathol Bacteriol*. 1931; 34: 545–579.
13. Laughlin LW, Meegan JM, Strausbaugh LJ, Morens DM, Watten RH. Epidemic Rift Valley fever in Egypt: observations of the spectrum of human illness. *Trans R Soc Trop Med Hyg*. 1979/01/01 ed. 1979; 73: 630–3. [https://doi.org/10.1016/0035-9203\(79\)90006-3](https://doi.org/10.1016/0035-9203(79)90006-3) PMID: 575446
14. Walsh J. Rift Valley fever rears its head. *Science*. 1988/06/10 ed. 1988; 240: 1397–9. <https://doi.org/10.1126/science.3375824> PMID: 3375824
15. Kwasnik M, Rozek W, Rola J. Rift Valley Fever—a Growing Threat To Humans and Animals. *J Vet Res*. 2021/04/06 ed. 2021; 65: 7–14. <https://doi.org/10.2478/jvetres-2021-0009> PMID: 33817390
16. Maurice Y. Premières constatations sérologiques sur l'incidence de la maladie de Wesselsbron et de la fièvre de la vallée du Rift chez les ovins et les ruminants sauvages du Tchad et du Cameroun. *Rev Elev Med Vet Pays Trop*. 1967; 20 (3): 395–405.
17. Zeller HG, Bessin R, Thiongane Y, Bapetel I, Teou K, Ala MG, et al. Rift Valley fever antibody prevalence in domestic ungulates in Cameroon and several west African countries (1989–1992) following the 1987 Mauritanian outbreak. *Res Virol*. 1995/01/01 ed. 1995; 146: 81–5. [https://doi.org/10.1016/0923-2516\(96\)80593-0](https://doi.org/10.1016/0923-2516(96)80593-0) PMID: 7754240
18. Sado FY, Tchegnna HS, Kamgang B, Djonabaye D, Nakoune E, McCall PJ, et al. Seroprevalence of Rift Valley fever virus in domestic ruminants of various origins in two markets of Yaounde, Cameroon. *PLoS Negl Trop Dis*. 2022/08/12 ed. 2022; 16: e0010683. <https://doi.org/10.1371/journal.pntd.0010683> PMID: 35951644
19. Ebogo-Belobo JT, Sadeuh-Mba SA, Mveng-Sanding GMA, Chavely GM, Groschup MH, Mbacham WF, et al. Serological evidence of the circulation of the Rift Valley fever virus in sheep and goats slaughtered in Yaounde, Cameroon. *Vet Med Sci*. 2022/05/21 ed. 2022; 8: 2114–2118. <https://doi.org/10.1002/vms3.848> PMID: 35594478
20. Gonzalez JP, Josse R, Johnson ED, Merlin M, Georges AJ, Abandja J, et al. Antibody prevalence against haemorrhagic fever viruses in randomized representative Central African populations. *Res Virol*. 1989/07/01 ed. 1989; 140: 319–31. [https://doi.org/10.1016/s0923-2516\(89\)80112-8](https://doi.org/10.1016/s0923-2516(89)80112-8) PMID: 2505350
21. Findlay G, Stefanopoulo G, Callum F. Présence d'anticorps contre le virus de la fièvre de la vallée du Rift dans le sang des africains. *Bull Soc Path Ex*. 1936; 29, 986.
22. Shimshony A, Barzilai R. Rift Valley fever. *Adv Vet Sci Comp Med*. 1983/01/01 ed. 1983; 27: 347–425. Available from: <https://www.ncbi.nlm.nih.gov/pubmed/6359835> PMID: 6359835
23. Maganga GD, Abessolo Ndong AL, Mikala Okouyi CS, Makiala Mandanda S, N'Diimabaka N, Pinto A, et al. Serological Evidence for the Circulation of Rift Valley Fever Virus in Domestic Small Ruminants in Southern Gabon. *Vector Borne Zoonotic Dis*. 2017/04/08 ed. 2017; 17: 443–446. <https://doi.org/10.1089/vbz.2016.2065> PMID: 28387600

24. Tshilenge GM, Mulumba MLK, Misinzo G, Noad R, Dundon WG. Rift Valley fever virus in small ruminants in the Democratic Republic of the Congo. *Onderstepoort J Vet Res*. 2019/11/13 ed. 2019; 86: e1–e5. <https://doi.org/10.4102/ojvr.v86i1.1737> PMID: 31714136
25. Halawi AD, Saasa N, Pongombo BL, Kajihara M, Chambaro HM, Hity M, et al. Seroprevalence of Rift Valley fever in cattle of smallholder farmers in Kwilu Province in the Democratic Republic of Congo. *Trop Anim Health Prod*. 2019/06/30 ed. 2019; 51: 2619–2627. <https://doi.org/10.1007/s11250-019-01978-z> PMID: 31250252
