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POPULATION HEALTH OF FALLOW DEER (*DAMA DAMA*) ON LITTLE ST. SIMONS ISLAND, GEORGIA, USA

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ABSTRACT: Fallow deer (*Dama dama*) were introduced to Little St. Simons Island, Georgia, USA in the 1920s and thrive at high population densities, to the exclusion of white-tailed deer (*Odocoileus virginina*). The presence of introduced pathogens and parasites as a result of their introduction is currently unknown, as is the impact of native disease on the exotic fallow deer. Hunter-killed fallow deer from 2003–2005 were necropsied and surveyed for evidence of infectious disease, parasitic agents, and toxicologic parameters. Fallow deer were positive for antibodies to bovine virus diarrhea virus I and II, bluetongue virus, and bovine adenovirus. Twenty species of bacteria were isolated from the internal organs, and 14 species of parasites were recovered including one abomasal nematode, *Spiculopteragia asymmetrica*, which is not known to occur in native North American ungulates. Concentrations of liver and copper were low, while lead, zinc, and iron were considered within normal levels. No clinical signs of disease were noted, and the overall health of the insular fallow deer was considered good.

Key words: Bacteria, barrier island, disease, exotic, fallow deer, parasite, ungulate.

INTRODUCTION

Populations of exotic ungulates have been established outside of their native range in many parts of the world (Chapman and Chapman, 1975; Feldhammer et al., 1988) prior to establishment of current regulations which ban their transport and release (e.g., GADNR, 2005). Concern exists regarding the potential detrimental impacts of these introductions on the health of livestock (e.g., cattle) and native wildlife species, as well as on the resource availability for native species. These concerns are magnified by reports that suggest that exotics may be more resistant to some diseases than are native species, thus conferring a competitive advantage over natives (Davidson and Crow, 1983; Davidson et al., 1985; Flynn et al., 1990).

The European subspecies of fallow deer (*Dama dama dama*) is a widely distributed member of the Cervidae, with worldwide introductions that include the United States (Chapman and Chapman, 1975). Fallow deer have been successfully introduced in Kentucky, California, Texas, and Georgia (Chapman and Chapman, 1975). Fallow deer were introduced to Little St. Simons Island (LSSI), Georgia in 1927 as a shipment of six individuals (four male, two female) from the Bronx Zoo in New York (Bronx Zoo files, unpubl.). Between 1919 and 1924, attempts to introduce sambar deer (*Cervus unicolor niger*), Barasingha deer (*Cervus duvauceli*), and red deer (*Cervus elaphus*) to LSSI were unsuccessful (Bronx Zoo files, unpubl.). The extent to which the fallow deer interacted with these other species before their extirpation is unknown. Similarly, the population status of the native white-tailed deer (*Odocoileus virginianus*) on LSSI at the time of the fallow deer introduction is also unknown. However, since that time, there have been only anecdotal records of white-tailed deer sightings on the island, with none reported from 2002–2006. Additionally, fallow deer coexisted for varying lengths of time with cattle, goats, or feral horses on LSSI between 1927 and 1996.

Health surveys have been conducted for introduced fallow deer populations in Kentucky (Davidson et al., 1985), California (Riemann et al., 1979), and Texas (Corn et al., 1990). In general, these surveys have reported that fallow deer were susceptible to exposure from para-

sites and diseases of native deer in North America, but were not impacted by these diseases. Corn et al. (1990) revealed high antibody prevalences in fallow deer for blue tongue virus (BTV; 59%) and epizootic hemorrhagic disease virus (EHDV; 64%) in Texas. *Amblyomma americanum* was the most commonly found (85%) ectoparasite in the Texas survey. Serum neutralization also revealed BTV antibodies in California fallow deer (Riemann et al., 1979) and BTV and EHDV antibodies in fallow deer from Kentucky (Davidson et al., 1985). Nine parasites, including the nonnative nematode *Spiculopteragia assymetrica*, parasitized fallow deer in Kentucky, but without noticeable pathogenic impacts. However, Davidson et al. (1985) relate lesions present in all five fallow deer sampled to previous infections of *Parelaphstrongylus tenuis*. Because the LSSI fallow herd represents an insular population that has historically coexisted with other domestic and exotic ungulates, we examined hunter-harvested fallow deer from 2003–2006 to evaluate the health status of this population. Diseased animals were not detected prior to this study; therefore, we conducted routine diagnostic testing with emphasis on ruminant (wild and domestic) diseases typically found in the southeastern United States, as determined from routine submissions to the University of Georgia Veterinary Diagnostic and Investigational Laboratory.

MATERIALS AND METHODS

Study site

Little St. Simons Island is a privately owned, undeveloped coastal barrier island located in Glynn County, Georgia, USA (ca. 31°16′N, 81°17′W). The island is comprised of approximately 1,730 ha of upland habitats (maritime forest, oak–pine forest, shrub–scrub) and approximately 3,470 ha of salt marshes. It is surrounded by water, with no land linkage. Elevations range from sea level to 9 m; however, most of the island is less than 3 m above mean sea level. Fallow deer are the only large feral mammal on LSSI, and population size is currently estimated at 500–700 animals. Mammalian predators of deer such as bobcats (*Lynx rufus*), foxes (*Vulpes vulpes* and *Urocyon cinereoargenteus*), and coyotes (*Canis latrans*) are absent. The only potential predator of fallow deer is the American alligator (*Alligator mississippians*). The island does not support populations of white-tailed deer or feral pigs (*Sus scrofa*), thus eliminating significant competition for food resources.

