Disease Vector Ecology Profile

Colombia

Office of the Deputy Under Secretary of Defense for Environmental Security






Defense Pest Management Information Analysis Center
Armed Forces Pest Management Board
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PREFACE

Disease Vector Ecology Profiles (DVEPs) summarize unclassified literature on medically important arthropods, vertebrates and plants that may adversely affect troops in specific countries or regions around the world. Primary emphasis is on the epidemiology of arthropod-borne diseases and the bionomics and control of disease vectors. DVEPs have proved to be of significant value to commanders, medical planners, preventive medicine personnel, and particularly medical entomologists. These persons use the information condensed in DVEPs to plan and implement prevention and control measures to protect deployed forces from disease, injury, and annoyance caused by vector and pest arthropods. Because the DVEP target audience is also responsible for protecting troops from venomous animals and poisonous plants, as well as zoonotic diseases for which arthropod vectors are unknown, limited material is provided on poisonous snakes, noxious plants, and diseases like hantavirus.

Vector-borne diseases are presented in two groups: those with immediate impact on military operations (incubation period < 15 days) and those with delayed impact on military operations (incubation period > 15 days). For each disease, information is presented on military importance, transmission cycle, vector profiles, and vector surveillance and suppression.

Similar information on venomous vertebrates and noxious plants is available in the Armed Forces Medical Intelligence Center's (AFMIC) Medical, Environmental, Disease, Intelligence, and Countermeasures (MEDIC) CD-ROM.

Contingency Operations Assistance: The Armed Forces Pest Management Board (AFPMB) is staffed with a Contingency Liaison Officer (CLO), who can help identify appropriate DoD personnel, equipment, and supplies necessary for vector surveillance and control during contingencies. Contact the CLO at Tel: (301) 295-8300, DSN: 295-8300, or Fax: (301) 295-7473.

Defense Pest Management Information Analysis Center (DPMIAC) Services: In addition to DVEPs, the DPMIAC publishes Technical Information Bulletins (TIBs), Technical Information Memoranda (TIMs), and the Military Pest Management Handbook (MPMH). The DPMIAC can provide online literature searches of databases on pest management, medical entomology, pest identification, pesticide toxicology, and other biomedical topics. DPMIAC also operates a home page on the Internet, from which documents of current operational interest, such as TIMs, DVEPs, and recent editions of TIBs can be downloaded. Customers can also conduct their own literature abstract data searches online. The home page address is: **http://www.afpmb.org**/. Contact DPMIAC at Tel: (301) 295-7476, DSN: 295-7476, or Fax: (301) 295-7483. Additional hard copies or diskettes of this publication are also available.

Other Sources of Information: The epidemiologies of arthropod-borne diseases are constantly changing, especially in developing countries undergoing rapid growth, ecological change, and/or large migrations of refugee populations resulting from civil strife. Therefore, DVEPs should be supplemented with the most current information on public health and geographical medicine. Current disease risk assessments, additional information on parasitic and infectious diseases, and other aspects of medical intelligence can be obtained from the Armed Forces Medical

Intelligence Center (AFMIC), Fort Detrick, Frederick, MD 21701, Tel: (301) 663-7511, DSN: 343-7511. Disease Risk Assessment Profiles (DISRAPs) and Vector Risk Assessment Profiles (VECTRAPs) for most countries in the world can be obtained from the Navy Preventive Medicine Information System (NAPMIS) by contacting the Navy Environmental Health Center (NEHC) at Tel: (804) 444-7575 ext. 456, DSN: 564-4657 ext. 456. Information is also available from the Defense Environmental Network and Information Exchange (DENIX) home page address: **http://denix.cecer.army.mil/denix/denix.html**

Specimen Identification Services: Specimen identification services and taxonomic keys can be obtained through the Walter Reed Biosystematics Unit (WRBU), Museum Support Center, MRC-534, Smithsonian Institution, Washington, DC 20560 USA; Tel: (301) 238-3165; Fax: (301) 238-3667; e-mail: wrbu@wrbu.si.edu

Emergency Procurement of Insect Repellents, Pesticides and Equipment: Deploying forces often need pesticides and equipment on short notice. The Defense Logistics Agency (DLA) has established the following Emergency Supply Operations Centers (ESOCs) to provide equipment and supplies to deploying forces:

For insect repellents, pesticides and respirators: Contact the Defense General Supply Center ESOC at Tel: (805) 275-4865, DSN: 695-4865. The ESOC is staffed seven days a week/24 hours a day.

For application equipment: Contact the Defense Construction Supply Center ESOC at Tel: (614) 238-2271/3191, DSN: 850-2271/3191.

For personal protection equipment (bednets, headnets, etc.): Contact the Defense Personnel Support Center at Tel: (215) 737-3042/3043, DSN: 444-3042/3043.

Every effort is made to ensure the accuracy of the information contained in DVEPs. Individuals having additional information, corrections, or suggestions, are encouraged to provide them to the Chief, DPMIAC, Armed Forces Pest Management Board, Forest Glen Section, Walter Reed Army Medical Center, Washington, DC 20307-5001; Tel: (301) 295-7476, DSN: 295-7476; Fax: (301) 295-7483.

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I. Executive Summary

Three Andean mountain chains (Cordillera Occidental, Cordillera Central, and Cordillera Oriental) run north and south dividing Colombia into the tropical coastal rainforest and the vast eastern lowland plains (Llanos Oriental). The land east of the Cordillera Oriental comprises about 60% of the country, but only 2% of the total population of 37.5 million people live there.

Malaria is transmitted by the bite of infected *Anopheles* mosquitoes. Risk is high year-round in rural areas below 800 m and less up to 2,700 m. More than half the cases occur in Antioquia Department. Countrywide, about 40% of the cases are *Plasmodium falciparum*, about 60% *P. vivax* and <1% *P. malaria*e. Ninety-eight percent of the malaria cases on the Pacific coast are *P. falciparum*. Chloroquine resistant *P. falciparum* has been reported in all areas and resistance to Fansidar® is widespread (Amazonia, Orinoquia, Cauca Valley, Caribbean regions), and to mefloquine and amodiaquine (Amazon Basin). *Plasmodium vivax* resistance to chloroquine, amodiaquine and primaquine has also been reported. The Andean highlands are risk free. Preventive measures against malaria include sanitation improvements to eliminate mosquito breeding sites, application of residual insecticides to vector resting sites and aerosol insecticides to screened living and sleeping areas, use of permethrin-impregnated bednets for sleeping, prompt and effective treatment of cases, conscientious use of chemoprophylaxis, and personal protective measures (**PPM**). **PPM** are outlined in Appendix H.

Dengue fever can debilitate its victims as early as 3 days following the bite of an infected *Aedes aegypti* mosquito. Risk is countrywide in urban and peri-urban areas below 1,800 m, particularly along the Atlantic coast, the slopes of the Cordillera Oriental, and in the Magdalena and Cauca River Valleys. Individuals who are serologically positive for the dengue virus are in greater danger of developing the more serious dengue hemorrhagic fever/dengue shock syndrome (DHF/DSS) if infected a second time. Eliminate larval breeding sources (artificial containers), spray interiors of tentage with permethrin, and use **PPM** against mosquitoes.

Venezuelan equine encephalitis (VEE), **Eastern equine encephalitis** (EEE), and **St. Louis encephalitis** (SLE) occur erratically and focally, primarily in jungle areas. In 1995, 14,156 cases of VEE were reported (26 deaths). Other causes of febrile illnesses include **Bussuquara**, **Caraparu**, **Guaroa**, **Ilheus**, **Mayaro**, **Oropouche**, **Wyeomyia**, **Vesicular Stomatitis**, and **Arboledas**. Oropouche has occurred as explosive epidemics. Mosquitoes, sand flies, and biting midges (*Culicoides*) have been implicated as vectors. Use **PPM** against flying/biting insects.

Leishmaniasis is a protozoan disease transmitted in the New World by *Lutzomyia* sand flies. The cutaneous form occurs year-round in forested highlands up to 1,800 m, while the visceral form has been reported in arid areas of the Magdalena River Valley below 900 m. Greatest risk is in forested Pacific coast areas, the lower Cauca River Valley, the Magdalena River Valley, and the eastern slopes of the Cordillera Oriental. Sand flies bite primarily during twilight hours and after dark, although they will bite during the day if disturbed while in their resting habitat. Because sand flies have very short flight ranges, barrier and area control with residual insecticides is very effective. Avoid bivouacking in dense, forested areas or close to human habitations, and use PPM, especially from sunset to sunrise.

Chagas disease, transmitted by the feces of kissing bugs, is associated with substandard housing (e.g., thatch construction, adobe shanties) prevalent in urban slums and rural areas. Enzootic foci occur in rural areas below 2,700 m within Norte de Santander Department and piedmont areas of the Cordillera Oriental. Bivouac away from domestic and peridomestic settings. Use **PPM** to protect against feeding kissing bugs.

Bartonellosis is a bacterial disease transmitted by sand flies along the eastern slopes of the Cordillera Occidental in the southwestern departments of NariZo, Cauca, and Valle del Cauca from 600 to 2,450 m. Control sand flies in bivouac sites and use PPM against biting sand flies.

Rocky Mountain spotted fever, a rickettsial disease, may be transmitted by ticks countrywide. Use **PPM**, check for ticks often using the buddy system, and remove attached ticks promptly.

Murine typhus is a rickettsial disease transmitted by fleas of commensal rodents. The highest rates are reported in the departments of Antioquia, Caldas, Cundinimarca, NariZo, and Norte de Santander. Avoid rodent-infested areas, employ good sanitation, control rodents and use **PPM**.

Epidemic typhus, a rickettsial disease transmitted by body lice, has been reported in NariZo Department. The disease proliferates under crowded and unsanitary conditions (war refugees, prisoners of war, and natural disasters). Avoid contact with local populations and use **PPM**.

Relapsing fever (tick-borne) is a spirochetal disease endemic from Barranquilla south along the Pacific coast and in the lowlands of eastern Colombia. Use **PPM** to prevent ticks from biting.

Sylvatic **yellow fever**, a virus transmitted by canopy-dwelling mosquitoes, is most prevalent in the central and northern valleys of the Magdalena River and in piedmont areas of the Cordillera Oriental from Ecuador to Venezuela. Use vaccine and **PPM** against day-biting mosquitoes.

Mansonellosis, **Onchocerciasis**, and **Bancroftian filariasis** are vector-borne filarial infections reported in the extreme SE, SW, and NW, respectively. Use **PPM** against black flies, biting midges (*Culicoides*), sand flies, and mosquitoes.

Leptospirosis is a spirochetal disease contracted when broken skin or mucous membranes come into contact with infected rodent urine or feces via contaminated surfaces, food, or water. Avoid rodent-infested habitats, bathing in potentially contaminated water, and drinking untreated water.

Rabies is endemic countrywide in cats, dogs, bats, and wild carnivores. Avoid handling pets and other animals. Report all animal bites immediately to medical personnel.

Venomous snakes, poisonous arthropods, and irritating and poisonous plants are present throughout Colombia. Ticks, chiggers and fleas are sources of significant discomfort. Severe irritation and scratching may result in secondary infections. Be alert for poisonous snakes and plants and use **PPM** against arthropods. Blouse trousers inside boots instead of using blousing garters.

II. Map of Colombia (CIA)

III. Country Profile

A. Geography. Colombia, located at the northern extreme of the Andes Mountains, is bordered to the northwest by Panama (255 km/158 mi) and the Caribbean/Atlantic (1,760 km/1,091 mi), on the east and northeast by Venezuela (2,050 km/1,271 mi), on the southeast by Brazil (1,643 km/1,019 mi), on the south by Peru (2,900 km/1,798 mi) and Ecuador (590 km/366 mi), and on the west by the Pacific Ocean (1,448 km/898 mi). The total landmass is 1,141,748 km^2/440,831 mi^2, slightly less than 3 times the size of Montana. There are 32 departments and the capital district of Bogot<. Colombia can be divided into four topographic regions: 1) The Caribbean/ Atlantic lowlands consist of plains extending north from the Panamanian border to an isolated mountain range, the Sierra Nevada de Santa Marta. Colombia's highest mountain, Pico Crist\bal Col\n, 5,775 m/18,947 ft, is located in this range, which isolates the semi-arid Peninsula de la Guajira. 2) The Pacific coastal lowlands (sea level to 330 m/1,000 ft) stretch from Ecuador to Panama as a narrow band 20-100 km/6-30 miles wide. These lowlands consist of jungles, swamps, and a low isolated mountain range less than 330 m/1,000 ft. 3) The Colombian Andes consist of three distinct ranges that run on a northeasterly axis through the western third of Colombia. Maximum elevations are 5,750 m/18,865 ft. Two of the ranges, the Cordillera Occidental and Cordillera Central, parallel the Pacific coastal lowlands and a third, the Cordillera Oriental, extends more inland to the northeast. The Central and Oriental ranges enclose a valley that drains north via the Rio Magdalena into the Caribbean/Atlantic lowland areas of Magdalena, Sucre, and Cesar Departments. Peaks of the cordillera have perpetual snow cover with a timberline of ca. 3,000 m/10,000 ft. 4) The eastern lowland plains of the north and jungle forest to the south comprise ca. 60% of the land area. This watershed drains the eastern slopes of the Andes into the Rio Orinoco in the north and into the Amazon to the south. Major rivers include the Rio Meta, Rio Vichada and Rio Guaviare, which are tributaries of the Rio Orinoco to the north, and the Rio Yar, Rio Apaporis, Rio Caquet<, and Rio Putumaya that drain into the Amazon to the south.

B. Climate. The tropical climate of Colombia predominates along the Pacific coast, with annual rainfall on the lower slopes of the western Andes exceeding 2,500 mm/100 inches. Some of the western slopes receive more than 5,000 mm/200 inches. All months are wet. Table 1 shows a weather summary for Andagoya, which is typical of these areas. The Caribbean coastal area is dryer, especially from December to March. The Central mountainous area of Colombia above 4,500 m/15,000 ft receives most precipitation as snow, while the lower valleys at the north end of the Andes receive an equal amount of precipitation as rain (between 1,000-2,500 mm/40-100 inches) throughout the year. Climatic data for Bogot< (Table 2) are typical of mountainous terrain 1,800-3,000 m/6,000-10,000 ft, where day and night temperatures range between 7°-11°C/45°-52°F at various times of the year. The lowlands east of the Andes have annual rainfall between 2,000-2,500 mm/80-100 inches, with two wet seasons, December to January and April to May. Manaus, Brazil (Table 3) is most representative of the wet equatorial lowlands (meteorological data for the eastern lowlands of Colombia are not available).

Table 1. 8-year meteorological summary at Andagoya (60 m/197 ft).

Month	Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Highest Temp. (°C)	35	36	36	36	36	34	36	35	35	36	35	35
Av. Maximum (°C)	32	32	32	32	32	32	32	32	32	32	31	31
Av. Minimum (°C)	24	24	24	24	24	23	23	23	23	23	23	23
Lowest Temp. (°C)	20	21	21	21	21	20	21	21	21	17	19	21
Av. Rainfall (mm)	635	544	495	663	647	655	592	592	625	577	569	495

Table 2. 10-year meteorological summary at Bogot< (2,645 m/8,678 ft).

Month	Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Highest Temp. (°C)	23	24	24	24	23	22	22	22	23	23	23	23
Av. Maximum (°C)	19	20	19	19	19	18	18	18	19	19	19	19
Av. Minimum (°C)	9	9	10	11	11	11	10	10	9	10	10	9
Lowest Temp. (°C)	4	6	6	7	7	7	7	7	7	6	7	4
Av. Rainfall (mm)	58	66	102	147	114	61	51	56	61	160	119	66

Table 3. 11-year meteorological summary at Manaus, Brazil (44 m/144 ft).

Month	Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Highest Temp. (°C)	37	38	36	34	35	35	35	37	37	38	37	38
Av. Maximum (°C)	31	31	31	31	31	31	32	33	33	33	33	32
Av. Minimum (°C)	24	24	24	24	24	24	24	24	24	24	24	24
Lowest Temp. (°C)	18	20	19	20	20	18	18	19	20	20	20	19
Av. Rainfall (mm)	249	231	262	221	170	84	58	38	46	107	142	203

C. Population/People. Seventy-five percent of the total population of 37,500,000 is concentrated in the central highland plateaus and basins of the Andes Mountains. Other populated areas include the lowland valleys of the Magdalena and Cauca Rivers in northern Colombia along the Caribbean/Atlantic. The Pacific coastal lowlands are sparsely populated, and only about 2% of the population lives in lowland areas east of the Cordillera Oriental, which comprise 60% of the country. Countrywide, there are an average of 32 persons per km^2/82 per mi^2. Ninety-five percent of the population is Roman Catholic, with ethnic groups totaling 58% Mestizo (mixed European and Indian), 20% White, 14% Mulatto, 4% Black, 3% mixed Black-Indian, and 1% Indian. The official language is Spanish.

D. Living and Sanitary Conditions. Although sewage systems serve close to 70% of the urban population and 25% of the rural population, a significant proportion of wastewater is discharged without treatment. Throughout the country, refuse is burned, dumped into bodies of water, or left in the open. Living conditions vary according to income and availability of building materials. In the countryside and urban shantytowns, huts and other makeshift dwellings predominate. About 60% of the urban population is believed to live in slums and squatter settlements, whereas affluent inhabitants live in homes that vary from old colonial stone

dwellings to ranch-style houses. Coastal swampland dwellings usually are of thatch, bamboo, or wood erected on stilts, with few basic services.

1. Pollution: Urban pollution sources include automobiles (estimated to produce 80% of the air pollution), industries, and energy production. Soil erosion and improperly treated waste contribute to the pollution of rivers and streams. Unregulated use of pesticides is increasing throughout Colombia. Crop irrigation water often is contaminated with raw sewage. Reportedly, 96% of the Atlantic coast's forest has been destroyed, resulting in damage to water sources and drying of river channels and marshes. Chemical pollutants and sewage releases have resulted in mangrove swamp destruction and water contamination. Ecological damage on the Atlantic coast is thought to be greater than on the Pacific coast.

2. Water: Surface water sources, including rivers, lakes and manmade reservoirs, are the predominant sources of drinking water for both rural and urban Colombians. Water treatment standards do not meet US drinking water standards. Municipal water treatment is subject to filtering and settling but not always to chlorination. Several Colombian cities also fluoridate their water supplies. There is a fairly extensive water distribution system in Colombia compared to other countries in South America. About 75% of the urban population and 50% of the rural population are supplied with water through in-house connections or public standpipes.

IV. Militarily Important Vector-Borne Diseases with Short Incubation Periods (<15 days)

A. Malaria. Malaria is a protozoan blood parasite transmitted to man by the bite of infective *Anopheles* mosquitoes. There are four species of human malaria (*Plasmodium falciparum, P. vivax, P. malariae, and P. ovale*) and all except *P. ovale* occur in Colombia. *Plasmodium falciparum* (malignant tertian), the most serious of the four malarias, frequently causes death (10% in untreated non-immune adults). *Plasmodium vivax* (benign tertian) and *P. malariae* are generally not life threatening but may result in complete debilitation during manifestations of clinical symptoms. Some *P. vivax* parasites may remain hidden in the liver cells (hypnozoites) for months or even years following the initial clinical disease. Consequently, relapses producing clinical malaria may occur months to years later. *Plasmodium malariae* parasites occasionally remain in the blood at undetectable levels, replicating at some later date and causing clinical disease. Relapses usually do not occur with *P. falciparum*. Lack of treatment, drug resistant strains, or inadequate therapy may result in relapses of all malaria species. Clinical symptoms of malaria vary with the species. Falciparum malaria may produce fever, chills, sweats, cough, diarrhea, respiratory distress and headache, often leading to more serious systemic complications. *Plasmodium vivax* and *P. malariae* cases may begin with general malaise and a slowly rising fever of several days duration, followed by shaking chills, rapidly rising temperature, usually accompanied by headache and nausea, and ends in profuse sweating. The fever subsides for a period and is followed by subsequent cycles of fever, chills, and sweating.

1. Military Impact and Historical Perspective. Case reporting in Colombia varies from year to year, but the annual number of cases has more than doubled since the middle 1980s, with 127,218 reported in 1994. These cases were based on positive blood smears. In the Andean countries, approximately three times as many cases were reportedly treated through governmental malaria control programs. Private sector care and self-treatment cases are not

included. The overall incidence increased from 36/1,000 in 1993 to 41/1,000 in 1994. Countrywide, *P. falciparum* constitutes approximately 40% of the cases, *P. vivax* just under 60%, and *P. malariae* <1%. Some areas along the Pacific coast report 98% of the cases as *P. falciparum*. Areas of high risk are those below 800 m, with less risk at elevations up to 2,700 m. Historically, malaria afflicted up to 40% of some Army units in Vietnam. Commanders lost 2 of every 5 men for substantial periods of time, seriously compromising their missions. The threat in Colombia is equally great, compounded by known *P. falciparum* resistance to some antimalarial drugs and known vector resistance to several pesticides. Malaria is largely preventable and casualties can be minimized with application of the measures outlined in paragraph IV.A.4.

2. Transmission Cycle. Humans are the sole reservoir for human malaria parasites (*P. vivax*, *P. falciparum*, *P. malariae* and *P. ovale*). Female mosquitoes of the genus *Anopheles* are the exclusive vectors of human malaria. During feeding, the mosquito ingests the sexual stages of the parasite (male and female gametocytes), which unite and form an ookinete that penetrates the midgut of the mosquito, forming an oocyst. The oocysts produce thousands of sporozoites, some of which migrate to the salivary glands and are injected into the human host on subsequent feedings. The time between ingestion of sexual gametocytes and liberation of sporozoites is dependent on the temperature and species of malaria parasite (8-35 days). Cooler temperatures that occur with increased altitudes will increase the time for development. Ingested sporozoites travel to the liver, where they invade the liver cells and divide, becoming asexual merozoites. After several division cycles, the merozoites leave the liver cells and parasitize individual red blood cells (RBCs). Most become schizonts that cyclically rupture the RBCs causing the characteristic symptom of "fever and chills". Some of the merozoites develop into sexual male and female gametocytes capable of infecting other *Anopheles* mosquitoes. Once infected, mosquitoes remain infective for life. Infected humans are potential reservoirs of infection as long as the sexual gametocytes are circulating in the blood. In vivax malaria, this is only 1-2 days after onset of symptoms as opposed to 10-12 days after onset in falciparum malaria.

3. Vector Ecology Profile. Important vectors of malaria in Colombia are *An. darlingi*, *An. albimanus*, *An. nuneztovari*, *An. pseudopunctipennis*, *An. aquasalis*, *An. agyritarsis*, *An. marajoara*, *An. neivae*, *An. triannulatus An. albitarsis*, *An. bellator*, *An. calderoni* and *An. braziliensis*. Relative importance may be determined by seasonal and/or geographic distribution. Vector ecology profiles for species with sufficient data are presented in **Appendix A.1**. Increasing knowledge of anopheline species in the Americas confirms the existence of complexes or sibling species. *Anopheles nuneztovari* has been shown to be a complex of sibling species that requires additional systematic and cytogenetic work. *Anopheles marajoara* was recently elevated from synonymy with *An. albitarsis*. Populations previously recognized as *An. punctimacula* along the Pacific coast and in the Cauca River Valley are currently considered *An. calderoni*. A list of mosquitoes that are known to occur in Colombia and important identification keys are presented in Appendices B.1 and G, respectively.

4. Vector Surveillance and Suppression. Larval and adult surveillance techniques are used to assess mosquito populations. Consult the vector ecology profiles in Appendix A.1 for mosquito species of concern and adapt larval surveys to breeding habitats of these species. Larval sampling using a white dipper equipped with a long handle will facilitate collection of *Anopheles*

larvae. Systematically performed, larval dipping will provide data on species composition and population dynamics on which to base control measures. Adult surveillance of potential anopheline vectors principally includes collection on animal or human bait or collection of resting adults using mechanical or mouth aspirators (Note: collections using human bait must be conducted under approved human-use protocols). Military operations may be more concerned with exophilic anopheline populations, necessitating the collection of mosquitoes away from domestic locations. Anthropophilic species can be collected in human dwellings using a mouth or powered aspirator and aided by a flashlight. Collections from whatever source are suitable for determining species composition and abundance, parity rates, and malaria parasite infection rates. The older the female anopheline mosquito population is in a malaria-endemic area, the greater the potential for transmission, since the man-mosquito-man cycle requires a minimum of 8-9 days. Malaria suppression should include elimination of gametocytes from blood of the human reservoir population, reduction of *Anopheles* mosquito populations, and prevention of mosquito bites. Mosquitoes are capable of acquiring *P. vivax* gametocytes from infected humans for 24-36 hours after treatment with chloroquine alone, but when treated concurrently with chloroquine and primaquine, the acquisition window is reduced to less than 4 hours. *Plasmodium falciparum* gametocytes are not killed by Fansidar®, quinine, or quinine plus tetracycline. Patients are known to infect mosquitoes for >30 days after successful treatment of erythrocytic stages. Primaquine is required to kill gametocytes and reduce transmission to new mosquito reservoirs. *Plasmodium falciparum* resistance to chloroquine is likely in all malarious areas. There is widespread resistance to Fansidar in the Amazonian, Orinoquian and Caribbean areas and in the Cauca River Valley, and unconfirmed reports of mefloquine resistance in southern Pacific coastal areas and the Amazon Basin. Cases of *P. vivax* in central Colombia are allegedly resistant to treatment with chloroquine and amodiaquine/primaquine (1989 reports). It is critically important for commanders to enforce medical protocols when antimalarial drugs are indicated. Consult TIM 24 for chemicals, equipment and procedures for controlling larval and adult mosquitoes. In 1980, WHO reported Dieldrin and HCH resistance in Colombian *An. albimanus*, *An. albitarsis* and *An. triannulatus*, and DDT resistance in *An. albimanus* and *An. albitarsis*. A WHO 1992 report added *An. darlingi* and *An. punctimacula* to the list of DDT-resistant anophelines but made no mention of Dieldrin and HCH resistance or any resistance in *An. triannulatus*. **Personal Protective Measures** to prevent mosquito bites include minimizing exposed skin by wearing permethrin-treated BDUs with sleeves down, particularly after sunset, using extended-duration deet repellent, arthropod repellent deet jackets, headnets treated with deet, and sleeping under permethrin-treated bednets whenever possible (TIM 36).

