



## Brief Report

Identification of novel *Theileria* genotypes from Grant's gazelleJanis Hooge<sup>a</sup>, Laryssa Howe<sup>a</sup>, Vanessa O. Ezenwa<sup>b,\*</sup><sup>a</sup> Institute of Veterinary, Animal and Biomedical Sciences, Massey University, Palmerston North 4442, New Zealand<sup>b</sup> Odum School of Ecology and Department of Infectious Diseases, College of Veterinary Medicine, University of Georgia, Athens, USA

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## ABSTRACT

Blood samples collected from Grant's gazelles (*Nanger granti*) in Kenya were screened for hemoparasites using a combination of microscopic and molecular techniques. All 69 blood smears examined by microscopy were positive for hemoparasites. In addition, *Theileria/Babesia* DNA was detected in all 65 samples screened by PCR for a ~450-base pair fragment of the V4 hypervariable region of the 18S rRNA gene. Sequencing and BLAST analysis of a subset of PCR amplicons revealed widespread co-infection (25/39) and the existence of two distinct Grant's gazelle *Theileria* subgroups. One group of 11 isolates clustered as a subgroup with previously identified *Theileria ovis* isolates from small ruminants from Europe, Asia and Africa; another group of 3 isolates clustered with previously identified *Theileria* spp. isolates from other African antelope. Based on extensive levels of sequence divergence (1.2–2%) from previously reported *Theileria* species within Kenya and worldwide, the *Theileria* isolates detected in Grant's gazelles appear to represent at least two novel *Theileria* genotypes.

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## 1. Introduction

*Theileria* and *Babesia* (Phylum: Apicomplexa) are two of the most economically important hemoparasites of livestock. Species in both genera are vectored by ticks and infect host erythrocytes. The diversity and distribution of *Theileria* and *Babesia* species in cattle have been studied in detail (Bishop et al., 2004; Bock et al., 2004), and with the advent of new molecular techniques, an increasing number of studies are characterizing these organisms in wild herbivores. For example, recent studies on African ungulates have examined *Theileria* or *Babesia* genotype diversity in buffalo (*Syncerus caffer* (Chaisi et al., 2011, 2014; Mans et al., 2011)), sable and roan antelope (*Hippotragus niger* and *H. equinus* (Oosthuizen et al., 2008, 2009)), tsessebe (*Damaliscus lunatus* (Brothers et al., 2011)), waterbuck (*Kobus defassa* (Githaka et al., 2014)), giraffe (*Giraffa camelopardalis* (Oosthuizen et al., 2009; Githaka et al., 2013)), and zebras (*Equus quagga* and *E. zebra* (Bhoora et al., 2010)). Novel parasite genotypes were reported in several of these hosts, suggesting that the full diversity of *Theileria* and *Babesia* species infecting wild African herbivores is only beginning to be understood.

In this study, we evaluated the occurrence of *Theileria* and *Babesia* species in Grant's gazelle (*Nanger granti*), a common antelope species found in East Africa. Grant's gazelle occupy similar habitat as a range

of plains herbivore species, and in some regions they also overlap extensively with livestock. A serological survey of Kenyan wildlife and livestock for the rickettsial hemoparasite, *Anaplasma*, found that four of five Grant's gazelles tested were positive for *Anaplasma* antibodies (Ngeranwa et al., 2008). Beyond this report, there is very little information on the hemoparasites infecting Grant's gazelle. Thus, we used a combination of microscopic and molecular techniques to estimate hemoparasite infection prevalence in this species and to identify target *Theileria* and/or *Babesia* parasites.

## 2. Materials and methods

## 2.1. Animal sampling

Grant's gazelles  $\geq 2$  years of age were captured at the Mpala Research Center (MRC), Laikipia, Kenya (0°17'N, 37°52'E) in January, February, and August 2009. Animals were captured using drive nets (Jan–Feb) or by helicopter using a hand-held net gun fired from the aircraft (August). Blood was collected from the jugular vein into 10 ml heparin tubes. Samples were kept on ice until transported to the laboratory where they were stored at –20 °C in the lab until processing. A 5-mm biopsy sample was also taken from the ear of each animal, and a residual drop of blood from the ear vein was collected into a capillary tube and used to make blood smears. Hematocrit (HCT) levels were used to estimate the possible impact of hemoparasite infection on anemia. To measure HCT, capillary tubes filled with heparin blood were spun in a microhematocrit centrifuge for 5 min and HCT values were estimated using a microhematocrit reader card. Animal protocols were approved by the University of

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Montana (#023-09VEDBS-051509) and the University of Georgia (#A2010 10–188) Animal Care and Use Committees.

