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Triage for the Biosphere: The Need and Rationale for Taxonomic Inventories and Phylogenetic Studies of Parasites

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ABSTRACT: A parasitological perspective in biodiversity survey and inventory provides powerful insights into the history, structure, and maintenance of the biosphere. Parasitology contributes a powerful conceptual paradigm or landscape that links ecology, systematics, evolution, biogeography, behavior, and an array of biological phenomena from the molecular to the organismal level across the continuum of microparasites to macroparasites and their vertebrate and invertebrate hosts. Effective survey and inventory can be strategically focused or can take a synoptic approach, such as that represented by the All Taxa Biodiversity Inventory. We argue that parasitology should be an integral component of any programs for biodiversity assessment on local, regional, or global scales. Taxonomists, who constitute the global taxasphere, hold the key to the development of effective surveys and inventories and eventual linkage to significant environmental and socioeconomic issues. The taxasphere is like a triage team. The "battlefield" is the biosphere, and the "war" is human activities that degrade the biosphere. Sadly, at the point in time that we realize we have documented only a tiny portion of the world's diversity, and want to document more, we find that one of the most rare and declining groups of biologists is the taxasphere. This taxonomic impediment, or critical lack of global taxonomic expertise recognized by Systematics Agenda 2000 and DIVERSITAS, prevents initiation and completion of biodiversity research programs at a critical juncture, where substantial components of global diversity are threatened. The Convention for Biological Diversity mandates that we document the biosphere more fully, and as a consequence, it is necessary to revitalize the taxasphere. One foundation for development of taxonomic expertise and knowledge is the Global Taxonomy Initiative and its 3 structural components: (1) systematic inventory, (2) predictive classifications, and (3) systematic knowledge bases. We argue that inclusion of parasites is critical to the success of the Global Taxonomy Initiative. Predictive databases that integrate ecological and phylogenetic knowledge from the study of parasites are synergistic, adding substantially greater ecological, historical, and biogeographic context for the study of the biosphere than that derived from data on free-living organisms alone.

KEY WORDS: biodiversity, biosphere, Global Taxonomy Initiative, inventory, parasites, phylogeny, survey, taxonomy.

A Biodiversity Perspective

Biodiversity represents a continuum across a variety of scales, encompassing numerical, ecological, and phylogenetic components within a temporal–spatial framework or fabric (Ricklefs, 1987; Barrowclough, 1992; Eldredge, 1992; Hoberg, 1997a). Any definition of biodiversity, then, must parallel this continuum across scales driven by habitats, ecosystems, and communities, genetic diversity in populations and species, genealogy and taxonomy, and history and geography. Different definitions are associated with an array of research programs for survey and inventory that seek different kinds of knowledge. They can (1) focus on local, regional, or global scales; (2) emphasize a specific taxon (e.g., host or parasite) or ecosystem; (3) be oriented in strategic or problem-based perspectives; or (4) be broadly synoptic, such as the approaches linked to the concept of the All Taxa Biodiversity Inventory (ATBI) (Janzen, 1993). Further continua are circumscribed within the sphere of strictly curiosity-based acquisition of knowledge, with eventual affiliation to economic and societal concerns. The scope of the problem may help determine the appropriate approach, but there is little question that the state of the biosphere should be a profound concern for science and society (Ehrlich and Wilson, 1991; Wilson, 1992; Smith et al., 1993; Savage, 1995).

We explore this intricate web to examine the

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critical contributions that emanate from an integrative and comparative approach that emphasizes a parasitological perspective. Parasitology is arguably the most integrative of all biological disciplines. Parasitology provides a powerful conceptual paradigm or landscape that links ecology, systematics, evolution, biogeography, behavior, and an array of biological phenomena from the molecular to the organismal level across the continuum of microparasites to macroparasites and their vertebrate and invertebrate hosts. Information from the study of parasites is synergistic, adding substantially greater ecological, historical, and biogeographic context for the study of the biosphere than information derived from free-living organisms alone.

Valuing Biodiversity

Humans work hard to preserve what they value and replace or ignore what they do not. This is true of furniture and gardens and should be true of biodiversity. This leads to a deceptively simple conclusion: the more species we value, the more species we will preserve. But value is a difficult concept to apply to biodiversity. Equating value with market price does not necessarily lead to sustainable use of resources, although this equation is a necessary component of socioeconomic development, especially in the biodiverse regions of the world. Attempting to define an intrinsic value for biodiversity underscores the fact that different groups of humans have different sets of values about biodiversity and have different degrees of biophilia (Wilson, 1984, 1988, 1992; Takacs, 1996; Brooks, 1998).

Biodiversity is valued in many different ways, not all of them mutually reinforcing (Ehrlich and Wilson, 1991; Pimentel et al., 1997). Concomitant with recognition of value is the necessity to develop a basic or baseline understanding of the components of biodiversity within the framework of a maximally informative system that focuses attention on the following: (1) societal, aesthetic, and intrinsic values (i.e., biophilia); (2) economic benefits and beneficial components (both historical and future); (3) ecosystem services and the value of information; and (4) patterns, processes, and distribution of pathogens and disease (Ehrlich and Wilson, 1991).

An appeal to the intrinsic value of biodiversity, for example, does not necessarily put food in people's stomachs or decrease infant mortality rates, the issues of most immediate importance for many people. Some species have value because they produce direct economic benefit, providing marketable products such as ecotourism and the raw materials for research and breeding stock (sourcing drugs and biocontrol agents from wildlands is familiar in many sectors, but the idea of paying wildlands, or the governments that administer them, for this service is a novel concept). Other species have value because they maintain ecosystem services, such as biodegradation of agricultural wastes and sequestration of carbon. Still other species have value because they provide the recreation—intellectual and physical—that contributes to a happy and adjusted populace. No one looks forward to living in a country congested with forest fire smoke or with oil-coated beaches, but in the absence of ecological alternatives that are also economical, people will choose to feed their families even if it means having to deal with massive local degradation of the biosphere. Using the resources provided by biodiversity or the application of a refined knowledge of the biosphere could provide these ecological and economically sound alternatives. Finally, many species have value because they are essential for the well-being and persistence of other species that have greater direct value.

Although economic and societal issues highlight the necessity to fully define the scope and depth of biodiversity within urban, agricultural, and natural ecosystems, elucidation of faunal structure and processes is also a prerequisite for understanding significant interactions at the interfaces of such systems, including the distribution of pathogens and emergence of disease (e.g., Davis, 1995, 1996; Epstein, 1997, 1999; Hoberg, 1997b; Brooks et al., 2000; Brooks, León-Règagnon, and Pérez-Ponce de León, 2000; Hoberg, Gardner, and Campbell, 1999; Hoberg et al., 1999). Pathogens and parasites have direct implications for human health, agriculture, natural systems, conservation practices, and the global economy through continued introductions and dissemination and our often limited knowledge of mechanisms that control distribution and emergence (Hoberg, 1997b).

Humans are concentrated in the world's most biodiverse regions, where they often live in conditions of poverty, poor education, and poor health. These people currently preserve only those species and ecosystems that enhance the immediate quality of their lives or those of their

children. This means, to most of them, domestic species and the habitats they occupy rather than wildlands. We are led to a deceptively straightforward proposition: link economic development to the preservation of wildlands and the species they contain, encouraging people to understand that the plant and animal species in the wildlands are as valuable as the more familiar domestic species. In this way, some wildlands may survive, not as the agroscape, but as another kind of cropland interdigitated with the agroscape. This proposition implies developing the economic and social potential of species living in wildlands, thus reducing demand for economic development of wildlands, into still more impoverished agroscape, which partly sustains yet more often starves people. Such a proposition assumes that at least some societies will conserve biodiversity on some portion of their landscape, if the wildlands generate intellectual and economic benefits that pay for their maintenance and contribute to national economic growth and sustainability. The preservation of biodiversity is thus driven, at present, more by social and economic development than technical expertise (Janzen, 1992; Brooks, 1998).

People interested in economic and social development of conserved wildlands can benefit from forming partnerships with the scientific community. Such partnerships are required to meld what scientists have long known and are still discovering through basic research with the pragmatic efforts of developing the wildlands as a third kind of major land use, alongside the urban and agricultural landscape. Being able to determine the multiple uses of species and their combinations requires technical and scientific expertise and social will (Parma, 1998).

If preservation is to be true and long-lasting, biodiversity conservation can occur only through nondestructive use of that biodiversity by a wide array of social sectors. Effective conservation efforts will simultaneously encompass biodiversity development and conservation projects (Soulé, 1991). This occurs by designating areas for wildland status, finding out what is in them, and putting that biodiversity to work. In this regard, a critical element of the scientific community is the *taxasphere* (Janzen, 1993), the global population of taxonomists and systematists.

These issues highlight the critical importance and rationale for biodiversity survey and inventory. Although inventory work is fundamentally important, we must remember that it is only a means to an end. Names attached to species revealed through intensive field and laboratory investigations must represent a significant amount of natural history, especially ecological, information for the stakeholders in national socioeconomic development to be able to assess the value of each species. The taxasphere may be likened to a triage team. The "battlefield" is the biosphere, and the "war" is human activities that tend to degrade the biosphere. In this war, every species is affected to a greater or lesser extent. Some are attacked directly through overexploitation and others indirectly through neglect. The triage teams survey parts of the battlefield as completely as possible, looking for "wounded" participants. They must be able to recognize all possible participants and the degree to which each has been affected (e.g., critical habitat requirements that are gone or going). The teams must then pass that information on to the decision makers, who are responsible for the optimal deployment of resources to save the maximum number of participants possible. Taxonomists communicate such information most efficiently through predictive classifications and electronic management of information.

Valuing Taxonomy

We already know much—and are learning more each day—about the importance of the documented portions of the biosphere. However, we have not documented, and thus do not understand, more than a fraction of that diversity, with only 1.7 million of an estimated 13 million to 14 million existing species currently described (Hawksworth, 1995). Consequently, we often have no idea what we might be losing and have only incomplete information on how to preserve what remains. Faced with our ignorance and gaps in knowledge, biologists react in a way that seems paradoxical. They often advocate extreme caution in development projects, simply because our ignorance may lead us to make mistakes and lose habitat and diversity both in the short- and long-term. At the same time, biologists understand that caution cannot impose stasis or inaction. We cannot be satisfied with slowing the rate at which species are lost or habitat is destroyed, because extinction is an irreversible process. We can never bring a species back once it is lost, and its potential to play

a role in the survival of our species is gone forever. Moreover, each species that becomes extinct may represent an irreversible loss of socioeconomic potential and may restrict our survival options for the future. Each species lost represents an irreversible loss of evolutionary potential, the very potential that has been the source of biotic recovery from past global ecological perturbations and environmental disasters (Jablonski, 1991). Many biologists, therefore, advocate immediate action to document the world's biodiversity. Sadly, at the point in time that we have realized that we have documented only a tiny portion of the world's diversity and want to document more, we find that one of the rarest and fastest declining groups of biologists is the taxasphere.

The fundamental units of biodiversity are genealogical information systems called *species,* which store and transmit information that leads to the emergence of ecosystems with their complex interactions. Who is trained to find and distinguish among those units? Taxonomists. Where do we find that information? In the descriptions, surveys, and revisions of taxonomists. The predictable parts of biological systems are the stable biological elements, both form and function, autecological and synecological, that have persisted through evolutionary time (Brooks, 1985a; Brooks and McLennan, 1991, 1993a; Brooks et al., 1995). This predictive power of taxonomy is embodied in the phylogenetic classifications of taxonomists (Simpson and Cracraft, 1995). The taxasphere includes some of the best-trained bioprospectors in the world, who are often highly skilled at finding particular species. Taxonomists are also versatile and opportunistic in the field because of their ability to recognize novelty. Their ability to determine evolutionary relationships and add value by making predictions based on those relationships can minimize the time and cost in research and development and planning and prioritization (Brooks et al., 1992).

Parasites in Biodiversity and Conservation Biology

In the realm of conservation biology, parasites have dual and conflicting significances. Pathogenic parasites can represent threats to the success of programs for management and recovery of threatened or endangered species (Dobson and May, 1986b; Scott, 1988; Lyles and Dobson, 1993; Holmes, 1996). Alternatively, parasites can control host populations, and they can play a central role in maintenance of genetic diversity and structuring of vertebrate and invertebrate communities (Windsor, 1995, 1996); in this latter role, parasites are significant and vital components of the biosphere. Additionally, it has been suggested that under special conditions (such as on islands) introduction of parasites and pathogens may be a viable method of controlling introduced mammals (Dobson, 1988).

The significance of biodiversity surveys and inventories in a phylogenetic–ecological context is apparent in documenting the abundance and species composition of faunas in protected host species and habitats. For example, in endangered Attwater's prairie chickens (*Tympanchus cupido attwateri*), the potential for interspecific interactions and sharing of pathogenic parasites with related species of grouse provided the rationale for comparative baseline studies of parasite diversity (Peterson, 1996). Definition of the parasite fauna, including recognition of new cryptic species, in Holarctic ruminants was necessary to identify the potential for impacts from circulation of endemic and exotic parasites among cervids and bovids in North America (Hoberg, Kocan, and Rickard, 2000). For wild ungulates, translocation and either introduction of parasites or exposure to novel pathogens remain major considerations in management decisions (Lankester and Fong, 1989; Samuel et al., 1992; Woodford and Rossiter, 1994; Hoberg, 1997b).

Parasites must be regarded as integral components of biodiversity; thus, there should be concerns about the ramifications of extinction both locally and globally (Wilson, 1984; Rósza, 1992; Windsor, 1995; Durden and Keirans, 1996). Coming full circle, the importance of accurate documentation for biodiversity and the world's parasite faunas is based on the intrinsic and extrinsic importance of parasites in healthy ecosystems, as agents of human disease, and at the nexi of natural and domestic, terrestrial, aquatic, and marine environments.

Parasites as Contemporary Ecological Indicators. At a higher level than the communities of parasites themselves, we recall that parasites track broadly and predictably through ecosystems. Parasites inform us of a myriad of interesting things about host ecology, behavior, and trophic interactions (Hoberg, 1996; Marcog-

liese, 1995; Marcogliese and Cone, 1997). Complex life cycles are integrated within intricate food webs, so parasites can be valuable indicators of trophic ecology, structure of food webs, food preferences, and foraging mode of hosts (Bartoli, 1989; Williams et al., 1992; Hoberg, 1996; Marcogliese and Cone, 1997). Within this ecological–trophic context, parasites can tell us the following: (1) trophic positions in food webs (what hosts eat and what eats them); (2) use of and time spent in different microhabitats (e.g., even though *Terrapene carolina* is mainly a terrestrial turtle, it hosts the digenean *Telorchis robustus,* which uses tadpoles as second intermediate hosts); (3) whether hosts are picking up parasites via host switching, and if so, which hosts might be in potential competition (e.g., guild associations were recognized in the Sea of Okhotsk based on examining parasites (Belogurov, 1966)); (4) whether any host harbors parasites that are likely to cause disease problems; (5) whether the host changes diet during its lifetime, including seasonally or regionally defined changes in food habits or prey availability (Bush, 1990; Hoberg, 1996); and (6) which hosts are residents and which are colonizers in the community. Because such a wide range of information can be gleaned from relatively little effort, parasites should be highly useful in all biodiversity studies. Additional special cases for the application of parasitological data are related to their use as contemporary biogeographic indicators. Analysis of parasite biogeography has been a powerful approach for identification of stocks or populations in fisheries management (Williams et al., 1992) and among marine mammals (Dailey and Vogelbein, 1991; Balbuena and Raga, 1994; Balbuena et al., 1995).

We can maximize the use of this information if we begin to think of parasites as biodiversity probes *par excellence* and as libraries of natural and geological history (Brooks et al., 1992; Gardner and Campbell, 1992; Hoberg, 1996). Parasites are admirably suited to augment the development of conservation strategies through the recognition of regions of critical diversity and evolutionary significance (Gardner and Campbell, 1992; Hoberg, 1997a).

The predictive power of parasitology in a phylogenetic context becomes increasingly important when attempts are made to elucidate impacts from natural and anthropogenic perturbations of faunas and ecosystems. In marine systems, climatological forcing, such as that linked to the El Niño–Southern Oscillation or to cyclical changes in atmospheric circulation, dramatically influences patterns of oceanic upwelling and water masses, which are reflected in food web structure and ultimately in parasite faunas. In such situations, parasites should be well suited to tracking variation in trophic dynamics and host distributions on the global scale (Hoberg, 1996). Knowledge of the evolution of a host–parasite assemblage can provide direct estimates of the history of ecological associations and can indicate the continuity of trophic assemblages through time.

Parasites as Historical Indicators. Manter (1966) made the most eloquent statement about the significance of parasites for understanding evolutionary and ecological phenomena:

> Parasites . . . furnish information about present-day habits and ecology of their individual hosts. These same parasites also hold promise of telling us something about host and geographical connections of long ago. They are simultaneously the products of an immediate environment and of a long ancestry reflecting associations of millions of years. The messages they carry are thus always bilingual and usually garbled. As our knowledge grows, studies based on adequate collections, correctly classified and correlated with knowledge of the hosts and life cycles involved should lead to a deciphering of the message now so obscure. Eventually there may be enough pieces to form a meaningful language which could be called parascript—the language of parasites which tells of themselves and their hosts both of today and yesteryear. (Manter, 1966)

Phylogenetic systematics provided the Rosetta stone for what are now called parascript studies (Brooks and McLennan, 1993a). In the past 2 decades, since formalization of the parascript concept (Brooks, 1977), the number of such studies has increased dramatically (see reviews in Brooks and McLennan, 1993a; Hoberg, 1997a). Today there is virtually no area of modern comparative evolutionary biology and historical ecology that has not been enriched by at least one parascript study.

The concept of parascript was based on the contention by Manter (1966) that parasites are powerful biological indicators of recent and ancient ecological associations and geographic distributions. Parasites tell stories about themselves and their hosts that involve evolutionary emergence of complex ecological associations throughout immense periods. These ideas dra-

matically influenced the development of a research program for comparative evolutionary biology of parasites (Brooks and McLennan, 1993a). Extending from the seminal study by Brooks (1977), this program has led to elaboration of methods for analyzing cospeciation, extinction, dispersal, and host switching in the evolution of parasite biotas (Brooks, 1979, 1981, 1990; Hoberg, Brooks, and Siegel-Causey, 1997), with the emergence of historical ecology (Brooks, 1985a; Brooks and McLennan, 1991) as the foundation for parascript investigations (Brooks and McLennan, 1993a, 1993b, 1993c). Because their geographic distributions are limited to those areas in which all obligate hosts are sympatric and synchronic, parasites are excellent systems for historical biogeographic studies. Thus, parasites, particularly those with complex life cycles, provide the linkage for examining the interaction of coevolution, colonization, and extinction on faunal structures and ecological continuity across deep temporal and broad geographic scales (Hoberg, 1997a; Hoberg, Gardner, and Campbell, 1999; Hoberg, Jones, and Bray, 1999).

The significance of historical reconstruction for current approaches in the assessment of biodiversity resides in the concept of the past as the key to the present (Hoberg, 1997a). Historical reconstruction allows identification of important centers for diversification (ancestral areas) and promotes a predictive framework to assess the importance of specific habitats, geographic regions, and biotas and recognition of areas of critical genealogical and ecological diversity. These are the realms of historical ecology (Brooks, 1985a; Brooks and McLennan, 1991) and historical biogeography. Although we have much to learn about the biosphere, historical studies of helminth parasite systems in piscine, amphibian, mammalian, and avian hosts have contributed context for understanding faunal structure across terrestrial (Platt, 1984; Gardner and Campbell, 1992; Hoberg and Lichtenfels, 1994), aquatic (Brooks et al., 1981; Klassen and Beverley-Burton, 1987, 1988; Kritsky et al., 1993), and marine environments (Klassen, 1992; Hoberg, 1995; Hoberg et al., 1998) (further reviewed in Brooks and McLennan, 1993a; Hoberg, 1997a). A historical ecological context, in conjunction with developing understanding of contemporary systems, is the basis for using parasites to illuminate local, regional, and global ecological perturbations.

Parasites as Mine Canaries Through Ecological and Evolutionary Time. Parasites track broadly and predictably through ecosystems, highlighting major trophic interactions among hosts that occupy multiple niches. As such, parasites may be sensitive indicators of subtle changes within ecosystems. This is especially true for parasites with heteroxenous life cycles. Severe reduction or disappearance of a population of only 1 of several obligate hosts will cause the parasite to disappear. Environmental pollution of benthic invertebrate faunas has resulted in elimination of many digeneans in fishes in some localities (Caballero y Rodriguez et al., 1992; Overstreet, 1997; Overstreet et al., 1996). Conversely, increased and detrimental levels of parasitism in molluscan intermediate hosts may result from changes in behavioral patterns and population density of seabirds, such as larids, that concentrate near areas of human activity in coastal zones (Bustnes and Galaktionov, 1999).

At the extreme, parasite extinction may precede host extinction, i.e., the sudden loss of a particular species of parasite in a given vertebrate host may result from degradation of faunal structure or from changes in host population density (Dogiel, 1961; Hoberg and McGee, 1982; Bush and Kennedy, 1994). As some hosts go extinct, some parasites will go extinct as well. Alternatively, some surviving parasites may be brought into contact with novel hosts as a result of ecological release. This new contact may produce disease, which is maladaptive for host and parasite, but given the alternative of extinction, from the parasite's standpoint it may represent a viable "strategy" to avoid extinction. From the host's standpoint, the cost of exposure to novel pathogens may be offset by the benefit of surviving a major environmental disaster. In the longer-term, coevolutionary dynamics might ameliorate, if not eliminate, the negative impacts of these once novel host–parasite associations, although we understand that reduction in virulence may not be correlated with length of association of pathogen and host (Ewald, 1995).

Thus, parasites can serve as indicators of ecosystem integrity and can be used to measure contemporary environmental perturbations. Nat-

ural and human alterations of ecosystems may be reflected in increases or decreases in abundance of a certain spectrum of the parasite fauna (Marcogliese, 1995). A particularly elegant study showing this was a documentation of the impact of acidification in riparian habitats on parasites of eels (*Anguilla rostrata*) in Nova Scotia (Cone et al., 1993; Marcogliese and Cone, 1996).

Aside from the effects of pollution, we predict substantial impacts on host–parasite systems and on changes in the distribution and emergence of pathogens and parasites from global climate change and global warming (Dobson and Carper, 1992; Epstein, 1997, 1999). Contemporary changes in distribution caused by global climate change have already been documented for anisakine nematodes in seals and fishes (Marcogliese et al., 1996). Disease outbreaks of elaphostrongyline nematodes in reindeer have been correlated with variation in summer temperatures (Handeland and Slettbakk, 1994). Historical studies of parasites can illuminate the influence of past climate variation on the distribution and diversification of parasites and their hosts (Hoberg, 1986, 1992, 1995).

Continued surveys and inventories of parasites become important components in documenting the impact of environmental change. As indicated by Marcogliese and Price (1997), "Parasitism is simply a reflection of the natural state of ecosystems, and healthy populations of organisms will play host to healthy populations of parasites."

Parasites and Assessing the Risk of Emerging Diseases. Parasites may act as agents of population control by causing acute or chronic disease in hosts (Scott, 1988; Gulland, 1995). Comprehensive inventories permit us to assess risk, to make predictions (for wildlife and game, agriculture and livestock, or public health), and to recognize endemic and introduced faunal elements (Hoberg, 1997b; Hoberg et al., 1999; Hoberg, Kocan, and Rickard, 2000). We are interested in interfaces between natural, agricultural, and urban ecosystems and the barriers that inhibit or the paths that promote introduction and dissemination of pathogens and parasites.

Inventories within a historical–phylogenetic context focus the range of questions to be examined. We can consider whether the same or related parasites occur only in related hosts or whether they occur in distantly related hosts with similar ecological habits. This is an ecological and evolutionary question with implications for emerging diseases. On the ecological side, we will find out which parasites are limited by host or geographic associations and which parasites are likely to disperse or colonize. On the evolutionary side, we can ask which groups have persisted through major environmental changes, either because their hosts survived or because the parasites successfully switched to alternative hosts. We might predict increasing host switching as hosts or host groups go extinct, i.e., the extinction of certain hosts might accelerate the emergence of pathogens and diseases rather than simply eliminating disease organisms.

Mechanisms for emergence have been well documented and generally are linked in some way to the breakdown of isolating barriers (Rausch, 1972; Dobson and May, 1986a, 1986b; Hoberg, 1997b). Factors that contribute to disease emergence include the following: (1) translocation, introduction, and dissemination of pathogens; (2) faunal disruption and ecological release (new hosts or new ecological situations); (3) increasing host population density (stress and reduced abilities for adaptation); and (4) amplification of parasite populations linked to environmental change, such as global warming. When dealing with complex systems, however, cause and effect are often difficult to distinguish. Limb deformities and mortality in anurans have been linked to infections by a species of *Ribeiroia,* a psilostomid digenean, which may be indicative of environmental pollution and its effect on populations of the snail intermediate hosts (Johnson et al., 1999). The ongoing reduction in anuran populations may further indicate the pervasive nature of emergence and the continued translocation and introduction of wildlife parasites on a global scale (Morell, 1999).

Introduction of parasites and pathogens with either wild or domesticated hosts is a major source of disease emergence. Establishment of introduced parasites has been documented for the following: (1) nematode faunas in ruminants across the Holarctic (Hoberg and Lichtenfels, 1994; Hoberg et al., 1999; Hoberg, Kocan, and Rickard, 2000); (2) parasites in freshwater and marine fishes (Kennedy, 1993; Scholz and Cappellaro, 1993; Barse and Secor, 1999); and (3) helminths in some avian hosts such as ratites

(Hoberg, Lloyd, and Omar, 1995). Many local parasite faunas are now mosaics of endemic and introduced species. A principal role for taxonomic inventories in such situations is to provide baseline information on the patterns of distribution for hosts, parasites, and pathogens as the foundation for prediction and prevention. Knowledge of the diversity and distribution of pathogens and parasites is important in limiting economic, societal, and biotic impacts and liability in management of endemic or exotic organisms (Hoberg, 1997b).

International Initiatives in Systematics and Biodiversity: Getting Parasitology Involved

The Convention on Biological Diversity (CBD) (Glowka et al., 1994) designated ecosystems management and sustainable development as the fundamental organizing principles for managing global biodiversity. Biologists and managers quickly realized that the current inventory of the world's species was far too limited to implement the mandate properly and that a critical shortage of trained taxonomists contributed directly to the problem. The United Nations Environment Program in biodiversity, called DIVERSITAS, coined the term *the taxonomic impediment* to refer to this critical lack of global taxonomic expertise, which prevents initiation and completion of biodiversity research programs (SA2000, 1994; Hoagland, 1996; Blackmore, 1996; PCAST, 1998). In North America, this concern led to Systematics Agenda 2000 (SA2000), an intensive professional inventory of the value of taxonomic expertise to this planet, and a set of recommendations for revitalizing the taxasphere and justifying the allocation of resources necessary to carry out such a revitalization (SA2000, 1994). In 1998, the Conference of the Parties to the CBD endorsed a Global Taxonomy Initiative (GTI) to improve taxonomic knowledge and capacity to further country needs and activities for the conservation, sustainable use, and equitable sharing of benefits and knowledge of biodiversity (GTI, 1999; http://research.amnh.org/biodiversity/acrobat/gti2.pdf). It appears that the solution to removing the taxonomic impediment in biodiversity planning is the revitalization of the taxasphere, and the rationale for revitalizing the taxasphere is the potential of taxonomic contributions for managing the biodiversity crisis. A foundation for development of taxonomic expertise and knowledge is the GTI and its 3 components: (1) systematic inventory, (2) predictive classifications, and (3) systematic knowledge bases, including collections.