Sample collection

Hunter-killed fallow deer ($n=68$) were sampled from LSSI during two hunting seasons, 2003–2004 (Year 1, $n=28$) and 2004–2005 (Year 2, $n=40$). The island was divided into hunting zones and the hunters distributed themselves throughout the island. Six separate hunts, lasting 3 to 5 days each, occurred at approximately 2–3 wk intervals between November and March. Hunters could harvest either sex, but according to LSSI harvest guidelines were discouraged from harvesting male deer ≤2 yr of age. Over the two hunting seasons, we obtained 24 adult males (AM), 19 adult females (AF), six yearling males (YM), seven yearling females (YF), and 11 fawns (F). All deer sampled were selected at the individual hunter's discretion.

All harvested deer were brought to a central processing station. Each deer was examined for external parasites and a necropsy was performed. Tissues collected included brain, eye, ear notch, tonsil, lymph nodes from head–throat region, heart, lung, thymus, blood (serum and plasma), liver, kidney, spleen, reproductive tract (whole for females, section of testicle for males), rumen, reticulum, omasum, abomasum, duodenum, jejunum, ileum, cecum, and spiral colon. Brain, eye, ear notch, and tonsil were not collected from deer identified for trophy mounts ($n=15$). A subset of fresh tissues was collected for bacteriologic testing. Another subset of tissues was collected frozen for virus isolation, fluorescent antibody testing, and toxicologic testing. A subset of each tissue was preserved in 10% buffered formalin for histologic analysis. To determine virus shedding, feces were collected for parasite analysis and electron microscopic examination. All collected tissues were transported to the University of Georgia, College of Veterinary Medicine, Veterinary Diagnostic and Investigational Laboratory (VDIL), Tifton, Georgia. Sex, weight (recorded from spring scale to nearest pound, then converted to kilograms) and ages of each deer were recorded. Age was estimated according to Severinghaus (1949) as modified for fallow deer (Murphy, 1995).

In December of 2006, an additional five adult (>2 yr of age) fallow deer were collected

as mentioned above and sampled for abomasal parasites (Eve and Kellogg, 1977). Whole abomasums were removed, sealed in plastic bags, and frozen. Abomasums were transported to the Southeastern Cooperative Wildlife Disease Study (SCWDS) at the University of Georgia, College of Veterinary Medicine, Athens, Georgia for processing.

Diagnostic testing

Histological examination: Formalin-fixed tissues were routinely processed and embedded in paraffin. One or more 5-μm-thick sections were cut from each paraffin block and placed on glass slides. The slides were stained with hematoxylin and eosin, cover-slipped, and viewed by light microscopy for histolopathologic changes in tissues.

Virus isolation: A 10% tissue homogenate in Earle's minimal essential media containing gentamycin was made for each deer. The homogenate was centrifuged for 10 min at 2,000 RPM (805 × G) and 4 C, supernatent filtered, and inoculated onto a preformed monolayer of Madin Darby bovine kidney (MDBK) and vero cells. Inoculated cells were incubated in a 5% CO_2 atmosphere at 37 C. Cells were examined daily for virus cytopathic effect (CPE). If no CPE was observed, aliquots of the first passage were transferred to a second preformed monolayer of MDBV and vero cells on day 7. If no CPE was observed after a second 7 days of passage, chambered slides of MDBK cells were made to examine cultures for noncytopathic bovine virus diarrhea virus (BVDV). Inoculated monolayers demonstrating virus CPE were passaged to chambered slides. All chamber slides were fixed in cold acetone and the appropriate FA tests, including BVDV, were done to confirm the isolates or to confirm any noncytopathic BVDV.

Fluorescent antibody (FA) testing: Virus-specific polyclonal conjugates (BTV, EHDV, bovine adenovirus [BA], and BVD) were applied to frozen sections of tissues (lung and spleen: BTV, EHDV, BA, BVDV; lung, spleen, kidney, and intestine: BVDV) collected at necropsy, and the slide was incubated in 5% CO_2 at 37 C for 30 min. The slides were rinsed twice, counter-stained with 0.5% Evans blue, dipped in distilled water, cover-slipped, and examined by fluorescent microscopy.

Serum neutralization: Serum neutralization assays were used to quantify antibodies for BVDV I and BVDV II. Antibody titer was the last dilution that provided complete protection of the monolayer.

Bluetongue serology was conducted using the BTV antibody competitive Elisa test kit (VMRD, Inc., Pullman, Washington, USA). According to manufacturer's kit insert, test sera are considered positive if they produced an optical density that is $<50\%$ of the mean of the negative control. Test sera that produced an optical density $\geq 50\%$ of the mean of the negative control are considered negative. Serology for EHDV was done with a commercially available agar gel immunodiffusion (AGID) test kit (Veterinary Diagnostic Technology, Wheat Ridge, Colorado).

*Bacterial culture (*routine*, Mycoplasma* sp.*, Leptospirosis* spp.*, Mycobacterium avium paratuberculosis):* Swabs of individual tissues (lung, spleen, liver, kidney, intestine) were streaked onto 5% bovine blood agar (BBA), Wilkins–Chalgren anaerobe agar, Mycoplasma agar, Lowenstein-Jensen agar slant, and Hektoen enteric agar (intestines only). Plates were examined daily for growth and subcultured onto BBA as needed. Bacterial colonies selected from pure cultures were stained using the Gram method. Cultures were inoculated into Sensititre (Trek Diagnostic Systems, Inc., Westlake, Ohio, USA) gram-negative AP80 or gram-positive AP90 auto-identification plates and allowed to incubate for 18 hr at 37 C prior to automated reading of the reactions per manufacturer directions.