## 2.2. Microscopic analysis

Immediately after collection of peripheral blood into capillary tubes, a thin smear was prepared on a glass slide, air-dried, and fixed with 100% methanol. Fixed slides were transported back to the laboratory where all slides were stained with Giemsa for 45 minutes. Presence or absence of infection was determined by examining each smear under oil immersion ( $\times 1000$ ) for up to 90 minutes concentrating on the monolayer. For each smear, the intensity of infection (i.e. the number of hemoparasites) was quantified by counting all organisms observed in the first 10 fields of view. Smears without any parasites in the first 10 fields were classified as having low infection intensities, and smears with parasites were classified as having high intensities.

## 2.3. Molecular analysis

DNA was extracted from 100  $\mu$ l of heparinized blood samples using the Qiagen DNeasy Kit (Qiagen, CA, USA) following the manufacturer's instructions. All extracted DNA samples were kept at  $-20^{\circ}\text{C}$  until further analysis was performed. A  $\sim 450$ -base pair (bp) fragment of the V4 hypervariable region of the 18S ribosomal RNA (rRNA) gene was amplified using primers RLB F2 [5'-GAC ACA GGG AGG TAG TGA CAA G-3'] and RLB R2 [5'-CTA AGA ATT TCA CCT CTA ACA GT-3'] to identify *Babesia* and *Theileria* species as described by Gubbels et al. (1999) with the following modifications. Briefly, a PCR amplification was performed in 50  $\mu$ l reactions containing approximately 50 ng of template genomic DNA, 0.2  $\mu$ M of each of the forward and reverse primer, 0.2  $\mu$ M of dNTP, 5  $\mu$ l 10X PCR buffer, 1.5 mM of  $\text{MgCl}_2$ , and 0.2  $\mu$ l (1 unit) of Platinum Taq DNA polymerase (Invitrogen, CA, USA). For negative controls, an equivalent volume of purified water was substituted for DNA. PCR reaction conditions involved a 4 minute initial denaturation at  $94^{\circ}\text{C}$ , followed by 35 cycles of  $94^{\circ}\text{C}$  for 20 seconds,  $57^{\circ}\text{C}$  for 30 seconds, and  $72^{\circ}\text{C}$  for 30 seconds and a final 10 minute extension at  $72^{\circ}\text{C}$ . PCR products were run on a 1.5% (w/v) ultra-pure agarose gel (Invitrogen) containing ethidium bromide at 100V for 45 minutes and visualized under UV light on a transilluminator. A subset of positive PCR amplicon samples was purified (PureLink PCR purification kit, Invitrogen) and subjected to automatic dye-terminator cycle sequencing with BigDye<sup>TM</sup> Terminator Version 3.1 Ready Reaction Cycle Sequencing kit and the ABI3730 Genetic Analyzer (Applied Biosystems, CA, USA) using both forward and reverse primers. Sequencing results were compared to the NCBI BLAST database to confirm *Theileria* or *Babesia* DNA amplification and identify relatedness.

Sequences from three gazelle isolates (GG1, GG2, and GG3) were aligned using Clustal W (Thompson et al., 1994) with 33 representative *Theileria* sp. sequences and a *Toxoplasma gondii* sequence available in GenBank and trimmed to the same length (400 base pairs) using Geneious Pro 4.8.5<sup>TM</sup> (Biomatters, Auckland, New Zealand). A Bayesian phylogenetic tree was generated in MrBayes version 3.1 (Ronquist and Huelsenbeck, 2003) using a general time-reversible model including invariable sites (GTR +I). The Bayesian phylogeny was obtained using 1 cold and 3 hot Monte Carlo Markov chains, which were sampled every 1000 generations over 2 million generations. Of these trees, 25% were discarded as burn-in material. The remaining trees were used to construct a majority consensus tree. Bootstrap percentages from the Bayesian analysis were added to the tree at the appropriate nodes. The sequence divergence between and within the different lineages was calculated using a Jukes–Cantor model of substitution implemented in the program PAUP\* 4.0 Beta version (Swofford, 2002). Sequences reported in this

paper are available in GenBank under accession numbers: KP641655, KP641656 and KP641657.