GTI Component 1: Systematic Inventory—Discovering and Naming the World's Species. Assessments by DIVERSITAS indicate that there are no more than 2,000 full-time professional taxonomists and perhaps an additional 5,000 people with some degree of taxonomic expertise on the planet. Furthermore, that population has a median age of 55 years. Concurrently, less than 10% of the terrestrial species and perhaps 1% of the world's marine species have been discovered and named. These factors act synergistically as the foundation of the taxonomic impediment (SA2000, 1994). Biodiversity inventories, as envisioned by the CBD, are biodiversity development and conservation projects, a means for restoring global taxonomic capacity, and opportunities to study the health, reproductive, and nutritional requirements and the ecology and evolution of a large number of wild species in natural, yet protected, environments.

What information should inventories provide? For each species, the following should be provided: (1) what is it (and how to distinguish it from others); (2) where does it occur; (3) what is its natural history; and (4) how do you get it when desired?

To what purposes should this information be put? (1) Monitoring global environmental health; (2) promoting socioeconomic development through sustainable use and equitable sharing of benefits and knowledge derived from diverse biological resources, including forestry, fisheries, and some components of agriculture; (3) developing of new products and ecotourism; and (4) providing risk assessment about the impact of introduced species and the source and impact of emergent diseases. Potential user groups for the information gathered include ecotourism, agricultural, pharmaceutical, and biotechnology prospecting companies; educational and scientific institutions; human, veterinary, and agricultural health experts; environmental monitoring and restoration programs; economists; and development agencies.

What general criteria do we follow in choosing particular inventory projects? They should (1) have high public visibility and approval in-

ternationally and locally; (2) be international in scope; (3) have high scientific value in both basic and applied terms; and (4) encourage group cohesion and cooperation, leading to the engagement of as many stakeholders as possible.

The CBD has mandated that each country embark on some form of national biodiversity inventory. National socioeconomic planners will determine the form of such an inventory. Once such a decision has been made, an immediate concern will be coordinating efforts. Every country in the world is now a debtor nation with respect to taxonomic expertise. As mentioned herein, the taxasphere sees the removal of the taxonomic impediment as an opportunity for the survival of the taxasphere and the biosphere. But because the taxasphere today consists of a relatively small, generally poorly funded and globally dispersed population of scientists, any national inventory project will require a multinational effort. Furthermore, most members of the taxasphere work in academia or museums, which represents an additional layer of cultural distinction. Academic and museum naturalists have a long history of self-motivated and self-directed, essentially solitary pursuit of knowledge. Specialists on the same groups of organisms may see each other as professional competitors rather than collaborators. Encouraging such people, representing different institutions and different countries, each with different personal career agendas, to collaborate is difficult but not impossible (Janzen and Hallwachs, 1994; Hoberg, Gardner, and Campbell, 1997). A 1993 National Science Foundation–sponsored conference in Philadelphia, Pennsylvania, U.S.A., brought leading taxonomists together to consider the feasibility of their cooperating to document those species useful to humans before they become extinct and to stave off the loss of a science of specialists who could identify them and learn about their natural histories. Faced with the immediacy of the crisis, the taxonomists present were able to cooperate strategically, even though there were and still are differences of opinion about tactics, primarily in the realm of inventory projects (Janzen and Hallwachs, 1994). Inventories can represent synoptic examinations of complex ecosystems or well-circumscribed, problem-driven projects. Synoptic examinations include the concept of the ATBI, documenting all species in a large conserved wildland site (Janzen, 1993; Janzen et al., 1993).

The ATBI concept was originally conceived to serve 2 functions. First, the most biodiverse terrestrial ecosystems in the world occur in tropical developing countries. Great diversity, many unknown species, and generally untapped biodiversity resources characterize tropical ecosystems. In such situations, recognizing that biodiversity programs may be simultaneously biodiversity development and conservation projects is critical. They can create a mechanism to preserve wildlands, build scientific infrastructure, and promote sustainable use of environmental resources. Socioeconomic development stemming from an ATBI is achieved by giving the neighbors of a conservation area a stake in preserving the local diversity; the more species that can be shown to be valuable, the more such opportunities exist, and the more species will be conserved.

Second, each site where an ATBI is carried out becomes a gigantic mine canary, where the effects of global environmental change could be monitored across significant numbers of species and large sectors of integrated ecosystems, giving us a true picture of the overall large-scale effects of such phenomena as global warming, biotic invasions, and habitat perturbation (Janzen, 1996, 1997; Janzen and Hallwachs, 1994). The information generated by an ATBI could be valuable for conservationists and land use planners, where conserved wildland choice is critical in the following ways: (1) Observing that a conserved wildland can be useful and used, national policy makers will be able to consider conserving wildland as an appropriate form of land use, on a par with agricultural and urban landscapes. Conservationists and economic development programs will become partners rather than adversaries. (2) An ATBI will aid conservationists who make site choices elsewhere, because it will generate a complete picture of a biodiverse landscape, a "known universe," by which biodiversity and ecosystem sampling schemes can be calibrated. (3) The data from an ATBI may enable us to answer difficult conservation questions based on correlations between the diversity of 2 or more taxa in a site or habitat array. (4) By accomplishing a project with high social approval, the taxasphere and biodiversity managers will feel more confident in making new partnerships with conservationists. (5) An ATBI will be a campus for people representing many stakeholders in biodiversity, many of those involved

directly in conservation site selection and biodiversity development in other places.

Inventories conducted to facilitate the use of wildland biodiversity by societies simultaneously benefit the taxasphere and those who desperately need food, shelter, education, and health care. The ATBI approach is not the only model for undertaking inventories, and many members of the taxasphere do not believe it represents the best use of limited taxonomic resources. The major selling points of an ATBI are the immediate socioeconomic benefits to local residents of a conserved wildland and the exciting "moonshot" nature of such a large-scale project. In addition, although there is currently a lack of funds for any ATBI, such a project could become important in the future.

Some doubt, given the state of the taxasphere, that it is in our best interests to concentrate a disproportionate amount of effort on a single site. They argue that the best way to revitalize the taxasphere globally is to initiate multiple inventory projects simultaneously throughout the world using available expertise. This emphasizes the concept of working locally and thinking globally. Furthermore, given that the goal of an ATBI may be national socioeconomic development, how can the taxasphere, especially through a GTI, give preference to one country's socioeconomic aspirations over another's? Wouldn't it be preferable to give individual countries a means of prioritizing their limited human and economic resources to make national inventories of priority taxa?

Critics of the more disseminated inventory approach suggest that it tends to reinforce taxonomic expertise in taxa for which there are already many taxonomists, and risks excluding interested taxonomists who do not happen to study one of the priority groups in one of the priority places. Advocates of the ATBI concept argue that the choice of which country to prefer will simply be a first-come, first-served phenomenon and that the expertise generated from the first ATBI will permit the second and succeeding ATBIs to be done faster and more cost-effectively. They also argue that, in contrast to an ATBI, which is focused within a single country, targeted inventories of selected taxa over wide geographic ranges will involve international and intranational planning and cooperation, something that is not guaranteed to happen in all parts of the world at any given time.

Clearly, there are good points made by people of goodwill on both sides of this issue. All agree that taxonomic inventories are fundamental to the future of biodiversity preservation and development (GTI, 1999), and that if we had unlimited funds, complete inventories of all targeted areas throughout the world would be ideal. This is what we refer to as strategic agreement. The realities of the situation, however, are that there will be only limited amounts of money available for national inventories and for revitalizing the taxasphere, even if the taxonomic impediment is regarded as a critical national priority or imperative such as that recently outlined for the United States (PCAST, 1998). We urge all parties to listen to each other, learn from each other, work together as much as possible, and take proactive stances that are environmentally, scientifically, and politically relevant.

PARASITES IN BIODIVERSITY SURVEYS AND INVENTORIES: It is clear from the variety of research programs supported by parasitological studies that parasites represent significant components of global biodiversity. Continued survey and inventory of the world's parasite faunas remain requisite for understanding basic issues in evolutionary biology, ecology, and biogeography. Documentation of parasite biodiversity is significant in an "applied" sense for elucidating solutions to ecological problems, examining emergence and re-emergence of pathogens, understanding interactions at the interface of ecosystems, and recognizing the impacts of global change. We will examine later 2 models for pursuing studies of parasite diversity in tropical and high-latitude boreal and arctic systems. Different strategies for acquiring biodiversity knowledge are dictated by prevailing social and economic situations.

Those who argue for inventories that focus on priority taxa suggest the following selection criteria: taxa should (1) be intrinsically important to humans, such as insect groups known to include important pollinators, biocontrol agents, or disease vectors; (2) be intrinsically important to ecosystems that humans want to preserve; (3) provide efficient means of learning something of importance; (4) be geographically widespread; and (5) provide opportunities for international networking of professionals, collaborative research, and training. In our opinion, it is easy to

justify the inclusion of parasites in any inventory project under all these guidelines.

Taxa should be intrinsically important to humans: Parasites are agents of disease in humans, livestock, and wildlife, with attendant socioeconomic significance. Parasites are significant components for assessing the risk of loss of biocontainment by introduced species, whether because of parasites of introduced species moving into the agricultural landscape or wildlands and switching to native hosts or because of parasites of native species moving out of the agricultural landscape or wildlands and infecting introduced, economically important host species. A special case involves the possibility of local residents and tourists sharing parasites and parasitic diseases between themselves and between humans and nonhuman hosts. Some parasite species may provide revenue as model systems for pharmaceutical companies or as biocontrol agents. Additionally, we must understand parasite biodiversity within the context of global change (Dobson and Carper, 1992; Hoberg, 1997b; Brooks et al., 2000; Brooks, León-Règagnon, and G. Pérez-Ponce de León, 2000; Hoberg, Kocan, and Rickard, 2000).

Taxa should be intrinsically important to ecosystems that humans want to preserve: Parasites are significant regulators of host populations (Scott, 1988; Gulland, 1995) and are potent agents that maintain ecosystems' integrity and stability (Minchella and Scott, 1991; Dobson and Hudson, 1986; Hudson et al., 1998). Complex feedback loops that involve parasites, herbivores, and habitat structure in ruminant grazing systems further indicate the significance of parasites as determinants of community structure (Grenfell, 1992). Parasites can also be important mediators of host behavior (Holmes and Bethel, 1972). Introduced parasites may have unpredictable and deleterious impacts on native species of hosts (Dobson and May, 1986a, 1986b; Woodford and Rossiter, 1994; Vitousek et al., 1996). It is, therefore, important to be able to quickly distinguish native from introduced parasite species (Hoberg, 1997b; Brooks, León-Règagnon, and Pérez-Ponce de León, 2000; Hoberg et al., 1999; Hoberg, Kocan, and Rickard, 2000).

Taxa should provide efficient means of learning something of importance: Parasites, especially those having complex life cycles involving more than 1 obligate host, are indicators of stable trophic structure in ecosystems (Marcogliese and Cone, 1997). This is because all the biotic components necessary for completion of the life cycle must co-occur regularly to maintain any given parasite species. Knowing the complement of parasite species inhabiting any given host thus provides a means of rapid assessment of the breadth and form of trophic interactions of host species.

Taxa should be geographically widespread: Many parasite taxa are widespread geographically. At the same time, they are highly localized with respect to infecting particular hosts, which themselves may be the focus of particular inventory activities.

Taxa should provide opportunity for international networking of professionals, collaborative research, and training: Parasite systematics is in serious trouble worldwide. Laboratory closures in the United Kingdom and elsewhere have eroded the infrastructure for taxonomy and systematics at a critical time. New survey opportunities and recognition of the importance of parasites may stimulate international collaboration and revitalization.

Parasites, therefore, fit a set of extrinsic criteria, indicating the importance of their inclusion as basic elements of surveys and inventories. Parasites are critically important as (1) ecological–trophic indicators (Marcogliese and Cone, 1997; Overstreet, 1997); (2) historical indicators of phylogeny, ecology, and biogeography (Brooks, 1985a; Brooks and McLennan, 1993a, and references therein; Pérez-Ponce de León, 1997; Pérez-Ponce de León, León-Règagnon, and Garcia-Prieto, 1997; Brooks et al., 2000; Brooks, León-Règagnon, and Pérez-Ponce de León, 2000); (3) contemporary and historical probes for biodiversity research (Brooks et al., 1992; Brooks et al., 2000; Brooks, León-Règagnon, and Pérez-Ponce de León, 2000; Gardner and Campbell, 1992; Hoberg, 1996, 1997a, and references therein; Pérez-Ponce de León, 1997; Pérez-Ponce de León, León-Règagnon, and Garcia-Prieto, 1997); and (4) model systems to explore theoretical issues and generalities in evolutionary biology, ecosystem and community structure, biogeography, adaptation and radiation, modes of speciation, and life history within a comparative framework (Price, 1980, 1986;

Esch, Bush, and Aho, 1990; Esch, Shostak et al., 1990; Brooks and McLennan, 1991, 1993a, 1993b, 1993c; Brooks et al., 2000; Brooks, León-Règagnon, and Pérez-Ponce de León, 2000; Ewald, 1995; Huxham et al., 1995; Poulin, 1995a, 1995b, 1997a, 1997b; Pérez-Ponce de León, 1997; Pérez-Ponce de León, León-Règagnon, and Garcia-Prieto, 1997). Substantial contributions by parasitological research to biodiversity inventories extend from the accretion of novel information from standard surveys established during the past 200 years to sophisticated research programs for systematics, ecology, biogeography, and evolutionary biology based on organismal and molecular approaches.

The ultimate value and comparability of these often disparate areas of research will be increased by standardized protocols and methods of collection, documentation, and reporting of information (Walther et al., 1995; Bush et al., 1997; Doster and Goater, 1997; Clayton and Walther, 1997; Hoberg, Kocan, and Rickard, 2000). Standardization will also emphasize the interdisciplinary linkages of parasitology within the biological sciences.

TROPICAL AND ARCTIC PARASITOLOGY—THE POWER OF INTEGRATED PARASITOLOGICAL RESEARCH:

The ATBI Model in the Tropics: The Area de Conservación Guanacaste (ACG) in northwestern Costa Rica (http://acguanacaste.ac.cr) is a biodiversity development area. Its purpose is to provide information about preserving species living within Costa Rica through sustainable use of at least some of them, while simultaneously representing a significant portion of national preserved wildlands (Janzen, 1992, 1993; Janzen et al., 1993). The ACG provides economic opportunity, involving local employment and training through various biodiversity-related activities, including taxonomic inventories. Equally, national and international socioeconomic development results from the findings of such inventories. The valuation of sustained use of biodiversity requires that the best technical and scientific knowledge be brought to bear on such inventories to make the findings of the inventory itself useful to as wide a range of stakeholders as possible. Moreover, local taxonomists must be trained for Costa Rica to be as self-sustaining as possible with respect to taxonomic expertise. An inventory of eukaryotic parasites inhabiting the 940 species of vertebrates living in the ACG began in 1997 (Brooks et al., 1999; Desser, 1997; Hoberg et al., 1998; Marques et al., 1997; Monks et al., 1997; Pérez-Ponce de León et al., 1998).

An ATBI assesses biotic resources synoptically and exhaustively. Strategically focused inventories, such as the one discussed next, are better for addressing specific environmental issues.

The Arctic Consortium Model: Large mammals, particularly ruminants, including muskoxen (*Ovibos moschatus*), caribou, and reindeer (*Rangifer tarandus*), represent keystone species for subsistence and maintenance of remote communities across the Holarctic. Parasite faunas, largely nematodes, of these ruminants had been considered to be well known. Since 1995, however, a new genus and 2 new species have been described from the central Canadian Arctic (Hoberg, Lloyd, and Omar, 1995; Hoberg et al., 1999). These projects highlight the importance of molecular data in the recognition of cryptic species (Anderson et al., 1998) but also demonstrate our poor level of knowledge about these systems, which is insufficient to understand the ecological control mechanisms for dissemination and host range. Additionally, poor documentation of faunal diversity, host distribution, and geographic range hinders development of predictions about impacts of global climate change and management practices, such as translocation and linkages to emergence of parasites (Hoberg, Kocan, and Rickard, 2000).

An initial step in the process of defining the fauna involved consolidating information in the form of comprehensive checklists and inventory for parasites in Holarctic Bovidae and Cervidae (Neilsen and Neiland, 1974; Hoberg, Kocan, and Rickard, 2000). These form the basis for strategic survey and inventory or targeted projects to examine the distribution of parasites and to assess the potential for parasitic disease emergence. Comprehensive collections from specific hosts or geographic localities during the past 20 to 30 years, such as those for Dall's sheep (*Ovis dalli*) (Neilsen and Neiland, 1974) and other northern ruminants, are baselines for comparison with contemporary surveys to document alteration in parasite distribution and abundance on local and regional scales. In this process, the utility of systematics and historical biogeogra-

phy to understand faunal structure is evident (Hoberg et al., 1999).

Studies of parasite diversity among large ruminants in the Arctic are consequential, because environmental perturbations attributable to global warming may be pronounced in that region. Synoptic data for parasite distribution in conjunction with studies of the intricacies of parasite biology contribute to the development of model systems to predict the biotic responses to ameliorating climatic conditions in the Arctic (Kutz et al., 2000).

The need to understand even relatively simple Arctic systems has led to development of a partnership for discovery that seeks to build a synergistic, complementary, and interdisciplinary linkage for parasitology, wildlife biology, and the dynamics of wildlife diseases with systematics and biogeography. An informal consortium links the University of Saskatchewan, the Department of Wildlife, Resources and Economic Development (Government of the Northwest Territories), the University of Alaska, and the Biosystematics and National Parasite Collection Unit of the Agricultural Research Service, U.S. Department of Agriculture, in studies of Arctic parasite biodiversity. Success of this approach depends on substantial input and approval from local communities in the North. The consortium is a powerful model for involvement and collaboration among academic scientists, government agencies, and native Inuit in the Arctic and also represents a general means of integrating the efforts of ecologists and systematists to treat a specific problem.

GTI Component 2: Predictive Classifications—What's in a Name? A crucial element in preserving biodiversity within the context of the CBD is managing information about the 1.7 million species currently known and the millions yet to be discovered and described. The framework for such information systems must include the capability of making predictions about the characteristics of species based on what we know about the biology of close relatives. Making such predictions requires knowledge of phylogenetic relationships. Phylogenetic classification systems are the most effective framework for predictive information systems about organisms and their place in the biosphere (Erwin, 1991; Brooks and McLennan, 1991, 1993a; SA2000, 1994; Humphries et al., 1995; Simpson and Cracraft, 1995; Brooks et al., 2000; Brooks, Leon-Regagnon, and Pérez-Ponce de León, 2000). Although systematists have made major strides in understanding the interrelationships of life, corroborated phylogenetic hypotheses are still lacking for many groups. DIVERSITAS and SA2000 propose to coordinate international research to achieve a phylogenetic framework for all life, resolved to the family level, by the year 2010.

The past decade has seen the integration of phylogenetic information in virtually all areas of evolutionary research (Brooks and McLennan, 1991; Harvey and Pagel, 1991), including historical ecology (Brooks, 1985a). Historical ecology is an interesting and important component of basic research in evolutionary biology and may also provide a means for placing a variety of important biodiversity information in a predictive framework. As a framework within which information from systematics and ecology can be integrated, historical ecology represents common ground that can serve the professional agendas of taxonomists and ecologists involved in biodiversity initiatives, while providing relevant data to conservation managers. For example, when plant taxonomists suggested that the sister species of the American yew tree might well have a compound similar to taxol, Taxotene was discovered. The interface of systematics and biodiversity has also been vital for the successful development of agriculture in this century (Miller and Rossman, 1995). The advent of such predictive applications for integrative data from systematics clearly drives the development of efficient and accessible systems for the storage, maintenance, and retrieval of such information.

PARASITES—A MAJOR COMPONENT OF BIO-COMPLEXITY: Since the advent of modern phylogenetic studies of parasites (Brooks, 1977), examination of these complex systems has included an assessment of the degree of congruence between host and parasite phylogeny as an indication of the form and duration of historical association between the host and parasite group. Interpretation of the current database (Brooks and McLennan, 1993a) suggests that about 50% of the host–parasite associations examined have resulted from cospeciation (Brooks, 1979), in which the ancestors of the host and the parasite were associated and have inherited (metaphorically speaking) their present ecological associa-

tion. The remainder can be attributed to speciation by host switching or colonization (Brooks, 1979). Significantly, in these systems, whether they were derived through cospeciation or colonization, there is no correlation between the degree of host specificity with definitive hosts and the age of coevolutionary associations (Brooks, 1979, 1981; Hoberg, 1986; Poulin, 1992; Brooks and McLennan, 1991, 1993a, 1993b, 1993c). These studies have emphasized that for parasitic helminths and their hosts, cospeciation is not a universal driving force behind diversification (Brooks and McLennan, 1993a and references therein; Pérez-Ponce de León and Brooks, 1995a, 1995b; León-Règagnon, 1998; León-Règagnon et al., 1996, 1998; Boeger and Kritsky, 1997; Hoberg, Brooks, and Siegel-Causey, 1997; Pérez-Ponce de León, León-Règagnon, and Mendoza-Garfias, 1997). Entire faunas have apparently originated by host switching and subsequent coevolution, e.g., the tetrabothriidean tapeworms among seabirds and marine mammals (Hoberg, 1997; Hoberg, Gardner, and Campbell, 1999; Hoberg, Jones, and Bray, 1999), and major taxa of eucestodes among terrestrial and aquatic vertebrates (Hoberg, Gardner, and Campbell, 1999; Hoberg, Jones, and Bray, 1999), the mazocraeidean monogenoideans among primary marine fishes (Boeger and Kritsky, 1997), and the absence of members of the Oligonchoinea (monogenoideans) in freshwater fishes (Boeger and Kritsky, 1997). Indeed, the importance of host switching has recently been emphasized in hypotheses for multiple origins of parasitism among the nematodes (Blaxter et al., 1998).

The discovery that there are no general patterns of host specificity correlated with patterns of speciation in parasitic groups supports the hypothesis that speciation and adaptation are always phylogenetically correlated, but neither is causally dependent on the other. This conclusion was also reached using studies of free-living organisms (Brooks and McLennan, 1991). The degree of host specificity shows no macroevolutionary regularities, i.e., one cannot estimate the degree of phylogenetic congruence between host and parasite phylogenies or the length of time hosts and parasites have been associated from observations of host specificity. Similar conclusions have been derived from investigations on the interaction of herbivores and plants in tropical systems (Janzen, 1973, 1980, 1985).

Phylogenetic approaches are thus requisite for elucidation of the complex histories of host–parasite assemblages and evaluation of a range of alternative hypotheses and predictions in the evolution of complex systems (Brooks et al., 2000; Brooks, León-Règagnon, and Pérez-Ponce de León, 2000). A diversity of model systems is necessary to resolve the intricacies of processes associated with cospeciation, particularly processes associated with host switching (Hoberg, Brooks, and Siegel-Causey, 1997, and references therein). In the future, we may be able to compare macroevolutionary patterns of association and search for generalities between host–parasite and other coevolutionary associations, such as phytophagous insects and their host plants or pollinators and their host plants. Comparative phylogenetic approaches are also a foundation for detailed studies in evolution and community structure. Next, we briefly review applications of parasitological data to these broader areas of biology.

Parasites and Comparative Biology:

Parasites are excellent systems for macroevolutionary studies of character evolution: Parasites are neither unusually simplified nor unusually adaptively plastic in their morphological traits (Brooks and McLennan, 1993a; Pérez-Ponce de León and Brooks, 1995a, 1995b; Pérez-Ponce de León, León-Règagnon, and Mendoza-Garfias, 1997; León-Règagnon, 1998; León-Règagnon et al., 1996, 1998). Parasite evolution has not been characterized by widespread loss of traits that would indicate that parasites have given up much evolutionary independence to their hosts or by widespread homoplasy, indicating that parasites are so simplified that their options for morphological innovation are limited evolutionarily.

Parasites are not degenerate, overspecialized, host–dependent creatures on the periphery of evolution (Brooks and McLennan, 1993a; Poulin, 1995b). They are successful, innovative creatures, many of which have persisted for a long time on this planet. This contention is supported by phylogenetically based estimates for the origins and age of the major groups of parasitic platyhelminths. Divergence of the common ancestor of the Aspidobothriidea and the Digenea, of the major lineages of monogenoideans, and of the common ancestor of the Gyrocotylidea and the Cestoidea (Amphilinidea +

Eucestoda) coincided with the divergence of the common ancestor of the Chondrichthyes and the common ancestor of the rest of the gnathostomous vertebrates (Brooks, 1985b). Complementary independent assessments within the eucestodes (Hoberg, Gardner, and Campbell, 1999; Hoberg, Jones, and Bray, 1999; Hoberg, Mariaux, and Brooks, 2000) and monogenoideans (Boeger and Kritsky, 1997) suggest origins extending to the Devonian from 350 million to 420 million or more years ago.

Other studies (Brooks et al., 1985, 1989) indicated that parasite ontogenies evolve as coherent units and that larval and adult morphological traits are phylogenetically congruent. The degree of adaptive response by each life cycle stage is thus constrained by common evolutionary history. Finally, although molecular systematics is in its infancy within parasitology, studies to date show that there is a high degree of concordance between phylogenies based on molecular and morphological traits, when proper phylogenetic methods are used and careful character analysis is performed (e.g., for ordinal-level relationships among the eucestodes, Hoberg et al., 1997; Hoberg, Mariaux, and Brooks, 2000; Mariaux, 1998). The merits of studies based on "total evidence" that combine morphological and molecular databases (Kluge, 1989, 1997, 1998a, 1998b; de Queiroz et al., 1995; Huelsenbeck et al., 1996; Sanderson et al., 1998) are also apparent (Hoberg, Mariaux, and Brooks, 2000). León-Règagnon et al. (1999) have recently emphasized this point, showing that a combination of molecular and morphological data could help resolve outstanding species-level taxonomic problems within a group of frog digeneans.

Parasites and the evolution of life history traits: The extent to which the individual components of reproductive biology, development, and ecology, as well as their complex interactions, can be highlighted and examined in parasite–host systems is impressive. Phylogenetic analysis also allows us to examine phylogenetically associated changes in reproductive and nonreproductive male and female characters. We can then ask questions, such as what are the costs and benefits of different reproductive strategies? Do male and female characters covary in either their origin or their loss (digeneans, monogenoideans)? What is the relationship between reproductive conservatism and reproductive flexibility in male or female characters (digeneans, monogenoideans)? What is the relationship between sexual reproduction and the appearance of character novelty (eucestodes)? And if asexual reproduction is good and sex is better, is sex combined with asexual reproduction the best (digeneans)? How does dioecy evolve in monoecious lineages (Platt and Brooks, 1997)? Recent studies (Morand, 1996a, 1996b; Poulin, 1992, 1995a, 1995b, 1997a, 1997b; Sasal et al., 1997, 1998; Sasal and Morand, 1998) confirm the suitability of parasite systems for studies of the evolution of life history strategies. Their results confirmed the assertion by Brooks and McLennan (1993a) that parasites show the same kinds of life history evolution as their closest free-living relatives.