Fecal float–gastrointestinal parasites: A mixture of 1 g of feces and 5 ml of water was strained with a 2×3-in single layer of gauze and mixed with Sheather's sugar solution, covered, and allowed to sit for approximately 1 hr. The coverslip was then placed on a slide and, using light microscopy, examined for parasite ova and oocysts.

Electron microscopy–virus shedding: Fecal samples were examined for virus shedding by negative-stain electron microscopy. Grids were examined for viruses or virus-like particles using a Zeiss EM 900 TEM (Carl Zeiss SMT, Peabody, Massachusetts) at 12,000× magnification or greater.

Abomasal parasite counts: The procedure for abomasal parasite counts was conducted according to Eve and Kellogg (1977). Each sample was thoroughly scanned by two persons. Collected worms were placed in a vial of 5% formalin and identified to species. Male worms were used for identification of species. Total counts (1,000 ml) were obtained by

multiplying the number of worms in the 50-ml subsample by 20.

Toxicology: The trace elements lead (Pb), copper (Cu), zinc (Zn), and iron (Fe) were measured based on routine testing protocol at the VDIL. A 3–5-g sample of liver was digested using 10 ml concentrated nitric acid (65%, Fisher Scientific, Fairlawn, New Jersey, USA) plus 10 ml distilled water. The digestates were analyzed by flame atomic absorption spectrophotometry (AAS, AAnalyst 100, Perkin Elmer, Norwalk, Connecticut, USA). Appropriate standards (Certified Reference Solution, Fisher Scientific) and controls (Standard Reference Material 2976 Mussel Tissue or 1577b Bovine Liver; National Institute of Standards and Technology, Gaithersburg, Maryland, USA) were employed for each analytical run. The average of triplicate analysis was reported. Detection limits (ppm) for these elements were: 0.01 Pb, 0.001 Cu, 0.0008 Zn, and 0.003 Fe.

Statistical analysis

Fisher's exact test was used to examine disease prevalence relationships between year, sex, and age. Significance was accepted at $\alpha \leq 0.05$. Age classifications were divided into young (<2 yr of age) and adult (>2 yr of age) animals. Toxicologic values were tested for normality and consequently log-transformed for analysis. Levels of copper and zinc from liver samples were examined for year, sex, and age effects using a three-way analysis of variance in a general linear model (PROC GLM, Statistical Analysis Software, Inc. [SAS], 2003, SAS Institute, Cary, North Carolina). The resulting data for lead levels were nonparametric; therefore, we used a Wilcoxon signed-rank test in SAS (Statistical Analysis Software Inc, 2003). Low sample size limited our ability to statistically analyze iron levels.

RESULTS

Deer age ranged from 0.5 to 8.5 yr with a median age of 3.5 yr. All animals examined were in moderate to excellent health condition, and no gross evidence of disease was noted in any animal. There was no significant difference in antibody prevalence of any of the disease agents tested between males and females, nor between young (<2 yr) and adult (>2 yr) deer using Fisher's exact test. There was no significant difference ($\alpha > 0.05$) between years for all tests; therefore, years were combined for analyses.

TABLE 1. Serologic profile of hunter-killed fallow deer from Little St. Simons Island, Georgia, USA, 2003–2005.

Agent	No. tested	No. positive (%)
Bovine virus diarrhea virus I	48	13 (27)
Bovine virus diarrhea virus II	32	7 (22)
Bluetongue virus	57	2 (4)
Epizootic hemorrhagic disease virus	48	0 (0)
Bovine adenovirus	58	11 (19)
Mycobacterium avium paratuberculosis	48	0 (0)
Leptospira spp.	8	0 (0)

Serology and virology: The ELISA tests disclosed antibodies to BTV in two of 57 samples (Table 1). None of the sera were positive for EHDV antibodies.

Fluorescent antibody tests for BA were positive for 11 of 58 deer sampled. All positive adenovirus results were classified as Adenovirus Group II. The ELISA antibody tests for *Mycobacterium avium paratuberculosis* (Johne's disease) were negative. Serum neutralization for BVDV I and BVDV II resulted in 13 of 48 positive and seven of 32 positive, respectively. Neutralization titers ranged from 4 to 64 for BVDV I and II. Titers of 50 or greater were considered indicative of previous exposure (Corn et al., 1990). Fluorescent antibody tests for either BVDV I or II on 29 samples revealed one positive result. Antigen-capture ELISA tests for BVDV disclosed four positive and six suspect results out of 16 samples tested. Ear notch tests for BVDV antigen on 48 samples resulted in one positive.

Electron microscopy: Fecal samples examined by negative-stain electron microscopy detected the presence of virus particles, consistent with bovine enterovirus (BEV), in four individuals.

Bacterial culture: Bacteria cultures were

TABLE 2. Bacteria cultured and percentage of isolates from internal organs (lung, liver, spleen, kidney, intestine) collected from hunter-harvested fallow deer on Little St. Simons Island, Georgia, USA.