## 3. Results

The blood smears of 69 individuals (41 males, 28 females) were examined for hemoparasites and at least one organism was detected in all of the smears. Some positive samples contained single organisms while others had paired or multiple organisms (Fig. 1); however, the parasites could not be distinguished taxonomically with this method. A smaller number of samples (12 of 69) had quantifiable levels of infection. A total of 83.3% (10 of 12) of these samples were males and 16.4% (2 of 12) were females. Infection intensity in these samples ranged from 1 to 15 parasites per 10 fields of view (mean = 5.2). There was no evidence that infection intensity was associated with differences in hematocrit (2-way ANOVA controlling for host sex: mean HCT (high) = 47.3 [n = 10], mean HCT (low) = 47.2 [n = 55],  $p = 0.93$ ).

Of the 69 samples examined by microscopy, 60 were available for molecular analysis. Five additional samples, for which we did not have smears, were available for PCR so in total we examined 65 samples by PCR. As with the smears, 100% of the blood samples tested by PCR were positive for hemoparasites (*Babesia/Theileria* spp.). Sequencing a random subset of the PCR amplicons revealed that the majority of samples (25 of 39 sequenced samples) had conspicuous overlapping peaks suggestive of co-infection. The conserved nature of the first 100 base pairs of the amplicons from co-infected animals confirmed the presence of multiple *Theileria* spp. Of the 14 non-coinfected amplicons, a BLAST search revealed a group of 11 isolates (represented by Grant's Gazelle [GG]1 and GG2 in Fig. 2) which were most similar (99%) to a large number of previously identified *Theileria ovis* isolates including those from Spain (GenBank DQ866845), Egypt (GenBank AB986194), Turkey, Tanzania, Sudan (GenBank AY260172, AY260173, AY260171 respectively) and China (GenBank FJ603460). A comparison of 18S rRNA sequences derived from these 11 gazelle isolates showed minor levels of heterogeneity (0.0–0.3%) at the nucleotide level (GG1 and GG2: Fig. 2, Table 1). Sequence divergence analysis also revealed extensive heterogeneity of 1.2–1.7% when compared to representative *T. ovis* isolates within