Parasites as model systems for studying adaptive radiations: Parasites have not experienced unusually high degrees of adaptive radiation but do show interesting patterns. Within the parasitic flatworms, the monogenoideans appear to have undergone adaptive radiation, whereas the digeneans and the eucestodes appear to have experienced evolutionary radiation that may or may not have been adaptive (Brooks and McLennan, 1993b, 1993c). It is important to realize, however, that a relatively species-poor sister group balances each species-rich group, so it is inaccurate to speak of parasites in general as having experienced high levels of adaptive radiations. The question of the relative extent of parasite adaptive radiations cannot be answered until we have comparable databases for free-living groups. At the moment, we can say that the monogenoideans, digeneans, and eucestodes, not unlike ostariophysan and percomorph fishes and passerine birds, provide a wealth of information about radiations, adaptive or not. This information, in turn, supports the hypothesis that such radiations were primarily a function of diversification of life cycle components. For example, 4 putative key innovations were identified during examination of the database for the parasitic flatworms (Brooks and McLennan, 1993a, 1993c): (1) the evolution of a direct life cycle (a developmental change, possibly caused by peramorphosis, with an ecological outcome); (2) the appearance of additional larval stages (a developmental change); (3) the appearance of asexual amplification of larval stages (a devel-

opmental change); and (4) the appearance of sexual amplification of reproductive output (again, a developmental change, this time involving the repetitious production of segments). The success of these key innovations was based on an interaction between the environment and populations, tempered by a background of substantial inherited constraints. Since both parasites and hosts have evolutionary tendencies and capabilities, parasite evolution will be historically correlated in some way with host evolution but will not necessarily be caused by it. Adaptive radiations, therefore, result from active interaction between parasite and environmental (host) characteristics rather than just from evolutionarily passive parasite responses to host characteristics.

The evolution of life cycles is the key element in the phylogenetic diversification and adaptive radiation of parasites. Life cycle patterns show a rich mosaic of diversification in reproductive, developmental, and ecological characteristics in a strongly phylogenetic context. Evolutionary radiations of parasite groups appear to involve, first, ontogenetic innovations, second, changes in adult reproductive structures, and third, ecological components of life cycles. The evolution of changes in the biology of the parasites dictates the changes in life cycle patterns, including patterns of host utilization, rather than the reverse. Species richness, therefore, is correlated with different phenomena in different groups of parasites. These phenomena include changes in ecological components of life cycles, production or amplification of dispersing larval or juvenile stages, and amplification of sexual reproductive output.

Parasites as systems for examining the modes of speciation: Price (1980) proposed that the evolution of many new parasite–host relationships occurred through colonization of new hosts. He interpreted this as an example of sympatric speciation, because the hosts had to overlap geographically for the switch to occur. This host-biased perspective changes when we view the speciation process from the perspective of the organism that is actually speciating. Brooks and McLennan (1993a) suggested that if one takes a worm's-eye view, different host species are like different island archipelagos, and speciation by host switching is better explained as a form of peripheral isolates, i.e., allopatric speciation, than as sympatric speciation. Recently, Funk (1998) has shown that if we consider speciation by host switching in the manner suggested by Brooks and McLennan (1993a), it is easier to understand how natural selection can play a role in producing isolating mechanisms in the populations living on or in different hosts. By this logic, true sympatric speciation in parasitic platyhelminths can be identified by finding sister species restricted to different parts of the same host. Rohde (1979) used similar reasoning to suggest that natural selection might play a role in determining the high degree of site specificity exhibited by many parasites.

The evidence collected to date suggests that vicariant and peripheral isolates speciation via host switching have played the dominant roles in the speciation of parasitic platyhelminths, including crocodilians and their digeneans, freshwater stingrays and their eucestodes, frogs and their digeneans, freshwater and marine turtles and their digeneans, marine fish and their digeneans and monogenoideans, and seabirds and pinnipeds and their eucestodes (see refs. in Brooks and McLennan, 1993a; also Hoberg, 1992, 1995; Hoberg and Adams, 1992; Pérez-Ponce de León and Brooks, 1995a, 1995b; Boeger and Kritsky, 1997; León-Règagnon, 1998; León-Règagnon et al., 1996, 1998). Tantalizing possibilities of sympatric speciation suggested by, for instance, ochetosomatid digeneans in snakes, some monogenoideans, and many oxyurid nematodes remain to be investigated phylogenetically.

Host isolation, and particularly isolation for definitive hosts, therefore drives speciation. Current evidence suggests that intermediate hosts are evolutionarily neutral. This seems to be a general pattern, emerging from phylogenetic studies of cestodes in avian and mammalian hosts in either terrestrial or marine environments (Hoberg, 1986, 1992, 1995; Hoberg and Adams, 1992; Hoberg et al., 2000). For example, among tapeworms of the genus *Taenia* (a group where detailed information is available for the life cycles of most species), speciation appears to be driven by host switching among definitive hosts exploiting prey within guild associations. A prediction that might follow from these findings suggests that ecological continuity and predictability are limited by transmission dynamics linked to intermediate hosts but that diversification is driven by predator–prey associations

(Hoberg et al., 2000). These studies indicate the necessity for having detailed phylogenetic and ecological data as the basis for examining patterns and process in speciation for hosts and parasites and at a higher level for evaluation of faunal and community history.

Parasites—paradigm systems for studies of community structure and evolution:

> One major advantage of parasite communities over others is that the habitat they live in, the host, has such a well defined structure. . .The host microcosm is replicated through time and space much more so than habitats for most other organisms. Therefore, the study of comparative community structure is very powerful. (Price, 1986)

There is, as yet, little overlap between parasite groups for which we have extensive community ecological information and groups for which we have extensive phylogenetic information (Poulin, 1995a, 1997b). In addition, differences in understanding between systematists and ecologists about the use of phylogenetic methods and the possible forms of phylogenetic components in community structure have led to unproductive and inappropriate polarization of perspectives (Bush et al., 1990). Perspectives on the primacy of ecological versus phylogenetic–historical determinants of faunal structure are changing, however, as researchers begin to recognize that communities are mosaics of species that evolved elsewhere and dispersed into the area (colonizers) and species that evolved in situ (residents or endemics) (Aho and Bush, 1993). Each parasite community represents a historically unique combination of colonizers and endemic species, in terms of both geographic dispersal and host switching (Brooks and McLennan, 1991, 1993a, 1993b, 1993c; Hoberg, 1997a). Because parasite communities are so well defined and so easily studied, parasitologists have an opportunity to assume a leadership role in the study of community evolution.

Parasites are developmentally and ecologically complex organisms subject to and constrained by the same rules that govern the evolution of all biological systems (Poulin, 1995b). This is the key to their value in predictive studies. Ernst Mayr (1957) recognized nearly half a century ago that the study of parasites "is not only valuable for the parasitologist, but is also a potential gold-mine for the evolutionist and general biologist."

Here at the end of the twentieth century, and in the midst of the biodiversity crisis, those sentiments are truer than ever. The ability to distinguish evolutionary colonizers from residents will permit us to recognize introduced species and to assess the risk that they may cause emergent diseases. The ability to distinguish evolutionary generalists from specialists will enable us to assess more fully the extent of biocontainment for any parasite being used as a biocontrol agent. Finally, the ability to distinguish the old from the recent components of ecosystem structure will help us assess what species are likely to respond to anthropogenic changes, in what order, in what ways, and to what extent.

GTI Component 3—Management of Systematic Knowledge Bases: Getting the Information to Those Who Can Use it Effectively. DIVERSITAS estimates that within 5 years electronic data handling and interlinked knowledge systems will become the principal medium for all activities associated with applying systematic information to biodiversity studies and policies. These efforts will require large databases on taxonomic information, specimens, and data in collections.

The taxasphere can contribute substantially in this area by developing 2 types of home pages: (1) phylogenetic home pages, providing the most up-to-date phylogenetic trees for all groups of parasites, interconnected in such a way that anyone can move from one taxonomic level to another (this will provide the predictive framework within which a variety of specialists can operate), and (2) species home pages, providing the following information for each targeted species: (i) what is it (and how to distinguish it from others), (ii) where is it, and (iii) what is its natural history. This process has been termed the creation of a biodiversity Yellow Pages for the Internet (Janzen and Hallwachs, 1994). These home pages will include electronic images, which can be used for other purposes, such as taxonomic descriptions and revisions or identification guides. These home pages would also include information about the known natural history of each species. Species home pages should be cross-linked to phylogenetics home pages so that we will eventually have a complete listing of the phylogenetic relationships of all species linked to their natural histories. Biodiversity information, irrespective of format, must eventu-

ally be linked back to a specimen-based reference system or biological collection.

GTI AND COLLECTIONS AS FUNDAMENTAL FOUNDATIONS FOR BIODIVERSITY: Biological collections are essential elements in the development of biodiversity information (Davis, 1996; GTI, 1999). Collections developed from inventory activities represent "a permanent, documentable record of specimen-based information" (GTI, 1999) that provides historical, contemporary, and predictive baselines for understanding the patterns and distribution of organisms in the biosphere. Collections are vital in defining the continuity of ecosystem or community structure and integrity. Collections allow detailed examination of spatial and temporal variation from local to global scales, directly linked to specimens and data for populations and species, and such biologically significant parameters as reproductive phenology, ecology, behavior, biogeography, and host associations. Biological collections are the context for developing and applying biodiversity information efficiently and effectively. Thus, the infrastructure for collections must be regarded as an integral facet of any developing programs for survey, inventory, and documentation of global biodiversity resources (GTI, 1999).

Conclusions

Both the taxasphere and the biosphere may be facing imminent extinction. We have insufficient taxonomic expertise across all components of diversity to address global needs for survey and inventory in a timely manner. Survival of the taxasphere depends in large part on making the cultural change from seeing ourselves in the traditional mode of collectors of things to being managers of information. It is no longer important, or even relevant, to have more specimens of a particular species in a collection than are found in any other collection; rather, it is important to know how much information is available about each species. Just as society is no longer willing to invest huge sums of money in classic set-aside conservation projects, neither is it willing to invest in ever-expanding museum collections that serve only as repositories of material accessible to an ever-decreasing number of specialists (Davis, 1996). Neither is society willing to invest enormous amounts of money in a taxasphere whose only concern is esoteric research. The ongoing internal conflict among systematic biologists about whether we should be seen as a service discipline or as an independent research discipline threatens to weaken the taxasphere in its efforts to make a significant contribution to society and thereby ensure its own survival.

Already too many ecologists and biodiversity managers believe that systematists are good only for providing names; at the same time, too many systematists believe that their only function should be generating phylogenies and cutting-edge comparative evolutionary studies. We must fully internalize the belief that the fundamental importance of systematic biology stems from its being both an essential service profession and an essential element of evolutionary biology. Ecologists have successfully grappled with similar issues (Lubchenco et al., 1991)

The call for more inventory work in biodiversity thus represents a tremendous opportunity and challenge for the taxasphere. Members of the taxasphere must understand that biodiversity preservation is based on such issues as economic development, conservation, solving major environmental challenges, and limiting the impacts of emerging pathogens and parasites. Inventories that seek to identity the critical components of biodiversity are necessary to achieve these goals, which are mutually beneficial and can be synergistic for the scientific community and the general population. They are an essential part of helping to save the biosphere by helping improve the socioeconomic status of as many people as possible.

The taxasphere has long assumed the responsibility of naming and classifying the species on this planet, and modern phylogenetic methods have produced maximally efficient modes of storing and transmitting information through classifications. The Internet gives us a powerful mechanism for disseminating enormous amounts of information quickly and widely. All those interested in preserving, managing, and sustainably using biodiversity should have a vested interest in supporting a strong taxasphere. Within the scientific community, however, taxonomists do not have a history of close and cordial interactions with other specialists. There are many reasons for this, some of which have been discussed (Brooks and McLennan, 1991), but we must overcome the historical constraints of sectarian competition for academic positions and prestige. The scientific community can help pre-

serve biodiversity effectively if each participant can give up something of his or her own immediate personal agenda to help achieve a greater good.

We return to the analogy of the taxasphere as a triage team, the biosphere as a "battlefield," and the "war" as human activities that degrade global biotic resources. The triage teams survey parts of the battlefield as completely as possible looking for "wounded" participants. All possible participants and the degree to which each has been affected must be recognized, and the taxasphere has the role of passing that information on to the decision makers who are responsible for the optimal deployment of resources. Names and critical life history and ecological information provided by taxonomists constitute the foundation for bringing a broad array of stakeholders in the national and international arena to understand the value of biodiversity.

DIVERSITAS has designated 2001 as the International Biodiversity Observation Year, which will, among other things, focus attention on the value of the taxasphere and promote a successful launch of the GTI. With an emphasis on involving local people in a variety of initiatives associated with this observation, the International Biodiversity Observation Year is an excellent opportunity for coalitions of international, national, and local political, social development, and environmental agencies to join together to provide a fuller inventory of the species on this planet.

One should never change a winning game and always change a losing game. So far we have been playing a losing game. On a global basis, people's lives are not improving, and we continue to lose large parts of the planet's biota. The 3-pronged action plan of the GTI represents a bold and assertive effort to change a losing game into a winning one. The comparative parasitological perspective using historical, ecological, and biogeographic information offers the potential for contributions toward recognizing, defining, and solving challenges to global biodiversity.

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Literature Cited

Aho, J. M., and A. O. Bush. 1993. Community richness in parasites of some freshwater fishes from North America. Pages 185–193 *in* R. E. Ricklefs and D. Schluter, eds. Species Diversity in Ecological Communities: Historical and Geographical Perspectives. University of Chicago Press, Chicago, Illinois.

Anderson, T. J. C., M. S. Blouin, and R. N. Beech. 1998. Population biology of parasitic nematodes: applications of genetic markers. Advances in Parasitology 41:219–283.

Balbuena, J. A., and J. A. Raga. 1994. Intestinal helminths as indicators of segregation and social structure of pods of long-finned pilot whales (*Globicephala melas*) off the Faeroe Islands. Canadian Journal of Zoology 72:443–448.

———, **F. J. Aznar, M. Fernandez, and J. A. Raga.** 1995. Parasites as indicators of social structure and stock identity of marine mammals. Pages 133–139 *in* A. S. Blix, L. Walloe, and O. Ultang, eds. Developments in Marine Biology, Vol. 4., Whales, Seals, Fish and Man. Elsevier, New York.

Barrowclough, G. F. 1992. Systematics, biodiversity and conservation biology. Pages 121–143 *in* N. Eldredge, ed. Systematics, Ecology and the Biodiversity Crisis. Columbia University Press, New York.

Barse, A. M., and D. H. Secor. 1999. An exotic nematode parasite of the American eel. Fisheries 24: 6–10.

Bartoli, P. 1989. Digenetic trematodes as bio-indicators for yellow-legged gulls in Corsica (western Mediterranean). Pages 251–259 *in* J. S. Aguilar, X. Monbailliu, and A. M. Paterson, eds. Status and Conservation of Seabird Ecogeography and

Mediterranean Action Plan. Society of Spanish Ornithology, Madrid.

Belogurov, O. I. 1966. Spetsifichnost' gel'mintov v otnoshenni definitivnykh khoziaev vzaimosiaz' gel'mintofauna zhivotnykh razlichnykh otriadov. Zoologicheskii Zhurnal 45:1449–1454.

Blackmore, S. 1996. Knowing the Earth's biodiversity: challenges for the infrastructure of systematic biology. Science 274:63–64.

Blaxter, M. L., P. De Ley, J. R. Garey, L. X. Liu, P. Scheldman, A. Vierstraete, J. R. Vanfleteren, L. Y. Mackey, M. Dorris, L. M. Frisse, J. T. Vida, and W. Kelley Thomas. 1998. A molecular evolutionary framework for the phylum Nematoda. Nature 392:71–75.

Boeger, W., and D. Kritsky. 1997. Coevolution of the Monogenoidea (Platyhelminthes) based on a revised hypothesis of parasite phylogeny. International Journal for Parasitology 27:1495–1511.

Brooks, D. R. 1977. Evolutionary history of some plagiorchoid trematodes of anurans. Systematic Zoology 26: 277–289.

———. 1979. Testing the context and extent of host-parasite coevolution. Systematic Zoology 28: 299–307.

———. 1981. Hennig's parasitological method: a proposed solution. Systematic Zoology 30:229–249.

———. 1985a. Historical ecology: a new approach to studying the evolution of ecological associations. Annals of the Missouri Botanical Garden 72:660–680.

———. 1985b. Phylogenetics and the future of helminth systematics. Journal of Parasitology 71: 719–727.

———. 1990. Parsimony analysis in historical biogeography and coevolution: methodological and theoretical update. Systematic Zoology 39:14–30.

———. 1998. Triage for the biosphere. Pages 71–80 *in* G. B. Softing, G. Benneh, K. Hindar, and A. Wijkman, eds. The Brundtland Commission's Report—10 Years. Scandinavian University Press, Oslo.

———, **S. M. Bandoni, C. A. MacDonald, and R. T. O'Grady.** 1989. Aspects of the phylogeny of the Trematoda Rudolphi 1808 (Platyhelminthes: Cercomeria). Canadian Journal of Zoology 67: 2609–2624.

———, **E. P. Hoberg, V. León-Règagnon, G. Pérez-Ponce de León, W. Boeger, D. Kritsky, and G. Klassen.** 2000. Putting the phylogenetic database to work. *In* D. T. J. Littlewood and R. A. Bray, eds. The Interrelationships of the Platyhelminthes. Taylor and Francis, London, U.K. (In press.)

———, **V. León-Règagnon, and G. Pérez-Ponce de León.** 2000. Parasitos y la biodiversidad. *In* M. H. Hernandez, A. N. Garcia, M. Ulloa, and F. Alvarez, eds. Enfoques Contemporaneos para el Estudio de la Biodiversidad. Universidad Nacional Autónoma de Mexico, Mexico City. (In press.)

———, **R. L. Mayden, and D. A. McLennan.** 1992. Phylogeny and biodiversity: conserving our evolutionary legacy. Trends in Ecology and Evolution 7:55–59.

———, **and D. A. McLennan.** 1991. Phylogeny, Ecology and Behavior: A Research Program in Comparative Biology. University of Chicago Press, Chicago, Illinois. 434 pp.

———, **and ———.** 1993a. Parascript: Parasites and the Language of Evolution. Smithsonian Institution Press, Washington, D.C. 430 pp.

———, **and ———.** 1993b. Historical ecology as a research program in macroevolution. Pages 76–113 *in* R. L. Mayden, ed. Systematics, Historical Ecology and North American Freshwater Fishes, Stanford University Press, Stanford, California.

———, **and ———.** 1993c. Historical ecology: examining phylogenetic components of community evolution. Pages 267–280 *in* R. Ricklefs and D. Schulter, eds. Species Diversity in Ecological Communities, University of Chicago Press, Chicago, Illinois.

———, ———, **J. M. Carpenter, S. G. Weller, and J. A. Coddington.** 1995. Systematics, ecology and behavior. Bioscience 45:687–695.

———, **R. T. O'Grady, and D. R. Glen.** 1985. Phylogenetic analysis of the Digenea (Platyhelminthes: Cercomeria) with comments on their adaptive radiation. Canadian Journal of Zoology 63: 411–443.

Brooks, D. R., L. Prieto, and G. Pérez-Ponce de León. 1999. Two new species of *Oochoristica* in *Ctenosaura* spp. from Costa Rica and Mexico. Journal of Parasitology 85:893–897.

———, **T. B. Thorson, and M. A. Mayes.** 1981. Freshwater stingrays (Potamotrygonidae) and their helminth parasites: testing hypotheses of evolution and coevolution. Pages 147–175 *in* V. A. Funk and D. R. Brooks, eds. Advances in Cladistics, New York Botanical Garden, New York.

Bush, A. O. 1990. Helminth communities in avian hosts: determinants of pattern. Pages 197–232 *in* G.W. Esch, A. O. Bush, and J. M. Aho, eds. Parasite Communities: Patterns and Processes. Chapman and Hall, London, U.K.

———, **J. M. Aho, and C. R. Kennedy.** 1990. Ecological versus phylogenetic determinants of helminth community richness. Evolutionary Ecology 4:1–20.

———, **and C. R. Kennedy.** 1994. Host fragmentation and helminth parasites: hedging your bets against extinction. International Journal for Parasitology 24:1333–1343.

———, **K. D. Lafferty, J. M. Lotz, and A. W. Shostak.** 1997. Parasitology meets ecology on its own terms: Margolis et al. revisited. Journal of Parasitology 83:575–583.

Bustnes, J. O., and K. Galaktionov. 1999. Anthropogenic influences on the infestation of intertidal gastropods by seabird trematode larvae on the southern Barents Sea coast. Marine Biology 133: 449–453.

Caballero y Rodriguez, G., C. Del Rio-Estrada, L. Rodriguez-Alvarez, and G. C. Tello-Sandoval. 1992. Trematodes as pollution level indicators by hydrogen sulphide in the Gulf of Mexico, and this acid's impact on public health. Annales del Instituto de Ciencias del Mar y Limnología 19:151–161.

Clayton, D. H., and B. A. Walther. 1997. Collection and quantification of arthropod parasites of birds. Pages 419–440 *in* D. H. Clayton and J. Moore, eds. Host-Parasite Evolution: General Principles and Avian Models. Oxford University Press, Oxford, U.K.

Cone, D. K., D. J. Marcogliese, and W. D. Watt. 1993. Metazoan parasite communities of yellow eels (*Anguilla rostrata*) in acidic and limed rivers of Nova Scotia. Canadian Journal of Zoology 71: 177–184.

Dailey, M., and W. K. Vogelbein. 1991. Parasite fauna of three species of Antarctic whales with reference to their use as potential stock indicators. Fisheries Bulletin 89:355–365.

Davis, G. 1995. Systematics and public health. Bioscience 45:705–714.

———. 1996. Collections of biological specimens essential for science and society. Association of Systematics Collections Newsletter 24:77–78, 88–90.

Desser, S. S. 1997. Blood parasites of the iguanid lizard, *Ctenosaura similis* from Costa Rica, with a description of a new species, *Hepatozoon gamezi*. Journal of Eukaryotic Microbiology 44:162–167.

Dobson, A. P. 1988. Restoring island ecosystems: the potential of parasites to control introduced animals. Conservation Biology 2:31–39.

———, **and R. Carper.** 1992. Global warming and potential changes in host-parasite and disease-vector relationships. Pages 201–217 *in* R. L. Peters and T. E. Lovejoy, eds. Global Warming and Biological Diversity. Yale University Press, New Haven, Connecticut.

———, **and P. J. Hudson.** 1986. Parasites, disease and the structure of ecological communities. Trends in Ecology and Evolution 1:11–15.

———, **and R. M. May.** 1986a. Disease and conservation. Pages 345–365 *in* M. E. Soulé, ed. Conservation Biology: The Science of Scarcity and Diversity. Sinauer Associates, Sunderland, Massachusetts.

———, **and** ———. **1986b.** Patterns of invasions by pathogens and parasites. Pages 58–76 *in* H. A. Moore and J. A. Drake, eds. Ecology of Biological Invasions of North America and Hawaii, Ecological Studies Vol. 58. Springer-Verlag, New York.

Dogiel, V. A. 1961. Ecology of the parasites of freshwater fishes. Pages 1–47 *in* V. A. Dogiel, G. K. Petruvshevski, and Y. I. Polyanski, eds. Parasitology of Fishes. Oliver and Boyd, Edinburgh, U.K.

Doster, G. L., and C. P. Goater. 1997. Collection and quantification of avian helminths and Protozoa. Pages 396–418 *in* D. H. Clayton and J. Moore, eds. Host-parasite Evolution: General Principles and Avian Models. Oxford University Press, Oxford, U.K.

Durden, L. A., and J. E. Keirans. 1996. Host-parasite coextinction and the plight of tick conservation. American Entomologist 42:87–91.

Ehrlich, P. R., and E. O. Wilson. 1991. Biodiversity studies: science and policy. Science 253: 758–762.

Eldredge, N. 1992. Where the twain meet: causal intersections and the genealogical and ecological realms. Pages 1–14 *in* N. Eldredge, ed. Systematics, Ecology and the Biodiversity Crisis. Columbia University Press, New York.

Epstein, P. R. 1997. Climate, ecology and human health. Consequences 3:3–19.

———. 1999. Climate and health. Science 285:347–348.

Erwin, T. L. 1991. An evolutionary basis for conservation strategies. Science 253:750–752.

Esch, G. W., A. O. Bush, and J. M. Aho. 1990. Parasite Communities: Patterns and Processes. Chapman and Hall, London, U.K. 335 pp.

———, **A. W. Shostak, D. J. Marcogliese, and T. M. Goater.** 1990. Patterns and processes in helminth parasite communities: an overview. Pages 1–19 *in* G. W. Esch, A. O. Bush, and J. M. Aho, eds. Parasite Communities: Patterns and Processes. Chapman and Hall, London, U.K.

Ewald, P. W. 1995. The evolution of virulence: a unifying link between parasitology and ecology. Journal of Parasitology 81:659–669.

Funk, D. J. 1998. Isolating a role for natural selection in speciation: host adaptation and sexual isolation in *Neochlamisus bebbianae* leaf beetles. Evolution 52:1744–1759.

Gardner, S. L., and M. Campbell. 1992. Parasites as probes for biodiversity. Journal of Parasitology 78:596–600.

Glowka, L., F. Burhenne-Guilmin, H. Synge, J. A. McNeely, and L. Gündling. 1994. A Guide to the Convention on Biological Diversity. International Union CN, Gland, Switzerland.

Grenfell, B. T. 1992. Parasitism and the dynamics of ungulate grazing systems. American Naturalist 139:907–929.

GTI (Global Taxonomy Initiative). 1999. Using systematic inventories to meet country and regional needs. A Report of the DIVERSITAS/ Systematics Agenda 2000 International Workshop, September 17–19, 1998, American Museum of Natural History. The Center for Biodiversity and Conservation, American Museum of Natural History, New York. 34 pp.

Gulland, F. M. D. 1995. The impact of infectious diseases on wild animal populations—a review. Pages 20–51 *in* B. T. Grenfell and A. P. Dobson, eds. Ecology of Infectious Diseases in Natural Populations. Cambridge University Press, Cambridge, U.K.

Handeland, K., and T. Slettbakk. 1994. Outbreaks of cerebrospinal elaphostrongylosis in reindeer (*Rangifer tarandus tarandus*) in Finnmark, Norway, and their relation to climatic conditions. Journal of Veterinary Medicine 41:407–410.

Harvey, P. H., and M. Pagel. 1991. The Comparative Method in Evolutionary Biology. Oxford University Press, Oxford, U.K.

Hawksworth, D. L. 1995. The resource base for biodiversity assessment. Pages 551–605 *in* V. H. Heywood and R. T. Watson, eds. Global Biodiversity Assessment. Cambridge University Press, Cambridge, U.K. 239 pp.

Hoagland, K. E. 1996. The taxonomic impediment and the Convention on Biodiversity. Association

of Systematics Collections Newsletter 24: 61–62, 66–67.

Hoberg, E. P. 1986. Evolution and historical biogeography of a parasite-host assemblage: *Alcataenia* spp. (Cyclophyllidea: Dilepididae) in Alcidae (Charadriiformes). Canadian Journal of Zoology 64:2576–2589.

———. 1992. Congruent and synchronic patterns in biogeography and speciation among seabirds, pinnipeds and cestodes. Journal of Parasitology 78: 601–615.

———. 1995. Historical biogeography and modes of speciation across high latitude seas of the Holarctic: concepts for host-parasite coevolution among the Phocini (Phocidae) and Tetrabothriidae (Eucestoda). Canadian Journal of Zoology 73:45–57.

———. 1996. Faunal diversity among avian parasite assemblages: the interaction of history, ecology, and biogeography in marine systems. Bulletin of the Scandinavian Society of Parasitology 6:65–89.

———. 1997a. Phylogeny and historical reconstruction: host-parasite systems as keystones in biogeography and ecology. Pages 243–261 *in* M. L. Reaka-Kudla, D. E. Wilson, and E. O. Wilson, eds. Biodiversity II: Understanding and Protecting Our Biological Resources. Joseph Henry Press, Washington, D.C.