Bacteria	*n*	No. of isolates (%)
Acinetobacter sp.	80	1 (5)
Bacillus sp.	80	6 (7.5)
Citrobacter freundii	80	7 (8.8)
Corynebacterium sp.	80	4 (5)
Enterobacter cloacae	80	13 (16.3)
Enterococcus faecalis	80	7 (8.8)
Escherichia coli	80	38 (47.5)
Hafnia alvei	80	7 (8.8)
Klebsiella pneumoniae	68	3 (4.4)
Moraxella spp.	48	2 (4.2)
Pantoea agglomerans	80	18 (22.5)
Pasteurella multocida	80	4 (5)
Pseudomonas aeruginosa	19	2 (10.5)
Pseudomonas sp.	80	8 (10)
Pseudomonas stutzeri	80	4 (5)
Shewanella putrefaciens	80	7 (8.8)
Staphylococcus sp.	80	6 (7.5)
Streptococcus alpha haemolytic	80	36 (45)
Streptococcus non-haemolytic	80	9 (11.3)
Streptococcus uberis	80	8 (10)

negative for *Leptospirosis* spp. and Johne's disease. Twenty species of bacteria were cultured from internal organs (intestines, lung, liver, spleen, and kidney; Table 2). Clinical disease was not associated with any of these bacteria. However, three deer had minimal to mild histologic changes that may have been associated with bacterial etiologies.

Histology: All fallow deer had mild to moderate eosinophilic enteritis. Rare renal calculi were noted in 24 individuals. Other histologic changes were noted, but were generally minimal to mild and in low prevalence ($n \leq 5$). These histologic changes included perivascular dermatitis (five deer), pneumonitis (three deer), splenitis (two deer), pyometra (one deer), and pneumonia (three deer). Pneumonitis was characterized by minimal expansion of the alveolar walls by rare neutrophils and macrophages. Splenitis was characterized by eosinophilic (both deer) and neutrophilic (one deer) infiltrates. Pyometra was characterized by uterine luminal aggregates of neutrophils and macrophages with no infectious agents seen and no mucosal invasion observed. Pneumonia was characterized by eosinophilic and granulomatous infiltrates that were most often associated with airways.

Parasites: Abomasal counts, fecal examination, and gross examination of the carcass revealed 14 species of parasites, including 10 nematodes and four arthropods. Five nematode species were recovered from four of the five abomasums sampled (Table 3). Overall parasite burdens from abomasal, fecal, and gross examinations were low. The abdominal worm *Setaria yehi* was found free in the abdomen of three fawns (<1 yr). A *Trichuris* sp. was present in one fawn. Larval forms of *Cephenemyia phobifer* were found in the retropharyngeal pouch

TABLE 3. Abomasal parasites of five fallow deer collected from Little St. Simons Island, Georgia, USA in December 2006. Average parasite count was 584.

Animal Number	1	2	3	4	5
Age in yr (sex[a])	4.5 (F)	3.5 (F)	2.5 (M)	3.5 (M)	2.5 (F)
Weight (kg)	42.7	31.3	49.9	54.5	33.6
Abomasal parasites					
Spiculopteragia asymmetrica	0	662	798	150	506
Mazamastrongylus odocoilei	0	0	0	0	39
Ostertagia mossi	0	142	42	150	78
Ostertagia ostertagi	0	0	0	0	78
Trichostrongylus askivali	0	236	0	0	39
Total counts	0	1,040	840	300	740

[a] F = female; M = male.

of five fawns and one 2.5-yr-old female. The nematode *Capillaria* sp. was found in the feces of one 2.5-yr-old male. Coccidia of the genus *Eimeria* was identified in fecal samples from two 1.5-yr-old females; this was the only protozoan parasite recovered.

Toxicology: Concentrations of copper and zinc did not differ between bucks and does ([Cu; $P=0.7987$], Zn [$P=0.8398$]), nor between young and adult deer (Cu [$P=0.5228$], Zn [$P=0.9465$]). Mean copper concentrations for young and adult bucks in our study were 6.47 and 7.73 ppm, respectively (Table 4). Ten of 55 samples examined for lead showed amounts greater than zero. Iron was only sampled from eleven deer in our study. Mean concentrations of iron in the liver of adult bucks ($n=6$) were higher than in does. Due to the low sample size tested for iron ($n=11$), samples were not statistically examined for year, sex, and age differences.

DISCUSSION

The fallow deer on LSSI are in good health condition, similar to reports from other fallow deer populations in the United States (Riemann et al. 1979; Davidson et al., 1985; Corn et al., 1990). We observed various potential pathogens, but only rare morphologic changes were noted and, when present, were of minimal to mild severity and not pathogen-specific. Histologic changes included pneumonitis ($n=3$), splenitis ($n=2$), pyometra ($n=1$), and pneumonia ($n=3$). In most cases, eosinophils were the primary component to the inflammatory infiltrate and were attributed to parasitism or similar hypersensitivity. However, in four deer, other cell types were noted and potentially infectious agents were identified. In these deer, it is possible that these other agents contributed either as primary or opportunistic invaders. For example, although pneumonitis was noted in three deer, only one deer was positive for pathogenic

TABLE 4. Concentrations (mean, SD, and range) in parts per million (ppm) of lead, copper, zinc, and iron in the liver of fallow deer (*Dama dama*) from Little St. Simons Island, Georgia, USA.