26. Digoutte JP. Rapport sur le fonctionnement technique de l'Institut Pasteur de Dakar. Année 1985. 1986.
27. Pourrut X, Nkoghe D, Souris M, Paupy C, Paweska J, Padilla C, et al. Rift Valley fever virus seroprevalence in human rural populations of Gabon. *PLoS Negl Trop Dis*. 2010; 4: e763. <https://doi.org/10.1371/journal.pntd.0000763> PMID: 20668541
28. LeBreton M, Umlauf S, Djoko CF, Daszak P, Burke DS, Kwenkam PY, et al. Rift Valley fever in goats, Cameroon. *Emerg Infect Dis*. 2006/05/24 ed. 2006; 12: 702–3. <https://doi.org/10.3201/eid1204.051428> PMID: 16715582
29. Cordelier R, Geoffroy B. Les moustiques de république Centrafricaine: Distribution, abondance et fréquence des Culicidés dans l'ouest du pays. Les arbovirus isolés. *Trav Doc L'Orstom*. 1976; 49: 105.
30. Digoutte JP, Jacobi JC, Robin Y, Gagnard VJ. [Zinga virus infection in man]. *Bull Soc Pathol Exot Fil*. 1974/09/01 ed. 1974; 67: 451–7. Available from: <https://www.ncbi.nlm.nih.gov/pubmed/4480466>.
31. Nakounne E, Selekon B, Morvan J. [Microbiological surveillance: viral hemorrhagic fever in Central African Republic: current serological data in man]. *Bull Soc Pathol Exot*. 2002/01/05 ed. 2000; 93: 340–7. Available from: <https://www.ncbi.nlm.nih.gov/pubmed/11775321>.
32. Bird BH, Bawiec DA, Ksiazek TG, Shoemaker TR, Nichol ST. Highly sensitive and broadly reactive quantitative reverse transcription-PCR assay for high-throughput detection of Rift Valley fever virus. *J Clin Microbiol*. 2007/09/07 ed. 2007; 45: 3506–13. <https://doi.org/10.1128/JCM.00936-07> PMID: 17804663
33. Johnston AR, Morikawa ML, Ntie S, Anthony NM. Evaluating DNA barcoding criteria using African duiker antelope (Cephalophinae) as a test case. *Conserv Genet*. 2011; 12: 1173–1182.
34. Bird BH, Khristova ML, Rollin PE, Ksiazek TG, Nichol ST. Complete genome analysis of 33 ecologically and biologically diverse Rift Valley fever virus strains reveals widespread virus movement and low genetic diversity due to recent common ancestry. *J Virol*. 2006/12/29 ed. 2007; 81: 2805–16. <https://doi.org/10.1128/JVI.02095-06> PMID: 17192303
35. Posada D, Crandall KA. MODELTEST: testing the model of DNA substitution. *Bioinformatics*. 1999/01/27 ed. 1998; 14: 817–8. <https://doi.org/10.1093/bioinformatics/14.9.817> PMID: 9918953
36. Guindon S, Dufayard JF, Lefort V, Anisimova M, Hordijk W, Gascuel O. New algorithms and methods to estimate maximum-likelihood phylogenies: assessing the performance of PhyML 3.0. *Syst Biol*. 2010/06/09 ed. 2010; 59: 307–21. <https://doi.org/10.1093/sysbio/syq010> PMID: 20525638
37. Kortekaas J, Kant J, Vloet R, Cetre-Sossah C, Marianneau P, Lacote S, et al. European ring trial to evaluate ELISAs for the diagnosis of infection with Rift Valley fever virus. *J Virol Methods*. 2012/10/02 ed. 2013; 187: 177–81. <https://doi.org/10.1016/j.jviromet.2012.09.016> PMID: 23022325
38. OIE. Manual of standards for diagnostic tests and vaccines for terrestrial animals (5th ed.). *Off Int Epizoot Paris Fr Pp* 185–194. 2004.
39. Grueber CE, Nakagawa S, Laws RJ, Jamieson IG. Multimodel inference in ecology and evolution: challenges and solutions. *J Evol Biol*. 2011/01/29 ed. 2011; 24: 699–711. <https://doi.org/10.1111/j.1420-9101.2010.02210.x> PMID: 21272107