———. 1997b. Parasite biodiversity and emerging pathogens: a role for systematics in limiting impacts on genetic resources. Pages 71–83 *in* K. E. Hoagland and A. Y. Rossman, eds. Global Genetic Resources: Access, Ownership and Intellectual Property Rights. Association of Systematics Collections, Washington, D.C.

———, **and A. M. Adams.** 1992. Phylogeny, historical biogeography and ecology of *Anophryocephalus* spp. (Eucestoda: Tetrabothriidae) among pinnipeds of the Holarctic during the late Tertiary and Pleistocene. Canadian Journal of Zoology 70: 703–719.

———, **D. R. Brooks, H. Molina-Ureña, and E. Erbe.** 1998. *Echinocephalus janzeni* n. sp. (Nematoda: Gnathostomatidae) in *Himantura pacifica* (Chondrichthyes: Myliobatiformes) from the Pacific coast of Costa Rica and Mexico, with historical biogeographic analysis of the genus. Journal of Parasitology 84:571–581.

———, ———, **and D. Siegel-Causey.** 1997. Host-parasite co-speciation: history, principles and prospects. Pages 212–235 *in* D. H. Clayton and J. Moore, eds. Host-parasite Evolution: General Principles and Avian Models. Oxford University Press, Oxford, U.K.

———, **S. L. Gardner, and R. A. Campbell.** 1997. Paradigm shifts and tapeworm systematics. Parasitology Today 13:161–162.

———, ———, **and** ———. 1999. Systematics of the Eucestoda: advances toward a new phylogenetic paradigm, and observations on the early diversification of tapeworms and vertebrates. Systematic Parasitology 42:1–12.

———, **A. Jones, and R. Bray.** 1999. Phylogenetic analysis among the families of the Cyclophyllidea (Eucestoda) based on comparative morphology, with new hypotheses for co-evolution in vertebrates. Systematic Parasitology 42:51–73.

———, ———, **R. L. Rausch, K. S. Eom, and S. L. Gardner.** 2000. A phylogenetic hypothesis for species of the genus *Taenia* (Eucestoda: Cyclophyllidea). Journal of Parasitology 86: In press.

———, **A. A. Kocan, and L. G. Rickard.** 2000. Gastrointestinal strongyles in wild ruminants from North America. *In* W. Samuel, M. Pybus, and A. A. Kocan, eds. Parasitic Diseases of Wild Mammals. Iowa State University Press, Ames. (In press.)

———, **and J. R. Lichtenfels.** 1994. Phylogenetic systematic analysis of the Trichostrongylidae (Nematoda) with an initial assessment of coevolution and biogeography. Journal of Parasitology 80:976–996.

———, **S. Lloyd, and H. Omar.** 1995. *Libyostrongylus dentatus* n. sp. (Nematoda: Trichostrongylidae) from ostriches in North America, with comments on the genera *Libyostrongylus* and *Paralibyostrongylus*. Journal of Parasitology 81:85–93.

———, **J. Mariaux, J.-L. Justine, D. R. Brooks, and P. J. Weekes.** 1997. Phylogeny of the orders of the Eucestoda (Cercomeromorphae) based on comparative morphology: historical perspectives and a new working hypothesis. Journal of Parasitology 83:1128–1147.

———, ———, **and D. R. Brooks.** 2000. Phylogeny among orders of the Eucestoda (Cercomeromorphae): integrating morphology, molecules and total evidence. *In* D.T. J. Littlewood and R. A. Bray, eds. Interrelationships of the Platyhelminthes. Taylor and Francis, London, U.K. (In press.)

———, **and S. G. McGee.** 1982. Helminth parasitism in raccoons, *Procyon lotor hirtus* Nelson and Goldman, in Saskatchewan. Canadian Journal of Zoology 60:53–57.

———, **K. Monsen, S. J. Kutz, and M. Blouin.** 1999. Structure, biodiversity and historical biogeography of nematode faunas in Holarctic ruminants: morphological and molecular diagnoses for *Teladorsagia boreoarcticus* n. sp. (Nematoda: Ostertagiinae), a dimorphic cryptic species in muskoxen (*Ovibos moschatus*). Journal of Parasitology 85:910–934.

———, **L. Polley, A. Gunn, and J. S. Nishi.** 1995. *Umingmakstrongylus pallikuukensis* gen. nov. et sp. nov. (Nematoda: Protostrongylidae) from muskoxen, *Ovibos moschatus*, in the central Canadian Arctic, with comments on biology and biogeography. Canadian Journal of Zoology 73: 2266–2282.

Holmes, J. C. 1996. Parasites as threats to biodiversity in shrinking ecosystems. Biodiversity and Conservation 5:975–983.

———, **and W. M. Bethel.** 1972. Modification of intermediate host behavior by parasites. Zoological Journal of the Linnean Society 1(supplement): 123–149.

Hudson, P. J., A. P. Dobson, and D. Newborn. 1998. Prevention of population cycles by parasite removal. Science 282:2256–2258.

Huelsenbeck, J. P., J. P. Bull, and C. W. Cunning-

ham. 1996. Combining data in phylogenetic analysis. Trends in Ecology and Evolution 11:152–158.

Humphries, C. J., P. H. Williams, and R. I. Vane-Wright. 1995. Measuring biodiversity value for conservation. Annual Review of Ecology and Systematics 26:93–111.

Huxham, M., D. Raffaelli, and A. Pike. 1995. Parasites and foodweb patterns. Journal of Animal Ecology 64:168–176.

Jablonski, D. 1991. Extinctions: a paleontological perspective. Science 253:754–757.

Janzen, D. H. 1973. Comments on host-specificity of tropical herbivores and its relevance to species richness. Pages 201–211 *in* V. H. Heywood, ed. Taxonomy and Ecology. Academic Press, New York.

———. 1980. When is it coevolution? Evolution 34: 611–612.

———. 1985. On ecological fitting. Oikos 45:308–310.

———. 1992. A south-north perspective on science in the management, use, and economic development of biodiversity. Pages 27–52 *in* O. T. Sandlund, K. Hindar, and A. H. D. Brown, eds. Conservation of Biodiversity for Sustainable Development. Scandinavian University Press, Oslo.

———. 1993. Taxonomy: universal and essential infrastructure for development and management of tropical wildland biodiversity. Pages 100–113 *in* O. T. Sandlund and P. J. Schei, eds. Proceedings of the Norway/UNEP Expert Conference on Biodiversity. NINA, Trondheim.

———. 1996. On the importance of systematic biology in biodiversity development. Association of Systematics Collections Newsletter 24:17, 23–28.

———. 1997. Wildland biodiversity management in the tropics. Pages 411–434 *in* M. L. Reaka-Kudla, D. E. Wilson, and E. O. Wilson, eds. Biodiversity II: Understanding and Protecting Our Biological Resources. Joseph Henry Press, Washington, D.C.

———, **and W. Hallwachs.** 1994. All Taxa Biodiversity Inventory (ATBI) of terrestrial systems: a generic protocol for preparing wildland biodiversity for non-damaging use. Report of a National Science Foundation Workshop, 16–18 April 1993, Philadelphia, Pennsylvania. 132 pp.

———, ———, **J. Jimenez, and R. Gámez.** 1993. The role of the parataxonomists, inventory managers and taxonomists in Costa Rica's national biodiversity inventory. Pages 223–254 *in* W. V. Reid et al., eds. Biodiversity Prospecting. World Resources Institute, Washington, D.C.

Johnson, P. T. J., K. B. Lunde, E. G. Ritchie, and A. E. Launer. 1999. The effect of trematode infection on amphibian limb development and survivorship. Science 284:802–804.

Kennedy, C. R. 1993. Introductions, spread and colonization of new localities by fish helminth and crustacean parasites in the British Isles: a perspective and appraisal. Journal of Fish Biology 43:287–301.

Klassen, G. J. 1992. Phylogeny and biogeography of ostracin boxfishes (Tetraodontiformes: Ostraciidae) and their gill parasitic *Haliotrema* sp. (Monogenea: Ancyrocephalidae): a study in host-parasite coevolution. Ph.D. Dissertation, University of Toronto, Canada. 366 pp.

———, **and M. Beverley-Burton.** 1987. Phylogenetic relationships of *Ligictaluridus* spp. (Monogenea: Ancyrocephalidae) and their ictalurid (Siluriformes) hosts: an hypothesis. Proceedings of the Helminthological Society of Washington 54:84–90.

———, **and** ———. 1988. North American freshwater ancyrocephalids (Monogenea) with articulating haptorial bars: host-parasite coevolution. Systematic Zoology 37:179–189.

Kluge, A. 1989. A concern for evidence and a phylogenetic hypothesis of relationships among *Epicartes* (Boidae, Serpentes). Systematic Zoology 38:7–25.

———. 1997. Testability and the refutation and corroboration of cladistic hypotheses. Cladistics 13: 81–96.

———. 1998a. Total evidence or taxonomic congruence: cladistics or consensus classification. Cladistics 14:151–158.

———. 1998b. Sophisticated falsification and research cycles: consequences for differential character weighting in phylogenetic systematics. Zoologica Scripta 26:349–360.

Kritsky, D., E. P. Hoberg, and K. B. Aubry. 1993. *Lagarocotyle salamandrae* n. gen. n. sp. (Monogenoidea, Polyonchoinea, Lagarocotylidea n. ord.) from the cloaca of *Rhyacotriton cascadae* Good and Wake (Caudata, Rhyacotritonidae) in Washington State. Journal of Parasitology 79:322–330.

Kutz, S. J., E. P. Hoberg, and L. Polley. 2000. Experimental infections of muskoxen (*Ovibos moschatus*) and domestic sheep with *Umingmakstrongylus pallikuukensis* (Nematoda: Protostrongylidae): parasite development, population structure and pathology. Canadian Journal of Zoology 78: (In press.)

Lankester, M. W., and D. Fong. 1989. Distribution of elaphostrongyline nematodes (Metastrongyloidea: Protostrongylidae) in Cervidae and possible effects of moving *Rangifer* spp. into and within North America. Alces 25:133–145.

León-Règagnon, V. 1998. *Machidatrema* n. gen. (Digenea: Hemiuridae: Bunocotylinae) and phylogenetic analysis of its species. Journal of Parasitology 84:140–146.

———, **D. R. Brooks, and G. Pérez-Ponce de León.** 1999. Differentiation of Mexican species of *Haematoloechus* Looss, 1899 (Digenea: Plagiorchiformes): combining molecular and morphological evidence. Journal of Parasitology 85:935–946.

———, **G. Pérez-Ponce de León, and D. R. Brooks.** 1996. Phylogenetic analysis of *Opisthadena* (Digenea: Hemiuridae). Journal of Parasitology 82: 1005–1010.

———, ———, **and** ———. 1998. Phylogenetic analysis of the Bunocotylinae Dollfus, 1950 (Digenea: Hemiuridae). Journal of Parasitology 84: 147–152.

Lubchenco, J., A. M. Olson, L. B. Brubaker, S. R. Carpenter, M. M. Holland, S. P. Hubbell, S. A.

Levin, J. A. MacMahon, P. A. Matson, J. M. Melillo, H. A. Mooney, C. H. Peterson, H. R. Pulliam, L. A. Real, P. Regal, and P. G. Risser. 1991. The sustainable biosphere initiative: an ecological research agenda. Ecology 72:371–412.

Lyles, A. M., and A. P. Dobson. 1993. Infectious disease and intensive management: population dynamics, threatened hosts, and their parasites. Journal of Zoo and Wildlife Medicine 24:315–326.

Manter, H. 1966. Parasites of fishes as biological indicators of recent and ancient conditions. Pages 59–71 *in* J. E. McCauley, ed. Host Parasite Relationships. Oregon State University Press, Corvallis, Oregon.

Marcogliese, D. J. 1995. The role of zooplankton in the transmission of helminth parasites to fish. Reviews in Fish Biology and Fisheries 5:336–371.

———, **F. Boily, and M. O. Hammill.** 1996. Distribution and abundance of stomach nematodes (Anisakidae) among grey seals (*Halichoerus grypus*) and harp seals (*Phoca groenlandica*) in the Gulf of St. Lawrence. Canadian Journal of Fisheries and Aquatic Sciences 53:2829–2836.

———, **and D. K. Cone.** 1996. On the distribution and abundance of eel parasites in Nova Scotia: influence of pH. Journal of Parasitology 82:389–399.

———, **and** ———. 1997. Food webs: a plea for parasites. Trends in Ecology and Evolution 12:320–325.

———, **and J. Price.** 1997. The paradox of parasites. Global Biodiversity 7:7–15.

Mariaux, J. 1998. A molecular phylogeny of the Eucestoda. Journal of Parasitology 84:114–125.

Marques, F., R. Centritto, and S. A. Stewart. 1997. Two new species of *Acanthobothrium* in *Narcine entemedor* (Rajiformes: Narcinidae) from the northwest coast of Guanacaste Peninsula, Costa Rica. Journal of Parasitology 83:927–931.

Mayr, E. 1957. Evolutionary aspects of host specificity among parasites of vertebrates. Pages 7–14 *in* J.G. Baer, ed. Premier Symposium sur la Spécificité Parasitaire des Parasites de Vertébrés. International Union of Biological Sciences and Université Neuchâtel, Paul Attinger, S.A., Neuchâtel, Switzerland.

Miller, D. R., and A. Y. Rossman. 1995. Systematics, biodiversity, and agriculture. BioScience 45:680–686.

Minchella, D., and M. E. Scott. 1991. Parasitism: a cryptic determinant of animal community structure. Trends in Ecology and Evolution 6:250–254.

Monks, S., F. Marques, V. León-Règagnon, and G. Pérez-Ponce de León. 1997. *Koronacantha pectinarius* comb. n. (Acanthocephala: Illiosentidae) from *Microlepidotus brevipinnis* (Haemulidae) and redescription of *Tegorhynchus brevis.* Journal of Parasitology 83:485–494.

Morand, S. 1996a. Life history traits in parasitic nematodes: a comparative approach for the search of invariants. Functional Ecology 10:210–218.

———. 1996b. Biodiversity of parasites in relation to their lifecycles. Pages 243–260 *in* M. E. Hochberg, J. Clobert, and R. Barbault, eds. Aspects of Genesis and Maintenance of Biological Diversity. Oxford University Press, Oxford, U.K.

Morell, V. 1999. Are pathogens felling frogs? Science 284:728–731.

Neilsen, C., and K. A. Neiland. 1974. Sheep Disease Report. Volume 14. Alaska Department of Fish and Game, Juneau, Alaska. 104 pp.

Overstreet, R. M. 1997. Parasitological data as monitors of environmental health. Parassitologia 39: 169–175.

———, **W. E. Hawkins, and T. L. Deardorff.** 1996. The western mosquitofish as an environmental sentinel: parasites and histological lesions. Pages 495–508 *in* M. R. Servos, K. R. Munkittrick, J. H. Carey, and G. J. Van der Kraak, eds. Environmental Fate and Effects of Pulp and Paper Mill Effluents. St. Lucie Press, Delray Beach, Florida.

Parma, A. M. 1998. What can adaptive management do for our fish, forests, food, and biodiversity. Integrative Biology 1:17–26.

PCAST (President's Committee of Advisors on Science and Technology). 1998. Teaming With Life: Investing in Science to Understand and Use America's Living Capital. PCAST Panel on Biodiversity and Ecosystems, Washington, D.C. 86 pp.

Pérez-Ponce de León, G. 1997. La taxonomia en Mexico: el papel de la sistematica filogenetica. Ciencia 48:33–39.

———, **and D. R. Brooks.** 1995a. Phylogenetic relationships of the genera of the Pronocephalidae Looss, 1902 (Digenea: Paramphistomiformes). Journal of Parasitology 81:267–277.

———, **and** ———. 1995b. Phylogenetic relationships of *Pyelosomum* Looss, 1899 (Digenea: Pronocephalidae). Journal of Parasitology 81:278–280.

———, **V. León-Règagnon, and L. Garcia-Prieto.** 1997. Que es la sistematica filogenetica. Ciencia y Desarollo 135:61–65.

———, ———, **and B. Mendoza-Garfias.** 1997. Análisis filogenético de la familia Pterinotrematidae Bychowsky y Nagibina, 1959 (Platyhelminthes: Cercomerida: Monogenea). Annales del Instituto de Biología de la Universidad Nacional Autónoma de México, Serie Zoología 68:193–205.

———, ———, **and S. Monks.** 1998. *Theletrum lamothei* sp. nov. (Digenea) parasite of *Echidna nocturna* from Cuajiniquil, Guanacaste, and other digenes of marine fishes from Costa Rica. Revista de Biología Tropical 46:159–168.

Peterson, M. J. 1996. The endangered Attwater's prairie chicken and an analysis of prairie grouse helminthic parasitism. Ecogeography 19:424–431.

Pimentel, D., C. Wilson, C. McCullum, R. Huang, P. Owen, J. Flack, Q. Tran, T. Saltman, and B. Cliff. 1997. Economic and environmental benefits of biodiversity. Bioscience 47: 747–756.

Platt, T. R. 1984. Evolution of the Elaphostrongylinae (Nematoda: Metastrongyloidea: Protostrongylidae) parasites of cervids (Mammalia). Proceedings of the Helminthological Society of Washington 51:196–204.

———, **and D. R. Brooks.** 1997. Evolution of the

schistosomes. Journal of Parasitology 83: 1035–1044.

Poulin, R. 1992. Determinants of host specificity in parasites of freshwater fish. International Journal of Parasitology 22:753–758.

———. 1995a. Phylogeny, ecology, and richness of parasite communities in vertebrates. Ecological Monographs 65:283–302.

———. 1995b. Evolution of parasite life history traits: myths and reality. Parasitology Today 11: 342–345.

———. 1997a. Species richness of parasite assemblages: evolution and patterns. Annual Review of Ecology and Systematics 28:341–358.

———. 1997b. Evolutionary Ecology of Parasites. Chapman and Hall, New York. 212 pp.

Price, P. W. 1980. Evolutionary Biology of Parasites. Princeton University Press, Princeton, New Jersey. 237 pp.

———. 1986. Evolution in parasite communities. International Journal for Parasitology 17:209–214.

Queiroz, A. de, M. J. Donoghue, and J. Kim. 1995. Separate versus combined analysis of phylogenetic evidence. Annual Review of Ecology and Systematics 26:657–681.

Rausch, R. L. 1972. Tropical problems in the Arctic: infectious and parasitic diseases, a common denominator. Industry and Tropical Health 7:63–70.

Ricklefs, R. E. 1987. Community diversity: relative roles of local and regional processes. Science 235: 167–171.

Rohde, K. 1979. A critical evaluation of intrinsic and extrinsic factors responsible for niche restriction in parasites. American Naturalist 114:648–671.

Rózsa, L. 1992. Endangered parasite species. International Journal for Parasitology 22:265–266.

SA2000 (Systematics Agenda 2000). 1994. Systematics Agenda 2000: Charting the Biosphere. Technical Report. American Museum of Natural History, New York. 34 pp.

Samuel, W. M., M. J. Pybus, D. A. Welch, and C. J. Wilke. 1992. Elk as a potential host for meningeal worm: implications for translocation. Journal of Wildlife Management 56:629–639.

Sanderson, M. J., A. Purvis, and C. Henze. 1998. Phylogenetic supertrees: assembling the trees of life. Trends in Ecology and Evolution 13:105–109.

Sasal, P., Y. Desdevuises, and S. Morand. 1998. Host-specialization and species diversity in fish parasites: phylogenetic conservatism? Ecography 21:639–643.

———, **and S. Morand.** 1998. Comparative analysis: a tool for studying Monogenean ecology and evolution. International Journal for Parasitology 28: 1637–1644.

———, ———, **and J.-F. Guegan.** 1997. Parasite species richness for fish of the Mediterranean Sea. Marine Ecology Progress Series 149:61–71.

Savage, J. M. 1995. Systematics and the biodiversity crisis. BioScience 45:673–679.

Scholz, T., and H. Cappellaro. 1993. The first record of *Corallobothrium parafimbiratum* Befus et Freeman, 1973 (Cestoda: Proteocephalidea), a parasite of North American catfishes (*Ictalurus* spp.), in Europe. Folia Parasitologica 40:105–108.

Scott, M. E. 1988. The impact of infection and disease on animal populations: implications for conservation biology. Conservation Biology 2:40–56.

Simpson, B. B., and J. Cracraft. 1995. Systematics: the science of biodiversity. BioScience 45: 670–672.

Smith, F. D. M., R. M. May, R. Pellew, Y. H. Johnson, and K. R. Walker. 1993. How much do we know about the current extinction rate. Trends in Ecology and Evolution 8:375–378.

Soulé, M. E. 1991. Conservation: tactics for a constant crisis. Science 253:744–750.

Takacs, D. 1996. The Idea of Biodiversity, Philosophies of Paradise. Johns Hopkins University Press, Baltimore, Maryland. 393 pp.

Vitousek, P. M., C. M. D'Antonio, L. L. Loope, and R. Westbrooks. 1996. Biological invasions as global environmental change. American Scientist 84:468–478.

Walther, B. A., P. Cotgreave, R. D. Price, R. D. Gregory, and D. H. Clayton. 1995. Sampling effort and parasite species richness. Parasitology Today 11:306–310.

Williams, H. H., K. MacKenzie, and A. M. McCarthy. 1992. Parasites as biological indicators of the population biology, migrations, diet and phylogenetics of fish. Reviews in Fish Biology and Fisheries 2:144–176.

Wilson, E. O. 1984. Biophilia. Harvard University Press, Cambridge, Massachusetts. 176 pp.

———. 1988. The current state of biological diversity. Pages 1–18 *in* E. O. Wilson, ed. Biodiversity. National Academy Press, Washington, D. C.

———. 1992. The Diversity of Life. Harvard University Press, Cambridge, Massachusetts. 424 pp.

Windsor, D. A. 1995. Equal rights for parasites. Conservation Biology 9:1–2.

———. 1996. Stand up for parasites. Trends in Ecology and Evolution 12:32.

Woodford, M. H., and P. B. Rossiter. 1994. Disease risks associated with wildlife translocation projects. Pages 178–200 *in* P. J. S. Olney, G. M. Mace, and A. T. C. Feistner, eds. Creative Conservation: Interactive Management of Wild and Captive Animals. Chapman and Hall, London, U.K.

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Natural Occurrence of *Diplostomum* sp. (Digenea: Diplostomatidae) in Adult Mudpuppies and Bullfrog Tadpoles from the St. Lawrence River, Québec

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ABSTRACT: Adult mudpuppies (*Necturus maculosus*) and bullfrog tadpoles (*Rana catesbeiana*) infected with the eyefluke *Diplostomum* sp. in the lenses were collected from the St. Lawrence River, Québec, Canada. Respective prevalence and mean abundance of *Diplostomum* sp. were 100% and 3.1 ± 1.7 in Lake St. François, 58.3% and 1.5 ± 1.8 in Lake St. Louis, and 53.8% and 0.7 ± 0.8 in Lake St. Pierre. No eyeflukes were observed in mudpuppies from the Richelieu River. Prevalence and mean abundance of *Diplostomum* sp. were significantly higher in mudpuppies from Lake St. François than in those from other sites. The high prevalence and abundance in Lake St. François may be because the regulated water levels may enhance snail intermediate host habitats. There was a significant negative correlation between mudpuppy length and number of eyeflukes per host when samples were pooled from the 3 sites where *Diplostomum* sp. was found. Mean length of infected mudpuppies from those 3 sites was significantly smaller than uninfected ones. Twenty-four (28%) of 86 mudpuppies had cataracts associated with infections of eyeflukes. Prevalence and mean abundance of *Diplostomum* sp. in bullfrog tadpoles collected from Lake St. Pierre were 14.3% and 0.1 ± 0.4 parasite per animal, much lower than observed for mudpuppies from the same lake. Higher occurrence of eyeflukes in mudpuppies compared with tadpoles is attributed to the greater age and more sedentary benthic nature of mudpuppies. This is the first report of amphibians naturally infected with *Diplostomum* sp. and only the second with eyeflukes in general.

KEY WORDS: *Diplostomum* sp., eyefluke, *Necturus maculosus,* mudpuppy, *Rana catesbeiana,* tadpole, amphibians, prevalence, abundance, St. Lawrence River, Canada.

The eyefluke *Diplostomum spathaceum* (Rudolphi, 1819) (Digenea: Diplostomatidae) is among the most common parasites of freshwater fishes worldwide (Chappell et al., 1994) and infects more than 100 species of fish belonging to diverse taxa (Chappell, 1995). Diplostome metacercariae are the most important pathogens of the eyes of fish, cause blindness, and lead to poor growth, emaciation, and death (Williams and Jones, 1994; Chappell, 1995).

While the host spectrum of *D. spathaceum* is without question diverse, its actual extent beyond fishes is not clear. For example, Ferguson (1943) successfully infected tadpoles and adults of the northern leopard frog, *Rana pipiens* Schreber, 1782, in addition to painted turtles (*Chrysemys picta* (Schneider, 1783)), with metacercariae of *D. spathaceum.* Morphologically, worms appeared normal in these "abnormal" hosts. Development to adulthood occurred when worms from frogs were administered to chicks (Ferguson, 1943), indicating that amphibians and reptiles may be able to function as intermediate hosts. Sweeting (1974) successfully established infections of *D. spathaceum* in the African clawed frog, *Xenopus laevis* (Daudin, 1802), and observed what appeared to be normal development of metacercariae.

The occurrence of *D. spathaceum* in natural amphibian populations is not known. However, in Mountain Lake, Virginia, U.S.A., the red-spotted newt, *Notophthalmus viridescens* (Rafinesque, 1820), is naturally infected with the fish eyefluke *Tylodelphys scheuringi* (Hughes, 1929) in its humors (Etges, 1961). No frogs or other salamanders were found infected, and experimental infections of tadpoles and adult frogs were unsuccessful (Etges, 1961).

Infection levels of diplostomatid eyeflukes in fish from the St. Lawrence River are believed to be high, given the frequency of cataracts and blindness in fish from the river (Fournier et al.,

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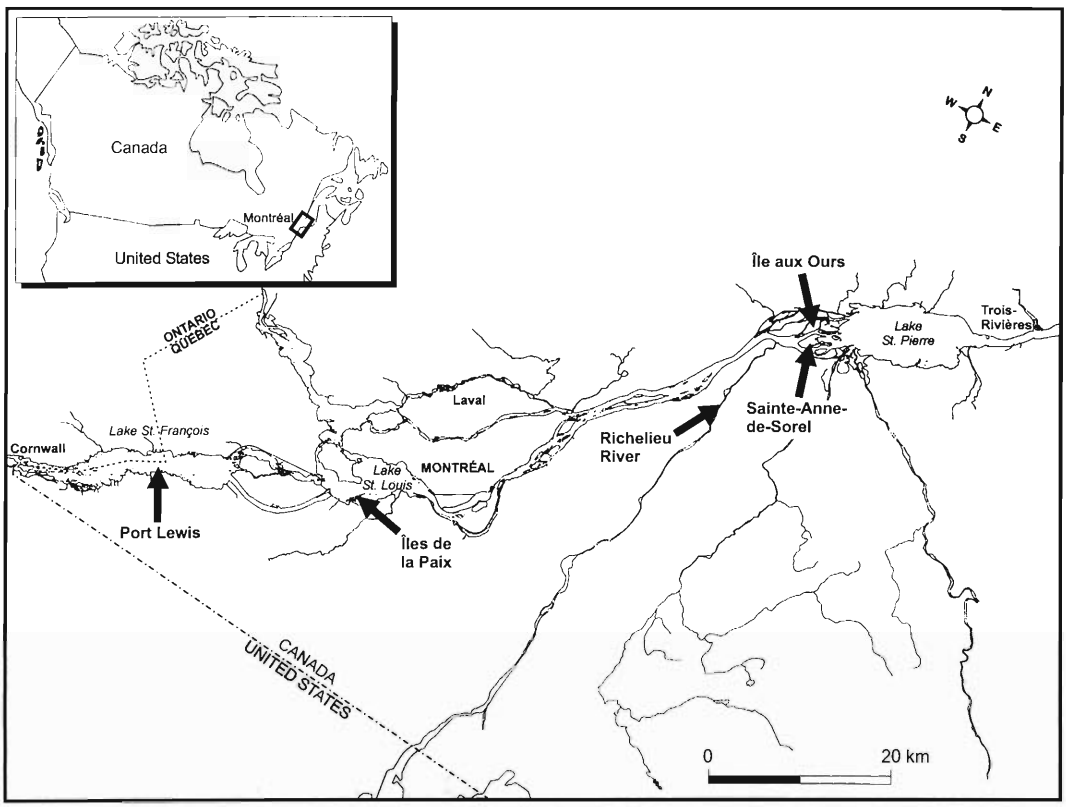

Figure 1. Map of the St. Lawrence River, Québec, Canada, depicting sampling localities and areas mentioned in the text. Adult mudpuppies (*Necturus maculosus*) were collected from Port Lewis, Îles de la Paix, Sainte-Anne-de-Sorel, and the Richelieu River during winter 1998. Bullfrog tadpoles (*Rana catesbeiana*) were collected from Île aux Ours in August 1998. Insert: Location of the sampling region is indicated on the map of Canada by a rectangle encompassing Montréal and the St. Lawrence River.