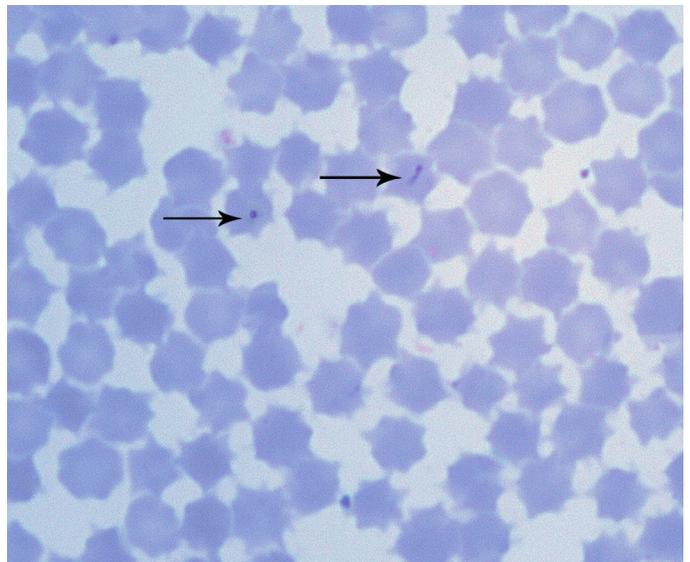
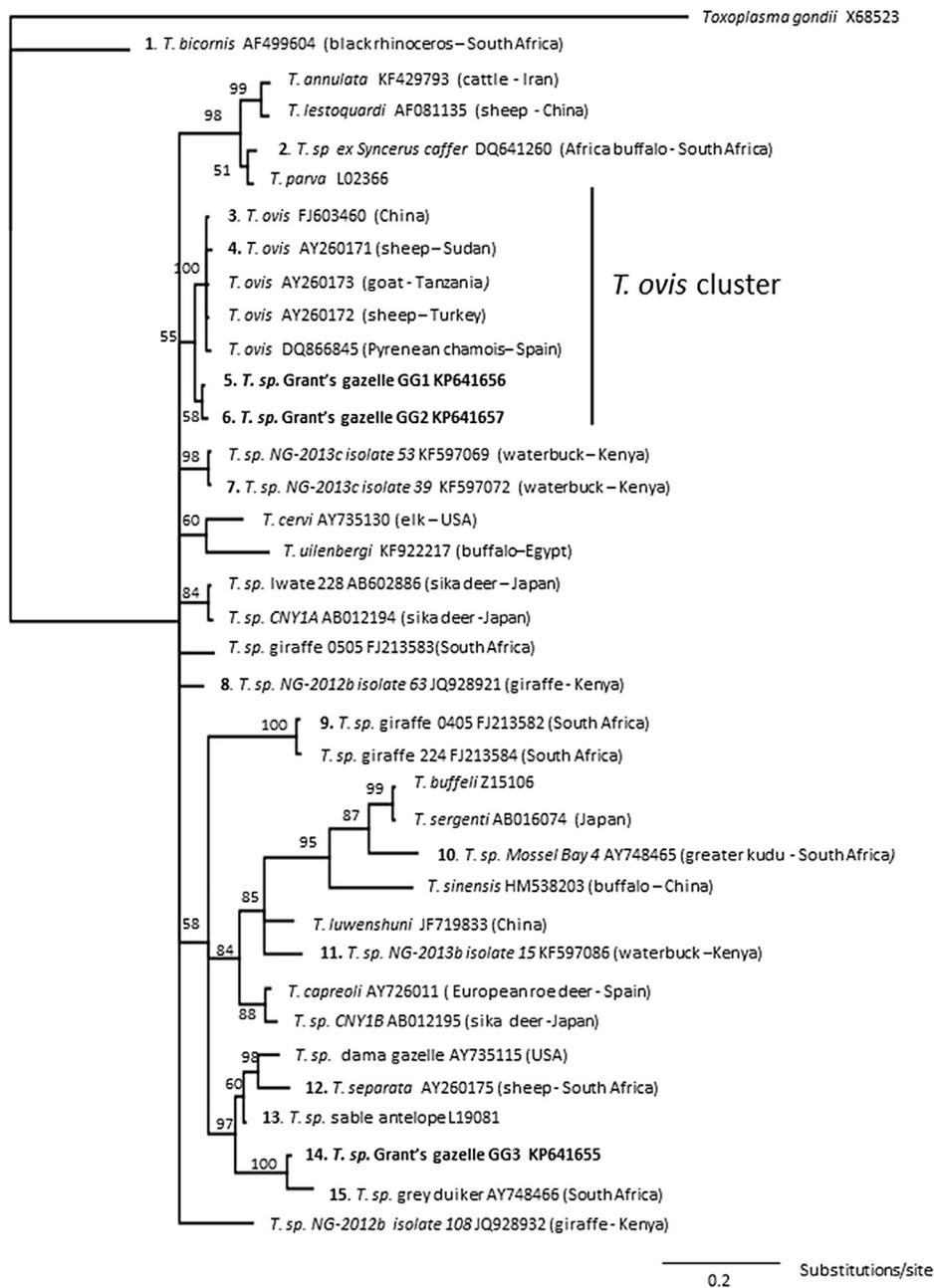


Fig. 1. Light microscopy of a blood smear stained with Giemsa showing single and paired hemoparasites (highlighted by arrows).



**Fig. 2.** Phylogenetic analysis of *Theileria* genotypes isolated from Grant's gazelles (*Nanger granti*) in Kenya. Bayesian analysis of a 400 nucleotide fragment of the 18S ribosomal RNA gene from 1 sequence of *Toxoplasma gondii*, 33 *Theileria* sequences from GenBank, and 3 representative *Theileria* sequences from Grant's gazelles (GG1, GG2, GG3) in this study (bold font). The tree is rooted on the lineage of *T.gondii*. Numbers above the branches indicate bootstrap support based on 1000 replicates. Host species, geographic location of isolation, and GenBank accession numbers of the sequences are provided where known. Numbered sequences are listed in Table 1.

the same cluster, suggesting that the gazelle isolates form a moderately supported (posterior probability of 0.55) distinct subgroup within the *T. ovis* cluster (Fig. 2, Table 1). When compared to other *Theileria* isolates from Kenya, the *T. ovis*-like isolates from Grant's gazelle had a sequence divergence of 3.8–5.2% from representative waterbuck isolates (GenBank KF597069, KF59707272 and KF597086) and 4.9–5.2% from a representative giraffe isolate (GenBank JQ928921) (Table 1).