B. Dengue Fever. Dengue fever (Flavivirus: Flaviviridae) is a viral illness transmitted in the Americas by the bite of the *Aedes aegypti* mosquito. Symptoms of dengue appear 3-14 days following the bite. They include sudden onset of fever, severe headache, muscle and joint aches, retro-orbital pain, loss of appetite, gastrointestinal upset and, in some cases, generalized erythema and rash. There are four serotypes of the dengue virus (DEN-1, 2, 3 and 4). Serotypes are extremely important when considering clinical illness. There is lifelong immunity to subsequent exposure to the same (homologous) serotype, whereas an individual is predisposed to the more serious and often fatal dengue hemorrhagic fever (DHF), or dengue shock syndrome (DSS), if exposed to a second and different (heterologous) serotype. There are no vaccines for protection against dengue virus.

1. Military Impact and Historical Perspective. Epidemics are likely with increased *Ae. aegypti* populations in urban endemic areas. Dengue was not present in Colombia prior to 1970. During 1980-1996, the total number of cases reported was 309,239 (annual range 774-82,272). Major outbreaks were reported in 1985 (82,272: single and first case of DHF) and 1992 (56,357: 496 cases of DHF). From 1980-1996, the average annual number of cases (excluding the two large outbreaks) was 11,174. DEN-1, 2, and 4 are known to occur in Colombia. The greatest potential for transmission exists along the Atlantic coast in the Magdalena River Valley, Cauca River Valley, Putumayo Intendancy and along the slopes of the Cordillera Oriental. Dengue fever is capable of causing many casualties, rendering soldiers unfit for duty for a week or more. While morbidity rates for nonimmune personnel are high, complications of DHF/DSS are uncommon. When operating in endemic urban and peri-urban locations, it is essential to use personal protective measures to prevent day-biting *Ae. aegypti.*

2. Transmission Cycle. Both humans and mosquitoes serve as reservoirs in the human-mosquito cycle. *Aedes aegypti*, the primary vector of dengue in the Americas, may acquire the virus from a viremic person immediately prior to and during the febrile period (about 6-7 days). The mosquito is able to transmit the virus 8-12+ days after acquisition, depending on temperature, and remains infective for life. Although *Aedes albopictus*, an additional potential vector of the dengue virus, is currently not found in Colombia, it occurs in Tabatinga, Brazil, near the eastern border of Colombia.

3. Vector Ecology Profile. The vector ecology profiles for *Ae. aegypti* and *Ae. albopictus* are presented in Appendix A.2.

4. Vector Surveillance and Suppression. Control of dengue fever is contingent upon managing populations of *Ae. aegypti* through surveillance, reduction of breeding sources, treatment with insecticides, and personal protection. Surveillance should include monitoring adult egg-laying activity by using black oviposition cups, conducting physical inspection of the interiors and exteriors of urban and peri-urban premises and encampment sites for artificial containers with *Ae. aegypti* larvae, and conducting landing counts with a mechanical or mouth aspirator (TB MED 561). Eliminate breeding sources (any receptacle that may hold fresh water) from premises and encampment sites. Broad area control of adult populations can be obtained using ultra-low-volume (ULV) equipment and thermal foggers. Consult TIM 24 for equipment and chemicals for control of mosquitoes. Cooler morning and evening hours are optimal times for applying fogs and aerosols, because at these times pesticides are less likely to be carried away by temperature induced updrafts. Wide spread resistance to organophosphates (chlorpyrifos, dimethoate, fenthion, malathion and temephos) has been reported for *Ae. aegypti* (WHO, 1992). Resistance to lambda-cyhalothrin has also been reported for this mosquito. The individual soldier can best prevent dengue fever by using **Personal Protective Measures** during the day, when *Ae. aegypti* frequently bite. Wear permethrin-impregnated BDUs and use extended-duration deet repellent on exposed skin surfaces (TIM 36). When possible, avoid the practice of stripping to the waist or wearing shorts to keep cool during the daytime.

C. Yellow Fever. Yellow fever is caused by a flavivirus transmitted by mosquitoes. Symptoms of yellow fever occur 3-6 days following the bite of an infected mosquito. Patients may be nearly asymptomatic, but symptoms generally begin with sudden onset of fever, chills, headache,

backache, generalized muscle pain, prostration and vomiting. Symptoms may progress to jaundice and hemorrhagic manifestations. Fatalities may exceed 50% in non-indigenous or non-immunized individuals. Yellow fever imparts a lifelong immunity to survivors.

1. Military Impact and Historical Perspective. The last documented case of urban yellow fever in Colombia was in 1929. Since that time, all reported cases have been traced to the sylvatic cycle. Yellow fever continues to sequester as focal enzootics and epizootics in the eastern plains of the Amazon and Orinoco River basins, forests at the base of the Cordillera Oriental, central and northern Magdalena Valley, Atrato and Catatumbo Basins, and along the Atlantic coast. The yellow fever vaccine offers American soldiers essentially 100% protection; however, host country soldiers or allies may not have the same protection and may require entomological support.

2. Transmission Cycle. Yellow fever is propagated in either an urban cycle or a sylvatic cycle. During the urban cycle, the virus is transmitted between mosquitoes and man. The sole mosquito vector is the domestic *Ae. aegypti.* Mosquitoes may acquire the virus from an infected person for a short time just before fever occurs and 3-5 days after onset. Mosquitoes require 9-12 days to become infective and remain so for life. The sylvatic cycle is a mosquito-monkey cycle, monkeys being the amplifying hosts. Primary vectors include canopy-dwelling species of *Haemagogus* and *Sabethes*, which transmit the virus to monkeys of the family Cebidae (howler monkeys, *Alouatta* spp.; and spider monkeys, *Cebus* spp., *Aotes* spp. and *Callithrix* spp.). Maintenance of the virus in the sylvatic cycle is enhanced by transovarial transmission in vector mosquitoes. Urban yellow fever occurs when humans enter the jungle cycle, contract the disease from *Haemagogus* or *Sabethes* mosquitoes, and return to urban areas where they infect *Ae. aegypti* that subsequently bite other urban residents. In the urban setting, humans are amplifying hosts for the virus.

3. Vector Ecology Profile. *Aedes aegypti* is the primary vector of yellow fever in the urban (mosquito-man) transmission cycle. *Haemagogus janthinomys* and *Sabethes chloropterus* are the vectors in the sylvatic cycle (monkey-mosquito-man) in Colombia. Vector ecology profiles for these species are presented in Appendix A.2.

4. Vector Surveillance and Suppression. Methods for surveillance and suppression of yellow fever in the urban environment are the same as for dengue. Transmission of sylvatic yellow fever can be prevented by vaccination and using **Personal Protective Measures** to prevent species of *Haemagogus* and *Sabethes* from biting. Wear permethrin-impregnated BDUs with sleeves rolled down and use extended-duration deet repellent on exposed skin surfaces, especially during the daytime when these mosquitoes actively bite (TIM 36 and Appendix H). When possible, avoid the practice of stripping to the waist or wearing shorts to keep cool during the daytime. Select bivouac areas as far from forests as the tactical situation permits.

D. Other Arboviral Fevers. Dengue fever, paragraph IV.B., and yellow fever, paragraph IV.C., are described in greater detail than the following arboviral diseases because of detailed knowledge of their epidemiologies and potential for producing severe hemorrhagic disease. In general, other arboviral fevers in Colombia are poorly understood; however, some have the potential for inflicting high morbidity, especially in non-immune personnel. Incapacitation can

occur rapidly, lasting a few days to several weeks. Although clinical symptoms differ slightly for each, they generally consist of fever, headache, dizziness, arthralgia, myalgia, often a rash, and occasional prostration. Other Colombian arboviruses include the Togaviridae (Alphaviruses): Eastern equine encephalitis (EEE), Mayaro, Venezuelan equine encephalitis (VEE), Western equine encephalitis (WEE); Flaviviridae (Flaviviruses): Bussuquara, Ilheus, St. Louis encephalitis (SLE); Bunyaviridae (Bunyaviruses) Group C: Caraparu, Bunyamwera Group: Wyeomyia, California Group: Guaroa, Simbu Group: Oropouche; and Rhabdoviridae: Vesicular Stomatitis. Arboviruses that occur in Colombia or that may be present east of the Andes in lowland tropical areas geographically contiguous with the Amazon Basin are listed in Appendix A.3. Potential vectors and reservoirs identified throughout the Amazon Basin (including those known in Colombia) are included in the appendix. Vaccines are not available for these arboviral diseases. Prevention of human cases requires control of mosquitoes, use of repellents (day and night) and bed nets, and avoidance of forest habitats when possible.

1. Oropouche. Oropouche occurs in either a sylvatic (forest) cycle or an urban cycle. Little is known about the sylvatic cycle, although primates, sloths, and possibly wild birds are implicated reservoirs. The biting midge, *Culicoides paraensis*, is a proven vector. *Culex quinquefasciatus* is also capable of transmission but considered to be of minor importance. Vector ecology profiles for *C. paraensis* and *Cx. quinquefasciatus* are presented in Appendix A.3. Epidemics involving hundreds of thousands of people have occurred in the Amazon Basin of Brazil. Oropouche virus may inflict rapid, extensive, and high morbidity in military personnel. Explosive outbreaks occur in association with urban areas and rural villages, especially those near banana and cacao crops. U.S. military entomologists in Brazil have noted that insecticide applications in cacao and banana plantations for other agricultural pests drastically reduce populations of *C. paraensis*. Residual pesticides can be directed at breeding areas at any time, while aerosol applications should be applied during peak host seeking activity, approximately an hour before sunset. Maintain good sanitation by eliminating wet and deteriorated vegetation (e.g., banana tree stalks and stumps, discarded cacao pods) that could provide ample breeding for *Culicoides*. Using extended-duration deet repellent and keeping trousers tucked into boots will prevent bites. Avoid resting on the ground, as individuals that lie on the ground in the life zone of *Culicoides paraensis* during the day are at greater risk of being bitten. Do not camp in or adjacent to banana or cacao plantations.

2. Venezuelan Equine Encephalitis. Mosquitoes transmit sylvatic and epizootic VEE. Sylvatic VEE is a mild clinical disease producing low-level viremia in equines, while epizootic VEE is more severe, killing most non-immunized equines and producing high viremias capable of infecting vector mosquitoes. Either form of VEE can produce severe clinical disease, encephalitis, and death in humans. Human cases of VEE usually follow the beginning of an epizootic in horses by about two weeks. Nearly all genera of mosquitoes are capable of transmitting VEE, but *Aedes scapularis*, *Ae. aegypti*, *Aedes taeniorhynchus*, *Mansonia tillitans*, and *Deinocerites pseudes* are incriminated vectors. Vaccinating the equine population and killing larval and adult mosquitoes with insecticides (TIM 24) controls VEE epidemics. Immunity to epizootic VEE in horses is complete within 5 days of vaccination. Nearly all genera of mosquitoes are capable of transmitting VEE, but are incriminated vectors. Human cases of VEE usually follow the beginning of an epizootic in equines by about two weeks. The likelihood of VEE occurring in troops during military operations is low unless an epizootic is

occurring in equine populations. Sporadic cases of sylvatic VEE are possible in forested areas inhabited by species of *Culex* (*Melanoconiun*) when virus is enzootic in rodent and bird populations. References for keys to the species of *Culex* (*Melanoconion*) are provided in Appendix G.

E. American Trypanosomiasis (Chagas Disease). Chagas disease is the second most important arthropod-borne disease in the world. This disease is caused by *Trypanosoma cruzi*, a flagellated protozoan that is transmitted by infected conenose or kissing bugs of the family Reduviidae. It is an insidious disease with variable symptoms in the acute phase, ranging from none to unilateral facial edema (RomaZa's sign) usually at the site of bite(s), fever, malaise, lymphadenopathy, and enlarged liver and spleen. Symptoms generally appear 5-14 days after infection. Chronic symptoms usually involve the heart, intestinal and autonomic nervous systems and persist for many years after the initial infection. Involvement of these organ systems results in extensive lingering morbidity and 20% fatalities. A second protozoan (*Trypanosoma rangeli*) is often present in vector species but has not been linked to human disease. Xenodiagnosis (blood feeding of "clean" bugs on a patient, waiting for several weeks, and examining the bug's feces for trypanosomes) is a method of diagnosis, especially during the chronic phase of the disease when the number of trypanosomes in the blood is low. An indirect fluorescent antibody technique is also available for testing blood and spinal fluid for *T. cruzi* antibodies. Trypanosomes present in blood may be transmitted during blood transfusions.

1. Military Impact and Historical Perspective. The disease is associated with poverty, primarily in rural areas of the Catatumbo River Basin in the northwest, the Magdalena River Valley in the central region, and the eastern slopes and piedmont of the Cordillera Oriental. Studies in Norte de Santander Department in 1985 indicate 15.6% of the houses were infested with the vector, 30% of the population studied had positive serologies, and 9% displayed abnormal heart function. *Trypanosoma cruzi* is found in animals and extradomiciliary vectors countrywide (excluding the Orinoco River Basin) below 2,700 m. It should be noted that sylvatic foci were recently identified in the northern Ecuadorian lowland provinces of Sucumbios and Napo bordering the Colombian departments of Putumayo and eastern Amazonas. Otherwise, little is known about the prevalence of the disease in the Orinoco River Basin. Since soldiers operate in forest habitats, the likelihood of exposure to Chagas disease by extradomiciliary vector species might be greater than exposure in a domestic setting if personal protective measures are not implemented. In addition, the disease has the potential to infect many troops that bivouac (sleep) adjacent to or in areas with substandard housing (e.g., makeshift and palm thatched construction). Avoid such areas and use **Personal Protective Measures** to prevent biting bugs and minimize risk. Because serious symptoms are delayed, the greatest impact will be on the individual soldier and the medical support system subsequent to deployment.

2. Transmission Cycle. Humans are important reservoirs of the parasite, *T. cruzi*, as are many domestic and peridomestic animals (e.g., dogs, cats, guinea pigs, and swine). Hosts differ depending on the endemic locality and vector species of Triatominae, Reduviidae (known as triatomines, conenose bugs, or kissing bugs). Some sylvatic species of triatomines will feed on humans who enter their habitats, but they are usually considered important only in maintaining the cycle in nature. When triatomines feed on infected humans, trypanosomes are ingested and

begin to replicate in the hindgut. The bug becomes infective 10-30 days after feeding on an infected host and remains so for life (as long as two years). Nymphal triatomines may also acquire trypanosomes while feeding on the infected blood of engorged bugs. All stages of the bug may become infective. Human infections occur when bugs defecate on the skin of their host during feeding and the infected feces are rubbed into the bite puncture or abrasions, or onto the mucosae of the eyes, mouth or nose. The most important vectors of Chagas disease live in association with man in domestic or peridomestic settings, inflict nearly painless bites, and defecate quickly during feeding. Species that have delayed patterns of defecation (e.g., feed and leave before defecating) are incapable of transmitting trypanosomes. Triatomines feed on humans only at night, particularly on the neck and face.

3. Vector Ecology Profile. Vector ecology profiles for the Colombian species *Rhodnius prolixus*, *Rhodnius pallescens*, *Triatoma dimidiata*, and *Triatoma maculata* are presented in Appendix A.4.

4. Vector Surveillance and Suppression. Surveillance of triatomines is best conducted with the aid of a flashlight during hours of darkness. Host-seeking bugs (adults and nymphs) can be detected crawling in the open, particularly around sleeping/bedding areas, guinea pig enclosures, and livestock shelters. Harborage for triatomine bugs such as cracks and crevices of adobe walls, wood and thatched roofs should be checked carefully. Triatomine fecal stains on walls are another sign of infestations. Light traps are ineffective as a surveillance tool for Reduviidae, although some sylvatic species are attracted to domestic light sources. Should chemical control become operationally feasible or necessary, spraying of interior surfaces of walls, cracks and crevices, overhead thatching, rafters, and under bed structures will control bug populations. Species of Reduviidae that occur in Colombia are listed in Appendix B.2. Risk of contacting vectors is greatest in or near dwellings with abundant cracks and crevices (e.g., thatch, dirt, or makeshift shanty construction) and domestic animal shelters (e.g., livestock, chickens, pigeons, etc.). Sylvatic species are potential biters also. Avoid sleeping near palm trees as these may harbor triatomines. During sleep, keep as much of the body covered as possible and use extended-duration deet repellent on exposed skin surfaces. Headnets and bednets used to prevent mosquito/sand fly-borne diseases also will exclude night-feeding triatomines (TIM 36).

F. Rocky Mountain Spotted Fever (RMSF). *Rickettsia rickettsii*, the causative agent of RMSF, is transmitted to man by the bite of an infected tick. This febrile illness (known as Tobia Fever in Colombia) begins with sudden onset of fever, severe headache (frontal and occipital), chills, deep muscle pain, aching in the lumbar region, and malaise. The most characteristic and constant symptom is a maculopapular rash that begins on the extremities (wrists and ankles) on the 2nd or 3rd day and spreads to all parts of the body. The incubation period from tick bite to first symptoms ranges from 2 to14 days. Fatalities occur in 3-5% of the cases between days 9 and 15 from onset. Early diagnosis and treatment are essential to ensure recovery.

1. Military Impact and Historical Perspective. The first cases of RMSF were reported in Colombia in 1937, and 362 cases were reported between 1950 and 1956. The vector is common throughout Colombia and the potential for contracting the disease through tick bites is comparable to that of endemic areas in the midwestern and eastern U.S. Follow the precautions for preventing tick bites (TIM 36) and for proper removal of ticks (Appendix H).

2. Transmission Cycle. The rickettsial organism is maintained in the tick population primarily by transovarial and transstadial transmission. Dogs, small rodents and ungulates are potential reservoirs of *R. rickettsii* but are considered to be of minor importance. All three stages of the tick (larva, nymph and adult) will feed readily on humans and each is capable of transmitting the agent during feeding. Ticks must feed for 4-6 hours before rickettsiae can be transmitted to the host. Ticks are infected for life (up to18 months).

3. Vector Ecology Profile. The principal Colombian vector of RMSF is the cayenne tick, *Amblyomma cajennense*. Experimental infection and transovarial/transstadial transmission have been demonstrated in the soft tick *Ornithodoros parkeri*, implicating it as a potential vector of RMSF. Both sexes of *Amblyomma cajennense* are haematophagous, and females require a blood meal for oviposition. This is a three-host tick with diverse host preferences. Larval and nymphal stages are found most frequently on small and intermediate-size hosts (e.g., rodents, marsupials, etc.), while adults prefer larger mammals (ungulates). Questing behavior is similar to that of other three-host ticks. Larval and nymphal ticks quest on lower surfaces (e.g., grass, leaves, and twigs), while adults crawl to greater heights on tall grasses, bushes, and shrubs. Questing activity is particularly great during cool mornings and evenings, when relative humidity is high in the ticks' questing zone. Questing is also greater following rain showers. Populations are typically higher in scrub areas with secondary growth, particularly along edges of forest clearings frequented by larger ungulates (deer). Ticks known to occur in Colombia and their hosts are listed in Appendix B.4.

4. Vector Surveillance and Suppression. Assessment of tick populations for species composition and population densities may be accomplished by tick drags and CO_2 baited traps (TIM 26). The former are made of white flannel cloth (30-40 inches square), either attached to a dowel for dragging or to a pole (as a flag) for waving over the surface of the ground or vegetation. Questing ticks will adhere to the flannel surface for easy collection. Expedient CO_2 traps can be made with a piece of white material 3-4 feet square (non-flannel) laid on the ground with a block of dry ice placed in the center. Flannel material will impede the movement of ticks toward the dry ice. Double-stick tape stuck on the sheet 360° around the piece of dry ice will entrap ticks as they advance toward the CO_2. Ticks also may be found adhering to the underside of the cloth. Broad area tick control is usually not practical or necessary except in areas where personnel are concentrated (command posts, permanent bivouac areas, garrisons, etc.). Provide area tick control with residual chemicals (TIM 24). The best personal defense against ticks is the permethrin-impregnated BDU with trousers tucked into boots. Ticks can penetrate under blousing garters. Use extended-duration repellent (deet) on exposed skin areas, especially on the back of the neck at the hairline (TIM 36). Check for ticks frequently using the buddy system and remove them promptly. Consult Appendix H for detailed guidelines on tick removal.

G. Murine Typhus. *Rickettsia typhi* (formerly *R. mooseri*), the causative agent of murine typhus, is transmitted in the feces or crushed bodies of infected fleas. Symptoms appear 6-14 days from time of infection. The disease is similar to epidemic typhus, but symptoms are milder and fatalities are rare. Symptoms include headache, high fever, chills, prostration and general body aches. Untreated cases result in < 5% mortality. Cases treated with appropriate antibiotics can expect complete recovery.

1. Military Impact and Historical Perspective. Murine typhus can be a serious debilitating illness. Cases may occur throughout the year. Incidence of the disease is highest in the departments of Antioquia, Caldas, Cundinimarca, NariZo and Norte de Santander, but murine typhus may occur in any areas where there are commensal rodents (*Rattus* spp.). Occasional cases may be expected, but impact on operations should be negligible.

2. Transmission Cycle. Murine typhus is a domiciliary disease associated with commensal rats and their fleas. The reservoirs are *Rattus rattus* and *Rattus norvegicus*. The rickettsiae are routinely transmitted between rodents by the fleas *Xenopsylla cheopis*, *Leptopsylla segnis*, *Nosopsyllus fasciatus* (occasionally), the louse *Polyplax spinulosa*, and the mite *Ornithonyssus bacoti.* Although all three species of fleas will bite man, only *X. cheopis* is considered important in transmission from rats to man. Rickettsial organisms are imbibed from an infected rat by *X. cheopis*, replicate in the epithelial cells of the midgut, escape into the midgut after 3-5 days, spread to the entire lining of the midgut within 7-9 days, and feces become infected by the 10th day. Fleas may continue to pass infected feces for 40 days. Although the primary mode of transmission is via infected feces, rickettsiae may be transmitted by the bite three weeks after the initial infection. Infected *X. cheopis* remain infective for life (5+ months). Unlike epidemic typhus, murine typhus rickettsiae do not kill the vectors, and their reservoir rodent hosts are also unaffected. During feeding, *X. cheopis* may pass infected feces onto the skin. These may be scratched or rubbed into the bite, abraded skin, or conjunctivae of the eyes and mucous membranes. Man is an accidental host and is not implicated in maintenance of the disease cycle. Endemic foci are generally found in urban areas but may be rural when commensal rodents are abundant.

3. Vector Ecology Profile. The major vector of murine typhus in Colombia is *X. cheopis*, whose profile is presented in Appendix A.5. A list of fleas and their hosts and references for their identification are provided in Appendices B.3 and I, respectively.

4. Vector Surveillance and Suppression. Populations of *Rattus* spp. are evident from the presence of burrows, rat runs, rat rubs, and rodent damage. *Rattus* populations can be evaluated with snap traps or cage traps baited with local fruits, vegetables, or rolled oats/peanut butter mixtures. Discourage commensal rodents in permanent encampments by eliminating sources of food, shelter and water. Bury putresible waste and garbage. If eradication is necessary, use rodenticides in accordance with guidelines (TIM 24 and TG 138). Wear permethrin-treated BDUs with trousers tucked into boots to protect against fleas. Fleas will crawl under blousing garters. Bivouac away from human habitations, sources of manmade rodent harborage, and agricultural grain harvest areas.

H. Epidemic Typhus. The agent of epidemic or louse-borne typhus, *Rickettsia prowazekii*, is transmitted in the feces of infected body lice (*Pediculus humanus humanus*) and not by the bite of the louse. Symptoms 7-14 days post-infection include headache followed by fever, chills, prostration and general body aches. A macular rash usually appears on the upper trunk and spreads to the entire body. Failure to treat with antibiotics may result in 10-40% fatalities.

1. Military Impact and Historical Perspective. Cases have been reported in NoriZo Department. Historically, most cases occur between March and September. Data are scanty after the late 1970s because the disease is no longer reportable. The potential for outbreaks among the indigenous population would be great during military operations that would result in concentrations of refugees or prisoners of war, but few cases would be expected among U.S. soldiers using routine **Personal Protective Measures** (TIM 36).