1996; Lair and Martineau, 1997; Mikaelian and Martineau, 1997). During a population study of mudpuppies, *Necturus maculosus* (Rafinesque, 1818), from the St. Lawrence River, we observed an individual with cataracts, and subsequently, an infection of *Diplostomum* sp. We then examined adult mudpuppies from 4 areas in the St. Lawrence River and 1 of its tributaries for eyeflukes. The mudpuppy is a long-lived, bottom-feeding, and strictly aquatic salamander, studied as a bioindicator of the St. Lawrence River (Bonin et al., 1995; Gendron et al., 1997). In addition, a single sample of bullfrog tadpoles (*Rana catesbeiana* Shaw, 1802) was collected from an area of high diplostome intensity (Marcogliese and Compagna, 1999) and examined for eyeflukes.

Materials and Methods

Mudpuppies were collected during a live trapping program with a small hoop net baited with dead fish placed at a depth of 1.5–2.5 m (Bonin et al., 1994) between the end of January and March 1998 from Port Lewis in Lake St. François (45°10′N; 74°17′W), Îles de la Paix in Lake St. Louis (45°20′N; 73°50′W), Sainte-Anne-de-Sorel in Lake St. Pierre (46°04′N; 73°03′W), and the Richelieu River (45°53′N; 73°09′W). The 3 lakes are formed from expansions of the St. Lawrence River, and the Richelieu River composes 1 of its tributaries (Fig. 1). Animals collected from Lake St. François (N = 36), Lake St. Louis (N = 27), and the Richelieu River (N = 23) were examined live for cataracts. Subsamples from these collections were examined directly for eyeflukes as follows. Animals from Lake St. François (N = 13) and the Richelieu River (N = 10) were transported live to the laboratory, where they were euthanized by cervical dislocation. The eyes were removed from the freshly killed animals, dissected, and examined with a stereomicroscope for parasites. Animals from Lake St. Louis (N = 12) were euthanized by cervical dislocation, fixed, and stored in 10% neutral buffered formalin, and their eyes removed, dissected, and examined with a stereomicroscope for parasites. No animals from Lake St. Pierre were examined for cataracts, but a sample

Table 1. Number (*N*), prevalence (P), and mean abundance (A ± SE) of *Diplostomum* sp. in the lenses of adult mudpuppies (*Necturus maculosus*) and bullfrog tadpoles (*Rana catesbeiana*) collected from localities in the St. Lawrence River and 1 of its tributaries in 1998.

	Lake St. François			Lake St. Louis			Lake St. Pierre			Richelieu River		
	N	P (%)	A ± SE	*N*	P (%)	A ± SE	*N*	P (%)	A ± SE	*N*	P (%)	A ± SE
Necturus maculosus	13	100	3.1 ± 1.7	12	58.3	1.5 ± 1.8	13	53.8	0.7 ± 0.8	10	0	0
Rana catesbeiana	—	—	—	—	—	—	35	14.3	0.1 ± 0.4	—	—	—

(N = 13) was collected and processed as that from Lake St. Louis.

Tadpoles (N = 35) of bullfrogs (*R. catesbeiana*) were collected from Île aux Ours in Lake St. Pierre (Fig. 1) using a beach seine measuring 22.6 m long by 1.15 m high, with a 3-mm mesh, in August 1998. Animals were euthanized by an overdose of anesthetic (MS 222), and their eyes were removed, dissected, and examined with a stereomicroscope for parasites.

Classification used herein adheres to that described by Gibson (1996). Metacercarial stages of diplostomes are difficult to identify to species, and resolution of the group's taxonomy is required before specimens can be assigned to species (Chappell, 1995; Gibson, 1996). In North America, metacercariae found in the lens of fish usually are considered to be *D. spathaceum*, and those in the vitreous humor to be other species, but these identifications must be regarded with caution (Gibson, 1996). The parasites found in this study correspond in terms of morphology and site within the host to *Diplostomum* sp. Experimental work where similar metacercariae were recovered from various fishes collected in the St. Lawrence River and fed to ring-billed gulls (*Larus delawarensis* Ord, 1815) suggests that 2 types of diplostomes occur in the river (*Diplostomum spathaceum indistinctum* (Guberlet, 1923) and *Diplostomum huronensis* (La Rue, 1927)) (J. D. McLaughlin, Concordia University, pers. comm.). The former typically occurs in the lens and the latter in the humor of the eyes of fish (Gibson, 1996). Unfortunately, the specific identification of eyeflukes from fishes and other vertebrates is problematic because most researchers working on surveys of these parasites do not have the capacity to rear these parasites in definitive hosts in the laboratory, especially when large numbers of metacercariae are involved. In addition, rearing metacercariae to adults in laboratory hosts such as chicks is problematic for 2 reasons. First, body dimensions of diplostomatid metacercariae can be affected by the species of host where they reside (Niewiadomska, 1987; Graczyk, 1991; Field and Irwin, 1995). Second, to minimize host-induced morphological variations, all hosts used by a parasite, including snails, fish, and birds, over the course of its life cycle must be identical (Field and Irwin, 1995). However, with metacercariae from wild-caught organisms such as fish or amphibians, it is often impossible to determine which snail hosts participated in their life cycles. For the various reasons listed above, numerous surveys simply record the parasites as *Diplostomum* sp. or *Diplostomulum* sp. (see Margolis and Arthur, 1979).

Data were not normally distributed and were compared using a nonparametric Kruskal-Wallis rank sum test, followed by pairwise comparisons using Tukey-Kramer HSD tests, using the JMP(TM) version 3.2.1 statistical package (SAS Institute, 1997). Statistical significance was set at a value of $P < 0.05$. Prevalence is defined as the proportion of animals infected in a sample, expressed as a percentage, and mean abundance is expressed as the mean number of parasites per host, infected and noninfected, in a sample (Bush et al., 1997).

Results

Adult mudpuppies from 3 of the 4 localities were infected with *Diplostomum* sp. in the lens of the eye. All animals from Lake St. François were infected, and abundance was significantly higher than at the other 3 localities (χ^2 = 24.65, df = 3, $P < 0.0001$) (Table 1). Prevalence was similar in mudpuppies from lakes St. Louis and St. Pierre, though mean abundance was higher in those from Lake St. Louis (Table 1). None of the mudpuppies examined from the Richelieu River was infected with eyeflukes. Multiple infections were much more common in mudpuppies from Lake St. François, with 10 of 13 animals having 2 or more parasites, and a maximum of 7 per host. In contrast, 5 of 12 from Lake St. Louis and 1 of 13 from Lake St. Pierre had 2 or more worms, with maxima of 6 and 3 worms per host, respectively. Prevalence of cataracts was 56% in mudpuppies from Lake St. François (N = 36), 11.1% in those from Lake St. Louis (N = 27), and 4.3% in those from the Richelieu River (N = 23). No mudpuppies from Lake St. Pierre were examined purposely for cataracts.

The correlation between total length of mudpuppies and total number of metacercariae per host was not significant ($P > 0.2$) at any of the 3 sites where *Diplostomum* sp. was found. There was a significant negative correlation between total number of parasites per host and total length of mudpuppies when animals were pooled from those 3 sites (r^2 = 0.303, P =

0.0003). Mean total length of infected hosts (246.0 ± 11.3 mm) was smaller than that of uninfected ones (306.2 ± 17.8 mm) ($\chi^2 = 6.88$; df = 1; $P = 0.0087$) when mudpuppies were pooled from the same 3 sites.

Five of 35 bullfrog tadpoles from Lake St. Pierre were infected, each with a single worm, giving a mean abundance of 0.1 ± 0.4 and a prevalence of 14.3% (Table 1).

Discussion

This is only the second report of amphibians from North American waters naturally infected with eyeflukes. Previously, red-spotted newts from Mountain Lake, Virginia, were found infected with *Tylodelphys scheuringi* at a prevalence of 100% (Etges, 1961). Adult mudpuppies and bullfrog tadpoles were infected with *Diplostomum* sp. at various localities in the St. Lawrence River. Mudpuppies were infected to a much greater degree than were tadpoles, probably due to their more sedentary benthic nature and their greater age. All mudpuppies collected were reproductive, making them at least 5 yr of age for males and 6 yr for females (Bonin et al., 1994). Among fishes, benthic species tend to be more heavily infected than pelagic ones. Cataracts are more prevalent in benthic fishes in the St. Lawrence River compared with pelagic foragers (Lair and Martineau, 1997). In addition, *D. spathaceum* metacercariae accumulate from year to year in hosts (Chappell et al., 1994), so older hosts tend to be more heavily infected. Benthic fish in the St. Lawrence River are more heavily infected than mudpuppies. Mean abundance of *Diplostomum* sp. in the white sucker (*Catostomus commersoni* (Lacépède, 1803)) aged 2–6 yr was 69.5 in Lake St. Louis and 22.0 in Lake St. Pierre, whereas in fish aged 7 yr or older, it was 167.0 and 62.9 in the 2 lakes, respectively (Marcogliese, unpubl.).

There is little information on geographic variation in infection levels within the St. Lawrence River system. In a survey of young-of-the-year fishes, no significant differences were found among sites (Marcogliese and Compagna, 1999), but among older fishes, infection levels were much higher in those from Lake St. Louis compared with Lake St. Pierre and near Québec City (Marcogliese, unpubl.). Data presented herein demonstrate that infection levels in mudpuppies from Lake St. François were significantly higher than in lakes St. Louis and St. Pierre. Moreover, there is a gradient in abundance declining downstream from west to east in the river. This cannot be directly correlated to the distribution of the definitive hosts, gulls and terns, as a large colony of ring-billed gulls consisting of 6156 pairs in 1997 is located near the sampling site in Lake St. Louis, but 3 larger colonies of ring-billed gulls, each consisting of more than 10,000 pairs, are situated downstream east of Lake St. Louis (P. Brousseau, Canadian Wildlife Service, pers. comm.). In addition, small colonies of common terns (*Sterna hirundo* Linnaeus, 1758), totaling 85 pairs in 1989, 108 pairs in 1997, and 138 pairs in 1997, as well as colonies of black terns (*Chlidonias niger* (Linnaeus, 1758)) occur in Lake St. François, Lake St. Louis, and Lake St. Pierre, respectively (Chapdelaine et al., 1999). Habitat in Lake St. François may be more suitable for the first intermediate hosts, lymnaeid snails. One important difference between Lake St. François and the other lakes is that water levels in this lake are heavily regulated, and do not fluctuate as much as in the other lakes. This stability may enhance snail populations and productivity. No worms were found in mudpuppies from the Richelieu River, although 1 mudpuppy was observed with cataracts. There is no information available on whether fish are infected with *Diplostomum* sp. in that river. Characteristics of that river may make it particularly unsuitable for the completion of the parasite's life cycle, in that definitive hosts or snail intermediate hosts are rare. There are no colonies of gulls or terns located on the river.

There was no relationship between body length and number of parasites among mudpuppies at any of the sites. When data were pooled from the 3 sites where *Diplostomum* sp. was found, there was a significant negative correlation between mudpuppy length and the number of parasites per host. Moreover, mean length of uninfected mudpuppies from those 3 sites was significantly greater than that of infected ones. These observations suggest that infections with *Diplostomum* sp. may be detrimental to mudpuppy growth, as was observed with infections in fish (Williams and Jones, 1994; Chappell, 1995). However, this conclusion may be premature. Our sample sizes are small. In addition, size of mudpuppies may be affected by pollution levels. For example, concentration of contaminants in mudpuppies varies with location in the

St. Lawrence River watershed (Bonin et al., 1995). Differences in mudpuppy size also could reflect some other aspect of habitat quality or age differences among the populations.

Prevalence of cataracts was high in Lake St. François, but extremely low in Lake St. Louis and the Richelieu River. This can be attributed to the higher prevalence and abundance of *Diplostomum* sp. in Lake St. François compared with the other sites. Yet, in both Lake St. François and Lake St. Louis, the prevalence of cataracts was much lower than the prevalence of eyeflukes. Thus, the presence of cataracts is not a reliable indicator of infection with eyeflukes, at least in mudpuppies. It is not known if the single mudpuppy possessing cataracts in the Richelieu River was infected with eyeflukes, or whether the cataracts resulted from another cause. Cataracts are caused by metacercariae, by dietary deficiency or excess, or by excessive exposure to sunlight, cold, or injury (Ferguson, 1989). In any case, the possibility of the presence of *Diplostomum* sp. in the Richelieu River cannot be dismissed.

The results demonstrate that animals other than fish become infected with metacercariae of *Diplostomum* sp. Given that amphibians develop cataracts (Ferguson, 1943; this study), concern for the health of aquatic fauna susceptible to blindness resulting from infection with eyeflukes must be extended beyond fish to include amphibians, especially in areas where *Diplostomum* sp. levels are high.

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Literature Cited

Bonin, J., J.-L. DesGranges, C. A. Bishop, J. Rodrigue, A. Gendron, and J. E. Elliott. 1995. Comparative study of contaminants in the mudpuppy (Amphibia) and the common snapping turtle (Reptilia), St. Lawrence River, Canada. Archives of Environmental Contamination and Toxicology 28:184–194.

———, ———, J. Rodrigue, and A. Gendron. 1994. Évaluation des possibilités d'utilisation du Necture tacheté (*Necturus maculosus*) comme bioindicateur de la pollution du fleuve Saint-Laurent. Série de rapports techniques no. 190. Service canadien de la faune, région du Québec. 58 pp.

Bush, A. O., K. D. Lafferty, J. M. Lotz, and A. W. Shostak. 1997. Parasitology meets ecology on its own terms: Margolis et al. revisited. Journal of Parasitology 83:575–583.

Chapdelaine, G., P. Brousseau, and J. F. Rail. 1999. Banque informatisée des oiseaux marins du Québec (BIOMQ). Service canadien de la faune, région du Québec, Environnement Canada.

Chappell, L. H. 1995. The biology of diplostomatid eyeflukes of fishes. Journal of Helminthology 69: 97–101.

———, L. J. Hardie, and C. J. Secombes. 1994. Diplostomiasis: the disease and host-parasite interactions. Pages 59–86 *in* A. W. Pike and J. W. Lewis, eds. Parasitic Diseases of Fish. Samara Publishing Ltd., Samara House, Tresaith, Dyfed, Great Britain.

Etges, F. J. 1961. Contributions to the life history of the brain fluke of newts and fish, *Diplostomulum scheuringi* Hughes, 1929 (Trematoda: Diplostomatidae). Journal of Parasitology 47:453–458.

Ferguson, H. W. 1989. Systematic Pathology of Fish. Iowa State University Press, Ames. 263 pp.

Ferguson, M. S. 1943. Development of eye flukes in the lenses of frogs, turtles, birds, and mammals. Journal of Parasitology 29:136–142.

Field, J. S., and S. W. B. Irwin. 1995. Life-cycle description and comparison of *Diplostomum spathaceum* (Rudolphi, 1819) and *D. pseudobaeri* (Razmaskin & Andrejak, 1978) from rainbow trout (*Oncorhynchus mykiss* Walbaum) maintained in identical hosts. Parasitology Research 81:505–517.

Fournier, D., F. Cotton, Y. Mailhot, D. Bourbeau, J. Leclerc, and P. Dumont. 1996. Rapport d'opération du réseau de suivi ichtyologique du fleuve Saint-Laurent: échantillonnage des communautés ichtyologiques des habitats lentiques du lac Saint-Pierre et de son archipel en 1995. Ministère de l'environnement et de la faune, Direction de la faune et des habitats, Direction régionale de la Mauricie-Bois-Francs, Direction régionale de la Montérégie. 59 pp.

Gendron, A. D., C. A. Bishop, R. Fortin, and A. Hontela. 1997. In vivo testing of the functional integrity of the corticosterone-producing axis in mudpuppy (Amphibia) exposed to chlorinated hydrocarbons in the wild. Environmental Toxicology and Chemistry 16:1694–1706.

Gibson, D. I. 1996. Guide to the Parasites of Fishes of Canada. Part IV. Trematoda. *in* L. Margolis and Z. Kabata, eds. NRC Research Press, Canadian Special Publication of Fisheries and Aquatic Sciences No. 124, Ottawa. 373 pp.

Graczyk, T. 1991. Variability of metacercariae of *Diplostomum spathaceum* (Rudolphi, 1819) (Trema-

toda, Diplostomidae). Acta Parasitologica Polonica 36:135–139.

Lair, S., and D. Martineau. 1997. Inventaire des conditions pathologiques chez les poissons du Saint-Laurent au site de Saint-Nicolas en 1994. Environnement Canada-Région du Québec, Conservation de l'environnement, Centre Saint-Laurent, Rapport scientifique et technique ST-140, Montréal. 72 pp.

Marcogliese, D. J., and S. Compagna. 1999. Diplostomatid eyeflukes in young-of-the-year and forage fishes in the St. Lawrence River, Québec. Journal of Aquatic Animal Health 11:275–282.

Margolis, L., and J. R. Arthur. 1979. Synopsis of the parasites of fishes of Canada. Bulletin of the Fisheries Research Board of Canada No. 199, Ottawa. 269 pp.

Mikaelian, I., and D. Martineau. 1997. Inventaire des conditions pathologiques chez les poissons du Saint-Laurent au site de Saint-Nicolas en 1995. Environnement Canada-Région du Québec, Conservation de l'Environnement, Centre Saint-Laurent, Rapport scientifique et technique ST-141, Montréal. 57 pp.

Niewiadomska, K. 1987. *Diplostomum paracaudum* (Iles, 1959) Shigin, 1977 (Digenea, Diplostomidae) and its larval stages—a new record from Poland. Acta Parasitologica Polonica 31:199–210.

SAS Institute. 1997. JMP™ Statistical Discovery Software, Version 3.2.1. Cary, North Carolina.

Sweeting, R. A. 1974. Investigations into natural and experimental infections of freshwater fish by the common eye-fluke *Diplostomum spathaceum* Rud. Parasitology 69:291–300.

Williams, H., and A. Jones. 1994. Parasitic Worms of Fish. Taylor & Francis Ltd., London. 593 pp.

Obituary Notice

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Assessment of Parenteral *Plagiorhynchus cylindraceus* (Acanthocephala) Infections in Shrews

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ABSTRACT: *Plagiorhynchus cylindraceus*, a common acanthocephalan parasite of passerine birds, does not require a paratenic host for completion of the life cycle, but extraintestinal (parenteral) infections do occur in short-tailed shrews (*Blarina brevicauda*). Examination of wild mammals trapped at 13 sites in and around Lincoln, Nebraska, U.S.A., revealed infections in short-tailed shrews and a masked shrew (*Sorex cinereus*) but not in any other species of mammals collected. Laboratory exposures of *B. brevicauda* and 5 other mammalian species that co-occur with short-tailed shrews at sites where shrews harbor extraintestinal *P. cylindraceus* infections resulted in infections only in short-tailed shrews and a single deer mouse (*Peromyscus maniculatus*). A cystacanth obtained from the mesentery of 1 of these shrews was infective when fed to a robin (*Turdus migratorius*), the usual definitive host. Intestinal histology and susceptibility of *P. maniculatus* to laboratory infections suggest that the absence of parenteral infections in mammals other than shrews is due to ecological circumstances rather than physiological or anatomical constraints. Laboratory exposures of 3 species of isopods and a survey of isopods collected from a site where infected shrews occur failed to reveal any species susceptible to *P. cylindraceus* other than the only known intermediate host, the terrestrial isopod *Armadillidium vulgare*. An analysis of the literature regarding diets and the fact that deer mice did not prey on *A. vulgare* in laboratory feeding trials suggest that other mammals co-occurring with shrews are unlikely to consume the intermediate host of *P. cylindraceus*.

KEY WORDS: *Plagiorhynchus cylindraceus*, Acanthocephala, cystacanths, shrews, *Blarina brevicauda*, extraintestinal infection, experimental infection, robin, *Turdus migratorius*, Nebraska, U.S.A.

As adults, acanthocephalans occur in the intestinal lumen of vertebrate definitive hosts; larvae develop in the hemocoel of arthropod intermediate hosts. Ingestion of the infected intermediate host by the definitive host completes the life cycle. Larval acanthocephalans of many species also occur as parenteral (extraintestinal) infections in the viscera of vertebrate hosts, but sexual maturity is not attained in these paratenic hosts. Paratenic hosts facilitate distribution across gaps in trophic levels between the intermediate host and predatory definitive hosts high in the food chain. Ewald et al. (1991) implicated 2 species of *Sorex* in the life cycle of *Centrorhynchus aluconis* (Müller, 1780) Lühe, 1911, which attains maturity in owls. Elkins and Nickol (1983) demonstrated that infection of raccoons, *Procyon lotor* (Linnaeus, 1758) Storr, 1780, with *Macracanthorhynchus ingens* (Linstow, 1789) Meyer, 1932, can be accomplished by ingestion of cystacanths occurring in mesenteries of green water snakes, *Nerodia cyclopion* (Dumeril, Bibron, and Dumeril, 1854) Rossman and Eberle, 1977. Several instances of paratenic hosts being incorporated into life cycles of acanthocephalans that infect piscivorous fish have been documented (Hasan and Qasim, 1960; Paperna and Zwerner, 1976).

Although it is clear that paratenic hosts play an important role in transmission of many acanthocephalans, the role of parenteral infections often is not explained easily by predator–prey relationships. Cystacanths frequently occur in hosts from which transmission is unlikely or impossible. For example, *M. ingens* occurs extraintestinally in armadillos (Radomski et al., 1991), in addition to water snakes, and *Mediorhynchus grandis* Van Cleave, 1916, a parasite of nonpredatory birds (usually icterids), can occur parenterally in shrews (Collins, 1971).

Adults of the acanthocephalan species *Plagiorhynchus cylindraceus* (Goeze, 1782) Schmidt and Kuntz, 1966, occur in passerine birds, especially robins (*Turdus migratorius* Linnaeus, 1766) and starlings (*Sturnus vulgaris* Linnaeus, 1758). Isopods are infected by ingesting parasite eggs passed from birds, and infective larvae develop in the hemocoel of *Armadillidium vulgare*

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(Latreille, 1804) Brandt and Ratzeburg, 1831, a terrestrial isopod (Schmidt and Olsen, 1964; Nickol and Dappen, 1982). Cystacanths of this acanthocephalan species also have been reported (Nickol and Oetinger, 1968) from the mesenteries of short-tailed shrews, *Blarina brevicauda* (Say, 1823) Baird, 1858, in New York state.

When cystacanths of *P. cylindraceus* were discovered in the viscera of short-tailed shrews of eastern Nebraska, a study to assess the significance of these parenteral forms was undertaken. The distribution of extra-intestinal forms among shrews and other co-occurring mammals was determined, infectivity of isopod-borne cystacanths to shrews and other co-occurring mammals was studied, and infectivity of mammal-borne cystacanths to robins was tested.

Materials and Methods

Acquisition and maintenance of *P. cylindraceus*

Gravid female worms obtained from robins and starlings in Lancaster County, Nebraska, were stored a maximum of 3 mo in tap water at 4 C. To infect isopods, egg suspensions were prepared by pulverizing stored worms in tap water. Each suspension was examined microscopically to ensure the presence of fully developed eggs.

A laboratory colony of isopods (*A. vulgare*) was maintained in covered plastic containers (32.5 × 17.5 × 9.0 cm) provided with 2 to 3 cm of soil, pieces of broken clay pots for shelter, a sponge moistened regularly to maintain humidity, and potato slices for food. Large pieces of potato were used to maintain humidity in some containers in place of the moistened sponge. These pieces of potato were allowed to sprout, and isopods were observed feeding regularly on the shoots as well as the potato itself.

To obtain laboratory-reared cystacanths, isopods less than 9.5 mm long (see Nickol and Dappen, 1982) were removed from the colony and held without food for 36 hr, after which they were allowed to feed on potato slices over which a suspension of *P. cylindraceus* eggs in water had been spread. Exposure was in covered wells (3.5 cm diameter × 1.0 cm deep) imprinted on a plastic plate. Fresh egg suspension was added to the potato slices after 24 hr. Except to add egg suspension, isopods were left undisturbed in the dark. After 36 to 48 hr of exposure, isopods were removed and isolated in a separate culture. Before use, cystacanths were allowed to develop at least 70 days in the isopods to ensure infectivity (Schmidt and Olsen, 1964).

Survey of mammals

Mammals at 13 sites located within 4 townships (North Bluff, Oak, West Lincoln, and Yankee Hill) in and around Lincoln, Nebraska, were surveyed to determine locations at which parenteral infections occur and to determine which species harbor cystacanths in nature. Mammals were trapped with medium-sized Sherman live traps baited with a mixture of peanut butter and oats, and all mammals caught were examined for *P. cylindraceus* cystacanths.

Laboratory exposure of mammals

To determine susceptibility to *P. cylindraceus* cystacanths, mammals of 6 species, collected at sites from which *P. cylindraceus* was absent in previous surveys, were administered cystacanths orally. The mammal species exposed were short-tailed shrews; European mice, *Mus musculus* Linnaeus, 1758; hispid pocket mice, *Perognathus hispidus* Baird, 1858; wood mice, *Peromyscus leucopus* (Rafinesque, 1818) Thomas, 1895; deer mice, *Peromyscus maniculatus* (Wagner, 1845) Bangs, 1898; and 13-lined ground squirrels, *Spermophilus tridecemlineatus* Mitchill, 1821.

To expose mammals, laboratory-reared cystacanths were pipetted to the back of the throat of lightly anesthetized (methoxyflurane) animals. Following exposure, each animal was placed into a receptacle lined with clean, tan-colored paper toweling for observation and recovery. After the animal recovered from anesthesia, it was returned to its normal housing. The recovery receptacle then was examined for cystacanths that were not ingested by the animal. The white cystacanths were highly visible on the paper towels, making possible an accurate determination of the number administered.