Three out of fourteen of the non-coinfected gazelle isolates (represented by GG3 in Fig. 2) were most similar (98%) to a *Theileria* sp. isolated from a common grey duiker (*Sylvicapra grimmia*) in South Africa (GenBank AY748466). Our phylogenetic reconstruction revealed that GG3 clusters with isolates identified from several species

of antelope (posterior probability of 97%); however, extensive heterogeneity ranging from 2.0% to 5.3% (GenBank AY748466, L19081, AY260175 and AY735115) was present, suggesting a significant level of diversity within the cluster (Fig. 2, Table 1). In addition, there is 5.8–6.1% sequence divergence between isolate GG3 and the two *T. ovis*-like isolates GG1 and GG2, suggesting the presence of at least two distinct *Theileria* genotypes circulating within a single population of Grant's gazelles.

#### 4. Discussion

We found evidence of widespread *Theileria* infections in Grant's gazelles from Kenya. This finding represents the first molecular-based

**Table 1**  
The sequence divergence (%) between 15 18S ribosomal RNA gene lineages of *Theileria*.

	1 <sup>a</sup>	2	3	4	5	6	7	8	9	10	11	12	13	14	15
1. <i>T. bicornis</i>	0.0 <sup>b</sup>	–	–	–	–	–	–	–	–	–	–	–	–	–	–
2. <i>T. sp. S. caffer</i>	10.3	0.0	–	–	–	–	–	–	–	–	–	–	–	–	–
3. <i>T. ovis</i> China	11.3	5.3	0.0	–	–	–	–	–	–	–	–	–	–	–	–
4. <i>T. ovis</i> Sudan	11.6	5.3	0.3	0.0	–	–	–	–	–	–	–	–	–	–	–
5. <b><i>T. sp. GG1</i></b>	10.2	5.0	1.2	1.5	0.0	–	–	–	–	–	–	–	–	–	–
6. <b><i>T. sp. GG2</i></b>	9.9	5.0	1.4	1.7	0.3	0.0	–	–	–	–	–	–	–	–	–
7. <i>T. sp. waterbuck</i> 39	11.7	6.2	4.1	4.4	3.8	3.8	0.0	–	–	–	–	–	–	–	–
8. <i>T. sp. giraffe</i> 63	11.3	7.3	5.2	5.5	4.9	5.2	5.2	0.0	–	–	–	–	–	–	–
9. <i>T. sp. giraffe</i> 0405	12.7	7.9	6.1	6.4	6.1	6.4	7.9	7.8	0.0	–	–	–	–	–	–
10. <i>T. sp. Mossel Bay</i>	12.0	8.6	8.2	8.5	7.6	7.6	7.4	10.2	10.2	0.0	–	–	–	–	–
11. <i>T. sp. waterbuck</i> 15	11.5	6.9	5.6	5.9	4.4	4.7	6.3	7.1	7.1	8.0	0.0	–	–	–	–
12. <i>T. separata</i>	12.6	8.0	6.1	6.4	5.3	5.6	6.4	8.5	7.9	8.0	6.3	0.0	–	–	–
13. <i>T. sp. sable antelope</i>	12.3	6.4	4.7	4.9	3.8	4.1	5.6	6.1	6.1	8.5	5.1	2.9	0.0	–	–
14. <b><i>T. sp. GG3</i></b>	13.4	8.2	6.7	7.0	5.8	6.1	7.3	8.4	7.0	8.8	6.2	4.1	3.8	0.0	–
15. <i>T. sp. grey duiker</i>	13.1	8.3	7.0	7.3	6.1	6.1	8.5	9.3	7.9	8.8	7.2	5.3	4.7	2.0	0.0

<sup>a</sup> Numbers correspond to the number assigned to each lineage in Fig. 2 where a phylogeny of the parasites and GenBank accession numbers are given. Shading indicates *T. ovis* cluster.