2. Transmission Cycle. Transmission is confined to the louse-human-louse cycle. Humans are the reservoir of *R. prowazekii* and the sole host for the human body louse, *P. h. humanus*. During the febrile period and for 2-3 days after, lice may acquire rickettsiae while feeding. The feces of the louse become infective 2-6 days after. The louse usually dies within two weeks from pathological effects of the rickettsia. During the life of the infective louse, rickettsiae are excreted in the feces while feeding and the feces (or the crushed louse) are subsequently rubbed into the bite or other abraded areas, infecting the individual. Infection may also occur by inhalation of infected louse feces. Some people suffer a mild form of the disease (Brill's disease) and become asymptomatic carriers, relapsing or introducing the disease into healthy populations many years later.

3. Vector Ecology Profile. Female lice produce 4-5 eggs per day. Eggs adhere to clothing and can characteristically be found in the seams. Eggs are not viable for more that four weeks and lice survive for only a few days without a blood meal; therefore, clothing discarded for a month or more poses no danger of infestation. Eggs hatch in about seven days and the nymphs undergo three molts, maturing to adults in 18 days. Male and female nymphs and adults each feed several times per day. Transfer of lice occurs by direct contact or via infested clothing. Unsanitary conditions and overcrowding caused by social or cultural events, natural disasters, or conflicts that result in concentration of refugees or prisoners of war, may result in epidemics. The disease is endemic in cooler climates associated with the inter-Andean cordilleras.

4. Vector Surveillance and Suppression. Surveillance for body lice involves examination of suspect individuals and their clothing. Outbreaks of epidemic typhus among local populations are indicative of infestations. Soldiers should avoid contact with local populations. Suppression of the disease by individual soldiers and prisoners of war includes application of permethrin to BDUs and personal clothing, and maintaining a high level of personal hygiene (frequent bathing and laundering of clothing). The DoD currently relies on permethrin-treated uniforms to repel and control body lice on soldiers and prisoners of war.

Refugees, Displaced Persons and Other Civilians. The Department of Defense no longer has the capability to mass delouse refugees, displaced persons, and other civilians using insecticidal dusts with powered application equipment. Although DoD does not have a delousing method using insecticidal dusts, a variety of other methods might be warranted to delouse refugees, displaced persons, and other civilians during emergencies. These methods include administering oral or topical pediculicides, washing or replacing clothing, and applying repellents. Since the specific methods may not all be approved in the U.S., their use during emergencies should be at the discretion of the Joint Task Force Surgeon after consultation with non-governmental organizations (such as the World Health Organization) and host nation officials. Current options, which may involve the use of laundry and bath facilities, pediculicide lotions and

shampoos (which require bath facilities following application), and clothing exchanges (possibly with permethrin-treated clothing) will create logistical burdens on U.S. forces. Control of body lice must be concurrent with louse-borne disease control. Additionally, U.S. soldiers may only use insecticides or pediculicides labeled for use by the U.S. Environmental Protection Agency or the U.S. Food and Drug Administration.

I. Relapsing Fever (tick-borne). The etiological agents of tick-borne (endemic) relapsing fever are species of *Borrelia* transmitted by soft-bodied ticks in the genus *Ornithodoros*. The *Borrelia* strains are specific for the area and species of host tick. Relapsing fever, as its name implies, is a series of fevers interrupted by afebrile periods. The initial incubation period is 5–15 days, and fevers last for 2-9 days followed by an afebrile period of 2-4 days. The febrile/afebrile sequence may continue for up to 10 cycles. Fatalities occur in 2-10% of untreated cases.

1. Military Impact and Historical Perspective. Surveys conducted prior to 1986 indicate that 26% of habitations harbored *Ornithodoros* ticks infected with *Borrelia venezuelensis*. A large percentage of ticks are reportedly infected. The disease is prevalent in the large river valleys of the Pacific coast and the lowlands of eastern Colombia. Exposure of military personnel to soft-bodied ticks is unlikely. Troops should avoid using the potentially tick infested dwellings of indigenous populations for shelter.

2. Transmission Cycle. The vectors of the spirochete *Borrelia venezuelensis* are the soft-bodied ticks *Ornithodoros rudis* and *O. talaje.* Both ticks are anthropophilic, but *O. talaje* prefers rodents. The reservoirs for the *Borrelia* are ticks, humans, rodents, and other animals. After feeding on an infected reservoir, the spirochete invades the hemocoel, salivary glands, coxal glands, and ovaries within 3-4 days. Transovarial transmission of the spirochete occurs, but infection is maintained mostly between ticks and their reservoir hosts. Once infected, ticks remain infective for life. Transmission occurs by the bite of infected ticks or from spirochete-laden coxal fluid that enters through broken or even intact skin.

3. Vector Ecology Profile. *Ornithodoros rudis* and *O. talaje* lay eggs following a blood meal. The eggs hatch into six-legged larvae and multiple nymphal molts occur during development to the adult stage. The ticks live for 2-5 years or more. All stages and both sexes feed at night. *Ornithodoros rudis* is considered a parasite of man and is less dependent upon rodents, whereas *O. talaje* feeds principally on rodents and man is an accidental host that becomes infected by entering an infested area. Both ticks feed quickly and return to their hiding places; however, unlike *O. rudis, O. talaje* inflicts a painful bite.

4. Vector Surveillance and Suppression. The need for surveillance and suppression is unlikely, but visual inspection of cracks, crevices, thatching, walls and floor spaces, etc. may reveal infestations, particularly in sleeping areas. Residual pesticides applied to infested sites will eliminate ticks (TIM 24). Troops should avoid using indigenous shelters or caves for overnight bivouac sites. Use **Personal Protective Measures** when potential exposure cannot be avoided, particularly at night when *Ornithodoros* ticks feed (TIM 36).

V. Militarily Important Vector-Borne Diseases with Long Incubation Periods (>15 days)

A. Leishmaniasis. Leishmaniasis, a potentially disfiguring and sometimes fatal disease, is caused by protozoan parasites in the genus *Leishmania* transmitted by bites of phlebotomine sand flies. All vectors of this disease in the New World are in the genus *Lutzomyia*. The disease may take several forms, characterized as cutaneous leishmaniasis (CL), mucocutaneous leishmaniasis (MCL), or visceral leishmaniasis (VL). Incubation in humans may take as little as ten days to more than six months. Symptoms include ulcerative skin lesions (CL), lesions in the mucosal areas of the mouth and/or nose (MCL), and internal pathological manifestations resulting in fever, lymphadenopathy, anemia, enlargement of the liver and spleen, and progressive emaciation and weakness (VL). Untreated VL usually results in death. Seven different species of the parasite have been reported in Colombia: *Leishmania braziliensis*, *L. colombiensis*, *L. guyanensis*, *L. panamensis*, *L. mexicana*, and *L. amazonensis*. Although the clinical manifestations are not necessarily indicative of the particular strain/species of parasite, *L. braziliensis* is prone to cause the severe disfiguring MCL or "espundia," while *L. chagasi* produces life-threatening VL. Entomological or tissue specimens can be submitted to: Instituto Nacional de Salud, Centro Administrativo Nacional, Departmento de Parasitologia, Avenida Eldorado, Carrera 50, Bogot<, Colombia for identification of *Leishmania* species.

1. Military Impact and Historical Perspective. Cutaneous and mucocutaneous leishmaniasis are endemic countrywide in tropical and semi-tropical forests up to 1,800 m. Visceral leishmaniasis is found in more arid, rocky areas below 900 m in the valleys of the Magdalena River tributaries. Risk is likely year-round. Areas free of leishmaniasis or at very low risk include the Catatumbo River Basin, areas above 1,800 m, and the mountain highlands of southern Colombia. There has been an increase in the number of cases reported during the 1990s, probably as a result of better reporting and increased movement of populations into newly cleared forest areas. The potential for many cases of leishmaniasis (CL, MCL, and VL) is greatest in the forested Pacific coastal areas, lower Cauca River Valley, Magdalena River Valley, and the eastern slopes of the Cordillera Oriental. Some cases of leishmaniasis have been reported from the Amazon Basin, but little is known of the incidence or epidemiology of the disease in this vast area. Intrusion into such unknown habitats, particularly during extended jungle operations, could result in exposure to forest-dwelling sand flies, with serious disease implications. The same can be said for jungle operations in better understood endemic areas. Although not immediately immobilizing, leishmaniasis would be detrimental for units and individual soldiers. Cases of the simplest form of the disease (CL) would typically require a three-week course of hospitalization and treatment. Isolates of *L. braziliensis* resistant to standard antimony treatment have been documented. Scarring that accompanies the cutaneous form of the disease can also contribute to poor unit morale. Units of American soldiers undergoing jungle training in Panama have experienced attack rates as high as 32%. Soldiers on night patrols that require constant starting and stopping, sitting in place for periods of time, and proceeding through multiple habitats are extremely vulnerable to biting sand flies. Keeping as much of the skin covered as possible and using extended-duration deet repellents are crucial preventive measures.

2. Transmission Cycle. New World leishmaniases are primarily zoonoses. Humans become infected incidentally when they enter the habitat of the vector and reservoir. When an infected

sand fly feeds on an infected host, ingested parasites develop and proliferate within the fly as motile promastigotes. The flies become infective 8-20 days after an infected blood meal. At subsequent feedings, the motile promastigotes are injected into the bite, are sequestered by macrophages and become non-motile amastigotes. The amastigotes multiply, eventually rupturing the macrophages and dispersing to infect other macrophages. Hosts can infect feeding sand flies for a few months to several years. Wild canines and domestic dogs are proven and significant reservoirs of *Leishmania chagasi*. Reservoirs of other parasite species may include sylvatic rodents, hydraxes, sloths, canines, and man. Reservoirs in some localities remain unknown. As the list of potential sand fly vectors continues to grow, new species of *Leishmania* are also being discovered. At least 14 species of *Leishmania* have been identified in the Americas. *Leishmania chagasi*, *L*. (*Viannia*) *brasiliensis*, *L*. (*V*.) *colombiensis*, *L*. (*V*.) *guyanensis*, *L*. (*V*.) *panamensis*, *L*. (*Leishmania*) *amazonensis* and *L*. (*L*.) *mexicana* have been documented in Colombia.

3. Vector Ecology Profile. Some man-biting sand fly species do not transmit *Leishmania* parasites. Important proven vectors identified in Colombia by the WHO Expert Committee on Control of Leishmaniasis (1990) include *Lutzomyia* (*Lutzomyia*) *longipalpis*, *Lu.* (*Nyssomyia*) *flaviscutellata*, *Lu*. (*N*.) *umbratilis*, (*N*.) *trapidoi* and *Lu*. *spinicrassa*. In addition, Killick-Kendrick, 1990 lists *Lu*. (*Nyssomyia*) *anduzei* and *Lu*. (*Psychodopygus*) *c*. *carrerai* as proven vectors of leishmaniasis occurring in Colombia. Vector ecology profiles for species with available data are presented in Appendix A.6. Sand fly species known to harbor various species of *Leishmania* are annotated in a listing of Colombian *Lutzomyia* species, Appendix B.5.

4. Vector Surveillance and Suppression. Mechanical or mouth aspirators can be used to collect sand flies resting on interior or exterior walls of dwellings, tree trunks, rock crevices, animal shelters, etc. Collecting with an aspirator using human bait has the advantage of capturing anthropophilic species, but is a dangerous practice in endemic areas. Sticky paper traps made from bond paper soaked in castor oil can be placed in burrows, rock hollows and other places frequented by sand flies. The flies stick to the oil and can easily be removed, counted, identified, and tested or examined for parasites. Some species of sand fly are readily attracted to light traps and can be successfully trapped in standard CDC light traps with fine mesh collecting bags. Various types of animal-baited traps can also be employed (Disney and Shannon Traps). When establishing semi-permanent or permanent bivouac sites, clear areas to provide a vegetation-free barrior zone of about 50 m between encampments and forested areas. Apply residual pesticides to perimeter vegetation. Since sand flies are weak flyers, these measures will provide an effective barrier between potential sand fly vectors and humans. Apply residual pesticides to walls of buildings or to tents (interior and exterior), particularly in bivouac bedding areas (TIM 36). Implement ULV spray operations during the peak-biting period (dusk and dawn) for quick knockdown of flying sand flies (TIM 24). Wear BDUs with trousers bloused and sleeves rolled down just prior to sunset and after dark, apply extended-duration deet repellents on exposed skin, and use fine mesh bednets when possible (TIM 36).

B. Bartonellosis. Bartonellosis (Carrion's disease) is caused by *Bartonella bacilliformis* and has two clinical manifestations: Oroya fever and verruga peruana (verruga warts). Sand flies of the genus *Lutzomyia* are suspected vectors of the bacterium. The symptoms of Oroya are fever, headache, myalgia, arthralgia and severe hemolytic anemia that often results in death if

untreated. Verruga peruana is a chronic form of the disease marked by rather painless nodular lesions that vary in degree of development. It may be preceded by Oroya fever by weeks to months, although verruga peruana may also occur without any symptoms of the febrile illness. Symptoms usually occur 16-22 days following the bite of an infected sand fly, although they may not materialize for 3-4 months. Man is the only known reservoir of *B. bacilliformis*, which may circulate in the blood for weeks prior to clinical symptoms and for years thereafter. The bacteria may be transmitted during blood transfusions. Untreated, fatalities are estimated at 10-20%; however, the bacterium is susceptible to antibiotic therapy.

1. Military Impact and Historical Perspective. The first epidemic (>6,000 cases) occurred in the vicinity of the Rio Putumayo and the city of Pasto, NariZo Department, following the Colombo-Peruvian War (1932-34), and extended into the 1940s, spreading to Bolivar, Cauca Department, in 1941. The disease disappeared after 1941 and did not reappear until 1988, when a single case was reported from Pradera (953 m), Valle del Cauca Department. Although little threat to troops exists, operations on the eastern slopes and in the foothills of the Western Cordillera in NariZo, Cauca, and Valle del Cauca Departments would warrant precautions. American troops with no immunity to this disease might well experience a repeat of the Peruvian epidemic of 1885 that resulted in hundreds of deaths among Chinese brought to Peru to build the railroad. Although readily treatable today, many cases of bartonellosis could occur rapidly among nonimmune personnel, resulting in considerable downtime.

2. Transmission Cycle. Little is known about the epidemiology or transmission of bartonellosis. Humans are the only known reservoir. The role of animal reservoirs (if any) is unknown, although *Bartonella bacilliformis* was isolated from a single species of *Phyllotis* (leaf-eared mice) in Peru in 1948. Domestic guinea pigs (*Cavia porcellus*) and chickens were implicated as reservoirs in an Ecuadorian focus. The sand fly *Lutzomyia colombiana* was the suspected vector in the 1936-44 outbreak in southern Colombia, but the development of the organism in sand flies has never been observed. Cases were reported between 600-2,800 m.

3. Vector Ecology Profile. Although never proven, man-biting sand flies are incriminated as the vectors of *Bartonella* organisms. *Lutzomyia colombiana*, the suspected primary vector of *B. bacilliformis* in Colombia, is an aggressive anthropophilic species, biting from dusk to dawn. This sand fly has been collected in NariZo, Cauca, and Valle del Cauca Departments along the Cordellera Occidental. High populations have been reported in the area of Samaniego, NariZo Department.

4. Vector Surveillance and Suppression. See leishmaniasis, paragraph V.A.4, for surveillance and suppression of sand flies. Since little is known regarding the vector species responsible for transmitting bartonellosis, all species of sand flies should be controlled. Screen blood supplies for *B. bacilliformis* in endemic areas.

C. Onchocerciasis. The causative agent of Onchocerciasis is a filarial worm, *Onchocerca volvulus*, transmitted by black flies. Onchocerciasis is characterized by fibrous nodular skin lesions, which occur primarily on the scapular areas of the trunk and on the iliac crest. After many years, untreated victims may experience impaired vision or blindness.

1. Military Impact and Historical Perspective. The first cases of onchocerciasis in Colombia were documented in 1965 at restricted foci on the western slope of the Cordillera Occidental, along the upper reaches of the Rio Micay near San Antonio, Cauca Department, and in NariZo Department. Lower in the Rio Micay drainage toward Lopez, the river slows and *Simulium exiguum* are less abundant than they are near San Antonio. From 1990-1995, Ivermectin (antihelminthic) was administered to the populace in the NariZo focus. 1997 reports for that area found no evidence of onchocerciasis. Similar programs are under way in less populated areas along the upper Rio Micay, although 51 cases were reported there in a population of 670 in 1996. Contraction of onchocerciasis by troops in the known focus is possible but would have no impact on military operations.

2. Transmission Cycle. Humans are the primary host/reservoir for the filarial worm, *Onchocerca volvulus*. Black flies (*Simulium* spp.) feeding on man ingest the microfilariae found in the skin. The microfilariae penetrate the thoracic muscles, develop into infective larvae (up to 14 days), migrate to the head capsule, and escape into the wound during feeding. The larvae develop within nodular skin lesions, but females do not produce microfilariae until they reach maturity (6-12 months). The female filarial worms remain in the nodules and produce microfilariae, which migrate to various regions of the body, occasionally locating in the eye and causing visual impairment or blindness. Heavy infections can build up only from exposure to repeated bites by infected *Simulium* over a prolonged period. Untreated victims can serve as reservoirs of microfilariae for as long as 15 years (life span of a single mature female larva).

3. Vector Ecology Profile. *Simulium exiguum* is considered the primary vector of *O. volvulus*. It is thought to have three sibling species in neighboring Ecuador. The vector ecology profile for *S. exiguum* is presented in Appendix A.7. Although *Onchocerca* spp. are not endemic in the Amazon and Orinoco Basins, *Simulium oyapockense* does occur in those areas and is a proven competent vector.

4. Vector Surveillance and Suppression. Small streams (less than 5 m across) do not generally support larval/pupal development of *S. exiguum*; therefore, surveillance of developing populations should be conducted in larger, rapidly flowing streams. This multivoltine species has two or more developmental cycles per year. Surveillance of aquatic habitats will provide information to determine timely control strategies coinciding with the larval stage and target species. Visual inspection of streams/rivers and sampling of larvae from all areas of a body of water (e.g., rocks, submerged vegetation and debris, etc.) will provide information on species composition, stage of development, and population dynamics. The most favorable time to control black fly populations is during the aquatic larval feeding stages (before pupation). Formulations of *Bacillus thuringiensis israeliensis* are economical, operationally feasible, and effective in controlling black fly populations in main river systems before they emerge as biting adults. Chemical control measures are largely ineffective for controlling adult populations. **Personal Protective Measures** include avoidance of open sunlit areas along streams, wearing permethrin-treated BDUs with sleeves down and using extended-duration deet repellent on exposed skin surfaces (TIM 36). Avoid wearing shorts during daytime recreational activities, as *S. exiguum* bite primarily below the waist.

D. Mansonellosis. Mansonellosis is an infection by the filarial worms *Mansonella ozzardi* or *M. perstans*, transmitted by the bite of black flies (*Simulium* spp.) or biting midges (*Culicoides* spp.). Most infected people are completely asymptomatic, but some suffer joint pains, headaches, poor circulation in the lower extremities, inguinal lymphadenopathy, and pruritic red spots. Microfilariae may also enter the eye, causing eye infections.

1. Military Impact and Historical Perspective. Mansonellosis is endemic in the departments of Guainia (*M. ozzardi* and *M. perstans*), Vaupes, Amazonas, Antioquia and Choco (*M. ozzardi*). Minor foci of *M. ozzardi* occur in Meta, Casanare, and Vichada Departments, but all cases are from migratory Indians from known endemic areas to the southeast. Infection should be considered in all these departments along rivers where potential vectors are present. The military impact is negligible, but a few individuals could depart the theater of operations with *Mansonella* infections that might later require treatment.

2. Transmission Cycle. Both species of *Mansonella* have similar developmental cycles. Microfilariae are imbibed from a human host during blood feeding by species of *Simulium* or *Culicoides*. The microfilariae migrate from the midgut to the thoracic muscles of the vector, where they develop into second-stage larvae. By the 6^{th} (*Simulium*) or 9-11^{th} (*Culicoides*) day post-feeding, mature third-stage larvae are present in the head capsule. During feeding, the mature larvae crawl out of the proboscis and enter the bite puncture. Differentiated males and females migrate to the peritoneal cavity, mate, and produce microfilariae. The microfilariae are non-periodic and may be found in the blood and skin at all times.

3. Vector Ecology Profile. Vectors are poorly understood, but at least four species of the *Simulium amazonicum* complex in Colombia have been shown to support development of *M. ozzardi* to the infective stage. *Simulium argentiscutum*, an anthropophilic species, has also been incriminated as a vector. Biting midges, *Culicoides insinuatus, C. furens, C. paraensis*, and *C. caprilesi* have been shown to harbor infective stages of *M. ozzardi*, although *Culicoides caprilesi* is probably unimportant in transmission because of low infection rates.

4. Vector Surveillance and Suppression. See onchocerciasis, paragraph V.C.4., for surveillance, control and prevention of black flies. Anthropophilic species of *Culicoides* may be collected off of human bait using aspirators. Times of day for biting are species dependent. Some species can readily be trapped with standard CDC light traps equipped with fine mesh bags or DDVP killing jars. Avoid open sunlit areas along streams, wear permethrin-treated BDUs with sleeves down, and use extended-duration deet repellent on exposed skin surfaces (TIM 36).

E. Bancroftian Filariasis. Bancroftian filariasis is caused by infection with a nematode worm, *Wuchereria bancrofti*, transmitted to man by mosquitoes. Most infected individuals remain asymptomatic for years. Delayed symptoms include blockage of the lymphatics by adult filarial worms (elephantiasis). This occurs only after continuous exposure to infected mosquitoes over long periods of time. Occasional acute recurrent fevers and lymphadenitis occur as a reaction to the nematodes.

1. Military Impact and Historical Perspective. Records indicate an extremely low incidence, but occasional cases of elephantiasis occur in the Cartagena area, Atlantico Department. There would be no immediate impact on military operations, but potential delayed infections by adult worms in the lymphatics of individual soldiers are possible.

2. Transmission Cycle. Filariasis is caused by a nematode passed between humans and mosquitoes. Although other mosquitoes are implicated, *Cx. quinquefasciatus* is the principal vector in Colombia. *Wuchereria bancrofti* microfilariae in the Americas circulate in the peripheral blood of humans only at night. The greatest numbers of microfilariae are found in peripheral blood between 10:00 P.M. and 2:00 A.M. This nocturnal periodicity coincides closely with the nocturnal feeding preferences of *Cx. quinquefasciatus*. Imbibed microfilariae penetrate the stomach wall and migrate to the thoracic muscles, develop to infective larvae in 12-14 days, and crawl out through the mosquito's proboscis during feeding. The larvae penetrate the bite puncture, travel to the lymphatic system, molt twice, and in 6 or more months begin to produce microfilariae. Nocturnal periodicity of circulating microfilariae may occur for 10 years or more in infected humans.

3. Vector Ecology Profile. The vector ecology profile for *Cx. quinquefasciatus* is presented in Appendix A.3.

4. Vector Surveillance and Suppression. The principal vector, *Cx. quinquefasciatus*, is moderately attracted to CDC light traps. It is attracted to foul water for oviposition, as it breeds in heavily polluted water high in organic content. Reiter gravid traps are much more effective in determining populations of gravid females. This vector is an urban mosquito and will not be found far from domestic settings. **Personal Protective Measures** are the same as for other mosquito-borne diseases (dengue, yellow fever, malaria, etc.). During permanent encampments, do not permit water to puddle around water storage containers, field showers, vehicle wash areas, etc. Populations of *Cx. quinquefasciatus* will rapidly proliferate in such manmade breeding areas.

VI. Other Diseases of Potential Military Significance

A. Leptospirosis. Leptospirosis is caused by a number of different bacteria belonging to the genus *Leptospira*. Over 200 serovars have been catagorized as pathogenic. Individual serovars are often associated with a particular host; however, humans are considered only incidental and dead-end hosts. Military troops are often exposed to *Leptospira interrogans*, serovar *icterohaemorrhagiae*, which occurs in wild rodent populations. Rodents pass the leptospires in their urine. Ingestion of contaminated food or water, or contact with abraded skin and mucous membranes are the usual modes of transmission. Symptoms appear 4-19 days following infection and include sudden onset of fever, headache, chills, and severe myalgia of the calves and thighs. More complicated symptoms may follow. Erroneous diagnoses often include encephalitis, meningitis, or influenza. Rodent excreta are commonly flushed from runs, burrows, etc. into the water systems, so leptospirosis frequently follows natural drainages. Flooding enhances the likelihood of transmission from animals to man in at least two ways: 1) it brings the maintenance host for the organism in closer contact with humans; and 2) it disseminates the infectious urine of host animals, resulting in exposure of many more persons.

As the rainy season ends and streams begin to dry up, the leptospires become concentrated and are an increased threat to those who may bathe in, or drink the water. Avoid wading, bathing, or drinking untreated water and ensure that sanitation is maintained to eliminate sources of food and harborage attractive to rodents.

B. Rabies. Rabies is a zoonotic viral disease transmitted through the saliva of animal bites. It is almost invariably fatal for humans. Cats, dogs and bats are the principal reservoirs. Rabies is frequently epizootic among bats, wild canines and other carnivorous mammals. Soldiers should avoid contact with bats, local dogs, cats, and other animals. Immediate reporting for medical treatment is paramount when bites occur. Units should not adopt feral animals as mascots.