Mammals were housed in standard mouse cages fitted with wire tops, water bottles, paper towels for nesting and shelter, and wood shavings. All nonsoricids were fed commercial hamster and gerbil food and observed to ensure that they were eating. Short-tailed shrews were provided additionally with a block of untreated wood (5 × 10 × 15–25 mm) and a small clay flower pot. The wood absorbed excess oil from the shrew's fur and provided shelter. The shrews deposited feces regularly within the flower pots, which were removed easily and cleaned. Shrews were fed 5 adult cockroaches, *Periplaneta americana* (Linnaeus, 1758) Burmeister, 1838, from a laboratory colony at each of 3 daily feedings.

Survey and susceptibility of isopods

A survey was conducted to determine what isopod species inhabit sites from which the infection was determined to be present in shrews. Isopods were collected by hand for 1 hr at night by flashlight, identified, and examined for cystacanths.

To investigate the extent of intermediate host specificity, isopods of 3 terrestrial species (*Armadillidium nasatum* Budde-Lund, 1885, *A. vulgare,* and *Metoponorthus pruinosus* (Brandt, 1833) Budde-Lund, 1879) were exposed to eggs of *P. cylindraceus.* All of these isopods were collected within Lancaster County, Nebraska.

Peromyscus feeding trials

Deer mice (*P. maniculatus*) were offered isopods (*A. vulgare*) as prey to determine the likelihood of their consuming an intermediate host in nature. After having food withheld for 4 hr, each of 6 deer mice was presented 20 isopods of assorted sizes for a period of 2 hr in 10-gallon aquaria. The bottom of each aquarium

Table 1. Number (*N*) of wild-caught mammals examined and prevalence (percentage infected [% Inf]) and mean intensity (Mean int)* of parenteral infections by *Plagiorhynchus cylindraceus*.

	All sites surveyed		Infection-free sites	Infection-present sites		
Species examined	*N*	% Inf	*N*	*N*	% Inf	Mean int
Lipotyphyla						
Blarina brevicauda	27	37	10	17	59	3.8
Sorex cinereus	9	11	4	5	20	2.0
Rodentia						
Microtus ochrogaster	16	0	8	8	0	—
Microtus pennsylvanicus	48	0	10	38	0	—
Mus musculus	25	0	4	21	0	—
Perognathus flavescens	3	0	3	0	—	—
Peromyscus leucopus	72	0	40	32	0	—
Peromyscus maniculatus	55	0	34	21	0	—
Reithrodontomys megalotis	10	0	6	4	0	—
Spermophilus tridecemlineatus	4	0	3	1	0	—
Carnivora						
Mustela nivalis	6	0	3	3	0	—

* Number of worms/number of infected shrews.

was covered by heavy paper with all edges taped down to prevent isopods from hiding. Water was available to the mice for the duration of the trial. The room housing the aquaria was left undisturbed in the dark for 2 hr, after which the deer mice were removed and the remaining isopods counted.

Measurements of external muscularis

To determine whether thickness of intestinal muscle could account for differences in susceptibility, the duodenum of each of 3 short-tailed shrews, meadow voles (*Microtus pennsylvanicus* (Ord, 1815) Rhoads, 1895), and deer mice was removed and fixed in neutral buffered 10% formalin. Tissues were imbedded in paraffin, cut in 6-μm cross-sections, stained with hematoxylin and eosin, and examined with light microscopy. The thickness of the external muscularis was measured in micrometers at 4 points along the intestine, with 2 measurements taken directly opposite on the cross-section at each point. Differences in thickness among species were sought by 1-way analysis of variance (ANOVA) of the resulting 24 measurements for each species.

Infectivity of mammal-borne cystacanths

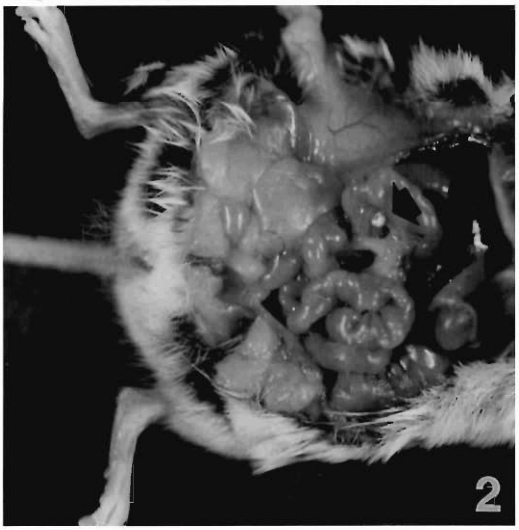
The infectivity of mammal-borne cystacanths was tested with laboratory exposures to robins. Robins were collected with mist nets (U.S. permit PRT-694828 and Nebraska permit 96–2) and housed in the laboratory where they were given food (Blankespoor, 1970) and water ad libitum. The robins were held for 3 wk for acclimation to captivity and to ensure that any worms naturally present would be old enough to distinguish from those fed in the trial. Cystacanths obtained from extraintestinal sites in mammals were pipetted directly into the esophagus of each bird to be exposed.

Results

Two hundred seventy-five mammals were collected at the 13 sites surveyed. Parenteral infections of *P. cylindraceus* occurred in 11 animals of 2 species. All infected animals were collected at either of 2 sites. Ten mammalian species were examined from these 2 sites, but *P. cylindraceus* was present only in short-tailed shrews and a masked shrew (Table 1). Ten of 17 short-tailed shrews were infected with 1 to 11 (mean intensity 3.8) extraintestinal cystacanths. The cystacanths were not found consistently in any specific location within the abdominal cavity; however, several had migrated through the peritoneal cavity and had extended their proboscides into abdominal muscle tissue. Some of these worms appeared vital and little changed from infective cystacanths found in isopods. Others were heavily encapsulated and appeared moribund. One of 5 masked shrews harbored 2 cystacanths, 1 encysted in the mesentery of the small intestine and the other unattached in the lumen of the small intestine.

Each of 27 mammals of 6 species was fed 5 to 16 cystacanths in the laboratory. Of the 12 short-tailed shrews fed, 7 cystacanths were recovered from the viscera of 3 (Fig. 1); 1 cystacanth was recovered from 1 of 7 deer mice (Fig. 2); and no other animal became infected (Table 2).

1



2

Figures 1, 2. Photographs of *Plagiorhynchus cylindraceus* cystacanths in viscera of laboratory-infected mammals. 1. Cystacanth (arrow) 3 days after infection in a short-tailed shrew, *Blarina brevicauda*. 2. Cystacanth (arrow) 14 days after infection in a deer mouse, *Peromyscus maniculatus*.

Two species of terrestrial isopods, *Trachelipus rathkei* (Brandt, 1833) Buddle-Lund, 1908 (n = 62) and *A. vulgare* (n = 2), were collected at a site from which infected shrews were obtained. None of the isopods was infected. To learn more about the susceptibility of isopods, 3 species of terrestrial isopod were exposed to *P. cylindraceus* eggs in the laboratory. Eighty percent of the exposed *A. vulgare* became infected (mean intensity = 2.98), but cystacanths were absent from all isopods of the other 2 species (Table 3). None of the isopods (*A. vulgare*) offered to 6 deer mice as food was consumed.

Measurement of the external muscularis of the duodenum revealed a mean thickness of 80 μm for short-tailed shrews, 72 μm for meadow voles, and 42 μm for deer mice (Table 4).

Two days after exposure, 1 of 3 robins that were fed cystacanths (4, 2, and 1 cystacanths) obtained from parenteral sites in laboratory-infected shrews harbored a 6-mm-long *P. cylindraceus* cystacanth. The other birds were uninfected.

Discussion

Presence at only 2 of 13 sites surveyed suggests that distribution of parenteral *P. cylindraceus* infections in mammals is highly localized within a broader geographical range of occurrence. The type of habitat does not appear to be the restricting factor, as the locations at which infections were present resembled infection-free sites more closely than each other. One of the infection sites is dry with thin cover and a flat terrain. The second infection site has moist soil with a thick cover of lush vegetation and a steep grade. The infection was absent from other sites surveyed that were similar to the infection sites.

In addition to having localized occurrence, parenteral *P. cylindraceus* infections appear to be restricted to certain individuals of the susceptible species. The laboratory infection of a deer mouse suggests that mammals other than shrews are susceptible. However, natural infections were found only in shrews even though mammals of other species, including deer mice, at the infection sites were examined. The thickness of the intestinal wall to be penetrated by a cystacanth for establishment of an extraintestinal infection does not seem to explain the restriction of hosts. Our measurements contained considerable variation and were from wild-caught animals, leaving several unaccountable variables, e.g., age and distention with chyme. Nevertheless, the 8 measurements from each of 3 animals of each of 3 species form a consistent pattern and ANOVA revealed a significant difference ($P < 0.01$) among the species. If our measurements are properly representative, short-tailed shrews possess a thicker external muscularis than do some uninfected species, e.g., deer mice and meadow voles.

Table 2. Occurrence of *Plagiorhynchus cylindraceus* in laboratory-exposed mammals.

Species exposed	Number of cystacanths administered	*N**	Number of cystacanths recovered
Lipotyphla			
Blarina brevicauda	16	1	0
	11	4	0, 0, 0, 5
	9	1	0
	7	4	0, 0, 0, 1
	5	2	0, 1
Rodentia			
Mus musculus	10	3	0, 0, 0
Perognathus hispidus	10	1	0
Peromyscus leucopus	10	6	0, 0, 0, 0, 0, 0
Peromyscus maniculatus	7	3	0, 0, 1
Spermophilus tridecemlineatus	10	2	0, 0

* Number of animals receiving the dose.

The restriction of infections to certain individuals is more likely because of interactions among birds, isopods, and these mammals than to inherent susceptibility or anatomical obstacles. Voles rarely use arthropods as prey (Rose and Birney, 1985). Deer mice and wood mice do so slightly more frequently (Hamilton, 1941; Whitaker and Ferraro, 1963). Because of the inclusion of arthropods, albeit rarely, in the diet of these animals, an occasional infection might be expected. Shrews, however, consume arthropods more commonly (Table 5) and, therefore, probably are exposed to infective cystacanths more frequently.

Interspecific differences among mammals that consume isopods might play a role in further limiting the distribution of *P. cylindraceus*. Apparently, *A. vulgare* is the only intermediate host for *P. cylindraceus*. It is the only species of isopod known to be infected in nature (Schmidt and Olsen, 1964), and examination of 62 *T. rathkei* collected from a site at which shrews were infected revealed no infection. Laboratory exposures of isopods of 2 additional local species (*A. nasatum* and *M. pruinosus*) failed to produce any cystacanths, whereas isopods of the species *A. vulgare* were infected readily. Porcellionid isopods are soft bodied and, according to Sutton (1980), cannot roll into a protective ball, whereas the exoskeleton of isopods belonging to the family Armadillidiidae is harder, and according to Sutton (1980), these isopods do roll up into a protective ball. Even after having food withheld for 2 hr, deer mice did not eat isopods (*A. vulgare*) offered in the laboratory feeding trial. This suggests that the isopod materials identified in dietary studies (Table 5) were not remains of infected isopods.

Table 3. Occurrence of *Plagiorhynchus cylindaceus* cystacanths in laboratory-exposed isopods.

Species exposed	Number examined	Number (%) infected	Intensity	
			Mean	Maximum
Armadillidium vulgarae				
Trial 1	50	38 (76)	3.03	10
Trial 2	20	18 (90)	2.89	7
Combined	70	56 (80)	2.98	10
Armadillidium nasatum				
Trial 1	22	0	—	—
Trial 2	16	0	—	—
Combined	38	0	—	—
Metoponorthus pruinosus				
Trial 1	7	0	—	—
Trial 2	10	0	—	—
Combined	17	0	—	—

Table 4. Thickness (micrometers) of external muscularis of 3 mammalian species.*

Species	Animal			Mean (SD)
	1	2	3	
Blarina brevicauda	89.37	58.12	92.50	80.00 (17.80)
Microtus pennsylvanicus	66.13	59.06	91.94	72.38 (17.30)
Peromyscus maniculatus	38.10	32.50	54.00	41.53 (11.15)

* Measurements were made for 3 animals of each species. For each animal, the thickness given is the mean of 8 measurements (2 measurements directly opposite each other at 4 points along the duodenum). Differences among species were significant ($P < 0.01$) by 1-way analysis of variance of the 24 individual measurements for each species.

The establishment in the intestine of a robin by a cystacanth removed from the viscera of a laboratory-exposed shrew demonstrates that parenteral cystacanths from mammals can be infective to definitive hosts. An infective cystacanth from an intermediate host is 3.0 to 4.4 mm long (Schmidt and Olsen, 1964), and parenteral cystacanths from our laboratory-infected shrews were 3.5 to 4.2 mm long. The worm recovered from the laboratory-infected robin measured 6.0 mm in length. Such growth during 2 days in the bird's intestine indicates successful establishment.

Despite the infectivity of extraintestinal cystacanths and occasional reports of passerine birds eating shrews and other small mammals (Powers, 1973; Penny and Knapton, 1977) we conclude that paratenic hosts are not important to *P. cylindraceus* populations. Comprehensive studies fail to identify small mammals as an important, or even minor, part of these birds' diets (Paszkowski, 1982; Wheelwright, 1986). Likewise, there is little evidence that paratenic hosts have played any meaningful role in facilitating wider host distribution for *P. cylindraceus*. There is little question that raptors and other flesh-eating birds could consume *P. cylindraceus* cystacanths (see Audubon, 1937, Plate 374). Dollfus and Golvan (1961) listed *Buteo buteo* Linnaeus, 1758, as a host for *P. cylindraceus,* and Ewald and Crompton (1993) found it in *Strix aluco* Linnaeus, 1758. Neither report, however, gives an indication of whether the worms reach maturity and produce eggs in those birds. Additionally, *P. cylindraceus* has been reported from several species of Corvidae. Rutkowska (1973) described eggs from females harbored by 1 of 500 jackdaws, *Coloeus monedula* = *Corvus monedula* Linnaeus, 1758, examined in Poland, but the other 8 records from corvids (Jones, 1928; Pemberton, 1961; Williams, 1961; Threlfall, 1965; Todd et al., 1967; Hendricks et

Table 5. Inclusion of isopods as prey by mammals that co-occurred at collection sites where *Plagiorhynchus cylindraceus* was present.

Species	Frequency*	Volume of diet (%)	Reference
Lipotyphyla			
Blarina brevicauda	3.7	No report	Hamilton, 1941
	6.7	1.4	Whitaker and Ferraro, 1963
	1.6	1.6	Whitaker and Mumford, 1972
Cryptotis parva	2.8	1.9	Whitaker and Mumford, 1972
Sorex cinereus	4.0	2.1	Whitaker and Mumford, 1972
Rodentia			
Microtus ochrogastor	0.0	—	Zimmerman, 1965
Microtus pennsylvanicus	0.0	—	Zimmerman, 1965
Mus musculus	0.0	—	Whitaker, 1966
Peromyscus leucopus	1.6	1.7	Whitaker and Ferraro, 1963
	0.0	—	Whitaker, 1966
Peromyscus maniculatus	2.0	No report	Hamilton, 1941
	0.0	—	Whitaker, 1966

* Percentage of animals with isopods present in diet.

al., 1969; Andrews and Threlfall, 1975; Lisitsyna, 1993) either report juveniles or give no indication of maturity.

Acanthocephalans belonging to all taxonomic classes and 6 of the 8 orders of the phylum have been reported as extraintestinal infections in vertebrates. Such parenteral infections are results of either specific adaptations serving to enhance transmission or historical events not currently maintained by natural selection. It is probable that this trait in *P. cylindraceus,* and perhaps in other species that occur parenterally in hosts from which transmission to a definitive host is impossible or unlikely, is a result of inheritance from an ancestor in which it might have had a selective advantage, rather than being an adaptation shaped by current selective forces.

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Literature Cited

Andrews, S. E., and W. Threlfall. 1975. Parasites of the common crow (*Corvus brachyrhynchos* Brehm, 1822) in insular Newfoundland. Proceedings of the Helminthological Society of Washington 42:24–28.

Audubon, J. J. 1937. The Birds of America. Macmillan Co., New York, New York, Plate 373.

Blankespoor, H. D. 1970. Host-parasite relationships of an avian trematode *Plagiorchis noblei* (Park, 1934). Ph.D. Thesis. Iowa State University, Ames, 180 pp.

Collins, G. D. 1971. *Mediorhynchus grandis* in a short-tailed shrew, *Blarina brevicauda,* from South Dakota. Journal of Parasitology 57:1038.

Dollfus, R. P., and Y. Golvan. 1961. Station Experimentale de Parasitologie de Richelieu (Indre-et-Loire). Contribution à la fauna parasitaire regionale. G. Acanthocephales. Annales de Parasitologie 36:314–322.

Elkins, C. A., and B. B. Nickol. 1983. The epizootiology of *Macracanthorhynchus ingens* in Louisiana. Journal of Parasitology 69:951–956.

Ewald, J. A., and D. W. T. Crompton. 1993. *Centrorhynchus aluconis* (Acanthocephala) and other helminth species in tawny owls (*Strix aluco*) in Great Britain. Journal of Parasitology 79:952–954.

———, ———, **I. Johnson, and R. C. Stoddart.** 1991. The occurrence of *Centrorhynchus* (Acanthocephala) in shrews (*Sorex araneus* and *Sorex minutus*) in the United Kingdom. Journal of Parasitology 77:485–487.

Hamilton, W. J. 1941. The food of small forest mammals in eastern United States. Journal of Mammalogy 21:250–263.

Hasan, R., and S. Z. Qasim. 1960. The occurrence of *Pallisentis basiri* Farooqi (Acanthocephala) in the liver of *Trichogaster chuna* (Ham.). Zeitschrift für Parasitenkunde 20:152–156.

Hendricks, L. D., R. Harkema, and G. C. Miller. 1969. Helminths of the crow, *Corvus brachyrhynchos* Brehm, 1822, in North Carolina. Proceedings of the Helminthological Society of Washington 36:150–152.

Jones, M. 1928. An acanthocephalid, *Plagiorhynchus formosus,* from the chicken and the robin. Journal of Agricultural Research 36:773–775.

Lisitsyna, O. I. 1993. Life cycle of *Prosthorhynchus cylindraceus* (Acanthocephala, Plagiorhynchidae). Vestnik Zoologii 1993:43–48. (In Russian.)

Nickol, B. B., and G. E. Dappen. 1982. *Armadillidium vulgare* (Isopoda) as an intermediate host of *Plagiorhynchus cylindraceus* (Acanthocephala) and isopod response to infection. Journal of Parasitology 68:570–575.

———, **and D. F. Oetinger.** 1968. *Prosthorhynchus formosus* from the short-tailed shrew (*Blarina brevicauda*) in New York state. Journal of Parasitology 54:456.

Paperna, I., and D. E. Zwerner. 1976. Parasites and diseases of striped bass, *Morone saxatilis* (Walbaum), from the lower Chesapeake Bay. Journal of Fish Biology 9:267–287.

Paszkowski, C. A. 1982. Vegetation, ground, and frugivorous foraging of the American robin. Auk 99: 701–709.

Pemberton, R. T. 1961. Helminth parasites of some British birds. Annals and Magazine of Natural History (Series 13) 3:455–463.

Penny, C., and R. W. Knapton. 1977. Record of an American robin killing a shrew. Canadian Field Naturalist 91:393.

Powers, L. R. 1973. Record of a robin feeding shrews to its nestlings. Condor 75:248.

Radomski, A. A., D. A. Osborn, D. B. Pence, M. I. Nelson, and R. J. Warren. 1991. Visceral helminths from an expanding insular population of the long-nosed armadillo (*Dasypus novemcinctus*). Journal of the Helminthological Society of Washington 58:1–6.

Rose, R. K., and E. C. Birney. 1985. Community ecology. Pages 310–339 *in* R. H. Tamarin, ed. Biology of New World *Microtus.* American Society of Mammalogists, Shippinsburg, Pennsylvania.

Rutkowska, M. A. 1973. A study of the helminth fauna of Corvidae in Poland. Acta Parasitologica Polonica 21:183–237.

Schmidt, G. D., and O. W. Olsen. 1964. Life cycle and development of *Prosthorhynchus formosus* (Van Cleave, 1918) Travassos, 1926, an acanthocephalan parasite of birds. Journal of Parasitology 50:721–730.

Sutton, S. L. 1980. Woodlice. Pergamon Press, Oxford, United Kingdom, 144 pp.

Threlfall, W. 1965. Helminth parasites and possible causes of death of some birds. Ibis 107:545–548.

Todd, K. S., J. V. Ernst, and D. M. Hammond. 1967. Parasites of the black-billed magpie, *Pica pica hudsonia* (Sabine, 1823) from northern Utah. Bulletin of the Wildlife Disease Association 3: 112–113.

Wheelwright, N. T. 1986. The diet of American robins. An analysis of U.S. Biological Survey records. Auk 103:710–725.

Whitaker, J. O., Jr. 1966. Food of *Mus musculus, Peromyscus maniculatus bairdi* and *Peromyscus leucopus* in Vigo County, Indiana. Journal of Mammalogy 47:473–486.

———, **and M. G. Ferraro.** 1963. Summer food of 220 short-tailed shrews from Ithaca, New York. Journal of Mammalogy 44:419.

———, **and R. E. Mumford.** 1972. Food and ectoparasites of Indiana shrews. Journal of Mammalogy 53:329–335.

Williams, I. C. 1961. A list of parasitic worms, including twenty-five new records from British birds. Annals and Magazine of Natural History (Series 13) 4:467–480.

Zimmerman, E. G. 1965. A comparison of habitat and food of two species of *Microtus*. Journal of Mammalogy 46:605–612.

Diagnostic Parasitology Course

The "Diagnostic Parasitology Course" is being offered July 31–August 11, 2000 at the Uniformed Services University of the Health Sciences, Bethesda, Maryland 20814-4799. This course will consist of a series of lectures and hands-on laboratory sessions covering the diagnosis of parasitic infections of humans. In addition to the examination of specimens, participants will be able to practice various methods used in the diagnosis of intestinal, blood, and tissue parasitic infections. Parasitic diseases encountered throughout the world will be included. Slide presentations and videotapes will be available for study. The course will be held at the University's campus, utilizing up-to-date lecture rooms and laboratory facilities. Microscopes will be available on a loan basis and laboratory supplies will be provided. Certain reference specimens will also be available for personal use.

The registration fee for the 2-week course is US$1,000 (This does not include lodging and meals). Enrollment is limited, so those interested should register as soon as possible. Previous laboratory experience is recommended.

For further information contact Dr. John H. Cross, (301) 295-3139 (e-mail: jcross@usuhs.mil) or Ms. Ellen Goldman, (301) 295-3129 (email: egoldman@usuhs.mil).

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Revision of the Genus *Pallisentis* (Acanthocephala: Quadrigyridae) with the Erection of Three New Subgenera, the Description of *Pallisentis* (*Brevitritospinus*) *vietnamensis* subgen. et sp. n., a Key to Species of *Pallisentis*, and the Description of a New Quadrigyrid Genus, *Pararaosentis* gen. n.

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ABSTRACT: The genus *Pallisentis* is revised. Golvan's 3 subgenera (*Farzandia, Neosentis, Pallisentis*) were distinguished solely by the number of hooks in proboscis hook circles, which proved to be a variable trait. Three new subgenera are erected based on the relative size of hooks in subsequent circles, the size of cement glands, and the number of their giant nuclei. A new species of *Pallisentis* is described from the snake head mullet, *Ophicephalus maculatus*, in Vietnam. A key to all 26 species of the genus *Pallisentis* accepted as valid and following our classification is included. A new quadrigyrid genus *Pararaosentis* is erected.

KEY WORDS: revision of *Pallisentis*; Acanthocephala, Quadrigyridae, new taxa, *Pallisentis* (*Brevitritospinus*) *vietnamensis* subgen. et sp. n., taxonomic key, *Pararaosentis* gen. n., snake head mullet, Vietnam.

The discovery of new species of the genus *Pallisentis* Van Cleave, 1928, from a Vietnamese mullet, *Ophicephalus maculatus* (Lacépède, 1802) (Channidae) necessitated the review of the current status of the genus and its component species. The confused taxonomic state was compounded by Golvan's (1959, 1994) subgeneric designations and assignments based on the variable character of the number of hooks in proboscis hook circles. Other problems of omissions, inconsistent assignments, and improper generic relegations necessitated the revision of the entire group, the creation of 3 new subgenera based on naturally consistent characters, and the creation of a key to all 26 species of *Pallisentis*.

Materials and Methods

Fifteen snake head mullets, *O. maculatus*, measuring 26–48 (mean, 36) cm in total length were examined for parasites. The fish were collected from waters around Hanoi, Vietnam, and purchased alive in a Hanoi fish market on 25 May 1998. Two fish were infected with about 20 worms, of which 9 were made available and described in this paper as a new species. These thin cylindrical worms were easily located in the pathologically enlarged upper intestine because of the apparent reddish inflammation at attachment sites. The worms were removed, extended in water, and fixed and shipped in 70% ethanol. Worms were stained in Mayer's acid carmine, dehydrated in ascending concentrations of ethanol, and whole-mounted in Canada balsam. Measurements are in micrometers unless otherwise stated. The range is followed by mean values (in parentheses). Width measurements refer to maximum width. Body (=trunk) length does not include neck, proboscis, or male bursa. Specimens have been deposited in the United States National Parasite Collection (USNPC), Beltsville, Maryland, U.S.A.

Results and Discussion

The Genus *Pallisentis* Van Cleave, 1928

Van Cleave (1928) created the genus *Pallisentis* in his new family Pallisentidae to accommodate *Pallisentis umbellatus* Van Cleave, 1928. Baylis (1933) synonymized the genera *Farzandia* Thapar, 1930, and *Neosentis* Van Cleave, 1928, despite Meyer's (1932) retention of *Farzandia* as an independent genus in a different family, Acanthogyridae. Petrochenko (1956) followed Meyer (1932). Baylis' (1933) synonymies were accepted by Harada (1935) and Yamaguti (1963) and currently remain valid; the genus *Pallisentis* was recognized in the family Quadrigyridae Van Cleave 1920, subfamily Pallisentinae Van Cleave, 1928.

In his generic diagnosis, Van Cleave used re-

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strictive traits that were basically descriptive of specific features of *P. umbellatus*. These included the number of proboscis hooks, collar and trunk spines, and giant nuclei of the cement gland, as well as the extent of distribution of the trunk spines, and the position of the testes. These same traits were used by subsequent workers, e.g., Petrochenko (1956) and Yamaguti (1963), despite the addition of more species adding more variability to the diagnostic criteria of the genus over the years. A new diagnosis of the genus *Pallisentis* is provided below.

Pallisentis Van Cleave, 1928, sensu lato

DIAGNOSIS: Quadrigyridae, Pallisentinae. Trunk slender, small-medium in length, with an anterior set of collar spines and a posterior set of trunk spines separated by region lacking spines. Collar spines arranged in a few closely set circles; circles of trunk spines more widely spaced and may extend to posterior end of males or females. Giant hypodermal nuclei may be present. Proboscis short, cylindroid–spheroid with 4 circles of 6–12 hooks each. Proboscis receptacle single-walled, with large cerebral ganglion near its base. Lemnisci long, cylindrical, equal or unequal. Testes ovoid–cylindrical, contiguous. Cement gland syncytial, medium-long, with few to many giant nuclei. Cement reservoir present. Saefftigen's pouch present or absent. Parasites of freshwater fishes in Asia.