	Lead			Copper			Zinc			Iron		
Age/Sex	*n*	x̄ (SD)	Range	*n*	x̄ (SD)	Range	*n*	x̄ (SD)	Range	*n*	x̄ (SD)	Range
Young (0–2 yr)												
F	12	0.02 (0.06)	0–0.22	12	12.35 (14.84)	7.76–53.53	12	36.93 (14.58)	18.69–60.30	1	76.86	–
M	8	0.27 (0.75)	0–2.13	8	6.47 (2.96)	3.53–13.01	8	50.42 (7.75)	43.63–67.89	–	–	–
Adult (>2 yr)												
F	14	0.7 (1.7)	0–5.59	14	12.01 (12.6)	2.33–46.79	14	49.07 (9.86)	21.89–65.60	4	77.92 (20.69)	54.77–104.79
M	21	0.1 (0.18)	0–0.51	21	7.73 (9.19)	1.65–34.64	21	51.98 (18.39)	20.62–110.56	6	133.4 (89.81)	57.87–301.98

organisms, including BVDV and BTV (antibodies). Further, this deer had very mild splenitis characterized by neutrophils and eosinophils. Although the eosinophils suggest a parasitic etiology or hypersensitivity, the neutrophils are more suggestive of an infectious etiology. Significant bacterial organisms were not cultured from the spleen or lung of this deer. One other case of splenitis was noted, and was mild, with only opportunistic organisms (*Klebsiella oxytoca*, *Citrobacter freundii*, *Streptococcus haemolytic*) isolated. In the pyometra case, the origin of the infection was not found. A uterine sample was not collected because, grossly, the uterus was unremarkable; however, opportunistic organisms (*Strepococcus uberis*, *Escherichia coli*, and *Pantoea agglomerans*) were cultured from the kidney and, thus, may have been a factor in the pyometra. Additionally, pneumonia was observed in this doe and was characterized by eosinophilic and granulomatous infiltrates presumed to be due to parasitism. However, opportunistic organisms similar to those cultured from the kidney were cultured from the lungs of this doe and, thus, may have contributed to the pulmonary changes. Finally, one deer with minimal to mild pulmonary changes was positive for adenovirus. It is unclear if the pulmonary changes were due to adenovirus or were incidental. In this deer, pulmonary changes were noted as involving both the interstitium (interstitial pneumonia) and airways (bronchopneumonia), with the latter being eosinophilic, granulomatous, and interpreted as due to parasitism.

Adenovirus infections in North American cervids have been mostly limited to mule deer (*Odocoileus hemionus*) and black-tailed deer (*O. h. columbianus*) in California (Davidson and Nettles, 1997). In 1993, high mortality of mule deer in Northern California, with fatal pulmonary edema, was linked to bovine adenovirus type 5 (Woods et al., 1996). Adenovirus type 5 was also associated with fatal pulmonary edema in white-tailed deer on an Iowa game farm (Sorden et al., 2000). Rarely has adenovirus infection been detected in fallow deer. Type 6 adenovirus was isolated from a single fallow deer after an outbreak of respiratory disease occurred in a Hungarian captive fallow deer herd (Boros et al., 1985). Antibodies to adenovirus group A have been found in fallow deer from Great Britain (Munro, 1994). The significance of the 19% seroprevalence of bovine adenovirus type 5 in LSSI fallow deer is unclear, given that gross or histologic changes consistent with bovine adenovirus type 5 were not found.

Hemorrhagic diseases caused by BTV and EHDV have infected several populations of fallow deer in the United States, with varying rates of antibody prevalence. Prevalence of antibodies to BTV and EHDV include 33% BTV in California (no tests were run for EHDV; Riemann et al., 1979) and 57% BTV and 64% EHDV in Texas (Corn et al., 1990). A disease survey of five adult, free-ranging fallow deer in Kentucky revealed antibodies to EHDV in two fallow deer and antibodies to BTV in one deer (Davidson et al., 1985). Four percent of the animals tested on LSSI had antibodies to BTV and none had antibodies to EHDV. Barrier island white-tailed deer populations showed a low annual incidence of BTV and EHDV, possibly making them susceptible to outbreaks should they occur (Stallknecht et al., 1991). Insular conditions of this population may have provided a degree of isolation from BTV and EHDV due to limited-vector flight distance and immigration–emigration barriers. Varying prevalence rates may be affected by host preference of the midge vector, *Culicoides* spp. (Odiawa et al., 1985). Future testing of *Culicoides* spp. and white-tailed deer from surrounding areas are needed to examine the presence of BTV and EHDV in coastal Glynn County, Georgia and surrounding areas. Further testing of LSSI fallow deer for BTV and EHDV to determine specific serotypes is also needed. The LSSI fallow deer may be inher-

ently resistant to the serotypes of BTV and EHDV currently in coastal Georgia.

Although BVDV has been rarely reported in fallow deer (Neilson et al., 2000), BVDV antibodies and antigen were detected in LSSI fallow deer. Serologic testing for antibodies to BVDV ranged from 0% from introduced fallow deer in California (Riemann et al., 1979) to 1% in Germany (Frölich, 1995) and 2% in Texas (Corn et al., 1990). Fluorescent antibody tests and ear notch tests resulted in one positive deer for each test, indicating a low infection rate for LSSI fallow deer. Serum neutralization indicated 27% seroprevalence for BVDV I, with nine of 13 positive results having titers ≤8. Of the seven positive results (22% antibody prevalence) for BVDV II, only two had titers ≤8. One deer was positive by antigen-capture ELISA and ear notch FA, but negative for both BVDV I and II using serum neutralization. This is similar to reports in cattle, where calves are persistently infected with BVDV until they becomes immune tolerant and then are seronegative (Lindberg and Alenius, 1999). The lack of clinical signs and the low rate of infection indicates that BVDV is not a major pathogen in LSSI fallow deer.