40. Bates D, Maechler M, Bolker B, Walker S. Linear Mixed-Effects Models using lme4. *J Stat Softw*. 2015;51.
41. Grobelaar AA, Weyer J, Leman PA, Kemp A, Paweska JT, Swanepoel R. Molecular epidemiology of Rift Valley fever virus. *Emerg Infect Dis*. 2011/12/17 ed. 2011; 17: 2270–6. <https://doi.org/10.3201/eid1712.111035> PMID: 22172568
42. Obame-Nkoghe J, Roiz D, Ngangue MF, Costantini C, Rahola N, Jiolle D, et al. Towards the invasion of wild and rural forested areas in Gabon (Central Africa) by the Asian tiger mosquito *Aedes albopictus*: Potential risks from the one health perspective. *PLoS Negl Trop Dis*. 2023/08/16 ed. 2023; 17: e0011501. <https://doi.org/10.1371/journal.pntd.0011501> PMID: 37585443
43. Makanga B, Costantini C, Rahola N, Yangari P, Rougeron V, Ayala D, et al. “Show me which parasites you carry and I will tell you what you eat”, or how to infer the trophic behavior of hematophagous arthropods feeding on wildlife. *Ecol Evol*. 2017/10/19 ed. 2017; 7: 7578–7584. <https://doi.org/10.1002/ece3.2769> PMID: 29043015
44. Ondo-Eyeghe A. La dot en Afrique: Cas pratique du Gabon. Éditions universitaires européennes; 2020.

45. Ross TM, Bhardwaj N, Bissel SJ, Hartman AL, Smith DR. Animal models of Rift Valley fever virus infection. *Virus Res.* 2011/11/17 ed. 2012; 163: 417–23. <https://doi.org/10.1016/j.virusres.2011.10.023> PMID: 22086058
46. Roulon-Doko P. The status of the goat in a society of hunters-gatherers-farmers, The case of the Gbaya 'bodo' of C.A.R. *J Afr.* 2008; 183–195. <https://doi.org/10.4000/africanistes.2504>
47. Service MW. The attraction of mosquitoes by animal baits in the Northern Guinea Savannah of Nigeria. *J Ent Soc Sth Afr.* 1964; 27, 29–36.
48. Lutomiah J, Omondi D, Masiga D, Mutai C, Mireji PO, Ongus J, et al. Blood meal analysis and virus detection in blood-fed mosquitoes collected during the 2006–2007 Rift Valley fever outbreak in Kenya. *Vector Borne Zoonotic Dis.* 2014/09/18 ed. 2014; 14: 656–64. <https://doi.org/10.1089/vbz.2013.1564> PMID: 25229704
49. Tchouassi DP, Okiro RO, Sang R, Cohnstaedt LW, McVey DS, Torto B. Mosquito host choices on live-stock amplifiers of Rift Valley fever virus in Kenya. *Parasit Vectors.* 2016/04/03 ed. 2016; 9: 184. <https://doi.org/10.1186/s13071-016-1473-x> PMID: 27036889
50. Hoogstraal H, Meegan JM, Khalil GM, Adham FK. The Rift Valley fever epizootic in Egypt 1977–78. 2. Ecological and entomological studies. *Trans R Soc Trop Med Hyg.* 1979/01/01 ed. 1979; 73: 624–9. [https://doi.org/10.1016/0035-9203\(79\)90005-1](https://doi.org/10.1016/0035-9203(79)90005-1) PMID: 44038
51. House C, Alexander KA, Kat PW, O'Brien SJ, Mangiafico J. Serum antibody to Rift Valley fever virus in African carnivores. *Ann N Acad Sci.* 1996/07/23 ed. 1996; 791: 345–9. <https://doi.org/10.1111/j.1749-6632.1996.tb53541.x> PMID: 8784515
52. Stoek F, Barry Y, Ba A, Schulz A, Rissmann M, Wylezich C, et al. Mosquito survey in Mauritania: Detection of Rift Valley fever virus and dengue virus and the determination of feeding patterns. *PLoS Negl Trop Dis.* 2022/04/16 ed. 2022; 16: e0010203. <https://doi.org/10.1371/journal.pntd.0010203> PMID: 35427361
53. Sousa CA, Pinto J, Almeida AP, Ferreira C, do Rosario VE, Charlwood JD. Dogs as a favored host choice of *Anopheles gambiae* sensu stricto (Diptera: Culicidae) of Sao Tome West Africa. *J Med Entomol.* 2001/03/28 ed. 2001; 38: 122–5. <https://doi.org/10.1603/0022-2585-38.1.122> PMID: 11268684
54. Wanji S, Tanke T, Atanga SN, Ajonina C, Nicholas T, Fontenille D. *Anopheles* species of the mount Cameroon region: biting habits, feeding behaviour and entomological inoculation rates. *Trop Med Int Health.* 2003/06/28 ed. 2003; 8: 643–9. <https://doi.org/10.1046/j.1365-3156.2003.01070.x> PMID: 12828548
55. Pereira-Dos-Santos T, Roiz D, Lourenco-de-Oliveira R, Paupy C. A Systematic Review: Is *Aedes albopictus* an Efficient Bridge Vector for Zoonotic Arboviruses? *Pathogens.* 2020/04/11 ed. 2020; 9. <https://doi.org/10.3390/pathogens9040266> PMID: 32272651