The Subgenera of *Pallisentis*

Based on the number of hooks in each of the proboscis hook circles, Golvan (1959) erected 3 subgenera of *Pallisentis*: *Farzandia* Thapar, 1931, with 10 hooks per circle, *Neosentis* Van Cleave, 1928, with 8 hooks per circle, and *Pallisentis* Van Cleave, 1928, with 6 hooks per circle. Some species were not assigned to a subgenus, and others could be relegated to more than 1 subgenus or not to any. By 1994 a greater number of species had been described and the number of nonassignments and exceptions increased disproportionately. Golvan (1994) additionally did not include 4 other species described earlier (footnote, Table 1). The character (number of hooks per circle) used by Golvan (1959) is inconsistent and showed variations even within the same species and, thus, should not be used for subgeneric assignment of species. Yamaguti (1963), Tadros (1966), Gupta and Verma (1980), Soota and Bhattacharya (1982), Gupta and Fatma (1986), and Chowhan et al. (1987) also rejected Golvan's (1959) system. Tadros (1966) and Soota and Bhattacharya (1982) also agreed with the above authors and evaluated other taxonomic characters of *Pallisentis*. We found the most consistent character to be the difference in the size of proboscis hooks in subsequent circles. Other characters of considerable consistency included the size of the cement gland and the number of its giant nuclei, the shape and distribution of trunk spines, and the presence or absence of Saefftigen's pouch. Based on the first 3 characters listed above, we designate herein 3 new subgenera. All characters (above) are used to construct the subsequent key to species.

Demidueterospinus subgen. n.

DIAGNOSIS: With the characters of the genus *Pallisentis* provided herein. Proboscis hooks in circle 2 about half as long as hooks in circle 1. Cement gland usually small, with few giant nuclei.

Taxonomic summary

TYPE SPECIES: *Pallisentis* (*D.*) *ophiocephali* (Thapar, 1931) Baylis, 1933.

OTHER SPECIES: *Pallisentis* (*D.*) *basiri* Farooqi, 1958; *Pallisentis* (*D.*) *panadei* Rai, 1967.

Remarks

Pallisentis basiri and *P. panadei* were synonymized with *Pallisentis colisai* Sarkar, 1954, by Soota and Bhattacharya (1982). We consider these species to be valid. These and other synonymies made by Soota and Bhattacharya (1982) did not acknowledge the species-specific differences that we outline in our key. Further, their tabulated data often did not match the narrative and were occasionally misplaced. These synonymies were also not accepted by Khan and Bilqees (1987) and were not followed by other workers. The redescription of *P. basiri* by Gupta and Fatma (1986) is inconsistent with the characteristics of that species and appears to be of another species. *Pallisentis ophiocephali* of Moravec and Sey (1989) from Vietnam is conspecific with our material from the same location and is described herein as a new species.

Brevitritospinus subgen. n.

DIAGNOSIS: With the characters of the genus *Pallisentis* provided herein. Proboscis hooks in circle 3 about half as long as hooks in circle 2.

Table 1. Present status of the subgenera and species of the genus *Pallisentis* according to Golvan (1959) (in parentheses) and Golvan (1994) based on the number of proboscis hooks per circle in species assigned to each subgenus.

Species and subgenera*	No. of proboscis hooks/circle	Remarks
Subgenus *Farzandia* Thapar, 1931	(10)	Thapar (1931) established genus *Farzandia* with *F. ophiocephali* as its type species
P. (F.) gaboes (MacCallum, 1918) Van Cleave, 1928	(10)	(=*Echinorhynchus gaboes* MacCallum, 1918): redescribed by Fernando and Furtado (1963), and Yamaguti (1954)
P. (F.) nagpurensis (Bhalerao, 1931) Baylis, 1933	(8–10)	Synonymized with *P. ophiocephali* by Soota and Bhattacharya (1982); redescribed by Kennedy 1989
Subgenus *Neosentis* Van Cleave, 1928	(8)	Baylis (1933) synonymized the genera *Farzandia* and *Neosentis* with *Pallisentis*
P. (N.) celatus (Van Cleave, 1928) Baylis, 1933	(8)	Type species of *Neosentis*; redescribed by Moravec and Sey (1989)
P. (N.) ophiocephali (Thapar, 1931) Baylis, 1933	(8–10)	*P. ophiocephali* of Moravec and Sey (1989), not *P. ophiocephali*
Subgenus *Pallisentis* Van Cleave, 1928	(6)	Nominal subgenus of genus *Pallisentis* Van Cleave, 1928
P. (P.) allahabadi Agarwal, 1958	8–10	Synonymized with *P. ophiocephali* by Soota and Bhattacharya (1982), redescribed by Jain and Gupta (1979)
P. (P.) basiri Farooqi, 1958	9	Needs a new subgenus (Tadros, 1966); synonymized with *P. colisai* by Soota and Bhattacharya (1982)
P. (P.) buckleyi Tadros, 1966	10	Improper assignment; included in subgenus *Farzandia* by Tadros (1966); synonymized with *P. colisai* by Soota and Bhattacharya (1982) and with *P. allahabadi* by Jain and Gupta (1979)
P. (P.) cavasii Gupta and Verma, 1980	6–10	Also agrees with subgenera *Farzandia* and *Neosentis*
P. (P.) colisai Sarkar, 1954	(10–12)	Improper assignment; fits subgenus *Farzandia* or new subgenus
P. (P.) fasciata Gupta and Verma, 1980	6–10	Also fits subgenera *Farzandia* and *Neosentis*
P. (P.) golvani Troncy and Vassiliades, 1973	6	Not a species of the genus *Pallisentis*
P. (P.) gomtii Gupta and Verma, 1980	8–10	Improper assignment; fits subgenera *Farzandia* and *Neosentis*
P. (P.) guntei Sahay, Nath, and Sinha, 1967	8–10	Improper assignment; synonymized with *P. colisai* by Soota and Bhattacharya (1982)
P. (P.) magnum Saeed and Bilqees, 1971	8–10	Improper assignment; synonymized with *P. ophicephali* by Soota and Bhattacharya (1982)
P. (P.) nandai Sarkar, 1953	8–10	Improper assignment; synonymized with *P. ophicephali* by Soota and Bhattacharya (1982)
P. (P.) panadei Rai, 1967	10	Improper assignment; synonymized with *P. colisai* by Soota and Bhattacharya (1982)
P. (P.) tetraodontae Troncy, 1978	6	Originally described as *P. golvani tetraodontae*; not a species of the genus *Pallisentis*
P. (P.) umbellatus Van Cleave, 1928	(6)	Type species of *Pallisentis* Van Cleave, 1928
Subgenus not assigned	6–12	As per Golvan (1959, 1994)
Pallisentis sp. Pearse, 1933	10	Originally reported as *Farzandia* sp. by Pearse (1933)
P. cholodkowskyi (Kostylew, 1928) Amin, 1985	3	Belongs in the genus *Acanthocephalorhynchoides* Meyer, 1932
P. garuai (Sahay, Sinha, and Ghosh, 1971) Jain and Gupta, 1979	6	(=*Devendrosentis garuai* Sahay, Sinha, and Ghosh, 1971)
P. guptai Gupta and Fatma, 1986	8	Agrees with subgenus *Neosentis*
P. kalriai Khan and Bilqees, 1985	10	Agrees with subgenus *Farzandia*
P. mehrai Gupta and Fatma, 1986	10–12	Agrees with subgenus *Farzandia* or an undefined subgenus
P. nandai Sarkar, 1953	(8–10)	Synonymized with *P. ophiocephali* by Soota and Bhattacharya (1982)
P. sindensis Khan and Bilqees, 1987	8	Agrees with subgenus *Neosentis*

* Golvan (1994) did not include *P. clupei* Gupta and Gupta, 1979 (8 hooks per circle), *P. reefoni* Mital and Lal, 1981 (10), *P. indica* Mital and Lal, 1981 (10), *P. singaporensis* Kahn and Ip, 1989 (10). He correctly reassigned "*P.* (?) *ussuriensis*" (Kostylew, 1941) Golvan, 1959 (=*Acanthocephalorhynchoides ussuriensis* Kostylew, 1941) to *Acanthocephalorhynchoides* Meyer, 1932.

Cement gland usually small with few giant nuclei.

Taxonomic summary

TYPE SPECIES: *Pallisentis* (*B.*) *allahabadi* Agarwal, 1958.

OTHER SPECIES: *Pallisentis* (*B.*) *cavasii* Gupta and Verma, 1980; *Pallisentis* (*B.*) *croftoni* Mital and Lal, 1981; *Pallisentis* (*B.*) *fasciata* Gupta and Verma, 1980; *Pallisentis* (*B.*) *guntei* Sahay, Nath, Sinha, 1967; *Pallisentis* (*B.*) *indica* Mital and Lal, 1981; *Pallisentis* (*B.*) *mehrai* Gupta and Fatma, 1986; *Pallisentis* (*B.*) *vietnamensis* sp. n. (this report).

Remarks

Pallisentis allahabadi and *P. guntei* were synonymized with *P. ophiocephala* and *P. colisai,* respectively, by Soota and Bhattacharya (1982). These synonymies did not acknowledge species-specific differences outlined in our key. The redescription of *P. allahabadi* by Jain and Gupta (1979) and their synonymization of *P. buckleyi* Tadros, 1966, with it are considered sound and are accepted; the key taxonomic characters are in agreement.

Pallisentis subgen. n. Van Cleave, 1928, sensu stricto

DIAGNOSIS: With the characters of the genus *Pallisentis* provided herein. Proboscis hooks gradually declining in size posteriorly; cement glands usually long with many giant nuclei.

Taxonomic summary

TYPE SPECIES: *Pallisentis* (*P.*) *umbellatus* van Cleave, 1928.

OTHER SPECIES: *Pallisentis* (*P.*) *celatus* (Van Cleave, 1928) Baylis, 1933; *Pallisentis* (*P.*) *colisai* Sarkar, 1954; *Pallisentis* (*P.*) *clupei* Gupta and Gupta, 1979; *Pallisentis* (*P.*) *gaboes* (MacCallum, 1918) Van Cleave, 1928; *Pallisentis* (*P.*) *garuei* (Sahay, Sinha, and Ghosh, 1971) Jain and Gupta, 1979; *Pallisentis* (*P.*) *gomtii* Gupta and Verma, 1980; *Pallisentis* (*P.*) *guptai* Gupta and Fatma, 1986; *Pallisentis* (*P.*) *kalriai* Khan and Bilqees, 1985; *Pallisentis* (*P.*) *magnum* Saeed and Bilqees, 1971; *Pallisentis* (*P.*) *nandai* Sarkar, 1953; *Pallisentis* (*P.*) *nagpurensis* (Bhalero, 1931) Baylis, 1931; *Pallisentis* (*P.*) *pesteri* (Tadros, 1966) Chowhan, Gupta, and Khera, 1987; *Pallisentis* (*P.*) *sindensis* Khan and Bilqees, 1987; *Pallisentis* (*P.*) *singaporensis* Khan and Ip, 1989.

Remarks

Pallisentis celatus was redescribed by Moravec and Sey (1989) from Vietnamese specimens. *Pallisentis gaboes* was provisionally and incompletely redescribed by Yamaguti (1954) and briefly referenced by Fernando and Furtado (1963). Khan and Ip (1988) referred to the proboscis armature of *P. gaboes* as similar to that of *P. singaporensis.* The synonymization of the genus *Devendrosentis* Sahay, Sinha, and Ghosh, 1971, with the genus *Pallisentis* and the assignment of *D. garuai* Sahay, Sinha, and Gosh, 1971, to the genus *Pallisentis* are accepted; the descriptions of the new genera are identical. Since none of their accounts was sufficiently similar to the original description, the incomplete redescription of *P. nagpurensis* by Jain and Gupta (1979) is considered questionable, that of Chowhan et al. (1987) uncertain, and that of Kennedy (1981) not of the same species. Based on comparability of cement gland structure, we accept the synonymy of the genus *Saccosentis* Tadros, 1966, with *Pallisentis* as proposed by Chowhan et al. (1987), and *Saccosentis pesteri* Tadros, 1966, is assigned to the genus *Pallisentis.* The synonymization of *P. magnum, P. nandai,* and *P. nagpurensis* with *P. ophiocephali* by Soota and Bhattacharya (1982) is not accepted, since these synonymies did not acknowledge the species-specific differences outlined in our key.

Other Taxonomic Assignments

Pallisentis cholodkowskyi (Kostylew, 1928) Amin, 1985 (=*Quadrigyrus cholodkowskyi* Kostylew, 1928) is assigned to the genus *Acanthocephalorhynchoides* Kostylew, 1941, based on proboscis and trunk spination patterns (see Williams et al. [1980] for additional information). Golvan (1994) made a similar assignment regarding *Acanthocephalorhynchoides ussuriensis* Kostylew, 1941.

Pallisentis tetraodontae Troncy, 1978, was described by Troncy (1978) as a subspecies of *Pallisentis golvani* Troncy and Vassiliadis, 1973. Golvan (1994) elevated it to species rank without justification. We have determined that *P. golvani* does not belong to the genus *Pallisentis* or any other known genus of the family Quadrigyridae Van Cleave, 1920 (see remarks). A

new genus is described below to accommodate *P. golvani.*

Pararaosentis n. gen.

DIAGNOSIS: Quadrigyridae, Pallisentinae. Trunk short with hypodermal nuclei and anterior constriction containing 1 set of minute spines arranged in a few complete circles, most anteriorly. Proboscis short, with 4 circles of small hooks gradually decreasing in length posteriorly. Proboscis receptacle single-walled, with large cerebral ganglion at its base. Male reproductive system compacted in posterior region. Testes short, robust, contiguous. Cement gland syncytial, small with few giant nuclei. Cement reservoir and Saefftigen's pouch present. Parasites of freshwater fishes in Africa.

Taxonomic summary

TYPE SPECIES: *Pararaosentis golvani* (Troncy and Vassiliades, 1973) n. comb. (=*Pallisentis golvani* Troncy and Vassiliades, 1973; *Pallisentis tetraodontae* Troncy, 1978).

Remarks

The type species does not belong in the genus *Pallisentis* because of its anterior trunk constriction, the presence of only 1 set of spines anteriorly, the noncylindrical form of its testes and cement gland, and its occurrence in African, not Asian, fishes. Furthermore, the trunk is short and lacks the anterior swelling of the long slender specimens of *Pallisentis.* The new genus is closest to the genus *Raosentis* Datta, 1947. In *Raosentis,* however, the trunk is not constricted anteriorly, and the proboscis hooks in the anterior 2 circles are longer and stouter than the hooks in posterior 2 circles and are separated from them by an unarmed area.

The characters on which Troncy (1978) based his assignment of *P. tetraodontae* as a subspecies of *P. golvani* are not significant enough to justify a subspecific status, and *P. tetraodontae* is herein relegated to a synonym of *P. golvani.*

Pallisentis (*Brevitritospinus*) *vietnamensis* sp. n. (Figs. 1–9)

Description

GENERAL: Shared characters (proboscis and hooks, proboscis receptable, trunk, and lemnisci) larger in females than in males (see Table 2 for measurements). Trunk curved ventrad, medium in length, slender, cylindrical with anterior swelling (Figs. 1, 5) and 83–137 long × 21–62 wide hypodermic nuclei in anterior half of trunk (0–5), posterior half (1–4), and in apical organ of proboscis (3). Proboscis truncated, wider than long, with conspicuous apical organ (Fig. 3). Proboscis hooks with shallow pluglike roots, in 4 circles of 10 hooks each. Hooks in first circle largest, hooks in second circle slightly smaller, hooks in third circle about half as long as hooks in second circle, hooks in fourth circle smallest (Figs. 3, 4). Neck very short (Figs. 3, 5, 9). Proboscis receptacle 5–6 times as long as proboscis, single-walled, with cerebral ganglion near its base (Figs. 1, 5). Lemnisci long, tubular, unequal, and with 1 giant nucleus each (Figs. 1, 5). Collar spines triangular, in 18–22 closely spaced circles beginning just behind a spineless area on anterior trunk (often interpreted as the neck) and overlapping and extending slightly posterior to the posterior half of the proboscis receptacle (Figs. 1, 5). Trunk spines triangular (Figs. 7, 8), in considerably more widely spaced circles extending to posterior end of females and to testes in males. Anterior trunk swelling covered by 17–20 circles of trunk spines. An unspined area separating trunk spines from collar spines (Figs. 1, 5). Unspined areas often occurring in posterior 2–3 circles of collar spines, and up to 5 or 6 times involving 2–6 circles of trunk spines throughout (Figs. 1, 5). Number of trunk spines decreasing to 1 or 2 in posteriormost circles where their size slightly decreases.

MALE: Based on 4 specimens. Reproductive system at posterior end of trunk. Testes oblong, contiguous; anterior testis larger than posterior. Cement gland rectangular, syncytial with 7–8 giant nuclei. Cement reservoir branching posteriorly into 2 ducts (Figs. 1, 2).

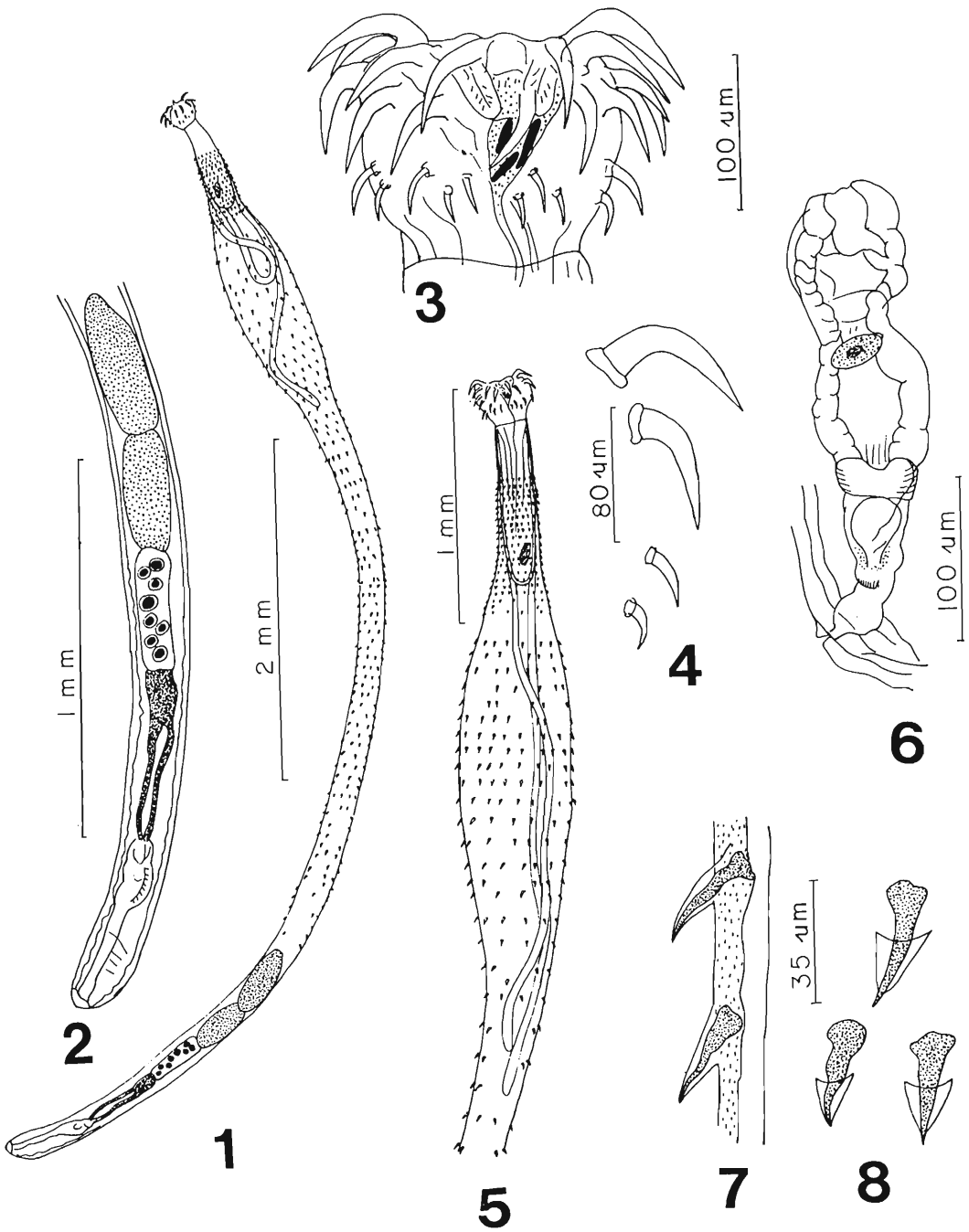
FEMALE: Based on 5 specimens. Reproductive system short, robust with the vaginal complex, uterus, and uterine bell of almost equal length; gonopore subterminal (Fig. 6). Eggs ovoid with concentric shells.

Taxonomic summary

TYPE HOST: Snake head fish (mullet), *Ophiocephalus maculatus* (Lacépède, 1802) (Channidae).

OTHER HOST: ca the be (Vietnamese name) *Acanthorhodeus fortunensis* (Cyprinidae) (only 1 juvenile found by Moravec and Sey, 1989).

SITE OF INFECTION: Upper intestine.



Figures 1–8. ***Pallisentis vietnamensis*** **sp. n. 1. Holotype male (note gaps in distribution of trunk spines). 2. Reproductive system of holotype male. 3. Proboscis of a paratype female (note 3 giant nuclei in apical organ). 4. One row of proboscis hooks from proboscis in Fig. 3. 5. Anterior end of a paratype female (note gaps in the distribution of collar and trunk spines). 6. Reproductive system of allotype female (note balloon-shaped vaginal gland and unripe egg). 7–8. Side and en face views of trunk spines from paratype in Fig. 5.**

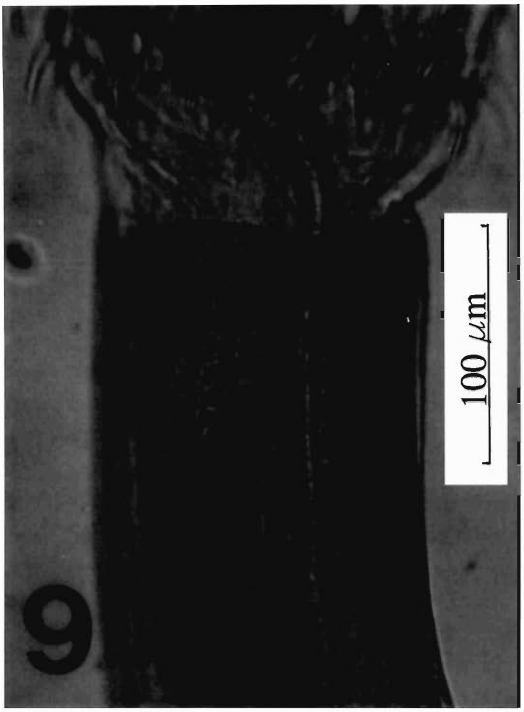


Figure 9. ***Pallisentis vietnamensis*** **sp. n. Anterior end of trunk of paratype female in Fig. 5, showing clear line of demarcation between the naked anterior end of the trunk and the very short neck at the base of the proboscis.**

TYPE LOCALITY: Lakes and Red River near Hanoi, Vietnam.

SPECIMENS DEPOSITED: USNPC No. 88635 (holotype male); No. 88636 (allotype female); No. 88637 (paratypes).

ETYMOLOGY: The new species is named for its geographical location in Vietnam.

Remarks

The identification of *P. vietnamensis* sp. n. as *P. ophiocephali* by Moravec and Sey (1989) overlooked the difference in proboscis hook size (these species belong to 2 different subgenera) and the fact that trunk spines of the latter species extend to the posterior ends of individuals of both sexes. Specimens believed to be conspecific with the new species by Moravec and Sey (1989) were previously reported by Ha (1969) from *O. maculatus*.

Of the 26 species of *Pallisentis* recognized as valid, *P. vietnamensis* sp. n. has the largest number of trunk spine circles in males (57–88) and females (120–149). The largest number of trunk spine circles in other species are 52 in *P. ophiocephali* males, 30–66 in *P. nagpurensis* males and females, and 28–32 and 36–76 in *P. garuei* males and females, repectively. The new species also has giant nuclei in the apical organ of the proboscis, a feature not reported in any other species of *Pallisentis* (Fig. 3). The female reproductive system is similar to that of *P. colisai* except that the uterus in the latter species is considerably longer.

The reference to *Pallisentis* sp. from threadfin shad, *Dorosoma petenense* (Günther, 1867), in Louisiana, U.S.A. by Arnold et al. (1968) is clearly in error, since *Pallisentis* occurs only in Asia.

Further differentiation between *P. vietnamensis* and the other 25 species of the genus *Pallisentis* is presented in the following key.

Key to Species of the Genus *Pallisentis* sensu lato

1. Proboscis hooks in second or third circle declining abruptly in size; cement gland usually small, with few giant nuclei 2
   Proboscis hooks gradually declining in size posteriorly; cement glands usually long, with many giant nuclei ..... Subgenus *Pallisentis* subgen. n. 12
2. Proboscis hooks in second circle about half as long as hooks in first circle ..... Subgenus *Demidueterospinus* subgen. n. 3
   Proboscis hooks in third circle about half as long as hooks in second circle ..... Subgenus *Brevitritospinus* subgen. n. 5
3. Trunk spines conical and extending to posterior end of males and females; Saefftigen's pouch absent ..... *Pallisentis* (*D.*) *ophiocephali* (Thapar, 1931) Baylis, 1933
   Trunk spines Y-shaped not extending to posterior end of males; Saefftigen's pouch present 4
4. Proboscis hooks in first circle 70–80 long; hook roots recurved, simple; lemnisci equal; testes equatorial, 580–620 (anterior) and 510–560 (posterior) long; cement gland 470–630 long; Saefftigen's pouch 320–390 long; female gonopore terminal ..... *Pallisentis* (*D.*) *panadei* Rai, 1967
   Proboscis hooks in first circle 100 long; hook roots stubby knobs; lemnisci unequal; testes pre-equatorial, 950 (ante-

Table 2. Morphometric characteristics of *Pallisentis* (*B.*) *vietnamensis* (measurements are in micrometers unless otherwise noted).