Electron microscopy revealed four positive results for BEV. Little information exists regarding bovine enteroviruses and deer. Ley et al. (2002) found BEV in 76% of cattle and 38% of white-tailed deer sampled in Maryland. Because infections in deer are believed to be asymptomatic, the presence of BEV in the feces of LSSI fallow deer likely has little significance.

Tissue histology revealed eosinophilic enteritis in all deer samples, likely due to internal parasite migration, although abomasal parasites levels were low in our study. Perivascular dermatitis noted was associated with tick bites. Although tick infestations were generally low on examined deer, increased levels of parasitism could cause stress and reduced vigor. White-tailed deer fawns are susceptible to fatal anemia from heavy infestations of hematophagic parasites such as ticks and mosquitoes (Strickland et al., 1981). In midsummer, we observed heavy infestations of mosquitoes and ticks on newborn fallow deer fawns.

Abomasal parasite counts (APC) have been used as an indicator of white-tailed deer health in the southeastern United States (Eve and Kellogg, 1977). Overall abomasum parasite burdens in the five samples from LSSI fallow deer (APC=584) were low (Table 3). It is interesting to note that one sampled deer had no abomasal parasites detected. This could be a result of extremely low worm burdens earlier in the year that were lost by the December collection. Abomasal parasite counts on white-tailed deer are typically conducted in late summer when parasite levels are greatest (M. Yabsley, pers. comm.). Our APC was conducted in December, due to the availability of harvested animals, and may under-represent the current intensity of parasitism in LSSI fallow deer. However, Osborne et al. (1992) reported APCs over five years of 732–2,362 for Blackbeard Island white-tailed deer in December (n=5–10 deer per year per collection). Abomasal parasite burdens of fallow deer appear within a range of other reports for fallow deer, including a previous report for LSSI (APC=152; SCWDS, 1969, unpublished data), Germany (APC=592; Barth and Matzke, 1984), Italy (APC=333; Ambrosi et al., 1993), Illinois (APC=421; SCWDS, 1995, unpublished data), and Spain (APC=719; Santin-Duran et al., 2004).

Two nematodes, *Spiculopteragia asymmetrica* and *Osteragia osteragia*, are not common in native white-tailed deer in the southeastern United States (Doster and Friend, 1971; Conti and Howerth, 1987). *Spiculopteragia asymmetrica* is one of the most common medium abomasum worms reported in fallow deer, having been reported from New Zealand (Andrews, 1973), Germany (Barth and Matzke, 1984), Italy (Ambrosi et al. 1993), and Spain (Santin-Duran et al., 2004). The

first record of *S. asymmetrica* in North American was recorded by Doster and Friend (1971) in fallow deer from LSSI, and our study indicates that it remains the most-abundant abomasum parasite. *Spiculopteragia assymetrica* is also present in populations of fallow deer in Kentucky (Davidson et al., 1985) and Illinois (M. Yabsley, pers. comm.). Although *Spiculopteragia assymetrica* has not been reported from white-tailed deer in the United States, it is listed as a parasite of white-tailed deer in New Zealand (Andrews, 1973).

Two abomasum parasites previously reported from LSSI, *Haemonchus contortus* and *Ostertagia dikmansi* (SCWDS 1969), were not found in our study. However, we recorded three species, *Mazamastrongylus odocoilei*, *Ostertagia ostertagi*, and *Trichostrongylus askival* that were not reported from the prior investigation.

Other endoparasites recovered (*Setaria yehi*, *Trichuris* sp., *Cephenemyia phobifera*, *Ostertagia mossi*, *Trichostrongylus askivali*, *Mazamastrongylus odocoilei*) are typical parasites of white-tailed deer in the southeastern United States (Davidson et al, 1985; Davidson and Nettles, 1997). To our knowledge, *C. phobifera*, *Trichuris* sp., and *T. askivali* have not been previously reported to parasitize fallow deer. Type specimens of *C. phobifera* recovered from LSSI fallow deer have been deposited in the Georgia Museum of Natural History entomology collections. Little is known about the role these parasites play in fallow deer health, but no significant clinical signs were observed in our examinations. The abdominal worm *Seteria yehi* was present in fawns but not in adult deer. Infections of white-tailed deer by *S. yehi* are most common among fawns and yearling deer (Prestwood and Pursglove, 1981). The presence of *S. yehi* in only fawns may indicate that fallow deer, like white-tailed deer in the Southeast, develop some immunity as they become adults.

An *Eimeria* sp. was the only protozoan parasite discovered in our study. They are known to infect a variety of ruminant hosts, and at least four species infect white-tailed deer (Kingston, 1991). An *Eimeria* sp. was reported from fallow deer in New Zealand (Mason, 1994), but is rarely reported in free-ranging populations in the United States. Infections of *Eimeria* spp. usually have no clinical effect, but could become pathogenic in stressed animals (Mason, 1994).