VII. Noxious/Venomous Animals and Plants of Military Significance

A. Arthropods. Identification keys for medically important arthropods are cited in Appendix G. Insect and arachnid bites and stings have the capacity to cause allergic reactions, create secondary infections and lower unit morale, not to mention their potential disease implications. U.S. Army trip reports filed by medical personnel (1997) indicate that insect bites and accompanying syndromes were a major complaint among soldiers and medical personnel in the Bolivian Army operating in lowland areas east of the Andes. Some of the lowland topography and climate of Colombia are somewhat comparable. Mosquitoes are major pests among the many biting or stinging arthropods of tropical and semi-tropical regions but are discussed above. Consult Appendix B.1. for mosquitoes that occur in Colombia. The following groups of arthropods include most of the major pests. Residual insecticides (chlorpyrifos, diazinon) applied to grounds and vegetation prior to encampment will eliminate many arthropod pests. Treat tentage periodically with permethrin and use d-phenothrin or other suitable aerosols within enclosed spaces (TIM 24). Avoid using pallets for tent flooring because these provide harborage for unwanted pests. The dual use of repellents on the skin (deet) and on the clothing (permethrin) (DoD Insect Repellent System), combined with the proper wearing of the BDU, will provide the individual soldier with nearly complete protection from most arthropods (TIM 36). Africanized honey bees, wasps, spiders, centipedes, scorpions, and other stinging or biting arthropods require additional precautions as noted below. When retiring, roll clothing tightly and stretch socks over boot tops to exclude crawling arthropods. Shake clothing and boots briskly before dressing and inspect for unwanted intruders. Foxholes are ideal pit traps for crawling arthropods (scorpions, centipedes, spiders, etc.) and snakes may also fall into them. Dug-in troops should always check foxholes for potentially dangerous animals. The following are the major groups of arthropod pests.

1. Ceratopogonidae (biting midges, no-see-ums, punkies). Some anthropophilic species of Ceratopogonidae (*Culicoides, Leptoconops*) are extremely annoying. Their bites may produce systemic allergic reactions as well as local irritation. Because of their small size, they may go unnoticed until an individual receives many bites. Breeding habits differ from species to species. Some are tree hole breeders, some breed in moist decaying vegetable material, while others are associated with the edges of streams, swamps, ponds and lakes, where adults lay eggs at the interface of moist soil and water. Development may occur in the water, wet soil, or in wet decomposing vegetation (see *Culicoides paraensis*, Appendix A.3). Massive emergence of

adults may occur in some species. *Culicoides furens* is a significant source of irritation in Pacific coastal areas associated with mangrove swamps.

2. Dipterans Causing Myiasis. The human bot fly (family Cuterebridae) and the primary screwworm (family Calliphoridae) are major causes of myiasis in humans throughout South America. The human bot fly (*Dermatobia hominis*), less than one centimeter in length, lays its eggs on other arthropods (usually diurnal blood-feeding dipterans, e.g., mosquitoes, deer flies, etc.). While the arthropod carrier feeds on a human host, the phoretic larva emerges onto the host's skin, penetrates, and begins to develop. The larva remains at the site of entry throughout its 3-4 month development, causing an irritating form of cutaneous myiasis. The adult female primary screwworm (*Cochliomyia hominivorax*) deposits numerous eggs in a few minutes on any area of broken skin (even sites as minor as a scratch or the site of a tick bite may be attractive). The eggs hatch in <24 hours and the larvae penetrate, feeding on live tissues. They feed for 3-6 days before pupating, causing significant pain and tissue damage. Females may also lay eggs in the nasal passages of sleeping humans. The larvae may invade the nasal cavities, sinuses, and eustachian tubes, causing severe damage. Human deaths have been reported from nasal infestations of the primary screwworm. Fortunately, screwworm flies do not fly after dark. Use extended-duration deet repellent on all exposed skin surfaces to prevent mosquitoes and other biting arthropods from gaining the contact required for *D. hominis* invasion. To prevent potential infestations of *C. hominivorax*, avoid sleeping during the day without headnets or bednets, and keep all cuts, scratches and open sores covered.

3. Lepidoptera (urticating moths/caterpillars). Numerous caterpillars in the tropics have poisonous urticating hairs that may cause serious dermatitis. Outbreaks of urticarial reactions have occurred periodically among military personnel in Panama. Investigations revealed that urticarial hairs of caterpillars had dropped into the local swimming pools, exposing swimmers. Avoid contact with caterpillars regardless of how harmless they might appear. Acute episodes of urticarial dermatitis caused by nocturnal moths of the genus *Hylesia* (Saturniidae) are frequently observed in some regions east of the Andes below 2,800 m/9,184 ft. Dermatitis is caused by poisonous abdominal setae of the female moths. Except indirectly, humans seldom contact these moths. Swarms of moths often invade villages at sunset and fly throughout the night around street or porch lights. The insects lose millions of setae that are carried by air currents and are ultimately inhaled or land on the skin of unsuspecting people. The moths emerge twice a year. Every 4-5 years an emergence cycle of immense proportions occurs, creating many cases of urticarial dermatitis. Clinically, lesions appear as an urticarial rash, especially on the neck and forearms. The rash remains for approximately a week. Lesions are aggravated by repeated nightly exposure to setae. Itching or rubbing the rash spreads the setae and exacerbates the condition. Newly exposed personnel are more vulnerable to an adverse reaction than previously "immunized" individuals. Oral antihistimines and steroids, and topical application of Natrium hyposulphite have proven useful.

4. Meloidae (blister beetles). Blister beetles produce cantharidin, a powerful vesicant that can cause blistering when it contacts exposed skin. Blister beetles are attracted to lights around sentry guard posts and frequently drop down soldiers' necks or onto their bare arms causing blisters. Such incidents are frequent among soldiers standing guard under streetlights in Panama.

Blisters usually result from rubbing or crushing the insects on the skin. Similar phototrophic behavior and blistering occurs in some species of beetles of the family Staphylinidae.

5. Simuliidae (black flies). Temperate Andean regions are not inhabited by anthropophilic species of black fly, although rivers and streams flowing out of the Andean foothills (toward the coast and toward the eastern lowlands) do produce black fly species that are annoying to humans. All anthropophilic black fly species in South America belong to the genus *Simulium*. *Simulium amazonicum, S. quadrivittatum, S. exiguum, S. metallicum, S. oyapokense* and *S. sanquineum* display some degree of anthropophily in Colombia. *Simulium* species reported in Colombia are listed in Appendix B.6.

6. Siphonaptera (fleas). Fleas can be an immense source of discomfort. Sensitivity to flea bites may vary from person to person. The most annoying fleas that commonly occur in Colombia are *Pulex simulans*, *P. irritans* (human flea), *Ctenocephalides canis* (dog flea), *Ctenocephalides felis felis* (cat flea), *Xenopsylla cheopis* (Oriental rat flea) and *Tunga penetrans* (sand flea, chigoe, jigger). *Pulex simulans* and *P. irritans* are frequently confused. *Xenopsylla cheopis*, the premier vector of plague, normally does not leave its common rodent hosts (*Rattus* spp.) in the domestic setting unless the rodents are exterminated without first chemically controlling the fleas. Fleas leaving dead hosts are quite mobile, jumping as much as 12 inches, and readily biting humans. *Xenopsylla cheopis* is most common where humidity is high. Dog and cat fleas are usually found in and about homes where animals roam free. Eggs are laid on the host and drop to the floor/ground, hatch and undergo three larval stages. When a dwelling is abandoned, the flea larvae will ultimately pupate and remain in a quiescent state for long periods of time. The vibrations of anyone entering such premises will stimulate a mass emergence of hungry adult fleas. Avoid using abandoned dwellings. *Pulex irritans*, although called the human flea, is a parasite of free-roaming domestic swine and is indiscriminate in its choice of hosts. This flea bites voraciously indoors and outdoors and is especially prevalent in domestic settings in higher Andean valleys. Although not encountered often, *T. penetrans* may infest primitive peridomestic settings. *Tunga penetrans*' ability to penetrate skin and remain embedded can cause extreme discomfort. Blousing trousers inside boots is essential in providing a barrier. Fleas will crawl under blousing garters.

7. Tabanidae (deer/horse flies). Species belonging to the genera *Chrysops*, *Haematopota* and *Lepiselaga* are important biting pests of man. Species of *Tabanus* are primarily zoophilic, but some may also bite humans. Deer and horse flies breed along rivers, lakes, swamps and other aquatic habitats. Although they are capable of extended flight, they usually remain close to their breeding grounds. Exposure is greatest close to sources of water and in low-lying areas. They inflict painful bites, and some species will bite through tight-fitting clothing such as t-shirts. Anthropophilic species are strictly diurnal and feed in open or forested areas, depending on the species. Skin repellents are deterrents but do not provide complete protection from biting tabanids. Avoid operating in wet swampy areas and other aquatic habitats when possible, and keep sleeves down when tabanids are active.

8. Chiggers and Ticks. Chiggers are parasitic larvae of the mite families Trombiculidae and Leuwenhoekiidae. Nymphal and adult stages are non-parasitic. Primary man-biting species include *Eutrombicula alfreddugesi*, *E. batatas*, and *E. tropica*. Other species of *Eutrombicula*

may also be a nuisance but the systematics of the genus is currently not well established in the region. Species of *Neotrombicula* may occasionally be involved in human infestations. Usual hosts for species affecting man are rodents, birds and reptiles, man being an accidental dead-end host. Chiggers do not burrow into the skin, or take a blood meal, but feed on liquefied tissue. Attached chiggers secrete powerful enzymes that disintegrate host skin cells. Eventually, the host tissue-response forms a feeding tube (stylostome) from which the mite imbibes dissolved tissue fluids. Intense itching is caused by the formation of the stylostome. Unlike chiggers, six-legged larval (seed) ticks and eight-legged nymphs of the family Ixodidae (hard ticks) each require a blood meal to molt to the next stage. Ticks penetrate the host with their mouthparts and remain attached for various periods, depending on the species. Refer to Appendix H for proper tick removal procedures. Although different species have different host preferences, mammals (small and large) or birds are the most common hosts. Individuals react differently to bites of these two groups of ectoparasites. The intense itching that often accompanies chigger and tick bites may lead to excoriated lesions that are subject to secondary infections. Their bites can become a serious morale problem for individuals, particularly those that are sensitized. The host-seeking behavior of chiggers and ticks is similar. The larvae of both families display a clustering behavior where hundreds will congregate on an object (leaf, twig, etc.) in a host questing posture. Potential hosts (man included) may brush against the cluster, becoming infested with hundreds of chiggers or larval seed ticks. Either may quest on vegetation up to several feet in height, but chiggers tend to remain closer to the ground (boot top height or less). These behaviors account for the spotty distribution of infestations, in which some people get badly bitten, while others receive only a few bites or none. Initial chigger infestations usually begin at ankle level. Chiggers have a propensity for attaching to humans under tight-fitting clothing (e.g., boot tops, belt line, bra lines) and areas of thin skin (e.g., behind knees, groin, peritoneal and axillary regions), while ticks are less selective but prefer hairlines. Species of Colombian ticks and their hosts are listed in Appendix B.4. Use extended-duration deet repellent on exposed skin and ensure that BDUs are impregnated with permethrin (TIM 36). Ticks and chiggers will crawl under trouser blousings, avoiding both the clothing and repellent barriers of the BDU. It is essential to tuck trousers inside boot tops to maximize the protective barrier.

9. Scorpions. Species of scorpions that occur in Colombia are listed in Appendix B.7. Most South American scorpions are considered no more toxic than common wasps and bees, although some people may be hypersensitive and suffer significant reactions. *Centruroides margaritatus* inflicts more stings than any other scorpion in Colombia but is not extremely poisonous. Two Colombian species whose stings cause severe reactions, high fevers, and sometimes death are *Centruroides noxius* and *C. suffusus*. Scorpions are most active at night and may crawl into clothing and bedding. See precautions paragraph VII.A. above.

10. Spiders. Brown recluse spiders (*Loxosceles laeta, L. rufescens*, and *L. rufipes*), black widow spiders (*Latrodectus* spp.), and banana spiders (*Phoneutria boliviensis* and *P. colombiana*), are common poisonous spiders that occur in Colombia. Banana spiders present the greatest potential for serious spider bites among troops that sleep on the ground. The nocturnal wandering habits of these solitary spiders bring them into frequent contact with people by crawling into clothing, shoes, bedding, tentage and equipment. Little has been written about *P. boliviensis* and *P. colombiana* but the Brazilian species *Phoneutria nigriventer* has a reputation for being extremely aggressive and inflicts many serious bites in eastern Brazil. *Phoneutria*

nigriventer and *P. reidyi* occur in the neighboring regions of Amazonas State, Brazil, and may also occur in the southeastern lowlands of Colombia. *Phoneutria* and *Latrodectus* venom is neurotoxic, while *Loxosceles* venom may cause severe necrotic lesions. Other species of spiders are capable of causing painful bites; however, their reactions are generally restricted to local pain, itching, and swelling. Prevent spider bites by following the guidelines in paragraph VII.A. above.

11. Centipedes. Centipedes in tropical countries attain considerable size and are capable of inflicting painful bites that cause swelling and local tenderness, but they are not considered dangerous. Their toxicity is comparable to that of a bee sting, although the acute pain is much greater and there is more tissue trauma at the sight of the bite. The width of cheliceral punctures may exceed ¾ of an inch.

12. Bees, Wasps and Hornets. The most significant threat among the Hymenoptera is the Africanized honey bee (AHB), endemic throughout Colombia. Several behavioral features of AHB make them especially dangerous to military personnel: 1) they are extremely aggressive and defensive of their hive; 2) they swarm and abscond excessively; and 3) they frequently build hives close to the ground in any protected cavity. Operations in wooded areas increase the risk of troops interacting with swarming AHB or their hives, particularly at night when they cannot be seen. If encounters occur, move away as swiftly as possible. Bees are not as aggressive away from the hive and have no method of homing in on an intruder at night. Avoid hives and swarming colonies of honey bees (TIM 34). Wasps and hornets are ever present in most localities and isolated stings can be expected. Bee sting kits should be available for individuals with known hypersensitivity to bee stings.

B. Snakes. The venomous terrestrial snakes of northwestern South America belong to the families Elapidae and Viperidae. Coral snakes, cobras, kraits, mambas, and sea snakes constitute the Elapidae but, of the terrestrial members, only coral snakes (*Leptomicrurus* spp. and *Micrurus* spp.) occur in the Neotropical Region. Although extremely toxic, coral snakes' timid, reclusive habits preclude them from being encountered often. Coral snakes will not bite unless handled or carelessly provoked, and their fangs are short, requiring a chewing action that delivers only small quantities of venom. Although they pose little threat, their infrequent bite victims suffer 50% fatalities. The family Viperidae contains six venomous genera (*Bothriechis*, *Bothriopsis*, *Bothrops*, *Crotalus*, *Lachesis*, and *Porthidium*) in northwestern South America. These snakes are frequently encountered and their bites may be life threatening. Arboreal or semi-arboreal genera include *Bothriechis* and most species of *Bothriopsis* and *Bothrops*. Arboreal species are especially dangerous because their fangs are very long, venom is copiously delivered, and victims are likely to be bitten on the head, arms and upper trunk areas. *Crotalus*, *Lachesis*, and *Porthidium* spp. are primarily ground dwellers. During floods, many of the ground-dwelling snakes (poisonous and nonpoisonous) may be concentrated along the high water line, increasing the risk of bites. Venoms of viperids vary in toxicity from species to species. Most deaths are caused by *Bothrops* spp., to which the fer de lance (*B. asper*) and barba amarilla (*B. atrox*) belong. Other extremely dangerous snakes include the bushmaster (*Lachesis muta* spp., rarely encountered because of its preference for nocturnal activity) and the tropical rattlesnake (*Crotalus durissus* spp.), reportedly the most dangerous snake in the Americas. Venomous snakes known to occur in Colombia are listed in Appendix C, together with distributional data.

Currently recognized species follow the systematic scheme of K.R.G. Welch, 1994, Snakes of the World: A Checklist. J. Coborn, 1991, The Atlas of Snakes of the World, is an excellent colored pictorial reference for many of the species that may be encountered. Sources of snakebite antivenoms are provided in Appendix D.

C. Plants. Plants that cause contact dermatitis are listed in Appendix E, and those that produce systemic toxic symptoms (and even death) when ingested are listed in Apprendix F. The components of each plant species (leaves, seeds, etc.) and chemicals that are thought to cause skin reactions or systemic poisoning are listed along side of each species. Plants most important to military personnel are *Toxicodendron* spp. and *Anacardium occidentale* (cashew nut). Poison ivy and poison oak in North America belong to the genus *Toxicodendron*. These are abundant at many CONUS installations, often causing skin reactions that require soldiers to be placed "on quarters" or occasionally hospitalized. The seriousness of lesions caused by poison ivy or poison oak is exacerbated in the tropics. The cashew nut is extremely toxic if eaten uncooked, and the resin in the plant and fruit can inflict extensive skin damage. Troops should be taught to recognize these plants and to avoid them.

VIII. Selected References

Military Publications

1966. Poisonous Snakes of the World, A Manual for Use by U.S. Amphibious Forces, NAVMED P-5099, BUMED, Department of the Navy, U.S. Gov. Print. Off., 212 pp.

1985. Technical Information Memorandum (TIM) 13. Ultra Low Volume Dispersal of Insecticides by Ground Equipment. AFPMB, 19 pp.

1998. TIM 26. Tick-Borne Diseases: Vector Surveillance and Control. AFPMB, 53 pp., Appendices A-J.

1998. TIM 24. Contingency Pest Management Pocket Guide. 5th Edition, AFPMB, 122 pp.

1991. Technical Guide (TG) 138. Guide to Commensal Rodent Control. U.S. Army Environmental Hygiene Agency.

1992. Technical Bulletin (TB MED) 561. Occupational and Environmental Health Pest Surveillance. Headquarters, Department of the Army, Washington, DC.

1993. TIM 31. Contingency Retrograde Washdowns: Cleaning and Inspection Procedures. AFPMB, 8 pp., Appendices A-H.

1995. TG 103. Prevention and Control of Plague. U.S. Army Center for Health Promotion and Preventive Medicine, 100 pp.

1995. TIM 34. Bee Resource Manual with Emphasis on the Africanized Honey Bee. AFPMB, 22 pp.

1996. TIM 36. Personal Protective Techniques Against Insects and Other Arthropods of Military Significance. AFPMB, 43 pp., 4 Appendices, Glossary.

Hamilton, D.R. 1995. Management of Snakebite in the Field. (unpublished document compiled by LTC Hamilton, filed as DPMIAC 162252).

Other Publications

Alexander, B. 1995. A Review of Bartonellosis in Ecuador and Colombia. Am. J. Trop. Med. Hyg., 52(4): 354-59.

Anonymous. 1980. Resistance of Vectors of Disease to Pesticides. WHO Technical Report Series No. 655, Fifth Report of the WHO Expert Committee on Vector Biology and Control, 58 pp.

Anonymous. 1990. Control of Leishmaniases. WHO Technical Report Series No.793, Report of a WHO Expert Committee, 158 pp.

Anonymous. 1992. Vector Resistance to Pesticides. WHO Technical Report Series No. 818, Fifteenth Report of the WHO Expert Committee on Vector Biology and Control, 62 pp.

Anonymous. 1995. Onchocerciasis and Its Control. WHO Techinical Report Series No. 852, Report of a WHO Expert Committee on Onchocerciasis Control.

Anonymous. 1995. Vector Control for Malaria and Other Mosquito-Borne Diseases. WHO Technical Report Series No. 857, Report of a WHO Study Group, 91 pp.

Barreto, P. 1969. The Species of Black Flies Found in Colombia (Diptera: Simuliidae). J. N.Y. Entomol. Soc., 77(1): 31-35.

Benenson, A.S. (Ed.). 1995. Control of Communicable Diseases Manual. 16th Edition, American Public Health Association, Washington, D.C., 577 pp.

Bequaert, J.C., and S. Renjifo-Salcedo. 1946. The Tabanidae of Colombia (Diptera). Psyche, 53(3-4): 52-88.

Blohm, H. 1962. Poisonous Plants of Venezuela. Harvard University Press, Cambridge. 136 pp.

Brenner, R.R. and A. de la M. Stoka. 1987. Chagas' Disease Vectors. Volume I. Taxonomic, Ecological, and Epidemiological Aspects. CRC Press, Inc., Boca Raton, Florida, 155 pp.

Campbell, J.A. and W.W. Lamar. 1993. The Venomous Reptiles of Latin America. Comstock Publishing Associates, Ithaca, New York, 425 pp.

Coborn, J. 1991. The Atlas of Snakes of the World. T.F.H. Publications, Inc., NJ, 591 pp.

Crosskey, R.W. 1990. The Natural History of Blackflies. British Museum (Natural History), John Wiley & Sons, New York, 711 pp.

Crosskey, R.W. and T.M. Howard. 1997. A New Taxonomic and Geographical Inventory of World Blackflies (Diptera: Simuliidae). The Natural History Museum, London, 144 pp.

D'Alessandro, A., P. Barreto and C. A. Duarte. 1971. Distribution of Triatome-Transmitted Trypanosomiasis in Colombia and New Records of the Bugs and Infections. J. Med. Entomol., 8: 159-172.

Doss, M.A., M.M. Farr, K.F. Roach & G. Anastos. 1978. Index-Catalogue of Medical and Veterinary Zoology, Special Publication No. 3. Ticks and Tickborne Diseases. IV. Geographical Distribution of Ticks. USDA, Washington, DC, 648 pp.

Fairchild, G.B. 1971. Family Tabanidae, Fascicle 28(1): 28-163, *In*: Papavero, N. (Ed.), A Catalogue of the Diptera of the Americas South of the United States. Museo de Zoologia, Universidade de Sat Paulo.

Fairchild, G.B. and J.F. Burger. 1994. A Catalog of the Tabanidae (Diptera) of the Americas South of the United States. Mem. Am. Ent. Inst. No. 55, 249 pp.

Gaffigan, T.V. and R.A. Ward. 1985. Index to the Second Supplement to "A Catalog of the Mosquitoes of the World (Diptera: Culicidae)." Mosq. Syst., 17(1): 52-63.

Golay, P., H.M. Smith, D.G. Broadley, J.R. Dixon, C. McCarthy, J-C. Rage, B. Sch@tti, and M. Toriba. 1993. Endoglyphs and Other Major Venomous Snakes of the World: A Check List. AXEMIOPS S.A., Herpetological Data Center, 478 pp.

Harwood R.F. and James, M.T. 1979. Entomology in Human and Animal Health. 7th Edition, MacMillan Publishing Company, Inc., New York, 548 pp.

Kettle, D.S. (Ed.). 1995. Medical and Veterinary Entomology, 2nd Edition, CAB International, University Press, Cambridge, 725 pp.

Killick-Kendrick, R. 1990. Phlebotomine Vectors of the Leishmaniases: A Review. Med. Vet. Entomol., 4: 1-24.

Kim, K.C. and R.W. Merritt. (Eds.) 1987 [1988]. Black Flies: Ecology, Population Management, and Annotated World List. Pennsylvania State University, University Park & London, 528 pp.

Knight, K.L. 1978. Supplement to "A Catalog of the Mosquitoes of the World (Diptera: Culicidae)." Thomas Say Foundation, Entomological Society of America, Supplement to Vol. 6, 107 pp.

Knight, K.L. and A. Stone. 1977. A Catalog of the Mosquitoes of the World (Diptera: Culicidae). 2nd edition. Thomas Say Foundation, Entomological Society of America, Vol. 6, 611 pp.

Lee, V. H., et. al. 1969. Haematophagous Arthropods of Rio Raposo, Valle, Colombia. Caldasia, Bogot<, 10(49): 441-58.

Lewis, R.E. 1972. Notes on the Geographical Distribution and Host Preferences in the Order Siphonaptera. Part 1. Pulicidae. Journal of Medical Entomology 9(6): 511-520.

Lewis, R.E. 1973. Notes on the Geographical Distribution and Host Preferences in the Order Siphonaptera. Part 2. Rhopalopsyllidae, Malacopsyllidae and Vermipsyllidae. Journal of Medical Entomology 10(3): 255-260.

Lewis, R.E. 1974a. Notes on the Geographical Distribution and Host Preferences in the Order Siphonaptera. Part 3. Hystrichopsyllidae. Journal of Medical Entomology 11(2): 147-167.

Lewis, R.E. 1974b. Notes on the Geographical Distribution and Host Preferences in the Order Siphonaptera. Part 4. Coptopsyllidae, Pygiopsyllidae, Stephanocircidae and Xiphiopsyllidae. Journal of Medical Entomology 11(4): 403-413.

Lewis, R.E. 1974c. Notes on the Geographical Distribution and Host Preferences in the Order Siphonaptera. Part 5. Ancistropsyllidae, Chimaeropsyllidae, Ischnopsyllidae, Leptopsyllidae and Macropsyllidae. Journal of Medical Entomology 11(5): 525-540.

Lewis, R.E. 1975. Notes on the Geographical Distribution and Host Preferences in the Order Siphonaptera. Part 6. Ceratophyllidae. Journal of Medical Entomology 11(6): 658-676.

Lucas, S. 1988. Spiders in Brazil. Toxicon, 26(9): 759-72.

Lucas, S.M. and J. Meier. 1995. Biology and Distribution of Spiders of Medical Importance. pp. 239-58, *In*: Handbook of Clinical Toxicology of Animal Venoms and Poisons, Meier, J. and J. White (Eds.), CRC Press, Boca Raton & New York, 752 pp.