<sup>b</sup> Sequence divergence was calculated using a Jukes–Cantor model of substitution. Lineages in bold are from this study.

report of this parasite group in the species. Historically, phylogenetic studies on apicomplexans have used the 18S rRNA gene which is highly conserved and enables genome analysis of differences between strains and species (Katzer et al., 1998; Allsopp and Allsopp, 2006; Lack et al., 2012). Here, we found that although organisms identified from Grant's gazelle clustered with known *Theileria* spp., they formed distinct sub-clades, with at least 1.2–2% nucleotide heterogeneity, that were divergent from previously reported species. In addition, the *Theileria* genotypes we detected in Grant's gazelles were clearly distinct from organisms that have been seen in other wild herbivore species in Kenya (Githaka et al., 2013, 2014). These results are consistent with previous studies that have examined variation in the 18S rRNA gene of various *Theileria* species and observed extensive (1.5–1.7%) sequence heterogeneity even within the same clade grouping (Mans et al., 2011; Chaisi et al., 2013, 2014). We also observed slight sequence heterogeneity (0.0–0.3%) between the *T. ovis*-like gazelle isolates themselves, suggesting that additional genetic markers may be needed (e.g. ITS or heat-shock proteins) to get a complete picture of the level of diversity within this lineage (Githaka et al., 2014; He et al., 2014).

It is not uncommon to find several distinct genotypes or species of *Theileria* present in one host species in the same region. For example, more than one distinct *Theileria* species was identified from waterbuck and giraffes in Kenya and South Africa (Chaisi et al., 2013, 2014; Githaka et al., 2013, 2014). In Grant's gazelles, the high prevalence of co-infection we observed (64% of samples) may reflect frequent habitat or host overlap between the two *Theileria* genotypes identified in this study and abundant opportunities for cross-species parasite transmission via shared vectors (Berggoetz et al., 2014). At our study site, Grant's gazelles co-mingle with up to 20 different species of wild ungulates (Ezenwa, 2003). More generally, patterns of interspecific association between Grant's and other herbivore species suggest that these gazelles occupy a central role in multi-species interaction networks which might facilitate cross-species parasite transmission (VanderWaal et al., 2014). This feature of gazelle behavior may also increase the opportunity for parasite recombination events to occur in generalist tick vectors resulting in minor levels of *Theileria* genome heterogeneity (Aktas et al., 2007; Katzer et al., 2011) as observed between the *T. ovis*-like isolates GG1 and GG2.

Although all of the individuals in our study were infected with *Theileria*, infection intensities were typically low. Only 17% of individuals had quantifiable levels of parasitemia, and roughly five times more males had detectable levels of infection when compared

to females. This sex difference could be a consequence of differences in tick exposure, or a function of higher male susceptibility to hemoparasite infection. The latter hypothesis is supported by previous studies showing that certain aspects of male gazelle behavior and physiology may increase their susceptibility to other parasite infections (Ezenwa, 2004; Ezenwa et al., 2012).

Based on a comparison of hematocrit levels, we found no evidence that high *Theileria* infection intensities were associated with anemia in Grant's gazelles. This result is not surprising, given that *T. ovis* isolates closely related to those we found in this study have been found to have low pathogenicity in livestock (Nagore et al., 2004; Gebrekidan et al., 2014). A lack of overt signs of pathogenicity is also common in many reports of hemoparasite infection in wildlife (Oosthuizen et al., 2008; Pfitzer et al., 2011). However, detailed studies of the effects of *Theileria* spp. infections on gazelle physiology, immunity, reproduction and survival would be needed to determine if these organisms affect Grant's gazelle health and fitness. Importantly, our finding of novel hemoparasites in gazelle that are closely related to *Theileria* species known to infect small ruminants raises the question of whether Grant's gazelles might act as sources of novel parasites that could negatively impact livestock. In our study region, Grant's gazelles freely share pasture with sheep and goats and host common tick vectors that also infest livestock (Walker et al., 2000). Spillover of *Theileria* spp. from one host species to others has previously been associated with clinical consequences in ungulates (Hofle et al., 2004; Nijhof et al., 2005). As such, determining whether the novel *Theileria* genotypes we detected in Grant's gazelles are also present in sympatric livestock, or whether these organisms pose a potential spillover risk are important next steps.

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## Conflict of interest

The authors declared that there is no conflict of interest.