High concentrations of lead in three individuals (2.13, 3.61, 5.59 ppm) exceeded normal levels (<1.0 ppm; Puls, 1994), but the source of this lead is unknown. Although mean concentrations of zinc were within the normal range (30–60 ppm; Puls, 1994), they were higher in all age and sex categories (Table 4) than in fallow deer from Slovania (means=31–32 ppm; Vengušt and Vengušt, 2004) and Illinois (mean=27.6 ppm; M. Yabsley, pers. comm.), as well as in red deer in Poland (mean=31 ppm; Falandysz et al., 2005). Iron concentrations were within the range (40–90 ppm) reported by Puls (1994). Mean concentrations of iron were lower in both sexes than that reported from Slovenia (Vengušt and Vengušt, 2004).

Mean concentrations of copper in LSSI fallow deer liver were below the normal range (25–80 ppm) provided by Puls (1994) and that reported from Slovenia (Vengušt and Vengušt, 2004) in all age and sex classifications, but were similar to that reported from Illinois (11.4 ppm; M. Yabsley, pers. comm). Copper deficiencies have been linked to health problems such as osteochondrosis and enzootic ataxia (Thompson et al., 1994; Audige et al., 1995). No clinical signs of copper deficiency have been observed in LSSI fallow deer, but the low levels found in this study suggest that the potential may exist and should be monitored.

The fallow deer population on LSSI demonstrates some exposure to the pathogens and parasites often associated with native white-tailed deer and cattle. At least one parasite, *S. asymmetrica*, was likely introduced, as this common parasite of

fallow deer has not previously been documented in North American white-tailed deer. Currently, there are no clinical signs of health-related problems, and the herd appears to be within carrying capacity. However, parasite loads and susceptibility to disease could increase if herd density increases. We recommend sustaining the herd at, or below, the current density. There is also no indication that human consumption of fallow deer venison from LSSI would be unsafe.

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LITERATURE CITED

Andrews, J. R. H. 1973. A host-parasite checklist of helminthes of wild ruminants in New Zealand. New Zealand Veterinary Journal 21: 43–47.

Ambrosi, M., M. T. Manfredi, and P. Lanfranchi. 1993. Pattern of abomasal helminths in fallow deer farming in Umbria (central Italy). Veterinary Parasitology 47: 81–86.

Audige, L., P. R. Wilson, R. S. Morris, and G. W. Davidson. 1995. Osteochondrosis, skeletal abnormalities and enzootic ataxia associated with copper deficiency in a farmed red deer (*Cervus* elaphus) herd. New Zealand Veterinary Journal 43: 70–76.

Barth, D., and P. Matzke. 1984. Gastro-intestinal nematodes of fallow deer (*Dama dama* L.) in Germany. Veterinary Parasitology 16: 173–176.

Boros, G., Z. Graf, M. Benkő, and A. Bartha. 1985. Isolation of bovine adenovirus from fallow deer (*Dama dama*). Acta Veterinaria Hungarica 33: 119–123.

Chapman, D., and N. Chapman. 1975. Fallow deer: Their history, distribution and biology. Terence Dalton Limited, Lavenham, Great Britain, 271 pp.

Conti, J. A., and E. W. Howerth. 1987. Ostertagiosis in a white-tailed deer due to *Ostertagia ostertagia*. Journal of Wildlife Diseases 23: 159–162.

Corn, J. L., D. M. Kavanaugh, D. A. Osborn, S. Demarais, D. M. Miller, and V. F. Nettles. 1990. Survey for diseases and parasites in exotic ruminants in Texas. Proceedings of the Annual Meeting of the U.S. Animal Health Association 94: 530–540.

Davidson, W. R., and C. B. Crow. 1983. Parasites, diseases, and health status of sympatric populations of sika deer and white-tailed deer in Maryland and Virginia. Journal of Wildlife Diseases 19: 345–348.

———, and V. F. Nettles. 1997. Field manual of wildlife diseases in the southeastern United States. Southeastern Cooperative Wildlife Disease Study, Athens, Georgia, 417 pp.

———, J. M. Crum, J. L. Blue, D. W. Sharp, and J. H. Phillips. 1985. Parasites, diseases, and health status of sympatric populations of fallow deer and white-tailed deer in Kentucky. Journal of Wildlife Diseases 21: 153–159.

Doster, G. L., and M. Friend. 1971. *Spiculopteragia* (Nematoda) from deer in North America. Journal of Parasitology 57: 468.

Eve, J. H., and F. E. Kellogg. 1977. Management implications of abomasal parasites in southeastern white-tailed deer. Journal of Wildlife Management 41: 169–177.

Falandysz, J., K. Szymczyk-Kobrzynska, A. Brzostowski, K. Zalewski, and A. Zasadowski. 2005. Concentrations of heavy metals in the tissues of red deer (*Cervus elaphus*) from the region of Warmia and Mazury, Poland. Food Additives and Contaminants 22: 141–149.

Feldhammer, G. A., K. C. Farris-Renner, and C. M. Barker. 1988. *Dama dama*. Mammalian Species No. 317: 1–8.

Flynn, L. B., S. M. Shea, J. C. Lewis, and R. L. Marchinton. 1990. Part III: Population statistics, health, and habitat use. *In* Ecology of sambar deer on St. Vincent National Wildlife Refuge, Florida. Bulletin Number 25, Tall Timbers Research Station, Tallahassee, Florida, pp. 63–96.

Frölich, K. 1995. Bovine virus diarrhea and mucosal disease in free-ranging and captive deer (Cervidae) in Germany. Journal of Wildlife Diseases 31: 247–250.