Marinkelle, C. J. 1972. Colombian Triatominae and Their Infestation with Trypanosomatid Flagellates. Mitt. Inst. Colombo-Alem<n Invest. Cient., 6: 13-29.

Mills, J.N., J.E. Childs, T. G. Ksiazek, C.J. Peters, and W.M. Velleca. 1995. Methods for Trapping & Sampling Small Mammals for Virologic Testing. U.S. Department of Health and Human Services, Centers for Disease Control and Prevention, Atlanta, GA, 61 pp.

Monath, T.P. (Ed.). 1988/89. The Arboviruses: Epidemiology and Ecology. Volumes I-V, CRC Press, Boca Raton, Florida.

Pipken, A. 1968. Domiciliary Reduviid Bugs and the Epidemiology of Chagas' Disease in Panama (Hemiptera: Reduviidae: Triatominae). J. Med. Entomol., 5: 107-124.

Polis, G.A. 1990. The Biology of Scorpions. Stanford University Press, Stanford, California, 587 pp.

Reinert, J.F. 1975. Mosquito Generic and Subgeneric Abbreviations (Diptera: Culicidae). Mosq. Syst., 7(2): 105-10.

Reinert, J.F. 1982. Abbreviations for Mosquito Generic and Subgeneric Taxa Established Since 1975 (Diptera: Culicidae). Mosq. Syst., 14(2): 124-26.

Reinert, J.F. 1991. Additional Abbreviations of Mosquito Subgenera: Names Established Since 1982 (Diptera: Culicidae). Mosq. Syst., 23(3): 209-10.

Roze, J.A. 1966. La Taxonomia y Zoogeografia de los Ofidios en Venezuela. Universidad Central de Venezuela, Caracas, 362 pp.

Roberts, D.R., L.L. Laughlin, P. Hsheih and L.J. Legters. 1997. DDT, Global Strategies, and a Malaria Control Crisis in South America. Emerg. Inf. Dis., 3(3): 295-302.

Ryckman, Raymond E. 1986. The Triatominae of South America: A Checklist with Synonymy (Hemiptera: Reduviidae: Triatominae). Bull. Soc. Vector Ecol., 11(2): 199-208.

Schultz, M.G. 1968. A History of Bartonellosis (Carrion's Disease). Am. J. Trop. Med. Hyg., 17: 503-515.

Shelley, A.J. 1988. Vector Aspects of the Epidemiology of Onchocerciasis in Latin America. Ann. Rev. Entomol., 30: 337-66.

Shelley, A.J. 1988. Biosystematics and Medical Importance of the *Simulium amazonicum* Group and the *S. exiguum* Complex in Latin America. *In*: Biosystematics of Haematophagous Insects, M.W. Service, (Ed.), Systematics Association Special Volume No. 37: 203-220.

Shelley, A.J., M. Arzube and C.A. Couch. 1989. The Simuliidae of the Santiago Onchocerciasis Focus of Ecuador. Bull. Br. Mus. Nat. Hist. (Ent.), 58(1): 79-130.

Shelley, A.J., C.A. Lowry, M. Maia-Herzog, A.P.A. Luna Dias and M.A.P. Moraes. 1997. Biosystematic Studies on the Simuliidae (Diptera) of the Amazonia Onchocerciasis Focus. Bull. Nat. Hist. Mus., London, (Ent.), 66(1): 1-121.

Ward, R.A. 1984. Second Supplement to "A Catalog of the Mosquitoes of the World (Diptera: Culicidae)." Mosq. Syst., 16(3): 227-70.

Ward, R.A. 1992. Third Supplement to "A Catalog of the Mosquitoes of the World (Diptera: Culicidae)." Mosq. Syst., 24(3): 177-230.

Welch, K.R.G. 1994. Snakes of the World, a Check List. Vol. 1, Venomous Snake. R & A Research and Information Limited, 135 pp.

Wilson, D.E. and D.M. Reeder. 1993. Mammal Species of the World: A Taxonomic and Geographic Reference. 2nd Edition, Smithsonian Institution Press, Washington, DC, 1206 pp.

Wygodzinsky, P. and S. Coscar\n. 1967. A Review of *Simulium* (*Pternaspatha*) Enderlein (Simuliidae, Diptera). Bull. Am. Mus. Nat. Hist., 136(2): 49-116.

Appendix A.1. Disease Vector Ecology Profile: Vectors of Malaria in Colombia.

VECTOR	GEOGRAPHIC DISTRIBUTION	TRANSMISSION SEASON	LARVAL HABITAT	BITING BEHAVIOR	RESTING BEHAVIOR	FLIGHT RANGE
Anopheles darlingi	Principal vector east of the Andes in forest and savanna lowlands of the Orinoco and Amazon Basins.	Most prevalent during rainy season. Populations decrease during the dry season. Biting activity peaks 30 minutes after sunset with a smaller peak just before sunrise.	Generally a riverine species, but also reported in clear water in streams, ponds, and swamps with algae and floating vegetation. Partial shade. Areas of secondary growth. Water temperatures may vary from 17-33°C. Requires pH near 7.0.	Exophagic[1] or endophagic[2].	Endophilic[3]. Rests on walls 10 minutes before biting. After feeding, rests on vertical surfaces within 2 m of floor, or on ceilings.	Routinely 200-1,500 m. Maximum: 1.6 km.
Anopheles pseudopunctipennis	High, warm, humid regions of the Cauca and Magdalena Valleys.	Populations increase at onset of dry season (June) and continue through September. Dusk and dawn.	Sunny pools left along rivers in the dry season. *Spirogira* required for larval development. Water temperatures between 18-20°C, pH. 7.5-8.5.	More anthropophilic than zoophilic. Exophagic or endophagic.	Prefers upper rafter areas indoors. Negatively phototrophic.	Females: 3-6 km. Males: 400-500 m.
Anopheles nuneztovari	Major vector in Caribbean coastal lowlands, Cordoba, Sucre, Magdalena and Atlantico Departments.	Most abundant during April and May, particularly where *An. darlingi* is absent. Most active 2200-2400 hours.	Along grassy margins of open marshy areas, ponds and lakes. Full sunshine or partial shade. Aquatic vegetation and algae often abundant.	Endophagic. Bites about equally in-doors and outdoors. Exits dwellings immediately after feeding.	Exophilic[4] first 2 hours after dark, but enters dwellings thereafter. Rests on wall 1 m or less from floor.	Unknown.
Anopheles albimanus	Pacific coastal plains along waterways. Caribbean coastal lowlands between Panama and Sierra Nevada de Santa Marta. Usually humid areas below 400 m.	Peak activity 2200-2400 hours.	Areas of secondary growth, full sunlight/partial shade. Clear to polluted water with scum/algae, springs, seepages, borrow pits, streams, canals and ponds. Associated vegetation: species of *Pistia, Elodea, Naias, Chara,* and *Utricularia*.	Anthropophilic or zoophilic.	Mostly exophilic, but will enter dwellings. Will rest in any protected wooded area.	Routinely 400-600 m. Maximum: 3 km.
Anopheles calderoni	Coastal lowlands below 250 m.	Unknown.	Small streams, irrigation canals, swamps associated with dense emergent vegetation, especially cattails (*Typhas* spp.). Prefers water temperatures of around 26°C.	Anthropophilic.	Unknown.	Unknown.
Anopheles aquasalis	Pacific coastal lowlands. Usually 200-300 m from coastline, but may extend 5-8 km along tidal estuaries.	Throughout year, but highest populations during low/moderate rainfall (Rio de Janeiro, Brazil). Weakly attracted to light traps. Crepuscular. Peak activity from dark to a few hours after sunset.	Prefers brackish water, 0.2-1.5% NaCl, but will breed in fresh water. Water free of plants in shade.	More zoophilic, but feeds on humans in absence of domestic animals.	Exophilic or endophilic resting on walls 1 m or less from floor.	Unknown in Colombia, but 350-2,000 m in Panama.

Appendix A.1. (Cont'd)

VECTOR	GEOGRAPHIC DISTRIBUTION	TRANSMISSION SEASON	LARVAL HABITAT	BITING BEHAVIOR	RESTING BEHAVIOR	FLIGHT RANGE
Anopheles neivai	Predominant in Pacific coastal regions in humid tropical forests up to 1,000 m (reported as high as 1,400 m in Panama)	Biting activity is bimodal from 1800-1900 hours, less in mornings 0500-0600 (September/ October).	Forests where abundant rainfall fills leaf axils of terrestrial and arboreal bromeliads. In Panama it breeds in bases of large wild pineapple leaves, *Ananas magdalenae*.	Anthropophilic and exophagic.	Exophilic.	Unknown.
Anopheles bellator	Important vector in Buenaventura vicinity and south along Pacific coast.	Evening biter, but also during the day in shade.	Leaf axils of terrestrial epiphytic bromeliads and in tree holes. Occurs in dryer areas of forest at higher elevations.	Feed mostly on humans and is exophagic.	Sylvatic mosquito, but will enter houses.	Unknown.
Anopheles agyritarsis	Secondary vector when in large numbers. Low and intermediate elevations up to 200 m. Most records are in foothills of the Cordillera Oriental and coastal plains.	Crepuscular with peak feeding in early morning and evening.	Stagnant ponds, rain puddles, springs, animal tracks, artificial containers. Full or partial shade. Often in sunny areas of secondary growth. Grasses and emergent aquatic vegetation.	Will feed on humans, but mostly zoophilic.	Exophilic.	Unknown.
Anopheles triannulatus	Potential vector, when found in large numbers, east of the Andes in lowland Departments of Aruca, Boyaca, and Meta.	Populations begin in May, peak in August, and are gone by late February. Peak between 1900-2000 hours.	Permanent ponds, lakes, slow streams, and river margins exposed to full sunlight, or partial shade. Clear freshwater associated with species of *Pistia stratiotes, Eichhornia, Azolla, Utricularia, Jussiaea, Elodea and Salvinia*.	Exophagic, anthropophilic and zoophilic, especially on bovines and equines.	Rarely found inside houses.	Unknown.
Anopheles marajoara	Secondary vector confined to tropical lowlands east of Andes below 400 m.	Throughout year, but many records reported in May-August and November (dry season). Biting habits unknown.	Large ground pools, small stream pools, swampy shores of lakes, small road puddles, small ponds, marshy depressions in swamps. Full sunlight and rarely partial shade. Most sites with muddy bottom and grassy or herbaceous vegetation. Associated aquatic plants include green algae and water hyacinth (*Ceratophyllum*).	Prefers larger hosts: humans and equines.	Unknown in Colombia, but exophilic in Panama and Venezuela, and endophilic both day and night in Amazonia and other areas of Brazil.	Females: 560-1500 m
Anopheles braziliensis	Potential vector east of the Andes throughout Colombia.	Predominantly at end of rainy season through dry season (May-September). Maximum activity in evenings or in middle of day during overcast days.	In secondary growth, in pastures, and forest clearings. Full sunlight or partial shade. Clear ponds and stagnant swamps with mud bottoms, grassy margins, floatage and/or algae.	Anthropophilic or zoophilic. Exophagic.	Unknown in Colombia, but endophilic in Amazonia, Brazil.	Unknown.

[1]Exophagic – bites outdoors. [2]Endophagic – bites indoors. [3]Endophilic – rests indoors. [4]Exophilic – rests outdoors.

Appendix A.2. Vector Ecology Profile: Vectors of Dengue and Yellow Fever in Colombia.

VECTOR	VIRUS	GEOGRAPHIC DISTRIBUTION	TRANSMISSION SEASON	LARVAL HABITAT	BITING BEHAVIOR	RESTING BEHAVIOR	FLIGHT RANGE
Aedes aegypti (*Aedes albopictus*)[1]	Dengue, Urban Yellow Fever	In urban, suburban, and rural communities of Atrato Basin, Cauca Valley, Magdalena Valley, Catatumbo Basin, and Atlantic coastal lowlands.	Populations increase at the onset of the rainy season when artificial containers are filled and vectors are prone to congregate more indoors. Biting occurs throughout daylight hours.	Almost exclusively in artificial containers associated with man, i.e., discarded tires, flower pots, vases, rain gutters, rain barrels, cisterns, etc. Occasionally breeds in leaf axils such as *Agave* spp. and banana palms. Females lay single eggs, 3 larval instars develop in 9 days (4-7 with ideal temperatures), pupae 1-5 days.	Agressively anthropophilic. Equally exophagic and endophagic.	Endophilic and exophilic. Rests during hours of darkness.	Usually less than 200 m. Maximum: ca. 2 km., especially when breeding areas are scarce.
Haemagogus janthinomys	Sylvatic Yellow Fever	Areas of primary rain forest throughout country, particularly in the Amazon, Orinoco, Catatumbo, Atrato and Magdalena River Basins.	Present year round, but population peak during the wettest season and decline during dryer spells. Bites during the daytime only primarily between 1200-1400 hours in forests.	Breeds in tree holes and bamboo stumps. The gonotrophic cycle lasts about 10 days and females may live as long as 95 days (average ca. 2 weeks). Over 75% of the eggs are laid between 1200-1600 hours with no eggs laid at night. Most eggs are laid during rainy periods.	Little biting activity outside of 1200-1400. Will leave canopy to bite at ground level, especially in damaged forest and along forest edges.	Canopy mosquito.	Unknown, but thought to be very limited.
Sabethes chloropterus	Sylvatic Yellow Fever	Areas of primary rain forest throughout country, particularly in the Amazon, Orinoco, Catatumbo, Atrato and Magdalena River Basins.	Adults in forest throughout year and most prevalent during rainy season (July-September in Panama). Adults found through dry season when species of *Haemagogus* are absent.	Tree hole breeders with preference for large cavities with small openings that hold water through dry season. Can survey for eggs with bamboo traps (tops closed with small hole through the side). Population drops in dry season.	Aggressive human biters. 10% of the specimens in a Panama study were collected at ground level using human bait.	Canopy mosquito.	Unknown.

[1] The vector ecology profile for *Ae. albopictus* differs little from that of *Ae. aegypti*. Although *Ae. albopictus* does not presently occur in Colombia, it can be expected to spread from western Brazil into Colombia.

Appendix A.3. Vector Ecology Profile: Vectors of Arboviruses Other Than Dengue or Yellow Fever in the Amazon Basin and Associated Northwestern Regions of South America.

The geographic distribution of vectors is given in broad terms and a vector may occur in only part of the country mentioned. Furthermore, the distribution given does not imply that any species is a vector over the whole of its range. Country names in parentheses after the arbovirus name indicate that the virus was isolated in that country from the vector listed in the same row of the table. "Geographic Distribution" indicates the countries in which the vector has been found, but does not necessarily indicate the distribution of the arbovirus.

VECTOR	ARBOVIRUS	ARBOVIRUS RESERVOIR	GEOGRAPHIC DISTRIBUTION	TRANSMISSION SEASON	LARVAL HABITAT	BITING BEHAVIOR	RESTING BEHAVIOR	FLIGHT RANGE
Aedes arborealis	Apeu (Brazil)	Marsupials (*Caluromys philander).*	Brazil, French Guiana, and Suriname.	During rainy season (November to March) in tropical rain forests.	Treeholes.	Known to bite humans.	Unknown.	Unknown.
Aedes hastatus	Western Equine Encephalitis (WEE) (Ecuador)	Epizootic transmission undefined, but passerine birds considered important reservoirs.	Argentina, Bolivia, Brazil, Colombia, Costa Rica, Ecuador, French Guiana, Honduras, Mexico, Panama, and Peru.	Unknown.	Temporary ground pools.	Bites humans by day in the forest.	Unknown.	Unknown.
Aedes scapularis	Venezuelan Equine Encephalitis (VEE) (Ecuador, Peru)	VEE: Many mammals and birds, but equines are key reservoirs with high viremias.	Argentina, Bolivia, Colombia, Cuba, Dominican Republic, Ecuador, French Guiana, Guyana, Haiti, Jamaica, Mexico, Panama, Paraguay, Peru, Puerto Rico, Suriname, Trinidad, United States, and Venezuela.	VEE virus activity begins at the end of the rainy season and disappears when the dry season is underway.	Temporary ground pools.	Feeds on birds and large mammals but prefers mammals. A vicious biter of humans, it feeds night or day in a wide variety of locations. It commonly moves indoors in areas that have been populated for long periods.	Unknown.	In one study, observed to move at least 4 km in 11 days.
Aedes septemstriatus	Apeu (Brazil)	Marsupials (*Caluromys philander).*	Brazil, Colombia, Costa Rica, Nicaragua, and Panama.	During the rainy season (November to March) in tropical rain forests.	Treeholes and broken bamboo.	Bites humans by day in the forest.	Unknown.	Unknown.
Aedes serratus	Oropouche	Primates (*Cebus*, *Alouatta*, *Saimiri*, *Saguinus*), sloths.	Argentina, Belize, Bolivia, Brazil, Colombia, Costa Rica, Ecuador, French Guiana, Guadaloupe, Guatemala, Guyana, Honduras, Jamaica, Mexico, Nicaragua, Panama, Peru, Puerto Rico, Suriname, Trinidad, and Venezuela.	Epidemics occur during the rainy season (November to March).	Temporary ground pools.	Bites humans by day in the forest, but prefers to bite at night in open areas. Prefers ground level and often enters buildings. Will also feed on chickens.	Unknown.	Unknown.

Appendix A.3. (Cont'd)

VECTOR	ARBOVIRUS	ARBOVIRUS RESERVOIR	GEOGRAPHIC DISTRIBUTION	TRANSMISSION SEASON	LARVAL HABITAT	BITING BEHAVIOR	RESTING BEHAVIOR	FLIGHT RANGE
Aedes taeniorhynchus	Oriboca VEE (Ecuador, Peru)	Oriboca: *Oryzomys capito*, *Proechimys guyanensis*, *Didelphis marsupialis.* VEE: Many mammals and birds, but equines are key reservoirs with high viremias.	Antigua, Bahamas, Belize, Brazil, Colombia, Costa Rica, Cuba, Dominican Republic, Ecuador, French Guiana, Guatemala, Guyana, Haiti, Jamaica, Mexico, Nicaragua, Panama, Peru, Puerto Rico, Suriname, St. Lucia, Trinidad, United States, and Venezuela.	Oriboca transmission occurs during the rainy season in tropical rain forests (November to March). VEE virus activity begins at the end of the rainy season and disappears during the dry season.	Coastal salt marshes and mangrove swamps.	Vicious biter of humans by day and night in many kinds of habitats; most active at dawn and dusk.	Rests in vegetation, emerging to bite when disturbed.	Flies up to 32 km.
Anopheles albitarsis Group	WEE (Ecuador)	Epizootic transmission undefined, but passerine birds considered important reservoirs.	Argentina, Bolivia, Brazil, Colombia, Costa Rica, Ecuador, French Guiana, Guatemala, Guyana, Panama, Paraguay, Suriname, Trinidad, Uruguay, and Venezuela.	Unknown.	Ground pools, pools along streams, swamps, and lakes all in full sunlight. Water with grassy margins.	Feeds on large mammals and refuses to feed on birds.	Unknown.	Unknown.
Coquillettidia arribalzagai	Oriboca	*Oryzomys capito*, *Proechimys guyanensis*, *Didelphis marsupialis.*	Brazil, Colombia, French Guiana, Nicaragua, Panama, Peru, and Suriname.	During the rainy season in tropical rain forests.	Larvae attach to roots of aquatic plants in permanent water.	Bites humans by day in forest.	Unknown.	Unknown.
Coquillettidia venezuelensis	Murutucu Oriboca Oropouche	Murutucu: *Oryzomys capito*, *Proechimys guyanensis*, *Nectomys squimipes*, *Didelphis marsupialis*, *Marmosa cinerea*, *Bradypus tridactylus*, *Artibeus literatus*, *Artibeus jamaicensis.* Oriboca: *Oryzomys capito*, *Proechimys guyanensis*, *Didelphis marsupialis.* Oropouche: Primates (*Cebus*, *Alouatta*, *Saimiri*, *Saguinus*), sloths (*Bradypus*), *Zygodontomys*, and possibly wild birds.	Argentina, Belize, Brazil, Colombia, Costa Rica, El Salvador, French Guiana, Guatemala, Guyana, Mexico, Nicaragua, Panama, Peru, Suriname, Trinidad, and Uruguay.	Murutucu and Oriboca transmission occur during the rainy season in tropical rain forests (November to March). Oropouche epidemics occur during the rainy season (November to March).	Larvae attach to roots of aquatic plants in permanent water pools.	Bites humans in the forest, especially where there is secondary growth.	Unknown.	Unknown.

Appendix A.3. (Cont'd)

VECTOR	ARBOVIRUS	ARBOVIRUS RESERVOIR	GEOGRAPHIC DISTRIBUTION	TRANSMISSION SEASON	LARVAL HABITAT	BITING BEHAVIOR	RESTING BEHAVIOR	FLIGHT RANGE
Culex coronator	Caraparu (Brazil, Panama)	*Oryzomys capito*, *Oryzomys laticeps*, *Proechimys guyanensis*, *Nectomys squamipes*, *Zygodontomys brevicauda*, *Heteromys anomalus*.	Argentina, Belize, Brazil, Bolivia, Colombia, French Guiana, Guatemala, Honduras, El Salvador, Mexico, Panama, Paraguay, Peru, Suriname, Trinidad, United States, and Venezuela.	During the rainy season (November to March) in tropical rain forests.	Ground pools, seeps, streams, artificial containers, bromeliads, and bamboo. Stagnant and slow-flowing water, shaded or unshaded.	Commonly considered not to feed on humans, but observed to be a major human biter in the Amazon Basin.	Unknown.	Unknown.
Culex gnomatus	VEE (Ecuador, Peru)	Many mammals and birds, but equines are key reservoirs with high viremias.	Brazil, Ecuador, and Peru.	VEE virus activity begins at the end of the rainy season and disappears during the dry season.	Unknown.	Unknown.	Unknown.	Unknown.
Culex nigripalpus	Caraparu (Brazil, Panama) EEE (Peru) St. Louis Encephalitis (SLE) (Colombia, Ecuador, Guatemala, Jamaica, Trinidad) Vesicular Stomatitis	Caraparu: *Oryzomys capito*, *Oryzomys laticeps*, *Proechimys guyanensis*, *Nectomys squamipes*, *Zygodontomys brevicauda*, *Heteromys anomalus*. EEE: Birds, particularly passerines. SLE: Wild birds. Vesicular Stomatitis: Poorly understood but primarily a disease of livestock (bovines and equines).	Bahamas, Barbados, Belize, Brazil, Colombia, Costa Rica, Cuba, Dominican Republic, Ecuador, French Guiana, Guatemala, Guyana, Jamaica, Mexico, Panama, Puerto Rico, Suriname, Trinidad, United States, and Venezuela.	Caraparu transmission occurs during the rainy season (November to March) in tropical rain forests. EEE virus activity occurs throughout the year (Peru). SLE: unknown.	Wide variety of habitats, including ground pools, ditches, grassy pools, crab holes, permanent pools in swamps, artificial containers, beaches, boats, and axils of bromeliads.	Feeds on humans, sometimes entering houses or tents.	Unknown.	Unknown.

Appendix A.3. (Cont'd)

VECTOR	ARBOVIRUS	ARBOVIRUS RESERVOIR	GEOGRAPHIC DISTRIBUTION	TRANSMISSION SEASON	LARVAL HABITAT	BITING BEHAVIOR	RESTING BEHAVIOR	FLIGHT RANGE
Culex ocossa	VEE (Ecuador, Peru) Apeu (Brazil) Caraparu (Brazil, Panama) Itaqui (Brazil, Venezuela) Marituba (Peru) Murutucu Oriboca	Apeu: Marsupials (*Caluromys philander).* Caraparu: *Oryzomys capito*, *Oryzomys laticeps*, *Proechimys guyanensis*, *Nectomys squamipes*, *Zygodontomys brevicauda*, *Heteromys anomalus.* Itaqui: *Oryzomys capito*, *Proechimys guyanensis*, *Nectomys squamipes*, *Marmosa murina*, *Metachirus nudicaudatus.* Marituba: Marsupials (*Didelphis marsupialis*). Murutucu: *Oryzomys capito*, *Proechimys guyanensis*, *Nectomys squamipes*, *Didelphis marsupialis*, *Marmosa cinerea*, *Bradypus tridactylus*, *Artibeus literatus*, *Artibeus jamaicensis.* Oriboca: *Oryzomys capito*, *Proechimys guyanensis*, *Didelphis marsupialis.*	Argentina, Brazil, Colombia, Ecuador, Guyana, Panama, Suriname, and Venezuela.	VEE virus activity begins at the end of the rainy season and disappears when the dry season is underway. Apeu, Caraparu, Itaqui, Marituba, Murutuca and Oriboca transmission occur during the rainy season (November to March) in tropical rain forests.	Permanent pools, always associated with aquatic plants such as *Pistia*.	Endophagic.	Commonly rests on screens of windows.	Unknown.
Culex pedroi	EEE (Peru)	Birds, particularly passerines.	Argentina, Brazil, Colombia, Costa Rica, Ecuador, French Guiana, Guatemala, Guyana, Mexico, Panama, Peru, Suriname, Tobago and Trinidad.	Virus activity occurs throughout the year (Peru).	Heavy shade in permanent bodies of water with abundant floatage.	Commonly bites humans but apparently prefers rodents. Has been known to feed on birds.	Unknown.	Unknown.