## References

- Aktas, M., Bendele, K.G., Altay, K., Dumanli, N., Tsuji, M., Holman, P.J., 2007. Sequence polymorphism in the ribosomal DNA internal transcribed spacers differs among *Theileria* species. *Vet. Parasitol.* 147, 221–230.
- Allsopp, M.T., Allsopp, B.A., 2006. Molecular sequence evidence for the reclassification of some *Babesia* species. *Ann. N. Y. Acad. Sci.* 1081, 509–517.
- Berggoetz, M., Schmid, M., Ston, D., Wyss, V., Chevillon, C., Pretorius, A.M., et al., 2014. Tick-borne pathogens in the blood of wild and domestic ungulates in South Africa: interplay of game and livestock. *Ticks Tick Borne Dis.* 5, 166–175.
- Bhoora, R., Buss, P., Guthrie, A.J., Penzhorn, B.L., Collins, N.E., 2010. Genetic diversity of piroplasmids in plains zebra (*Equus quagga burchellii*) and Cape mountain zebra (*Equus zebra zebra*) in South Africa. *Vet. Parasitol.* 174, 145–149.
- Bishop, R., Musoke, A., Morzaria, S., Gardner, M., Nene, V., 2004. *Theileria*: intracellular protozoan parasites of wild and domestic ruminants transmitted by ixodid ticks. *Parasitology* 129 (Suppl.), S271–S283.
- Bock, R., Jackson, L., de Vos, A., Jorgensen, W., 2004. Babesiosis of cattle. *Parasitology* 129 (Suppl.), S247–S269.
- Brothers, P.S., Collins, N.E., Oosthuizen, M.C., Bhoora, R., Troskie, M., Penzhorn, B.L., 2011. Occurrence of blood-borne tick-transmitted parasites in common tsessebe (*Damaliscus lunatus*) antelope in Northern Cape Province, South Africa. *Vet. Parasitol.* 183, 160–165.
- Chaisi, M.E., Sibeko, K.P., Collins, N.E., Potgieter, F.T., Oosthuizen, M.C., 2011. Identification of *Theileria parva* and *Theileria* sp (buffalo) 18S rRNA gene sequence variants in the African Buffalo (*Syncerus caffer*) in southern Africa. *Vet. Parasitol.* 182, 150–162.
- Chaisi, M.E., Collins, N.E., Potgieter, F.T., Oosthuizen, M.C., 2013. Sequence variation identified in the 18S rRNA gene of *Theileria mutans* and *Theileria velifera* from the African buffalo (*Syncerus caffer*). *Vet. Parasitol.* 191, 132–137.
- Chaisi, M.E., Collins, N.E., Oosthuizen, M.C., 2014. Phylogeny of *Theileria buffeli* genotypes identified in the South African buffalo (*Syncerus caffer*) population. *Vet. Parasitol.* 204, 87–95.
- Ezenwa, V.O., 2003. Habitat overlap and gastrointestinal parasitism in sympatric African bovines. *Parasitology* 126, 379–388.
- Ezenwa, V.O., 2004. Host social behavior and parasitic infection: a multifactorial approach. *Behav. Ecol.* 15, 446–454.
- Ezenwa, V.O., Ekernas, L.S., Creel, S., 2012. Unravelling complex associations between testosterone and parasite infection in the wild. *Funct. Ecol.* 26, 123–133.
- Gebrekidan, H., Hailu, A., Kassahun, A., Rohousova, I., Maia, C., Talmi-Frank, D., et al., 2014. *Theileria* infection in domestic ruminants in northern Ethiopia. *Vet. Parasitol.* 200, 31–38.
- Githaka, N., Konnai, S., Skilton, R., Kariuki, E., Kanduma, E., Murata, S., et al., 2013. Genotypic variations in field isolates of *Theileria* species infecting giraffes (*Giraffa camelopardalis tippelskirchi* and *Giraffa camelopardalis reticulata*) in Kenya. *Parasitol. Int.* 62, 448–453.
- Githaka, N., Konnai, S., Bishop, R., Odongo, D., Lekolool, I., Kariuki, E., et al., 2014. Identification and sequence characterization of novel *Theileria* genotypes from the waterbuck (*Kobus defassa*) in a *Theileria parva*-endemic area in Kenya. *Vet. Parasitol.* 202, 180–193.
- Gubbels, J.M., de Vos, A.P., van der Weide, M., Viseras, J., Schouls, L.M., de Vries, E., et al., 1999. Simultaneous detection of bovine *Theileria* and *Babesia* species by reverse line blot hybridization. *J. Clin. Microbiol.* 37, 1782–1789.