Georgia Department of Natural Resources (GADNR). 2005. Importation of Cervids. Regulation 391-4-9-05. Wildlife Resources Division.

Kingston, N. 1991. Protozoan parasites. *In* Diseases and parasites of white-tailed deer, W. R. Davidson (ed.). Misc. Publ. No. 7 of Tall Timbers Research Station, Tallahassee, Florida, pp. 214–236.

LEY, V., J. HIGGINS, AND R. FAYER. 2002. Bovine enteroviruses as indicators of fecal contamination. Applied and Environmental Microbiology 68: 3455–3461.

LINDBERG, A. L., AND S. ALENIUS. 1999. Principles for eradication of bovine viral diarrhea virus (BVDV) infections in cattle populations. Veterinary Microbiology 64: 197–222.

MASON, P. 1994. Parasites of deer in New Zealand. New Zealand Journal of Zoology 21: 39–47.

MUNRO, R. 1994. Respiratory System. *In* Management and diseases of deer: A handbook for the veterinary surgeon, T. L. Alexander and D. Buxton (eds.). The Veterinary Deer Society, London, England, 109 pp.

MURPHY, B. 1994. Determining the age of deer. Guns & Game 2 (6): 42–48.

NIELSEN, S. S., L. ROENSHOLT, AND V. BITSCH. 2000. Bovine virus diarrhea virus in free-living deer from Denmark. Journal of Wildlife Diseases 36: 584–587.

ODIAWA, G., J. L. BLUE, D. E. TYLER, AND E. B. SHOTTS. 1985. Bluetongue and epizootic hemorrhagic disease in ruminants in Georgia: Survey by serotest and virologic isolation. American Journal of Veterinary Research 46: 2193–2196.

OSBORNE, J. S., A. S. JOHNSON, P. E. HALE, R. L. MARCHINTON, C. V. VANSANT, AND J. M. WENTWORTH. 1992. Population ecology of the Blackbeard Island white-tailed deer. Bulletin Number 26. Tall Timbers Research Station, Tallahassee, Florida, 108 pp.

PRESTWOOD, A. K., AND S. R. PURSGLOVE, JR. 1981. Gastrointestinal nematodes. *In* Diseases and parasites of white-tailed deer, W. R. Davidson (ed.). Misc. Publ. No. 7. Tall Timbers Research Station, Tallahassee, Florida, pp. 318–350.

PULS, R. 1994. Mineral levels in animal health. Diagnostic data, 2nd Edition. Sherpa International, Clearbrook, British Columbia, Canada, 356 pp.

RIEMANN, H. P., R. RUPPANNER, P. WILLEBERG, C. E. FRANTI, W. H. ELLIOTT, R. A. FISHER, O. A. BRUNETTI, J. H. AHO, J. A. HOWARTH, AND D. E. BEHYMER. 1979. Serologic profile of exotic deer at Point Reyes National Seashore. Journal of the American Veterinary Medical Association 175: 911–913.

SANTÍN-DURÁN, M., J. M. ALUNDA, E. P. HOBERG, AND C. DE LA FUENTE. 2004. Abomasal parasites in wild sympatric cervids, red deer, *Cervus elaphus*, and fallow deer, *Dama dama*, from three localities across central and western Spain: Relationship to host density and park management. Journal of Parasitology 90: 1378–1386.

SEVERINGHAUS, C. W. 1949. Tooth development and wear as a criteria of age in white-tailed deer. Journal of Wildlife Management 13: 195–216.

SORDEN, S. D., L. W. WOODS, AND H. D. LEHMKUHL. 2000. Fatal pulmonary edema in white-tailed deer (*Odocoileus virginianus*) associated with adenovirus infection. Journal of Veterinary Diagnostic Investigation 12: 378–380.

STALLKNECHT, D. E., J. L. BLUE, E. A. ROLLOR, III, V. F. NETTLES, W. R. DAVIDSON, AND J. R. PEARSON. 1991. Precipitating antibodies to epizootic hemorrhagic disease and bluetongue viruses in white-tailed deer in the southeastern United States. Journal of Wildlife Diseases 27: 238–247.

STRICKLAND, R. K., R. R. GERRISH, AND J. S. SMITH. 1981. Arthropods. *In* Diseases and parasites of white-tailed deer, W. R. Davidson (ed.). Tall Timbers Research Station Misc. Publ. No. 7, Tallahassee, Florida, pp. 363–389.

THOMPSON, K. G., L. AUDIGE, D. G. ARTHUR, A. F. JULIAN, M. B. ORR, K. D. MCSPORRAN, AND P. R. WILSON. 1994. Osteochondrosis associated with copper deficiency in young farmed red deer and wapiti × red deer hybrids. New Zealand Veterinary Journal 42: 137–143.

VENGUŠT, G., AND A. VENGUŠT. 2004. Some trace minerals as well as trace and toxic elements in livers of fallow deer (*Dama dama*) in Slovenia. European Journal of Wildlife Research 50: 59–61.

WOODS, L. W., P. K. SWIFT, B. C. BARR, M. C. HORZINKE, R. W. NORDHAUSEN, M. H. STILLIAN, J. F. PATTON, M. N. OLIVER, K. R. JONES, AND N. J. MACLACHLAN. 1996. Systemic adenovirus infection associated with high mortality in mule deer (*Odocoileus hemionus*) in California. Veterinary Pathology 33: 125–132.

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