Appendix A.3. (Cont'd)

VECTOR	ARBOVIRUS	ARBOVIRUS RESERVOIR	GEOGRAPHIC DISTRIBUTION	TRANSMISSION SEASON	LARVAL HABITAT	BITING BEHAVIOR	RESTING BEHAVIOR	FLIGHT RANGE
Culex portesi	Bimiti (Peru) Guama (Guama, Colombia) Itaqui (Brazil, Venezuela) Marituba (Peru) Murutucu Oriboca	Bimiti: *Oryzomys laticeps*, *Zygodontomys brevicauda*, *Heteromys anomalus*, *Proechimys guyanensis*. Guama: *Oryzomys capito*, *Proechimys guyanensis*, *Nectomys squamipes*, *Oryzomys laticeps*, *Heteromys anomalus*, *Zygodontomys brevicauda*, *Coendou* spp. Itaqui: *Oryzomys capito*, *Proechimys guyanensis*, *Nectomys squamipes*, *Marmosa murina*, *Metachirus nudicaudatus*. Marituba: Marsupials (*Didelphis marsupialis*). Murutucu: *Oryzomys capito*, *Proechimys guyanensis*, *Nectomys squamipes*, *Didelphis marsupialis*, *Marmosa cinerea*, *Bradypus tridactylus*, *Artibeus literatus*, *Artibeus jamaicensis*. Oriboca: *Oryzomys capito*, *Proechimys guyanensis*, *Didelphis marsupialis*.	Brazil, Colombia, French Guiana, Peru, Suriname, Trinidad, and Venezuela.	Bimiti virus activity begins at the end of the rainy season (March) and disappears during the dry season (May). Guama transmission occurs during the rainy season in tropical rain forests (November to March). Incubation period <10 days. Itaqui, Marituba, Murutucu and Oriboca transmission occurs during the rainy season (Novembers to March) in tropical rain forests.	Lowland swamp forests at elevations from sea level to 30 m. Larvae also found in deep shade of tree buttresses, root caves, and leafy swamp margins.	Unknown.	Unknown.	Unknown.
Culex quinquefasciatus	Oropouche	Primates (*Cebus*, *Alouatta*, *Saimiri*, *Saguinus*), sloths.	Associated with human settlements and widely distributed throughout the tropical and subtropical regions of the world.	Year-round where temperatures are favorable for mosquito development, but especially during the dry season when organic material concentrates in breeding areas.	Stagnant/ polluted water high in organic content, in ground seeps or in artificial containers. Breeds in clean and brackish water.	Preference for avian blood but will feed readily on mammals, including humans. Bites throughout night, but especially a few hours before and after midnight.	Rests during day in dark humid shelters, e.g., culverts, cellars, outhouses, chicken houses.	Routinely 200-300 m. Maximum: 1.3 km, but Hawaiian studies show that 4 km is common.

Appendix A.3. (Cont'd)

VECTOR	ARBOVIRUS	ARBOVIRUS RESERVOIR	GEOGRAPHIC DISTRIBUTION	TRANSMISSION SEASON	LARVAL HABITAT	BITING BEHAVIOR	RESTING BEHAVIOR	FLIGHT RANGE
Culex spissipes	Bimiti (Peru) Caraparu (Brazil, Panama) Itaqui (Brazil, Venezuela) Oriboca	Bimiti: *Oryzomys laticeps*, *Zygodontomys brevicauda*, *Heteromys anomalus*, *Proechimys guyanensis*. Caraparu: *Oryzomys capito*, *Oryzomys laticeps*, *Proechimys guyanensis*, *Nectomys squamipes*, *Zygodontomys brevicauda*, *Heteromys anomalus*. Itaqui: *Oryzomys capito*, *Proechimys guyanensis*, *Nectomys squamipes*, *Marmosa murina*, *Metachirus nudicaudatus*. Oriboca: *Oryzomys capito*, *Proechimys guyanensis*, *Didelphis marsupialis*.	Belize, Bolivia, Brazil, Colombia, Ecuador, French Guiana, Guatemala, Honduras, Mexico, Panama, Peru, Suriname, Trinidad, and Venezuela.	Bimiti virus activity begins at the end of the rainy season (March) and disappears during the dry season (May). Caraparu, Itaqui and Oriboca transmission occur during the rainy season (November to March) in tropical rain forests.	Heavily or partially shaded margins of lakes in forests, margins of swamps, and in ground pools. Water is usually permanent and fresh, with abundant grassy and floating aquatic vegetation, or with dense accumulations of fallen leaves.	Has been collected at night in mouse-baited traps.	Unknown.	Unknown.
Culex taeniopus	Bimiti (Peru) Guama (Colombia, Peru) Ossa (Panama)	Bimiti: *Oryzomys laticeps*, *Zygodontomys brevicauda*, *Heteromys anomalus*, *Proechimys guyanensis*. Guama: *Oryzomys capito*, *Proechimys guyanensis*, *Nectomys squamipes*, *Oryzomys laticeps*, *Heteromys anomalus*, *Zygodontomys brevicauda*, *Coendou* spp. Ossa: *Proechimys semispinosus*.	Bahamas, Belize, Cayman Islands, Colombia, Costa Rica, Dominican Republic, French Guiana, Guatemala, Honduras, Jamaica, Mexico, Nicaragua, Panama, Puerto Rico, and Venezuela.	Bimiti virus activity begins at the end of the rainy season (March) and disappears when dry season is underway (May). Guama transmission occurs during the rainy season in tropical rain forests (November to March). Incubation period <10 days. Ossa transmission occurs during the rainy season in tropical rain forests (November to March).	Found in stagnant water in swamps and forests.	Unknown.	Unknown.	Unknown.

Appendix A.3. (Cont'd)

VECTOR	ARBOVIRUS	ARBOVIRUS RESERVOIR	GEOGRAPHIC DISTRIBUTION	TRANSMISSION SEASON	LARVAL HABITAT	BITING BEHAVIOR	RESTING BEHAVIOR	FLIGHT RANGE
Culex vomerifer	Caraparu (Brazil, Panama) Guama (Colombia, Peru) Itaqui (Brazil, Venezuela) Murututucu Ossa (Panama)	Caraparu: *Oryzomys capito*, *Oryzomys laticeps*, *Proechimys guyanensis*, *Nectomys squamipes*, *Zygodontomys brevicauda*, *Heteromys anomalus*. Guama: *Oryzomys capito*, *Proechimys guyanensis*, *Nectomys squamipes*, *Oryzomys laticeps*, *Heteromys anomalus*, *Zygodontomys brevicauda*, *Coendou* spp. Itaqui: *Oryzomys capito*, *Proechimys guyanensis*, *Nectomys squamipes*, *Marmosa murina*, *Metachirus nudicaudatus*. Murututucu: *Oryzomys capito*, *Proechimys guyanensis*, *Nectomys squamipes*, *Didelphis marsupialis*, *Marmosa cinerea*, *Bradypus tridactylus*, *Artibeus literatus*, *Artibeus jamaicensis*. Ossa: *Proechimys semispinosus*.	Brazil, Colombia, Ecuador, French Guiana, Panama, Peru, Trinidad, and Venezuela.	Caraparu transmission occurs during the rainy season (November to March) in tropical rain forests. Guama transmission occurs during the rainy season in tropical rain forests (November to March). Incubation period <10 days. Itaqui, Murututucu and Ossa transmission occur during the rainy season (November to March) in tropical rain forests.	Treeholes, most often found in the forest canopy.	Unknown.	Unknown.	Unknown.
Culex (*Melanoconion)* spp.	SLE (Ecuador) VEE (Ecuador, Peru)	SLE: Wild birds. VEE: Many mammals and birds, but equines are key reservoirs with high viremias.	Unknown.	SLE: Unknown. VEE: Virus activity begins at the end of the rainy season and disappears during the dry season.	Unknown.	Unknown.	Unknown.	Unknown.

Appendix A.3. (Cont'd)

VECTOR	ARBOVIRUS	ARBOVIRUS RESERVOIR	GEOGRAPHIC DISTRIBUTION	TRANSMISSION SEASON	LARVAL HABITAT	BITING BEHAVIOR	RESTING BEHAVIOR	FLIGHT RANGE
Haemagogus janthinomys	Mayaro (Bolivia, Colombia, Peru)	Callithricid primates (*Callithrix argentata* and *Callithrix humeralifer*) and passerine birds.	Argentina, Bolivia, Brazil, Colombia, Costa Rica, Ecuador, French Guiana, Guyana, Honduras, Nicaragua, Panama, Paraguay, Peru, Suriname, Tobago and Trinidad, and Venezuela.	Disease found mainly in forests.	Treeholes, most often found in the forest canopy.	Bites humans during the day in the canopy of undisturbed rain forest.	Unknown.	Unknown.
Limatus durhamii	Caraparu (Brazil, Panama)	*Oryzomys capito*, *Oryzomys laticeps*, *Proechimys guyanensis*, *Nectomys squamipes*, *Zygodontomys brevicauda*, *Heteromys anomalus*.	Argentina, Belize, Bolivia, Brazil, Costa Rica, Dominican Republic, Ecuador, El Salvador, French Guiana, Guadaloupe, Grenada, Guyana, Honduras, Mexico, Nicaragua, Panama, Peru, Suriname, Trinidad, and Venezuela.	During the rainy season (November to March) in tropical rain forests.	Fallen leaves and small containers with abundant decomposing plant matter.	Bites humans in disturbed forests during the day.	Unknown.	Unknown.
Limatus flavisetosus	Mayaro (Bolivia, Colombia, Peru) Wyeomyia (Colombia)	Callithricid primates (*Callithrix argentata* and *Callithrix humeralifer*) and passerine birds. *Wyeomyia*: "Mosquito."	Bolivia, Brazil, Colombia, French Guiana, Peru, and Suriname.	Disease found mainly in forests. *Wyeomyia*: Unknown.	Fallen leaves and small containers with abundant decomposed plant matter.	Bites humans mainly during the day at ground level in the forest.	Unknown.	Unknown.
Limatus spp.	Guama	*Oryzomys capito*, *Proechimys guyanensis*, *Nectomys squamipes*, *Oryzomys laticeps*, *Heteromys anomalus*, *Zygodontomys brevicauda*, *Coendou* spp.	Colombia, Peru.	During the rainy season in tropical rain forests (November to March). Incubation period <10 days.	Unknown.	Unknown.	Unknown.	Unknown.
Mansonia indubitans (Ecuador)	Vesicular Stomatitis	Poorly understood but primarily a disease of livestock (bovines and equines).	Bolivia, Brazil, Ecuador, Panama, Peru, Trinidad, and Uruguay.	Virus activity begins at the end of the rainy season and disappears when the dry season is underway.	Permanent water with abundant vegetation. Larvae use siphon to penetrate roots of aquatic plants for air.	Bites humans day or night, sometimes indoors. Vicious biter.	Unknown.	Unknown.

Appendix A.3. (Cont'd)

VECTOR	ARBOVIRUS	ARBOVIRUS RESERVOIR	GEOGRAPHIC DISTRIBUTION	TRANSMISSION SEASON	LARVAL HABITAT	BITING BEHAVIOR	RESTING BEHAVIOR	FLIGHT RANGE
Mansonia spp.	Guama (Colombia, Peru) WEE (Ecuador)	Guama: *Oryzomys capito*, *Proechimys guyanensis*, *Nectomys squamipes*, *Oryzomys laticeps*, *Heteromys anomalus*, *Zygodontomys brevicauda*, *Coendou* spp. WEE: Epizootic transmission undefined, but passerine birds considered important reservoirs.	Guama: Colombia, Ecuador, Peru. WEE: Ecuador, Peru.	Guama: During the rainy season in tropical rain forests (November to March). Incubation period <10 days. WEE: unknown.	Unknown.	Unknown.	Unknown.	Unknown.
Psorophora albigenu	EEE (Peru) VEE (Peru)	EEE: Birds, particularly passerines. VEE: Many mammals and birds, but equines are key reservoirs with high viremias.	Argentina, Bolivia, Brazil, Ecuador, Paraguay, Peru, and Venezuela.	EEE virus activity throughout the year (Peru). VEE virus activity begins at the end of the rainy season and disappears when the dry season is underway.	Heavily shaded temporary ground pools.	Unknown.	Unknown.	Unknown.
Psorophora albipes (Colombia)	Mayaro (Bolivia, Colombia, Peru)	Callithricid primates (*Callithrix argentata* and *Callithrix humeralifer*) and passerine birds.	Bolivia, Brazil, Guatemala, Colombia, Honduras, Mexico, Peru, Suriname, Trinidad, and Venezuela.	Disease found mainly in forests.	Temporary ground pools.	Bites humans primarily during the day in the forest. Can be the dominant biting species.	Unknown.	Unknown.
Psorophora ferox	EEE (Peru) Ilheus (Colombia) Mayaro (Bolivia, Colombia, Peru) Oriboca	EEE: Birds, particularly passerines. Ilheus: Unknown. Mayaro: Callithricid primates (*Callithrix argentata* and *Callithrix humeralifer*) and passerine birds. Oriboca: *Oryzomys capito*, *Proechimys guyanensis*, *Didelphis marsupialis.*	Canada south to Argentina.	EEE Virus activity throughout the year (Peru). Ilheus: Unknown. Mayaro transmission occurs mainly in forests. Oriboca transmission occurs during the rainy season in tropical rain forests.	Temporary, shaded ground pools in forests.	Bites humans at ground level, usually in the forest during the day. A vicious biter of any warm-blooded animal, waits in vegetation and emerges to bite. Sometimes bites indoors.	Unknown.	Usually remains near larval site but has been observed to fly up to 2 km.

Appendix A.3. (Cont'd)

VECTOR	ARBOVIRUS	ARBOVIRUS RESERVOIR	GEOGRAPHIC DISTRIBUTION	TRANSMISSION SEASON	LARVAL HABITAT	BITING BEHAVIOR	RESTING BEHAVIOR	FLIGHT RANGE
Psorophora pallescens	WEE (Ecuador)	Epizootic transmission undefined, but passerine birds considered important reservoirs.	Argentina, Bolivia, Ecuador and Paraguay.	Unknown.	Predacious on other mosquito larvae in temporary ground pools.	Has been observed to feed primarily on cattle, but also other large mammals and on chickens.	Unknown.	Unknown.
Psorophora spp.	Guama (Colombia, Peru)	*Oryzomys capito*, *Proechimys guyanensis*, *Nectomys squamipes*, *Oryzomys laticeps*, *Heteromys anomalus*, *Zygodontomys brevicauda*, *Coendou* spp.	Colombia and Peru.	During the rainy season in tropical rain forests (November to March). Incubation period <10 days.	Unknown.	Unknown.	Unknown.	Unknown.
Trichoprosopon digitatum	Bussuquara (Panama) SLE (Colombia, Ecuador) Wyeomyia (Colombia)	Bussuquara and Wyeomyia: "Mosquito". SLE: Wild birds.	Belize, Brazil, Colombia, Costa Rica, Ecuador, French Guiana, Guatemala, Guyana, Mexico, Nicaragua, Panama, Peru, Suriname, and Venezuela.	Unknown.	Bamboo internodes, fallen fruits or nuts, fallen leaves, artificial containers (cans, tires, dishes, etc.), treeholes, *Heliconia* flower bracts, and leaf axils of bromeliads.	Bites humans, especially at ground level in the forest during the day, with greatest numbers in the evening. A vicious biter.	Unknown.	Unknown.
Trichoprosopon spp.	Guama (Colombia, Peru)	*Oryzomys capito*, *Proechimys guyanensis*, *Nectomys squamipes*, *Oryzomys laticeps*, *Heteromys anomalus*, *Zygodontomys brevicauda*, *Coendou* spp.	Colombia, Peru.	During the rainy season in tropical rain forests (November to March). Incubation period <10 days.	Unknown.	Unknown.	Unknown.	Unknown.

Appendix A.3. (Cont'd)

VECTOR	ARBOVIRUS	ARBOVIRUS RESERVOIR	GEOGRAPHIC DISTRIBUTION	TRANSMISSION SEASON	LARVAL HABITAT	BITING BEHAVIOR	RESTING BEHAVIOR	FLIGHT RANGE
Wyeomyia aporonoma	Mayaro (Bolivia, Colombia, Peru) Wyeomyia (Colombia)	Mayaro: Callithricid primates (*Callithrix argentata* and *Callithrix humeralifer*) and passerine birds. Wyeomyia: "Mosquito."	Belize, Bolivia, Brazil, Colombia, Costa Rica, El Salvador, French Guiana, Grenada, Guatemala, Guyana, Honduras, Mexico, Panama, St. Vincent, and Venezuela.	Mayaro transmission is suspected to occur year-round, mainly in forests. Wyeomyia: unknown.	Leaf axils of terrestrial bromeliads.	Bites humans in the forest during the day in the canopy or at ground level.	Unknown.	Unknown.
Wyeomyia medioalbipes	Caraparu (Brazil, Panama) Wyeomyia (Colombia)	Caraparu: *Oryzomys capito*, *Oryzomys laticeps*, *Proechimys guyanensis*, *Nectomys squamipes*, *Zygodontomys brevicauda*, *Heteromys anomalus*. Wyeomyia: "Mosquito."	Brazil, Colombia, Panama, Suriname, and Trinidad.	Caraparu transmission occurs during the rainy season (November to March) in tropical rain forests. Wyeomyia: unknown.	Leaf axils of terrestrial bromeliads.	Unknown.	Unknown.	Unknown.
"Mosquitoes"	Guaroa (Colombia, Peru)	Human isolate.	Colombia, Peru.	Unknown.	Unknown.	Unknown.	Unknown.	Unknown.
Culicoides paraensis (Diptera: Ceratopogonidae)	Oropouche	Primates (*Cebus*, *Alouatta*, *Saimiri*, *Saguinus*), sloths.	From sea level to elevations where tropical rain forests begin.	Epidemics occur during the rainy season (November to March).	Eggs are laid in decaying vegetable matter. Decaying banana stocks, cut-off banana stumps and piled up cacao pods are primary breeding sources in peridomestic settings and plantations. Rains provide moisture required for larval development in the decaying vegetation.	Exophagic or endophagic. Bites lower extremities, especially ankles. Inflicts painful bites capable of causing severe tissue reactions. Strictly daytime biters. Small peak at noon and large peak beginning 1 hour before and continuing to sunset. Increased activity right after rain showers.	Endophilic and exophilic.	Unknown, but probably less than 1 km.

Appendix A.3. (Cont'd)

VECTOR	ARBOVIRUS	ARBOVIRUS RESERVOIR	GEOGRAPHIC DISTRIBUTION	TRANSMISSION SEASON	LARVAL HABITAT	BITING BEHAVIOR	RESTING BEHAVIOR	FLIGHT RANGE
Lutzomyia spp. (Diptera: Psychodidae)	Arboledas (Colombia) Buenaventura (Colombia) Guama (Colombia, Peru) Mariquita (Colombia) Vesicular Stomatitis	Arboledas and buenaventura: unknown. Guama: *Oryzomys capito*, *Proechimys guyanensis*, *Nectomys squamipes*, *Oryzomys laticeps*, *Heteromys anomalus*, *Zygodontomys brevicauda*, *Coendou* spp. Mariquita: unknown. Vesicular Stomatitis: Poorly understood but primarily a disease of livestock (bovines and equines).	Colombia, Peru.	Arboledas, Buenaventura and Mariquita transmission coincides with increases in sand fly populations during the rainy season (November to March). Guama transmission occurs during the rainy season in tropical rain forests (November to March). Incubation period <10 days. Vesicular stomatitis virus activity begins at the end of the rainy season and disappears when the dry season is underway.	Unknown.	Unknown.	Unknown.	Unknown.
Simulium exiguum (Diptera: Simuliidae)	Vesicular Stomatitis	Poorly understood but primarily a disease of livestock (bovines and equines).	Colombia	Virus activity begins at the end of the rainy season and disappears during the dry season.	Unknown.	Unknown.	Unknown.	Unknown.

Appendix A.4. Disease Vector Ecology Profile: Reduviid Vectors of Chagas Disease in Colombia.

VECTOR	GEOGRAPHIC DISTRIBUTION	POTENTIAL HOSTS	TRANSMISSION SEASON	BIONOMICS/HABITAT INFORMATION	BITING BEHAVIOR
Rhodnius prolixus	Primary vector in central and eastern Colombia, unknown in coastal regions and in southern areas of Colombia. Populations diminish toward lowlands of northern Magdalena Valley. Not found infected with *T. cruzi* above 2,600 m.	Unknown.	Unknown.	Well established in human habitations. Peridomestic in chicken houses and pigeon coops. Associated with bird (*Jabira mycteria* and *Mycteria americana*). Bird may transport egg in their feathers to new locations. Two generations per year. Commonly collected on palms. Palm fronds used by local people to construct houses may start the domestic cycle or re-infest following eradication.	Strongly anthropophilic, weakly zoophilic. Aggressive feeder. Painless bite without reaction. Defecates within 10 minutes of feeding.
Rhodnius pallescens	Atlantic coastal areas from Panama border. Primary vector in neighboring Panama.	Unknown.	Unknown.	Increasingly becoming domestic. Peridomestic in pig pens, hollow trees, etc. Associated with opossum, *Didelphis marsupialis*, an established *T. cruzi* reservoir. Breeds commonly in palm trees. Requires moist climate. Relative humidity >60%. Peak populations coincide with rainy season.	Anthropophilic.
Triatoma dimidiata	Exclusively along Pacific Coast. Not found infected with *T. cruzi* above 2,300 m.	Unknown.	Unknown.	Domestic/peridomestic in urban and rural structures. Prefers densely forested areas. Infected with *T. cruzi* for life. Maintenance of sylvatic cycle of *T. cruzi* is more important than domestic contact with man. Frequently covered with soil particles. Common in burrows of armadillo. Attracted to lights.	Strongly zoophilic, weakly anthropophilic. Timid feeder. Feeds 10-20 minutes. Flagellates in feces 12-15 days post infected meal. Produces pruritis and erythema.
Triatoma maculata	Atlantic coastal area of Guajira Department.	Unknown.	Unknown.	Infected *T. maculata* < 1%. Sometimes domestic, usually peridomestic in chicken houses, pigeon coops, livestock shelters and even under stones. Has been collected in bird nests. Second only to *T. infestans* as vector of Chagas disease in Venezuela.	Anthropophilic.

Appendix A.5. Disease Vector Ecology Profile: Flea Vector of Murine Typhus in Colombia.

VECTOR	GEOGRAPHIC DISTRIBUTION	POTENTIAL HOSTS	TRANSMISSION SEASON	BIONOMICS/HABITAT INFORMATION	BITING BEHAVIOR
Xenopsylla cheopis	Primary vector at elevations below 2,800 m countrywide although *X. cheopis* will occur at higher elevations.	*Rattus rattus, Rattus norvegicus, Cavia porcellus.*	Unknown.	300-400 ovoid white eggs are deposited in the nest or burrow of the host at a rate of 2-6/day. The eggs hatch in 9-13 days. Three larval stadia are legless and eyeless, lasting 32-34 days. Pupae spin a silken cocoon and adults emerge in 25-30 days. Development occurs in nest. Adults live up to 158 days at 20°C and 90-94% relative humidity.	Voracious feeder, feeding frequently for short periods. Proventricular blockage occurs below 27°C. Blockage occurs in 12-21 days after ingesting plague bacilli. Found to feed readily on 75 different hosts, including man. Can jump 20 cm. Burrowing *Rattus* spp. usually harbor more *X. cheopis* than those confined to surface habitats.

Appendix A.6. Disease Vector Ecology Profile: Sand Fly Vectors of Leishmaniasis in Colombia.

VECTOR	GEOGRAPHIC DISTRIBUTION	POTENTIAL HOSTS	TRANSMISSION SEASON	BIONOMICS/HABITAT INFORMATION	BITING BEHAVIOR
Lutzomyia longipalpis	Principal vector of *L. chagasi* in northern Colombian lowlands below 900 m and in Andean valleys, particularly the Magdalena River Valley. Arid and semi-arid regions in close association with humans and domestic animals.	Unknown.	Throughout year with bimodal annual abundance: small peak Oct/Nov and large peak Apr/May. Increases activity shortly after sunset (1830-2330), continues until just after sunrise. Increased activity in moonlight. Decreased activity below 24°C.	Populations associated with primary forest destruction and development of farmland. Developmental cycle from egg to adult 20-41 days. Maximum flight range: ca. 0.5 km, but usual 50 to 300 m. Rests indoors and among rocky areas. Eggs laid in crevices. 25°C is optimal lab rearing temperature.	Feeding activity associated with domestic animals and their shelters. Exophagic, but rests indoor after feeding. Multiple feedings during gonotrophic cycle. Feeds on large animals and is more zoophilic than anthropophilic. Will feed on birds, domestic animals and humans. *Proechimys* and *Oryzomys* are natural hosts.
Lutzomyia flaviscutellata	Isolated populations identified in Department of Cesar.	Unknown.	Populations present throughout year, but significant population begins towards end of dry season and declines as rainy season commences. Biting collections have been made during the first four hours after dark.	Found in dry secondary forests. Egg to adult averages 40.5 days. Males emerge before females. Females and males live 17-41 days (ave. 27) and 2-12 days (ave. 6), respectively. Requires blood meal for egg development. Lay 165 eggs/female. Optimal temperature range for rearing larvae is 20-26°C and for adults is 23-27°C at 95-98% relative humidity.	Strongly attracted to rodents (especially *Proechimys*) and marsupials, but will bite humans entering their habitat. Females will feed as many as four times during lifespan.
Lutzomyia umbratilis	Southeastern lowland Amazon Basin department of Amazonas.	Unknown.	Unknown.	Adults found in primary rain forests. Adults collectible from ground level to 1.5 m on tree trunks. In French Guiana commonly found on trunks of Niamboka (*Pouteria guianensis*) and Kopi (*Goupia glabra*) trees. Associated with the sand fly *Lutzomyia rorotaensis* (French Guiana).	Anthropophilic.
Lutzomyia spinicrassa	Suspected vector of *L. braziliensis* in mountainous coffee growing areas.	Unknown.	Populations peak just before rainy season (February through April) and again at onset of 2nd wet season (September through November). Day or night biter.	Readily attracted to light traps suspended from ground level to 16 meters. Collected intradomicilary. Attracted to human bait.	Known to feed on humans, chickens, opossums, pigs, and horses.