- He, L., Yu, Q., Zhang, W.J., Zhang, Q.L., Fan, L.Z., Miao, X.Y., et al., 2014. Molecular cloning and characterization of a novel heat shock protein 20 of *Babesia orientalis*. *Vet. Parasitol.* 204, 177–183.
- Hofle, U., Vicente, J., Nagore, D., Hurtado, A., Pena, A., de la Fuente, J., et al., 2004. The risks of translocating wildlife Pathogenic infection with *Theileria* sp and *Elaeophora elaphi* in an imported red deer. *Vet. Parasitol.* 126, 387–395.
- Katzer, F., McKellar, S., Kirvar, E., Shiels, B., 1998. Phylogenetic analysis of *Theileria* and *Babesia equi* in relation to the establishment of parasite populations within novel host species and the development of diagnostic tests. *Mol. Biochem. Parasitol.* 95, 33–44.
- Katzer, F., Lizundia, R., Ngugi, D., Blake, D., McKeever, D., 2011. Construction of a genetic map for *Theileria parva*: identification of hotspots of recombination. *Int. J. Parasitol.* 41, 669–675.
- Lack, J.B., Reichard, M.V., Van Den Bussche, R.A., 2012. Phylogeny and evolution of the Piroplasmida as inferred from 18S rRNA sequences. *Int. J. Parasitol.* 42, 353–363.
- Mans, B.J., Pienaar, R., Latif, A.A., Potgieter, F.T., 2011. Diversity in the 18S SSU rRNA V4 hyper-variable region of *Theileria* spp. in Cape buffalo (*Syncerus caffer*) and cattle from Southern Africa. *Parasitology* 138, 766–779.
- Nagore, D., Garcia-Sanmartin, J., Garcia-Perez, A.L., Juste, R.A., Hurtado, A., 2004. Identification, genetic diversity and prevalence of *Theileria* and *Babesia* species in a sheep population from Northern Spain. *Int. J. Parasitol.* 34, 1059–1067.
- Ngeranwa, J.J., Shompole, S.P., Venter, E.H., Wambugu, A., Crafford, J.E., Penzhorn, B.L., 2008. Detection of *Anaplasma* antibodies in wildlife and domestic species in wildlife-livestock interface areas of Kenya by major surface protein 5 competitive inhibition enzyme-linked immunosorbent assay. *Onderstepoort J. Vet. Res.* 75, 199–205.
- Nijhof, A.M., Pillay, V., Steyl, J., Prozesky, L., Stoltz, W.H., Lawrence, J.A., et al., 2005. Molecular characterization of *Theileria* species associated with mortality in four species of African antelopes. *J. Clin. Microbiol.* 43, 5907–5911.
- Oosthuizen, M.C., Zweygarth, E., Collins, N.E., Troskie, M., Penzhorn, B.L., 2008. Identification of a novel *Babesia* sp from a sable antelope (*Hippotragus niger* Harris, 1838). *J. Clin. Microbiol.* 46, 2247–2251.
- Oosthuizen, M.C., Allsopp, B.A., Troskie, M., Collins, N.E., Penzhorn, B.L., 2009. Identification of novel *Babesia* and *Theileria* species in South African giraffe (*Giraffa camelopardalis*, Linnaeus, 1758) and roan antelope (*Hippotragus equinus*, Desmarest 1804). *Vet. Parasitol.* 163, 39–46.
- Pfister, S., Oosthuizen, M.C., Bosman, A.M., Vorster, I., Penzhorn, B.L., 2011. Tick-borne blood parasites in nyala (*Tragelaphus angasii*, Gray 1849) from KwaZulu-Natal, South Africa. *Vet. Parasitol.* 176, 126–131.
- Ronquist, F., Huelsenbeck, J.P., 2003. MrBayes 3: bayesian phylogenetic inference under mixed models. *Bioinformatics* 19, 1572–1574.
- Swofford, D.L., 2002. PAUP\*: Phylogenetic Analysis Using Parsimony (and Other Methods). Sinauer Associates, Sunderland.
- Thompson, J.D., Higgins, D.G., Gibson, T.J., 1994. Clustal-W – Improving the sensitivity of progressive multiple sequence alignment through sequence weighting, position-specific gap penalties and weight matrix choice. *Nucleic Acids Res.* 22, 4673–4680.
- VanderWaal, K.L., Atwill, E.R., Isbell, L.A., McCowan, B., 2014. Quantifying microbe transmission networks for wild and domestic ungulates in Kenya. *Biol. Conserv.* 169, 136–146.
- Walker, J.B., Keirans, J.E., Horak, I.G., 2000. The Genus *Rhipicephalus* (Acari, Ixodidae): A Guide to the Brown Ticks of the World. Cambridge University Press. 643 p.