Appendix A.6. (Cont'd)

VECTOR	GEOGRAPHIC DISTRIBUTION	POTENTIAL HOSTS	TRANSMISSION SEASON	BIONOMICS/HABITAT INFORMATION	BITING BEHAVIOR
Lutzomyia hartmanni	Limited to forested coastal areas with high annual rainfall from sea level to 700 m.	Unknown.	Abundant in forest habitats and in clearings. Biting activity greater in canopy than on ground. Peak activity 1900-0300 hours in forest at ground level and between 1900-2300 in clearings. Peak activity in canopy between 2100-0300.	Females observed to lay 12 and 27 eggs. Development from egg to adult is 53 days. Larvae feed at or below the surface of their food supply (organic detritis).	Anthropophilic.
Lutzomyia gomezi	Widely distributed species considered vector of *L. panamensis* in Panama in mature lowland forests (sea level to 800 m) and cultivated and cleared areas.	Unknown.	Populations most prevalent in dry season January-April (Panama). Greatest biting activity near forest clearings from 1800-2000 hours and small peak 0500-0700. Biting not observed in deep forest.	Readily enters dwelling <200 m from forests. Adapted to dry conditions. Easily collected with light traps and aspirators at ground level to 16 m. Maximum flight range: 157 m (one report 1 km), usual <20 m. Autogeny occurs in some females. Lay average of 29 eggs and require 34 days to adult emergence. Adults emerge at hilltop areas associated with large *Anacardium* trees. Larvae feed at the surface of organic detritis.	Often mates before feeding and feeds rapidly in < 1 minute. Feeds on humans, horses chickens, and a variety of mammals. Bites indoors and outdoors. Concentrated in clearings and prevalent at forest edges and in open pastures.
Lutzomyia evansi	Vector of *L. chagasi* in northern Colombia.	Unknown.	Populations peak in July and August.	Unknown.	Human bait preferred over dogs or opossums.
Lutzomyia trapidoi	Primarily in forested areas of high rainfall along coastal lowlands (sea level to 800 m) from Panama south.	Unknown.	Major peaks occur in July and in December/ January, but prevalent throughout year (Panama). Bites in under story during daytime. Peaks at night from 2000-2100 and again from 0300-0400 hours. Other reports between 2300-0500.	Readily attracted to light traps. Mature-forest inhabitants. Maximum flight range: 200 m, usual <57m. Reluctant to enter clearings. Males rest in leaf litter, and females on tree trunks between .6-2 m (Panama). 21 eggs/cycle. Egg to adult 33-47 days. Undergoes up to 4 gonotrophic cycles (autogeny not reported).	Canopy biter, but bites in understory during daytime. Migrates upward at night.

Appendix A.7. Disease Vector Ecology Profile: Black Fly Vector of Onchocerciasis in Colombia.

VECTOR	GEOGRAPHIC DISTRIBUTION	TRANSMISSION SEASON	LARVAL HABITAT	BITING BEHAVIOR	RESTING BEHAVIOR	FLIGHT RANGE
Simulium exiguum	Pacific coastal region along upper river drainage of the Rio Micay, near San Antonio, department of Cauca, and in drainage areas of the department of NariZo.	Peak in August at the end of second dry season. Parasite development is more rapid during the dry season, thus greater transmission. Throughout day with bimodal peaks first hour after daylight and in afternoon in low wind conditions.	Low altitudes (<100 m) in middle reaches of fast-flowing, large (5 meters or greater) sunlit rivers or up to 2,000 m in small, slow flowing streams. Stones, wood debris, and submerged vegetation in deep parts of river. Water running over shale rock beds.	Anthropophilic in absence of equines or bovines. 80% of bites are below the waist. Actively feed in open sunlight, less in shade.	On vegetation along streams (breeding areas) and at forest edges.	Maximum: 3-4 km

Appendix B: Arthropod Species

Appendix B.1: Species of Mosquitoes Reported from Colombia*

Aedeomyia (*Aedeomyia*)
- *squamipennis*

Aedes (*Howardina*)
- *eleanorae*
- *leei*
- *marinkellei*
- *osornoi*
- *pseudodominicii*
- *septemstriatus*
- *sexlineatus*
- *whitmorei*

Aedes (*Ochlerotatus*)
- *angustivittatus*
- *comitatus*
- *condolescens*
- *crinifer*
- *deficiens*
- *euiris*
- *euplocamus*
- *fulvus*
- *hastatus*
- *hortator*
- *milleri*
- *pectinatus*
- *scapularis*
- *scutellalbum*
- *serratus*
- *taeniorhynchus*

Aedes (*Protomacleaya*)
- *argyrothorax*
- *berlini*
- *braziliensis*
- *buenaventura*
- *insolitus*
- *terrens*
- *zavortinki*

Aedes (*Stegomyia*)
- *aegypti*

Anopheles (*Anopheles*)
- *apicimacula*
- *calderoni*
- *eiseni*
- *mattogrossensis*
- *mediopunctatus*
- *neomaculipalpus*
- *peryassui*
- *pseudomaculipes*
- *pseudopunctipennis*
- *punctimacula*
- *vestipennis*

Anopheles (*Kerteszia*)
- *bambusicolus*
- *bellator*
- *boliviensis*
- *cruzii*
- *homunculus*
- *lepidotus*
- *neivai*

Anopheles (*Lophopodomyia*)
- *gilesi*
- *oiketorakras*
- *squamifemur*

Anopheles (*Nyssorhynchus*)
- *albimanus*
- *albitarsis*
- *allopha*
- *aquasalis*
- *argyritarsis*
- *benarrochi*
- *braziliensis*
- *darlingi*
- *evansae*
- *marajoara*

nuneztovari
oswaldoi
parvus
rangeli
triannulatus
trinkae

Anopheles (*Stethomyia*)
kompi
nimbus
thomasi

Chagasia bathana
bonneae
fajardi

Coquillettidia (*Rhynchotaenia*)
albicosta
arribalzagae
hermanoi
juxtamansonia
lynchi
nigricans
venezuelensis

Culex (*Aedinus*)
amazonensis

Culex (*Anoedioporpa*)
bamborum
browni
conservator
corrigani

Culex (*Belkinomyia*)
elridgei

Culex (*Carrollia*)
antunesi
bihaicola
bonnei
infoliatus
kompi
metempsytus
secundus
urichii
wilsoni

Culex (*Culex*)
abnormalis
alani
archegus
bickleyi
bonneae
brevispinosus
camposi
chidesteri
chitae
coronator
inflictus
maracayensis
mollis
nigripalpus
ousqua
quinquefasciatus
spinosus
stigmatosoma
thriambus
usquatissimus
usquatus

Culex (*Lutzia*)
allostigma
bigoti

Culex (*Melanoconion*)
adamesi
albinensis
alogistus
bastagarius
batesi
caudelli
comatus
commevynensis
comminutor
conspirator
distinguendus
dunni
eastor
educator
elevator
epanatasis

erraticus
ferreri
garcesi
iolambdis
kummi
lucifugus
mistura
ocossa
pedroi
phlogistus
pilosus
quasihibridus
rooti
serratimarge
spissipes
sursumptor
taeniopus
theobaldi
vomerifer
zeteki

Culex (*Microculex*)
carioca
chryselatus
daumastocampa
elongatus
imitator
jenningsi
kukenan

Culex (*Phenacomyia*)
corniger

Culex (Subgenus Uncertain)
flochi
ocellatus

Deinocerites atlanticus
barretoi
colombianus
curiche
dyari
melanophylum
pseudes

Galindomyia leei

Haemagogus (*Conopostegus*)
clarki

Haemagogus (*Haemagogus*)
anastasionis
andinus
boshelli
celeste
chalcospilans
equinus
janthinomys
lucifer

Johnbelkinia longipes
ulopus

Limatus asulleptus
durhamii

Mansonia (*Mansonia*)
humeralis
pseudotitillans
titillans
wilsoni

Orthopodomyia fascipes
phyllozoa

Psorophora (*Grabhamia*)
cingulata
confinnis

Psorophora (*Janthinosoma*)
albigenu
ferox
pilosus
pseudoalbipes

Psorophora (*Psorophora*)
ciliata
cilipes
lineata

Runchomyia (*Ctenogoeldia*)
magna

Sabethes (*Peytonulus*)
ignotus
xenismus
Sabethes (*Sabethes*)
albiprivus
belisarioi
cyaneus
quasicyaneus
tarsopus

Sabethes (*Sabethinus*)
intermedius

Sabethes (*Sabethoides*)
chloropterus

Shannoniana fluviatilis

Toxorhynchites (*Lynchiella*)
bambusicola

Wyeomyia (*Cruzmyia*)
kummi
mattinglyi

Wyeomyia (*Dendromyia*)
complosa
jocosa
pseudopecten
ypsipola

Wyeomyia aporonoma
chalcocephala
clasoleuca
melanocephala
moerbista
personata
serratoria
guadeloupensis
haemorrhoidalis
hypoptes
theobaldi

Trichoprosopon compressum
digitatum
pallidiventer

Uranotaenia (*Uranotaenia*)
apicalis
calosomata
geometrica
hystera
lowii
pulcherrima
riverai

Wyeomyia (*Atunesmyia*)
colombiana
flavifacies

Wyeomyia (*Dodecamyia*)
aphobema

Wyeomyia (*Exallomyia*)
tarsata

Wyeomyia (*Wyeomyia*)
celaenocephala
hosautos
scotino

***References: Knight and Stone 1977, Knight 1978**

Appendix B.2: Species of Kissing Bugs Reported from Colombia*

Belminus rugulosus [2]

Cavernicola pilosa [2]

Eratyrus cuspidatas [2]
Eratyrus mucronatus [2] (east of Andes)

Microtriatoma trinidadensis [2]

Panstrongylus geniculatus
Panstrongylus rufotuberculatus

Rhodnius brethesi
Rhodnius dalessandroi
Rhodnius neivai
Rhodnius pallescens [1]
Rhodnius pictipes
Rhodnius prolixus [1]
Rhodnius robustus

Triatoma dimidiata capitata [1]
Triatoma maculata[1]
Triatoma nigromaculata
Triatoma venosa

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[1] Anthropophilic species that colonize human habitations and are naturally infected with *Trypanosoma cruzi*.
[2] Little or no association with man.

***Reference: Brenner and Stoka 1987**

Appendix B.3: Species of Fleas and Their Hosts Reported from Colombia *

Flea Species	Hosts
Ceratophyllidae	
Plusaetis apollinaris	*Cavia* (guinea pigs), *Mustela* (weasels)
Plusaetis equatoris	*Akodon* (grass mice), *Oryzomys* (rice rats), *Rhipidomys* (climbing mice)
Plusaetis smiti	*Oryzomys* (rice rats), *Thomasomys* (Thomas's paramo mice)
Ctenophthalmidae	
Adoratopsylla antiquorum discreta	*Monodidelphis* (opossums)
Adoratopsylla intermedia copha [2]	Marsupials
Neotyphloceras rosenbergi [2]	Multiple hosts
Leptopsyllidae	
Leptopsylla segnis [1]	*Mus* (house/rice field mice)
Pulicidae	
Ctenocephalides canis [1]	*Canis* (dogs), *Felis* (cats)
Ctenocephalides felis felis [1]	*Canis* (dogs), *Felis* (cats), *Rattus* (rats)
Pulex irritans	*Canis* (dogs), *Cavia* (guinea pigs), *Conepatus* (hog-nosed skunks), *Felis* (cats), humans, *Lagostomus* (plains viscacha)
Pulex simulans	*Cavia* (guinea pigs)
Tunga penetrans [1]	*Canis* (dogs), *Cavia* (guinea pigs), *Felis* (cats), *Rattus* (rats), *Sus* (swine)
Xenopsylla cheopis [1]	*Cavia* (guinea pigs), *Rattus* (rats)
Pygiopsyllidae	
Ctenidiosomus rex	*Oryzomys* (rice rats), *Rhipidomys* (climbing mice, *Thomasomys* (Thomas's paramo mice)
Ctenidiosomus traubi	*Caenolestes* (common shrew opossums)
Rhopalopsyllidae	
Ayeshaepsylla thurmanni	*Oryzomys* (rice rats)
Gephyropsylla klagesi samuelis	*Proechimys* (spiny rats)

Gephyropsylla klagesi klagesi	*Dasyprocta* (agoutis), *Proechimys* (spiny rats)
Polygenis bohlsi bohlsi	*Oligoryzomys* (rice rats)
Polygenis caucensis	*Oryzomys* (rice rats)
Polygenis delpontei	*Oryzomys* (rice rats)
Polygenis peronis	*Heteromys* (forest spiny pocket mice)
Polygenis pradoi	*Melanomys*
Polygenis roberti beebei	*Oryzomys* (rice rats)
Polygenis trapidoi trapidoi	*Oligoryzomys* (rice rats)
Polygenis hopkinsi	*Oryzomys* (rice rats)
Rhopalopsyllus australis australis	*Dasyprocta* (agoutis), *Proechimys* (spiny rats)
Rhopalopsyllus australis tamoyus	*Proechimys* (spiny rats)
Rhopalopsyllus australis tupinus	*Speothos* (bush dog)
Rhopalopsyllus lugubris cryptoctenes	*Agouti* (pacas)
Rhopalopsyllus saevus	Didelphid marsupials
Scolopsyllus colombianus	*Oryzomys* (rice rats)
Tetrapsyllus comis	*Akodon* (grass mice), *Sigmodon* (cotton rats), *Thomasomys* (Thomas's paramo mice)

Stephanocircidae

Cleopsylla monticola	*Caenolestes* (common shrew opossums)
Plocopsylla thor	*Oryzomys* (rice rats), *Thomasomys* (Thomas's paramo mice)

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[1]Anthropophilic.
[2]Found naturally infected with plague.
[3]Transmitted plague experimentally in the laboratory.
Genera of known host(s) and their common names are listed to the right of each species.
Bat and bird fleas have not been implicated in disease transmission and are not included.

***Reference: Johnson 1957, Lewis 1972, 1973, 1974a, b, c, 1975**

Appendix B.4: Species of Ticks and Their Hosts Reported from Colombia*

Tick Species	Hosts
Argasidae	
Antricola mexicanus	*Corollia perspicillata, Desmodus rotundus, Mormoops megalophylla, Pteronotus personata* (bats)
Argas magnus	
Argas miniatus	poultry
Argas persicus	*Phyllostomus discolor* (bat)
Ornithodoros azteci	*Carollia perspicillata, Desmodus rotundus* (bats)
Ornithodoros brodyi	*Corollia perspicillata, Natalus tumidirostris* (bats)
Ornithodoros hasei	*Artibeus jamaicensis, Noctilio labialis, Phyllostomus discolor, Rhogeessa parvula* (bats)
Ornithodoros marinkellei	*Pteronotus parnellii, Pteronotus personata* (bats)
Ornithodoros marmosae	*Marmosa* (opossum)
Ornithodoros peropteryx	*Peropteryx macrotis* (bat)
Ornithodoros puertoricensis	
Ornithodoros rossi	*Desmodus rotundus, Glossophaga longirostris, Micronycteris megalotis* (bats)
Ornithodoros rudis	*Hoplomys gymnurus* (thick-spined rat)
Ornithodoros talaje	
Ornithodoros yumatensis	*Pteropteryx macrotis*
Otobius megnini	*Odocoileus* (deer)
Ixodidae	
Amblyomma auricularium	dog, *Dasypus kappleri* (armadillo), *Dasypus sabanicola* (armadillo), *Myrmecophaga* (anteater)

Amblyomma cajennense	humans, cattle, horse, *Agouti paca* (paca), *Bos indicus* (ox), *B. taurus* (ox), *Didelphis marsupialis* (opossum), *Marmosa robinsoni* (opossum), *Myrmecophaga tridactyla* (anteater)
Amblyomma calcaratum	*Tamandua tetradactyla* (anteater)
Amblyomma coelebs	*Myrmecophaga tridactyla*
Amblyomma crassum	*Testudo, Geochelone denticulata*
Amblyomma dissimile	*Bufo marinus* (toad), *Caiman sclerops* (caiman), *Constrictor constrictor, Boa constrictor*
Amblyomma geayi	*Caluromys* (opossum)
Amblyomma humerale	*Geochelone denticulata*
Amblyomma incisum	*Myrmecophaga*
Amblyomma longirostre	porcupines
Amblyomma maculatum	cattle, dog
Amblyomma multipunctum	*Tapirus pinchaque* (tapir)
Amblyomma naponense	peccary, *Didelphis marsupialis, Heteromys australis* (spiny pocket mouse), *Myrmecophaga*
Amblyomma nodosum	*Ramphocelus carbo*
Amblyomma oblongoguttatum	*Dasyprocta fuliginosa* (agouti), *Tayassu pecari* (collared peccary)
Amblyomma ovale	dog, cattle, toad, *Eira, Nasua nasua* (coatis)
Amblyomma pacae	*Agouti paca* (paca)
Amblyomma parvum	*Dasypus sabanicola* (armadillo)
Amblyomma pecarium	peccary
Amblyomma rotundatum	*Bufo marinus* (toad)
Amblyomma sabanerae	*Rhinoclemmys annulata*
Amblyomma scalpturatum	*Sus scrofa* (domestic pig)
Amblyomma tapirellum	peccary, *Alouatta palliata* (howler monkey)
Amblyomma tigrinum	*Agouti paca* (paca), *Bos indicus, Quiscalus lugubris*
Amblyomma triste	dog, deer
Amblyomma varium	*Bradypus griseus, B. tridactylus* (sloths)
Anocentor nitens	donkey, horse, mule, *Bos taurus*
Boophilus microplus	human, cattle, deer, *Bos taurus*
Dermacentor imitans	human, *Tayassu pecari* (collared peccary)
Haemaphysalis juxtakochi	*Mazama* (brocket deer)
Haemaphysalis leporispalustris	rabbit
Haemaphysalis montgomeryi	
Hyalomma lusitanicum (introduced)	cattle
Ixodes affinis	

Ixodes andinus (probable)	*Oryzomys albigularis* (rice rat)
Ixodes auritulus	*Atlapetes pallidinucha, Thrauris cyanocephala*
Ixodes bequaerti	*Diglossa cyanea*
Ixodes boliviensis	*Tremarctos ornatus* (spectacled bear)
Ixodes lasallei	*Dasyprocta* (agouti)
Ixodes loricatus	*Metachirus nudicaudatus* (opossum), *Oryzomys capito* (rice rat)
Ixodes luciae	*Dasyprocta punctata* (agouti), *Didelphis marsupialis*
Ixodes montoyanus	*Pudu* (deer)
Ixodes pararicinus	
Ixodes tapirus	*Tapirus pinchaque* (tapir)
Ixodes tropicalis	*Oryzomys laticeps* (rice rat)
Ixodes venezuelensis	*Oryzomys caliginosus* (rice rat)
Rhipicephalus sanguineus	dog, mule, rabbit, *Phaseolus vulgaris*

***Reference: Doss et al. 1978**

Appendix B.5: Species of Sand Flies Reported from Colombia*

Lutzomyia (*Coromyia*)
- *isovespertilionis*
- *vespertilionis*

Lutzomyia (*Dampofomyia*)
- *insolita*[2]
- *rosabali*

Lutzomyia (Species Group *Saulensis*)
- *saulensis*

Lutzomyia (*Evandromyia*)
- *cerqueirai*
- *infraspinosa*
- *monstruosa*

Lutzomyia (*Helcocyrtomyia*)
- *cirrita*
- *erwindonaldoi*[1]
- *hartmanni* [1, 7]
- *osornoi*
- *sanguinaria*[3]
- *scorzai*
- *strictivilla*[1]
- *tortura*[1]
- *Lu.* sp. of Pichinde

Lutzomyia (*Lutzomyia*)
- *araracuarensis*
- *bifoliata*
- *cruciata*[1,2]
- *evangelistai*
- *falcata*
- *gomezi*[1]
- *longipalpus*[1,6]
- *lichyi*[1]
- *marinkellei*
- *sherlocki*

Lutzomyia (*Micropygomyia*)
- *atroclavata*[1]
- *cayennensis cayennensis*
- *micropyga*
- *yencanensis*
- *venezuelensis*

Lutzomyia (Species Group *Oswaldoi*)
- *Lu.* sp. #2
- *rorotaensis*
- *trinidadensis*

Lutzomyia (Species Group *Pilosa*)
- *pilosa*

Lutzomyia (*Nyssomyia*)
- *anduzei*[1,2,8]
- *antunesi*[1]
- *flaviscutellata*[1]
- *hernandezi*
- *olmeca bicolor*[1]
- *richardwardi*[1]
- *trapidoi*[1,11]
- *umbratilis*[1,4,8,13]
- *ylephiletor*[1,5,10, 11]
- *yuilli yuilli*[1]

Lutzomyia (*Pintomyia*)
- *christenseni*
- *damascenoi*

Lutzomyia (*Pressatia*)
- *calcarata*
- *camposi*
- *choti*
- *dysponeta*
- *Lu.* sp. # 1
- *tricantha*

Lutzomyia (Species Group *Baityi*)
- *baityi*
- *gorbitzi*

Lutzomyia (*Psathyromyia*)
- *abonnenci*
- *campbelli*[2]
- *cratifer*[2]
- *cuzquena*
- *dasymera*
- *dendrophyla*[1]
- *lutziana*
- *punctigeniculata*
- *scaffi*
- *shannoni*[1]
- *undulata*

Lutzomyia (Species Group *Aragaoi*)
- *aragaoi*
- *barrettoi barrettoi*
- *barrettoi majuscula*
- *carpenteri*
- *runoides*

Lutzomyia (Species Group *Dreisbachi*)
- *dresbachi*
- *aclydifera*

Lutzomyia (*Psychodopygus*)
- *amazonensis*
- *ayrozai*[1]
- *bernalei*
- *bispinosa*
- *carrerai carrerai*
- *carrerai thula*
- *chagasi*[1]
- *claustrei*[3]
- *corossoniensis*
- *davisi*[1]
- *fairtigi*
- *geniculata*[1]
- *hirsuta hirsuta*[1,3]
- *Lu.* sp. of Tres Esquinas
- *nocticola*
- *panamensis*[1,5, 11]
- *paraensis*
- *parimaensis*
- *recurva*

Lutzomyia (*Sciopemyia*)
- *preclara*
- *sordellii*

Lutzomyia (Species Group *Migonei*)
- *dubitans*
- *migonei*[1]
- *sericea*[2]
- *walkeri*

Lutzomyia (Species Group *Verrucarum*)
- *andina*[1]
- *colombiana*[1,12?]
- *disiuncta*
- *evansi*[1,6]
- *guasitownsandi*
- *longiflocosa*[1]
- *moralesi*
- *nevesi*[1]
- *nuneztovari*[1,3]
- *ovallesi*[1,5]
- *sauroida*[1,5?]
- *serrana*[1]
- *spinicrassa*[1,2,5]
- *torvida*[1]
- *townsendii*[1,5?]
- *youngi*[2]

Lutzomyia (*Trichophoromyia*)
- *auraensis*
- *bettinii*[2]
- *cellulana*
- *howardi*
- *reburra*
- *ubiquitalis*[1,9]

Lutzomyia (*Trichopygomyia*)
- *conviti*
- *ferroae*
- *longispina*
- *martinezi*
- *triramula*
- *wagleyi*
- *witoto*

Lutzomyia (*Viannamyia*)
caprina
furcata
tuberculata

pia[1]
ponsi
rangeliana
Lu. sp. of Anchicaya

Ungrouped *Lutzomyia*
ignacioi

Warileya rotundipennis

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[1] Anthropophilic
[2] Sand fly likely occurs, but not confirmed
[3] *Leishmania* (species not confirmed)
[4] *Leishmania amazonensis*
[5] *Leishmania brasiliensis*
[6] *Leishmania chagasi*
[7] *Leishmania colombiensis*
[8] *Leishmania guyenensis*
[9] *Leishmania lainsoni*
[10] *Leishmania mexicana*
[11] *Leishmania panamensis*
[12] Bartonellosis
[13] Phlebovirus

***Reference: Young and Duncan 1994**

Appendix B.6: Species of Black Flies Reported from Colombia*

Simulium (*Cerqueirellum*)
- *amazonicum*
- *argentiscutum*
- *oyapockense*
- *sanguineum*

Simulium (*Coscaroniellum*)
- *quadrifidum*

Simulium (*Ectemnapsis*)
- *bicoloratum*
- *bicornutum*
- *furcillatum*
- *ignescens*
- *pifanoi*
- *romanai*
- *tolimaense*

Simulium (*Hemicnetha*)
- *mexicanum*
- *muiscorum*
- *paynei*
- *rubrithorax*
- *seriatum*

Simulium (*Inaequalium*)
- *subnigrum*

Simulium (*Notolepria*)
- *exiguum*

Simulium (*Psaroniocompsa*)
- *incrustatum*
- *jujuyense*

Simulium (*Psilopelmia*)
- *dinellii*
- *escomeli*
- *gabaldoni*
- *haematopotum*
- *lutzianum*
- *ochraceum*
- *quadrivittatum*
- *rorotaense*
- *samboni*
- *shewellianum*

Simulium (*Simulium*)
- *matteabranchium*
- *metallicum*

Simulium (*Trichodagmia*)
- *lahillei*
- *orbitale*

Simulium not placed in subgenus
- *scutellatum*
- *tarsatum*

***Reference: Kim and Merritt 1987**

Appendix B.7: Species of Scorpions Reported from Colombia*

Buthidae

Ananteris gongonae (Pacific coastal)
Ananteris columbianus (Atlantic coastal)

Centruroides gracilis (Pacific coastal)
Centruroides margaritatus (Western Colombia – inflicts many stings)
Centruroides noxius (more common in lowlands)
Centruroides suffusus (more common in lowlands)

Rhopalurus laticauda (Atlantic coastal)

Tityus asthenes (Pacific coastal)
Tityus clathratus (Department of Vichada)
Tityus columbianus (Bogot<-Boyaca and Magdalena-Cauca Region)
Tityus engelkei (Atlantic coastal)
Tityus festae (Atlantic coastal)
Tityus filodendron (Amazonian Region)
Tityus forcipula (Pacific coastal)
Tityus fuhrmanni (Magdalena-Cauca Region)
Tityus intermedius (Pacific coastal)
Tityus macrochirus (Bogot<-Boyaca and Magdalena-Cauca Region)
Tityus nematochirus (Bogot<-Boyaca and Magdalena-Cauca Region)
Tityus pachyurus (Pacific Region)
Tityus parvulus (Magdalena and Santa Marta Regions)
Tityus sastnei (Pacific coastal)
Tityus tayrona (Atlantic coastal)
Tityus urbinae (Amazonian Region)

Chactidae

Brotheas camposi (Amazonian Region)
Brotheas lichyi (Amazonian Region)
Brotheas mingueti (Amazonian Region)

Chactas aequinoctialis (Atlantic coastal)
Chactas brevicaudatus (Atlantic coastal)
Chactas keyserlingi (Bogot<-Boyaca)
Chactas mauniesi (Pacific coastal)
Chactas oxfordi (Atlantic coastal)
Chactas reticulatus (Magdalena-Cauca Region)
Chactas scabrimanus (Bogot<-Boyaca)

Chactas vanbenedeni (Pacific coastal)

Teuthraustes adrianae (Amazonian Region)
Teuthraustes carmelinae (Amazonian Region)

Diplocentridae

Tarsoporosus klugeri anchicaya (Pacific coastal)
Tarsoporosus klugeri klugere (coastal bordering Venezuela)

Ischnuridae

Opisthacanthus lepturus (Pacific coastal)

***Reference: Polis 1990**

Appendix C: Species of Venomous Snakes from Colombia*, **

Elapidae

Leptomicrurus narduccii (Amazonian slopes of Andes in southern Colombia)
Leptomicrurus schmidti (considered *L. scutiventris* by some herpetologists)

Micrurus ancoralis (Pacific coastal Colombia)
Micrurus bocourti (northern Colombia)
Micrurus clarki (western Colombia)
Micrurus dissoleucus ssp. (northern Colombia)
Micrurus dumerilii spp.
Micrurus filiformis (Amazonian lowlands of southeastern Colombia)
Micrurus hemprichii (upper Amazon drainages)
Micrurus isozonus (Meta Department)
Micrurus langsdorffi (upper Amazon region of southern Colombia)
Micrurus lemniscatus [6] (Amazon region)
Micrurus mipartitus [6, 10] (black-ringed coral snake, Pacific coast)
Micrurus multiscutatus (Cauca Department)
Micrurus nigrocinctus (Pacific coast in northern Colombia)
Micrurus psyches (Meta Department)
Micrurus putumayensis (southeast Colombia)
Micrurus sangilensis (northern Colombia)
Micrurus spixii [6] (upper Amazon region of southern Colombia)
Micrurus spurrelli (Choco Department)
Micrurus steindachneri (likely occurs, but not confirmed)
Micrurus surinamensis (eastern Colombia)

Viperidae

Agkistrodon bilineatus (water mocassin, cantil)

Bothriechis schlegelii [6] (eye-lash pit viper, baracara, palm viper)
Bothriopsis albocarinata (likely occurs on eastern versant of Andes, but not confirmed)
Bothriopsis bilineata (emerald pit viper)
Bothriopsis punctata
Bothriopsis taeniata (east of Andes)

Bothrops asper [5, 6, 7, 10] (fer de lance, coastal areas)
Bothrops atrox [2, 6, 8, 9, 10, 11] (barba amarilla, equatorial forests)
Bothrops brazili (equatorial forests)
Bothrops castelnaudi (robo de chucha, Cauca Valley and east of Andes, 1,000-1,500 m)
Bothrops lansbergi (hog nosed viper, banana plantations of Santa Marta area)
Bothrops microphthalmus (Antioquia and Cauca Departments)

Bothrops monticelli (robo de chucha, Cauca Valley and east of Andes, 1,000-1,500 m)
Bothrops neglectus (robo de raton, Colombian-Brazilian border)
Bothrops pulcher (Pacific lowlands)
Bothrops xanthogrammus (likely occurs on Pacific Andean slopes, but not confirmed)

Crotalus durissus cumanensis [1, 2, 3, 4, 5, 6, 8, 9, 11] (cascabel or tropical rattlesnake, northeast Colombia)

Lachesis mutus [2, 4, 6, 9, 10, 11] (bushmaster, equatoria forests and Pacific slopes)

Porthidium hyoprora (forests of southern Colombia)
Porthidium lansbergii
Porthidium nasutum (hog-nosed viper)

Footnotes – Antivenoms available from corresponding antivenom providers may be found in Appendix D.

***Reference: Campbell and Lamar 1993**
****Additional information on venomous snakes is available on AFMIC'S MEDIC CD-ROM**

Appendix D: Sources of Snake Antivenoms*

Argentina - 1
Instituto Nacional de Microbiologa
"Dr. Carlos G. Malbran"
Av. Velez Sarsfield 563
Buenos Aires, Argentina

Brazil – 2
Fundahno Ezequiel Dias
Rua Conde Pereira
80-Gameleira 30550
Belo Horizonte-MG
Brazil
TEL: (031) 332-2077
FAX: (031) 332-2534
TELEX: 392417 FEDS BR

Brazil – 3
Institutos Vital Brazil S.A.
Caixa Postal 28
Niteroi
Rio de Janeiro, Brazil
TEL: 55212558688

Brazil - 4
Instituto Butantan
Av. Dr. Vital Brazil, 1500
Caixa Postal 65
CEP 01051
Sno Paulo, SP, Brazil
FAX: (011) 815-1505
TELEX: (011) 83325 BUTA BR

Colombia - 5
Instituto Nacional de Salud
Av. Eldorado con Carrera 50, Zona 6
Bogot<, Colombia
FAX: 57-1-2220975
TEL: 57-1-2220577, ext. 147

Costa Rica - 6
Instituto Clodomiro Picado
Facultad de Microbiologia
Universidad de Costa Rica
Ciudad Universitaria "Rodrigo Facio"
San JosJ, Costa Rica
FAX: (506) 29-31-35
TEL: (506) 29-03-44

Ecuador - 7
Instituto Nacional de Higiene y
Medicina Tropical
"Leopoldo Izquieta PJrez"
Casilla Postal 3961
Guayaquil, Ecuador

Mexico – 8
Zapata Laboratories
Mexico City, Mexico
TEL: 592-82-70
TEL: 561-12-11
TEL: 592-88-93

Pennsylvania, U.S.A. – 9
Wyeth International Ltd.
P.O. Box 8299
Philadelphia, PA 19101-1245
TEL: (215) 688-4400

Peru - 10
Instituto Nacional de Hygiene
Lima, Peru

Peru – 11
Institutos Nacionales de Salud
Departamento de Animales Venenosos
Calle Capac Yupanqui 1400
Apartado 451
Lima, Peru
TEL: (51) 14416141
TEL: (51) 14678212
TEL: (51) 14311130

***Additional information on antivenoms is available on AFMIC's MEDIC CD-ROM**

Appendix E: Plants Of Colombia That Cause Contact Dermatitis*

Agave spp., motua, penca (sap of leaves - saponin)
Ammannia spp., aquatic plants
Anacardium occidentale, cashew cut (nut, bark, leaves - anacardic acid)
Calophyllum inophyllum, aceite maria
Caloptropis spp., found in dry open areas, milkweed (milky sap)
Comocladia spp.
Croton spp., sangre de grado (resinous oil)
Dalechampia spp., vines in disturbed areas
Daphne spp. (sap - mezercin)
Euphorbia spp. (sap - euphorbin)
Hippomane mancinella, manzanillo (milky latex and fruit)
Hura spp., ceiba amarilla, ceiba de leche, ceiba de agua (sap)
Malpighia spp., found in dry deciduous forests
Mucuna pruriens
Ricinus spp., castor bean (dust of seeds)
Schinus spp., found in inter-Andean valleys
Sterculia spp., teta vieja, camajoru
Thevetia peruviana, found in inter-Andean valleys (seeds, leaves, stems and roots)
Toxicodendron spp., manzanillo (poison oak/ivy)
Urera spp., urticating nettle

***Additional information on vegetation is available on AFMIC's MEDIC CD-ROM**

Appendix F: Plants Of Colombia That Are Toxic When Ingested*

Ageratina altissima
Anacardium occidentale, cashew nut (nut and shell - anacardic acid)
Blighia sapida
Brugmansia spp., tree-like (seeds)
Calophyllum inophyllum, aceite maria
Caloptropis spp., milkweed, found in dry open areas
Coriaria spp., sprawling shrub in open areas and woody liana in forests (small fruits – corianmyratine)
Crotalaria spp.
Croton spp., algodoncillo
Daphne spp.
Datura spp. (seeds-scopolamine and hyoscyamine)
Dioscorea bulbifera (bulbs, if eaten uncooked)
Duranta spp., weedy herbs in dry areas
Euphorbia spp. (sap and seeds)
Heliotropium spp., heliotrope
Hippomane mancinella, manzanillo (milky latex and fruit)
Hura spp., tronador, sandbox, ceiba amarilla, ceiba de leche, ceiba de agua (sap, seeds and bark)
Jacquinia spp.
Jatropha spp., ortiga, nettle (seeds)
Manihot esculenta (uncooked roots - hydrocyanic acid)
Melia spp.
Phytolacca spp.
Pilocarpus spp. (pilocarpine nitrate poisoning)
Pilea spp., found in cloud forests, some are epiphytic
Ricinus spp. castor bean (seeds, leaves and stems)
Sapium spp., cauchillo
Schinus spp., found in inter-Andean valleys
Solandra spp., found in cloud forests and lowland wet forests
Solanum spp. (fruits and leaves)
Strychnos spp., canopy lianas (contains curare alkaloids)
Thevetia peruviana, found in inter-Andean valleys (seeds, leaves, stems and roots)
Toxicodendron spp., manzanillo, found in middle elevation forests, poison oak/ivy

***Additional information on vegetation is available on AFMIC's MEDIC CD-ROM**

Appendix G: Selected List of Identification Keys

Argasidae/Ixodidae

Fairchild, G.V., G.M. Kohls and V.J. Tipton. 1966. The Ticks of Panama (Acarina: Ixodoidea), pp. 167-219. *In*: R.L. Wenzel and V.J. Tipton (Ed.), Ectoparasites of Panama. Field Museum of Natural History, Chicago.

Jones, E. K., and C. M. Clifford. 1972. The Systematics of the Subfamily Ornithodorinae (Acarina: Argasidae). V. A Revised Key to Larval Argasidae of the Western Hemisphere and Description of Seven New Species of *Ornithodoros*. Ann. Entomol. Soc. Am., 65(3): 730-40.

Jones, E.K., C.M. Clifford, J.E. Keirans and G.M. Kohls. 1972. The Ticks of Venezuela (Acarina: Ixodoidea) with a Key to the Species of *Amblyomma* in the Western Hemisphere. Brigham Young Univ. Sci. Bull., Biol. Ser., 17(4): 1-40.

Keirans, J.E., H. Hoogstraal and C.M. Clifford. 1979. Observations on the Subgenus *Argas* (Ixodoidea: Argasidae: *Argas*). 16. *Argas* (*A*.) *moreli*, New Species, and Keys to Neotropical Species of the Subgenus. J. Med. Entomol., 15(3): 246-52.

Culicidae

Arnell, J.H. 1973. Mosquito Studies (Diptera, Culicidae). XXXII. A Revision of the Genus *Haemagogus*. Contrib. Am. Entomol. Inst., 10(2): 1-174.

Dodge, H.R. 1962. Supergeneric Groups of Mosquitoes. Mosq. News, 22(4): 365-68.

Faran, M.E. 1980. Mosquito Studies (Diptera, Culicidae) XXXIV. A Revision of the Albimanus Section of the Subgenus *Nyssorhynchus* of *Anopheles*. Contrib. Am. Entomol. Inst., 15(7): 1-215.

Faran, M.E. and K.J. Linthicum. 1981. A Handbook of the Amazonian Species of *Anopheles* (*Nyssorhynchus*) (Diptera: Culicidae). Mosq. Syst., 13(1): 1-81.

Gorham, J.R., C.J. Stojanovich and H.G. Scott. 1973. Illustrated Key to the Anopheline Mosquitoes of Western South America. Mosq. Syst., 5: 97-123.

Kumm, H.W., E. Osorno-Mesa and J. Boshell-Manrique. 1946. Studies of Mosquitoes of the Genus *Haemagogus* in Colombia (Diptera, Culicidae). Am. J. Hyg., 43: 13-28.

Lane, J. 1953. Neotropical Culicidae. Volumes I & II, Sno Paulo, Univ. Sno Paulo.

Levi-Castillo, R. 1951. Los Mosquitos del Genero *Haemagogus* Williston, 1896 en America del sur. Editorial "Don Bosco," Cuenca, Ecuador. 76 pp.

Linthicum, K.J. 1988. A Revision of the *Argyritarsis* Section of the Subgenus *Nyssorhynchus* of *Anopheles*. Mosq. Syst., 20(2): 98-271.

Pecor, J.E., V.L. Mallampalli, R.E. Harbach and E.L. Peyton. 1992. Catalog and Illustrated Review of the Subgenus *Melanoconion* of *Culex* (Diptera: Culicidae). Contrib. Am. Entomol. Inst., 27: 1-228.

Sirivanakarn, S. 1982(1983). A Review of the Systematics and Proposed Scheme of Internal Classification of the New World Subgenus *Melanoconion* of *Culex* (Diptera: Culicidae). Mosq. Syst., 14(4): 265-333.

Zavortink, J.J. 1970. Mosquito Studies (Diptera, Culicidae) XIX. The Treehole *Anopheles* of the New World. Contrib. Am. Entomol. Inst., 5(2): 135.

Zavortink, J.J. 1973. Mosquito Studies (Diptera, Culicidae) XXIX. A Review of the Subgenus *Kerteszia* of *Anopheles*. Contrib. Am. Entomol. Inst., 9(3): 1-54.

Mammalia

DeBlase, A.F. and R.E. Martin. 1974. A Manual of Mammalogy, with Keys to Families of the World. Wm. C. Brown Company Publishers, Dubuque, Iowa, 329 pp. (Mammal trapping and ectoparasite collecting techniques, study skin preparations, and keys to family level)

Eisenberg, J.F. 1989. Mammals of the Neotropics, the Northern Cone. Vol. 1, Panama, Colombia, Venezuela, Guyana, Suriname, French Guiana. Univ. Chicago Press, Chicago, 449 pp. (Generic keys and index to common names)

Emmons, L.H. and F. Feer. 1997. Neotropical Rainforest Mammals, A Field Guide, 2nd Edition, Univ. Chicago Press, Chicago, 307 pp. (Family and generic keys – excellent detailed color illustrations of many species)

Fisler, G.F. 1970. Keys to Identification of the Orders and Families of Living Mammals of the World. Los Angeles County Museum of Natural History, Sci. Ser., 25(2): 1-29. (Order and family keys)

Lawlor, T.E. 1976. Handbood of the Orders and Families of Living Mammals. MAD River Press, Eureka, California, 244 pp.

Plants

Gentry, A.H. 1993. A Field Guide to the Families and Genera of Woody Plants of Northwest South America (Colombia, Ecuador, Peru) with Supplementary Notes on Herbaceous Taxa, Univ. Chicago Press, Chicago, 895 pp.

Psychodidae

Young, D.G. and M.A. Duncan. 1994. Guide to the Identification and Geographic Distribution of *Lutozomyia* Sand Flies in Mexico, the West Indies, Central and South America (Diptera: Psychodidae). Mem. Am. Entomol. Inst. No. 54, 881 pp.

Reduviidae

Lent, H., and P. Wygodzinsky. 1979. Revision of the Triatominae (Hemiptera, Reduviidae) and Their Significance as Vectors of Chagas' Disease. Bull. Am. Mus. Nat. Hist., 163(3): pp. 125-520.

Simuliidae

Coscar\n, S. 1987. El GJnero *Simulium* Latreille en la Regi\n Neotropical: An<lisis de los Grupos Supraespec?ficos, Especies que los Integran y Distributi\n Geogr<fica (Simuliidae, Diptera). Museu Paraense Em?lio Goeldi, BelJm, Brazil. 112 pp. (In Spanish).

Coscar\n, S. 1991. Simuliidae. Fauna de Agua Dulce de la Republica Argentina.38. (Insecta, Diptera, Simuliidae), Fascicle 2, 304 pp. +78 pp. of unnumbered figures (In Spanish).

Coscar\n, S. and P. Wygodzinsky. 1972. Taxonomy and Distribution of the Blackfly Subgenus *Simulium* (*Pternaspatha*) Enderlein (Simuliidae, Diptera, Insecta). Bull. Am. Mus. Nat. Hist., 147: 199-240.

Shelley, A.J., M. Arzube and C.A. Couch. 1989. The Simuliidae (Diptera) of the Santiago Onchocerciasis Focus of Ecuador. Bull. Br. Mus. Nat. Hist. (Ent.), 58(1): 79-130.

Shelley, A.J., C.A. Lowry, M. Maia-Herzog, A.P.A. Luna Dias and M.A.P. Moraes. 1997. Biosystematic Studies on the Simuliidae (Diptera) of the Amazonia Onchocerciasis Focus. Bull. Nat. Hist. Mus., London (Ent.), 66(1): 1-121.

Shelley, A.J., R.R. Pinger and M.A.P. Moraes. 1982. The Taxonomy, Biology and Medical Importance of *Simulium amazonicum* Goeldi (Diptera: Simulidae), with a Review of Related Species. Bull. Brit. Mus. (Nat. Hist.), 44(1): 1-29.

Siphonaptera

Hopkins, G.H.E. and M. Rothschild. 1953. An Illustrated Catalogue of the Rothschild Collection of Fleas (Siphonaptera) in the British Museum (Natural History). I. Tungidae and Pulicidae. British Museum, Natural History, London, 361 pp. + 45 plates.

Johnson, P.T. 1957. A Classification of the Siphonaptera of South America. Mem. Ent. Soc. Wash. No. 5., Entomological Society of Washington, Washington, DC. 298 pp.

Smit, F.G.A.M. 1987. An Illustrated Catalogue of the Rothschild Collection of Fleas (Siphonaptera) in the British Museum (Natural History). Volume VII. Malacopsyllidae and Rhopalopsyllidae. Oxford University Press, London, 380 pp. + 5 plates.

Tipton, V.J. and E. Mendez. 1966. Fleas (Siphonaptera) of Panama. In: Ectoparasites of Panama, Wenzel, R.L. and V.J. Tipton, eds., Field Museum of Natural History, pp.289-338, + plates 47-93.

Snakes

Peters, J.A. 1972. The Snakes of Ecuador, A Check List and Key. Bull., Mus. Comp. Zool., 122(9): 491-541.

Tabanidae

Coscar\n, S. and N. Papavero. 1993. An Illustrated Manual for the Identification of the Neotropical Genera and Subgenera of Tabanidae (Diptera). Museu Paraense Emilio Goeldi, Belem. 150 pp.

Fairchild, G.B. 1969. Notes on Neotropical Tabanidae XII. Classification and Distribution, with Keys to Genera and Subgenera. Arquivos de Zoologica., Sno Paulo, 17(4): 199-255.

Fairchild, G.B. and J.F. Burger. 1994. A Catalog of the Tabanidae (Diptera) of the Americas South of the United States. Mem. Am. Entomol. Inst. No. 55, 249 pp.

Wilkerson, R.C. 1979. Horse Flies of the Colombian Departments of Choco, Valle and Cauca. Cespedesia 8: 87-433.

Trombiculidae

Brennan, J.M. and M.L. Goff. 1977. Keys to the Genera of Chiggers of the Western Hemisphere (Acarina: Trombiculidae). J. Parasitol., 63(3): 554-66.

Appendix H: Personal Protective Measures

Personal protective measures are the first line of defense against arthropod-borne disease and, in some cases, may be the only protection for deployed military personnel. Proper wearing of the uniform and appropriate use of repellents can provide high levels of protection against blood-sucking arthropods. The uniform fabric provides a significant mechanical barrier to mosquitoes and other blood-sucking insects. Therefore, the uniform should be worn to cover as much skin as possible if weather and physical activity permit. When personnel are operating in tick-infested areas, they should tuck their pant legs into their boots to prevent access to the skin by ticks, chiggers, and other crawling arthropods. They should also check themselves frequently for ticks and immediately remove any that are found. If a tick has attached, seek assistance from medical authorities for proper removal or follow these guidelines from TIM 36, Appendix C:

1. **Grasp the tick's mouthparts** where they enter the skin, using pointed tweezers.

2. **Pull out** slowly and steadily with gentle force.

    a. Pull in the reverse of the direction in which the mouthparts are inserted, as you would for a splinter.

    b. **Be patient** – The long, central mouthpart (called the hypostome) is inserted in the skin. It is covered with sharp barbs, sometimes making removal difficult and time consuming.

    c. Many hard ticks secrete a cement-like substance during feeding. This material helps secure their mouthparts firmly in the flesh and adds to the difficulty of removal.

    d. It is important to continue to pull steadily until the tick can be eased out of the skin.

    e. **Do not** pull back sharply, as this may tear the mouthparts from the body of the tick, leaving them embedded in the skin. If this happens, don't panic. Embedded mouthparts are comparable to having a splinter in your skin. However, to prevent secondary infection, it is best to remove them. Seek medical assistance if necessary.

    f. **Do not** squeeze or crush the body of the tick because this may force infective body fluids through the mouthparts and into the wound.

    g. **Do not** apply substances like petroleum jelly, fingernail polish remover, repellents, pesticides, or a lighted match to the tick while it is attached. These materials are either ineffective or, worse, may agitate the tick and cause it to salivate or regurgitate infective fluid into the wound site.

h. If tweezers are not available, grasp the tick's mouthparts between your fingernails, and remove the tick carefully by hand. Be sure to wash you hands -- especially under your fingernails -- to prevent possible contamination by infective material from the tick.

3. Following removal of the tick, **wash the wound** (and your hands) with soap and water and **apply an antiseptic.**

4. **Save the tick** in a jar, vial, small plastic bag, or other container for identification should you later develop disease symptoms. Preserve the tick by either adding some alcohol to the jar or by keeping it in a freezer. Storing a tick in water will not preserve it. Identification of the tick will help the physician's diagnosis and treatment, since many tick-borne diseases are transmitted only by certain species.

5. **Discard** the tick after one month; all known tick-borne diseases will generally display symptoms within this time period.

Newly developed repellents provide military personnel with unprecedented levels of protection. An aerosol formulation of permethrin (NSN 6840-01-278-1336) can be applied to the uniform according to label directions, but not to the skin. This will impart both repellent and insecticidal properties to the uniform material that will be retained through numerous washings. An extended formulation lotion of N, N-diethyl-m-toluamide (deet) (NSN 6840-01-284-3982) has been developed to replace the 2 oz. bottles of 75% deet in alcohol. This lotion contains 33% active ingredient. It is less irritating to the skin, has less odor and is generally more acceptable to the user. A properly worn Battle Dress Uniform (BDU) impregnated with permethrin, combined with use of extended duration deet on exposed skin, has been demonstrated to provide nearly 100% protection against a variety of blood-sucking arthropods. This dual strategy is termed the DoD INSECT REPELLENT SYSTEM. In addition, permethrin may be applied to bednets, tents, and other field items as appropriate. Complete details regarding these and other personal protective measures are provided in TIM 36, *Personal Protective Techniques Against Insects and Other Arthropods of Military Significance* (1996).

Appendix I: Points of Contact for Colombia

U.S. Embassy
Consular Section
Calle 22-d Bis, No. 47-51
Avenida El Dorado and Carrera 50
Bogot<, Colombia
Country/City Code: 011-57-1
Telephone: 315-0811
Fax: 315-2197

U.S. Embassy
Consular Section
Calle 77, No. 68-15
Barranquilla, Colombia
Country/City Code: 011-57-5
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