	Moravec and Sey (1989) (9 males, 5 females)	Present paper (4 males, 5 females)
Males		
Trunk (mm)	6.74–14.6 × 0.408–0.503	7.04–14.20 (9.03) × 0.29–0.42 (0.37)†
Hypodermal/nuclei	NG*	3 (apical organ), 0–5 (anterior), 3–4 (posterior)
Proboscis	163–177 × 204–245	130–145 (135) × 167–187 (178)
First circle hooks	81–84	80–88 (85)
Second circle hooks	72–75	75 (75)
Third circle hooks	36–42	32–38 (35)
Fourth circle hooks	30	25–28 (26)
Neck	NG	22–32 (29) × 112–135 (123)
Proboscis receptacle	340–517 × NG	458–728 (855) × 146–156 (152)
Brain	NG	100–137 (120) × 50–75 (64)
Anterior spineless area	272–367 × 177–204 (called neck)	187–312 (236) × 135–156 (146)
Lemniscus		
Long (mm)	1.06–1.90 × NG	2.49 × 0.04–0.06
Short (mm)	Only 1 measurement given	1.87 × 0.03–0.05
Collar spines		
Circles/no. per circle	18–21/20–22	20–21/10–22
Length	27–30	17–25 (21) (anterior), 20–32 (26) (posterior)
Trunk spines	Extend to testes	Extend to testes
Circles/no. per circle	57–86/NG	75–88 (82)/1–16
Length	21–30	30–57 (37) (anterior), 35–65 (44) (posterior)
Anterior testis	449–767 × 218–313	406–988 (616) × 146–198 (161)
Posterior testis	449–721 × 218–313	333–551 (424) × 125–166 (148)
Cement gland	NG (550 × 180, Fig. 2C)	395–728 (504) × 94–156 (119)
No. nuclei	8	7–8 (usually 8)
Cement reservoir	NG (370 × 180, Fig. 2C)	187–499 (304) × 83–187 (117)
Cement duct	NG (400, Fig. 2C)	364–572 (455)
Bursa	NG × 258	238 × 135 (n = 1)
Females		
Trunk (mm)	14.42–20.54 × 0.54–0.57	11.02–19.40 (15.81) × 0.32-0.47 (0.38)
Subcuticular nuclei	NG	3 (apical organ), 0–2 (anterior), 1–3 (posterior)
Proboscis	177–204 × 218–258	142–175 (156) × 177–210 (197)
First circle hooks	87–99	75–95 (85)
Second circle hooks	75–94	70–87 (77)
Third circle hooks	39	33–45 (38)
Fourth circle hooks	30	25–32 (28)
Neck	NG	25–32 (28) × 130–145 (135)
Proboscis receptacle	NG	655–728 (699) × 135–187 (168)
Brain	NG	125–150 (135) × 45–80 (61)
Anterior spineless area	258–422 × 190–218 (called "neck")	228–260 (239) × 146–187 (169)
Lemniscus		
Long (mm)	NG	2.08–2.67(2.50) × 0.04–0.07 (0.05)
Short (mm)	NG	1.56–2.50 (2.13) × 0.04–0.06 (0.05)
Collar spines		
Circles/no. per circle	21/22–24	19–22/6–23
Length	24–30	20–25 (23) (anterior), 23–27 (25) (posterior)
Trunk spines	Extend to posterior end	Extend to posterior end
Circles/no. per circle	120/NG	125–149 (138)/1–17
Length	24–30	32–38 (36) (anterior), 33–43 (37) (posterior)
Reproductive system	NG (Fig. 2I of another species)	364–458 (410)
Gonopore	Subterminal	Subterminal
Eggs	75–84 × 30–33 (ripe) 50 × 25 (Fig. 2J)	42–57 (50) × 18–22 (20) (unripe)

* NG = not given.
† Range (mean).

rior) and 700 (posterior) long; cement gland 900 long; Saefftigen's pouch 770 long; female gonopore subterminal ..... *Pallisentis* (*D.*) *basiri* Farooqi, 1958

5. Trunk spines conical 6
   Trunk spines Y-shaped 9
6. Trunk spines in many circles, 57–88 in males and 120–149 in females; Saefftigen's pouch absent ..... *Pallisentis* (*B.*) *vietnamensis* sp. n.
   Trunk spines in fewer circles, up to 27 in males and 36 in females; Saefftigen's pouch present 7
7. Trunk small, up to 2.0 mm long in males and 4.5 mm long in females; proboscis hooks in anterior 2 circles similar in size; trunk with 14–18 circles of spines each with 17–24 spines; cement gland less than 200 long ..... *Pallisentis* (*B.*) *guntei* Sahay, Nath, and Sinha 1967
   Trunk larger, 3.4–6.9 mm long in males and 7.3–15.6 mm long in females; proboscis hooks in second circle slightly smaller that hooks in first circle; trunk with 20–27 circles of spines each with up to 12 spines; cement gland 400–973 long 8
8. Female gonopore terminal; length of testes 733–910 (anterior), 785–925 (posterior); cement gland 863–973, and cement reservoir 580–816 ..... *Pallisentis* (*B.*) *croftoni* Mital and Lal, 1981
   Female gonopore subterminal; length of testes 475 (anterior), 437 (posterior), cement gland 400, and cement reservoir 361 ..... *Pallisentis* (*B.*) *allahabadi* Agarwal, 1958
9. Trunk spines extending to posterior end of males and females; proboscis hooks 10–12 per circle; hooks in anterior circle larger than 100 ..... *Pallisentis* (*B.*) *mehrai* Gupta and Fatma, 1986
   Trunk spines not extending to posterior end of males or females; proboscis hooks 6–10 per circle; hooks in anterior circle shorter than 100 10
10. Females less than 4.0 mm long; lemnisci ending well above anterior testis, testis small, up to 225 (anterior) and 200 (posterior) long; cement gland small, 200–230 long, with 6–8 giant nuclei ..... *Pallisentis* (*B.*) *cavasii* Gupta and Verma, 1980
    Females longer than 4.0 mm long; lemnisci may reach anterior testis; testes between 200 and 910 long; cement glands between 172 and 926 long, with 10–18 giant nuclei each 11
11. Proboscis hooks 10 per circle; female proboscis receptacle more than 700 mm long; lemnisci ending well above anterior testis ..... *Pallisentis* (*B.*) *indica* Mital and Lal, 1981
    Proboscis hooks 6–10 per circle; female proboscis receptacle less than 400 long; lemnisci extending to mid-anterior testis ..... *Pallisentis (B.) fasciata* Gupta and Verma, 1980
12. Trunk spines conical or Y-shaped, extending to posterior end of at least 1 sex 13
    Trunk spines only conical, not extending to posterior end of either sex 16
13. Trunk spines conical, extending to posterior end in females only; testes post-equatorial 14
    Trunk spines conical or Y-shaped, extending to posterior end of both males and females; testes equatorial 15
14. Proboscis hooks in first circle less than 100 long; proboscis receptacle less than 500 long; cement gland with 20–30 giant nuclei; female gonopore subterminal ..... *Pallisentis* (*P.*) *nagpurensis* (Bhalero, 1931) Baylis, 1933
    Proboscis hooks in first circle 100 or more long; proboscis receptacle more than 800 long; cement gland with 9–16 giant nuclei; female gonopore terminal ..... *Pallisentis* (*P.*) *clupei* Gupta and Gupta, 1979
15. Trunk spines conical, in 28–32 circles in males and 36–76 circles in females; neck separated from proboscis by transverse circular muscle band; cement gland longer than 1.6 mm ..... *Pallisentis* (*P.*) *garuei* (Sahay, Sinha and Ghosh, 1971) Jain and Gupta, 1979
    Trunk spines Y-shaped, in 16–20 circles in males and 25–30 circles in females; no muscle band between neck and proboscis; cement gland less than 0.6 mm long ..... *Pallisentis* (*P.*) *guptai* Gupta and Fatma, 1986
16. Males with Saefftigen's pouch 17
    Males lacking Saefftigen's pouch 21

17. Trunk spines appearing continuous with collar spines ..... *Pallisentis* (*P.*) *magnum* Saeed and Bilqees, 1971
    Trunk spines well separated from collar spines 18
18. Proboscis hooks 10 per circle, each embedded in thickened cuticular rim; trunk spines with cuticular comblike thickening; males with additional circles of posttesticular trunk spines; testes preequatorial ..... *Pallisentis* (*P.*) *kalriai* Khan and Bilqees, 1985
    Proboscis hooks 8–10 per circle; no cuticular thickening at base of proboscis hooks or trunk spines; no posttesticular trunk spines; testes not preequatorial 19
19. Female trunk spines in 36–73 circles, extending to just anterior to posterior end; lemnisci unequal; testes small, less than 0.5 mm long ..... *Pallisentis* (*P.*) *gomtii* Gupta and Verma, 1980
    Female trunk spines in 10–20 circles, extending only to anterior third of trunk; lemnisci equal; testes large, more than 1.0 mm long 20
20. Proboscis hooks 8 per circle; testes equatorial; cement gland 2.2–3.0 mm long ..... *Pallisentis* (*P.*) *sindensis* Khan and Bilqees, 1987
    Proboscis hooks 10 per circle; testes postequatorial; cement gland short, 0.7–1.6 long ..... *Pallisentis* (*P.*) *gaboes* (MacCallum, 1918) Van Cleave, 1928
21. Proboscis hooks 6–7 per circle 22
    Proboscis hooks 8–12 per circle 23
22. Proboscis hooks 6 per circle; anterior hooks 89–119 long; cement gland with 16 giant nuclei ..... *Pallisentis* (*P.*) *umbellatus* Van Cleave, 1928
    Proboscis hooks 7 per circle; anterior hooks 60–70 long; cement gland with 10–12 nuclei ..... *Pallisentis pesteri* (Tadros, 1966) Showhan, Gupta, and Khera, 1987
23. Cement gland with 12–14 giant nuclei; lemnisci equal 24
    Cement gland with 23–25 giant nuclei; lemnisci unequal 25
24. Proboscis hooks 8 per circle; collar spines in 6–7 circles each with 29–40 spines; trunk spines in 8–13 circles each with 30–41 spines with sclerotized, large, variably shaped beds; testes longer than 0.7 mm ..... *Pallisentis* (*P.*) *celatus* Van Cleave, 1928
    Proboscis hooks 10–12 per circle; collar spines in 15–17 circles each with 18–20 spines; trunk spines in 21–22 (males), 67 (females) circles each with 16–20 simple triangular spines; testes 0.28–0.42 long ..... *Pallisentis* (*P.*) *colisai* Sakkar, 1954
25. Proboscis hooks 93, 80, 60, 33 long (from anterior); trunk spines in 44–55 circles, each with 16–20 spines; female gonopore posteroventral ..... *Pallisentis* (*P.*) *nandai* Sarkar, 1953
    Proboscis hooks 62–64, 49–54, 36–46, 24–28 long (from anterior); trunk spines in 25–26 circles, each with 10 spines; female gonopore terminal ..... *Pallisentis* (*P.*) *singaporensis* Khan and Ip, 1988

Literature Cited

Arnold, J. G., Jr., H. E. Schafer, and R. L. Vulliet. 1968 The parasites of the freshwater fishes of Louisiana. Proceedings of the Southeastern Association of Game and Fish Commission Annual Conference 21:531–543.

Baylis, M. A. 1933. On some parasitic worms from Java, with remarks on the acanthocephalan genus *Pallisentis*. Annals and Magazine of Natural History, Series 10 12:443–449.

Chowhan, J. S., N. K. Gupta, and S. Khera. 1987. On two species of the genus *Pallisentis* Van Cleave, 1928 from fishes of Gobindsagar Lake and synonymy of the genus *Saccosentis* Tadros, 1966. Research Bulletin of the Panjab University, Science 38:21–26.

Farooqi, H. U. 1958. A new species of the genus *Pallisentis* from a fresh-water eel. Zeitschrift für Parasitenkunde 18:457–464.

Fernando, C. H., and J. I. Furtado. 1963. A study of some helminth parasites of freshwater fishes in Ceylon. Zeitschrift für Parasitenkunde 23:141–163.

Golvan, Y. J. 1959. Le phylum de Acanthocephala. Deuxième note. La classe de Eoacanthocephala (Van Cleave, 1936). Annales de Parasitologie Humaine et Comparée 34:5–52.

———. 1994. Nomenclature of the Acanthocephala. Research and Reviews in Parasitology 54:135–205.

Gupta, S. P., and S. L. Verma. 1980. On three new Acanthocephala parasites of the genus *Pallisentis* Van Cleave, 1928 from freshwater fishes of Lucknow. Helminthologia 17:269–282.

Gupta, V., and S. Fatma. 1986. On some acanthocephalan parasites (Family Quadrigyridae Van Cleave, 1920) from fishes of Uttar Pradesh and

Tamil Nadu. Indian Journal of Helminthology 37: 149–180.

Ha, K. 1969. Parasite fauna of some freshwater fishes in North Vietnam and measures against the most important fish diseases. Avtoreferat Kanditatska Dissertasia Zoologia Institut AN S.S.S.R., Leningrad, 18 pp. (In Russian.)

Harada, I. 1935. Zur Acanthocephalenfauna von Japan. Memoirs of the Faculty of Science and Agriculture, Taihoku Imperial University of Formosa, Japan 14:7–23.

Jain, M., and N. K. Gupta. 1979. On two already known species of the genus *Pallisentis* Van Cleave, 1928 (Acanthocephala) and discussion on the validity of *Pallisentis buckleyi* Tadros, 1966 and genus *Devendrosentis* Sahay, Sinha et Ghosgh, 1971. Helminthologia 16:173–183.

Kennedy, M. J. 1981. *Pallisentis nagpurensis* (Bhalerao, 1931) (Acanthocephala: Quadrigyridae) from the gabus, *Ophiocephalus striatus,* from Depok, Indonesia. Canadian Journal of Zoology 59: 1847–1851.

Khan, A., and F. M. Bilqees. 1987. Two new Acanthocephala species from freshwater fishes of Kalri Lake. Pakistan Journal of Zoology 19:263–271.

Khan, M. M., and Y. K. Ip. 1988. *Pallisentis singaporensis* new species (Acanthocephala: Quadrigyridae) from the mudskipper, *Periophthalmodon schlosseri* in Singapore. Journal of the Singapore National Academy of Science 17:24–27.

Meyer, A. 1932. Acanthocephala. Dr. H. G. Bronns, Klassen und Ordnungen des Tier-Reichs, Leipzig 4:1–332.

Moravec, F., and O. Sey. 1989. Acanthocephalans of freshwater fishes from North Vietnam. Věstnik Československé Společnosti Zoologické 53:89–106.

Pearse, A. S. 1933. Parasites of Siamese fishes and crustaceans. Journal of the Siamese Society of Natural History (Suppl. 9):179–191 (+ 30 figs.)

Petrochenko, V. I. 1956. Acanthocephala of domestic and wild animals. Vol. 1. Izdatel'stvo Academii Nauk S.S.S.R., Moscow. (English translation by Israel Program for Scientific Translations Ltd., 1971, 465 pp.)

Soota, T. D., and S. B. Bhattacharya. 1982. On the validity of the species of the genus *Pallisentis* Van Cleave, 1928 (Acanthocephala: Pallisentidae) from the Indian subcontinent. Records of the Zoological Survey of India 80:157–167.

Tadros, G. 1966. On three new Acanthocephala of the genera *Pallisentis* Van Cleave, *Saccosentis* gen. nov. and *Acanthocephalus* Koelreuther, from fish. Journal of Helminthology 40:155–180.

Troncy, P. M. 1978. Nouvelles observations sur les parasites de poissons du bassin tchadien. Bulletin de l'Institut Fondamental d'Afrique Noire 40: 528–552.

Van Cleave, H. J. 1928. Acanthocephala from China. I. New species and new genera from Chinese fishes. Parasitology 20:1–9.

Williams, J. S., D. I. Gibson, and A. Sadighian. 1980. Some helminth-parasites of Iranian freshwater fishes. Journal of Natural History 14:685–699.

Yamaguti, S. 1954. Parasitic worms from Celebes. Part 8. Acanthocephala. Acta Medicinae Okayama 8:406–413.

———. 1963. Systema Helminthum. Vol. V. Acanthocephala. Interscience Publishers, New York, 423 pp.

Comp. Parasitol.
67(1), 2000 pp. 51–59

Two New Species of *Popovastrongylus* Mawson, 1977 (Nematoda: Cloacinidae) from Macropodid Marsupials in Australia

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ABSTRACT: The cephalic anatomy of *Popovastrongylus wallabiae* (Johnson and Mawson) is described, giving additional morphological details. New species of *Popovastrongylus* (Nematoda: Cloacinidae: Cloacininae) are described. *Popovastrongylus tasmaniensis* sp. n. from *Thylogale billardierii* (Desmarest) from Tasmania, Australia, has an oval mouth opening and buccal capsule, and the intestinal wall extends anteriorly to surround the esophageal bulb. *Popovastrongylus pluteus* sp. n. from *Macropus robustus* Gould from New South Wales, Australia, is similar to *Popovastrongylus pearsoni* (Johnson and Mawson) in having, among other characters, a shelf-like projection in the buccal capsule. It differs from *P. pearsoni* in having a circular mouth opening and buccal capsule rather than a quadrangular mouth opening and slightly oval buccal capsule. Species of *Popovastrongylus* infect mainly pademelons, *Thylogale* spp., and the smaller wallabies, *Macropus rufogriseus* Desmarest, *Macropus irma* (Desmarest), and *Macropus eugenii* (Desmarest). It also occurs in the larger kangaroos, *Macropus rufus* (Desmarest), *Macropus giganteus* Shaw, and *M. robustus*, in northern Australia where it is uncommon. In southern Australia the only kangaroo hosts known are *Macropus fuliginosus* (Desmarest) on Kangaroo Island, off the shore of South Australia, *M. robustus* in New South Wales, and an accidental infection of *M. robustus* in the Australian Capital Territory.

KEY WORDS: Nematoda, marsupials, macropodids, *Popovastrongylus wallabiae*, *Popovastrongylus tasmaniensis* sp. n., *Popovastrongylus pluteus* sp. n., *Macropus robustus*, *Thylogale billardierii*, taxonomy, Australia.

More than 40 genera of the strongylid family Cloacinidae (Stossich, 1899) are found in the large herbivorous marsupials, kangaroos, wallabies, and wombats of Australia, Irian Jaya, and Papua New Guinea (Beveridge, 1987). *Popovastrongylus* Mawson, 1977, was erected to contain those species occurring in the stomachs of macropodid marsupials (kangaroos and wallabies) that had, among other characters, 4 submedian papillae and 2 amphids borne on a cephalic collar, a circular to oval mouth opening, and a cylindrical to oval buccal capsule with a thick transparent inner layer that may form a shelf-like structure in the lumen. Mawson (1977) included 3 species, *Popovastrongylus wallabiae* (Johnston and Mawson, 1939), the type species, *Popovastrongylus pearsoni* (Johnston and Mawson, 1940), and *Popovastrongylus irma* Mawson, 1977, in the new genus. Subsequently Beveridge (1986) revised the group, expanding the generic definition to encompass a quadrilateral, triangular, or small and triradiate mouth opening and to include a labial collar, internal to the cephalic collar, the buccal capsule sclerotized, often with annular thickening, and the lining inflated and/or forming a shelf. He redescribed *P. pearsoni*, indicating additional features not given in the earlier descriptions by Johnston and Mawson (1939) and Mawson (1971, 1977) and described 2 new species, *Popovastrongylus macropodis* Beveridge, 1986, and *Popovastrongylus thylogale* Beveridge, 1986.

Known hosts for species of *Popovastrongylus* are *Macropus rufogriseus* (Desmarest, 1817) (the red-necked wallaby); *Macropus fuliginosus* (Desmarest, 1817) (the western grey kangaroo); *Macropus eugenii* (Desmarest, 1817) (the tammar wallaby); *Macropus irma* (Jourdan, 1837) (the western brush wallaby); *Macropus rufus* (Desmarest, 1822) (the red kangaroo); *Macropus giganteus* Shaw, 1790 (the eastern grey kangaroo); *Macropus robustus* Gould, 1841 (the common wallaroo); *Thylogale stigmatica* Gould, 1860 (the red-legged pademelon); *Thylogale brunii* (Schreber, 1778) (the dusky pademelon); *Thylogale thetis* (Lesson, 1827) (the red-necked pademelon); and *Petrogale persephone* Maynes, 1982 (the Proserpine rock-wallaby).

Collections of material from *M. robustus* and *Thylogale billardierii* (Desmarest, 1822) in the South Australian Museum, Adelaide (SAMA), were found to have 2 new species of *Popovastrongylus*, which are described in this paper. The cephalic anatomy of *P. wallabiae*, examined for comparative purposes, is also described in greater detail than previously given.

Materials and Methods

No details of hosts, beyond those given below, are known, and there is no record of host bodies having been deposited in any museum. All the parasite material studied had been deposited in the SAMA. Its preservation history is not known, but probably it was fixed in 5%–10% formalin before being stored in 70% ethanol. Specimens were cleared for study in temporary wet mounts in lactophenol prior to examination with the aid of interference contrast light microscopy. Measurements were made with the aid of an ocular micrometer or drawing tube and map measurer. Unless otherwise stated, measurements are given in micrometers as a range followed by the mean in parentheses. Drawings were prepared with the aid of a drawing tube. Terminology used follows that of Beveridge (1986).

Results

Popovastrongylus wallabiae (Johnston and Mawson, 1939) Mawson, 1977 (Figs. 1–6)

SYNONYMS: *Macropostrongylus wallabiae* Johnson and Mawson, 1939; *Gelanostrongylus wallabiae* Popova, 1952.

GENERAL: Cloacinidae: Cloacininae. With characters of the genus *Popovastrongylus* as described by Johnston and Mawson (1939) and Mawson (1977) and redefined by Beveridge (1986). Comparative measurements of specimens from New South Wales and Tasmania are given in Table 1.

DESCRIPTION OF CEPHALIC END: Mouth opening quadrangular in apical view, surrounded by elevated, finely striated labial collar, indented at corners on external margin by submedian papillae; amphids on lateral projections external to labial collar; submedian papillae each with 2 short, medially directed setae; cephalic collar present posterior to labial collar, bearing papillae and amphids. Buccal capsule approximately cylindrical, longer than wide, thickened anteriorly; internal lining of buccal capsule thick, transparent, almost occluding lumen anteriorly but not forming shelf-like projection; outer wall of buccal capsule sclerotized, refractile, thickened in mid region, nonstriated; buccal capsule circular in cross section.

TYPE SPECIMENS: Holotype male, allotype female, SAMA AHC V2832.

TYPE HOST: *Macropus rufogriseus* (Desmarest, 1817).

SITE OF INFECTION: Stomach.

LOCALITY: Bathurst District, New South Wales.

SPECIMENS STUDIED: Types from *M. rufogriseus*: Southern Queensland: 3 female, 1 male, no other collection data, SAMA AHC 6004; from Tasmania: 3 male, 2 female from Launceston, 18 April 1973, SAMA AHC 6006; 3 male, 1 female from Pipers River, collector D. Obendorf 26 January 1982, SAMA AHC 16410.

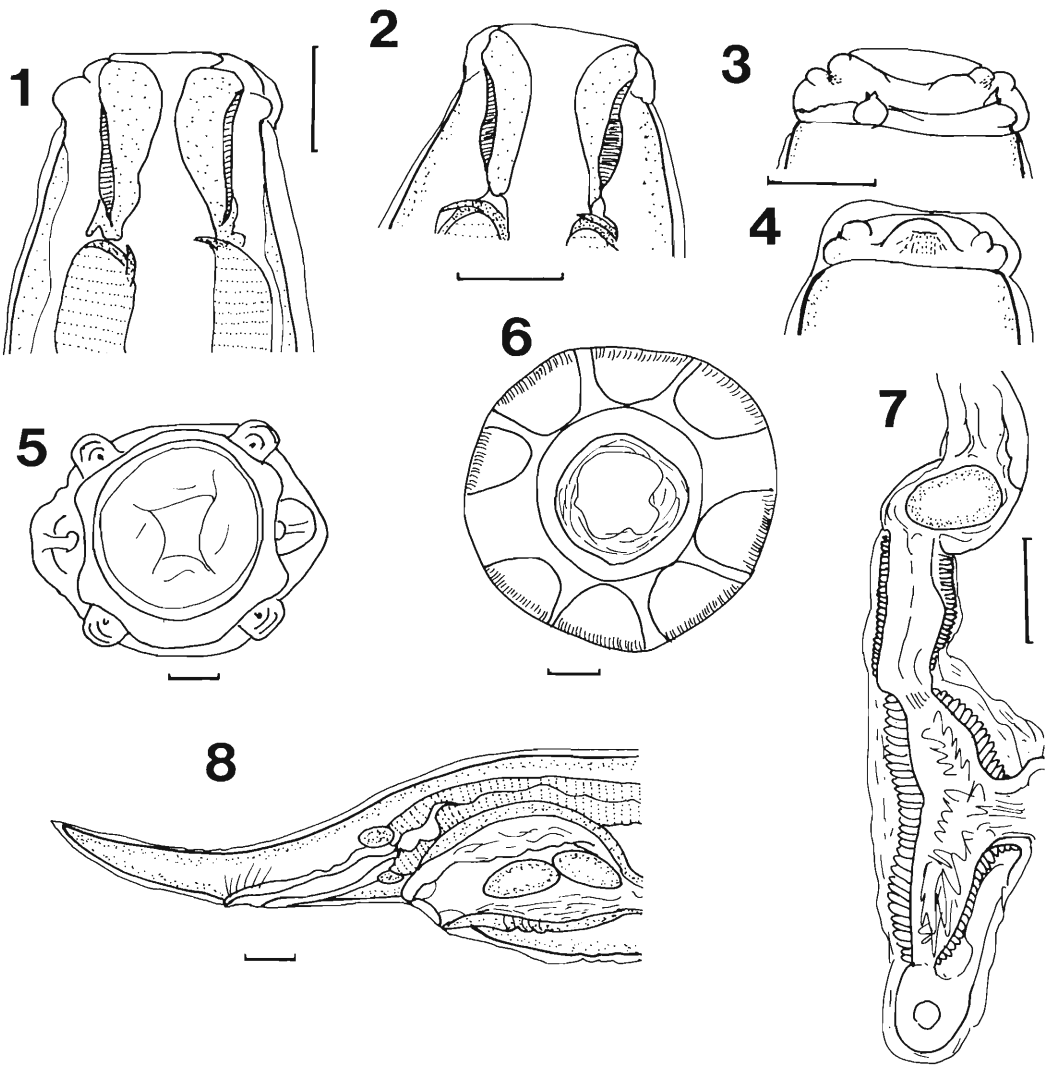
REMARKS: This species was clearly differentiated from *P. pearsoni,* the other species of *Popovastrongylus* occurring in *M. rufogriseus,* by Mawson (1977). Furthermore, Beveridge (1986, pp. 263–264, Fig. 3A, B), in his redescription of the cephalic end of *P. pearsoni,* noted and figured a shelf-like projection of the inner lining of the buccal capsule. Mawson (1977) described a narrow shelf toward the anterior end of the buccal capsule of the type, but not other specimens of *P. wallabiae.* Careful examination of specimens of *P. wallabiae* in this study has shown that a shelf-like projection is not present, but the inner lining of the buccal capsule is thickest at the anterior end.

Mawson (1977) listed *Macropostrongylus wallabiae* Johnston and Mawson, 1939 p. 526 from *Wallabia bicolor* Desmarest, 1804, as a synonym of *P. wallabiae.* This is in error, as *M. wallabiae* was described by Johnston and Mawson (1939) from *Macropus ruficollis* (=*M. rufogriseus).* The material from *Macropus ualabatus* (=*W. bicolor*), originally described as *Macropostrongylus dissimilis* Johnston and Mawson, 1939, was subsequently identified as *Arundelia dissimilis* (Johnston and Mawson, 1939) by Mawson (1977). *Wallabia bicolor* therefore is not a host for *P. wallabiae.*

Popovastrongylus tasmaniensis sp. n. (Figs. 7–21)

Description

GENERAL DESCRIPTION: Small worms, body covered with numerous fine transverse striations; mouth opening oval; surrounded by elevated, finely striated collar, indented on external margin; cephalic collar present, posterior to labial collar, bearing 2 amphids and 4 cephalic papillae, each with 2 prominent setae. Buccal capsule cylindrical, oval in cross-section, slightly longer than wide; walls sclerotized, refractile internal lining thick, transparent, expanded anteriorly. Esophageal corpus long, cylindrical; isthmus short; bulb ovoid; deirids anterior to nerve ring, excretory pore in mid-esophageal position, pos-



Figures 1–8. ***Popovastrongylus wallabiae*** **from** ***Macropus rufogriseus*****. 1. Cephalic end, optical section (ventral view). 2. Cephalic end, optical section (lateral view). 3. Cephalic collar (lateral view). 4. Cephalic collar (ventral view). 5. Mouth opening (apical view). 6. Buccal capsule, optical transverse section showing thickened internal lining. 7, 8.** ***Popovastrongylus tasmaniensis*** **sp. n. from** ***Thylogale billardierii*****. 7. Ovejector (lateral view). 8. Female tail (lateral view). Scale bars: Figures 1–4 = 25 μm; Figures 5, 6 = 10 μm; Figure 7 = 50 μm; Figure 8 = 200 μm.**

terior to nerve ring. Intestinal wall extending anteriorly, surrounding esophageal bulb.

MALES (measurements of 10 specimens): Length 6.5–9.0 (7.5) mm; width 310–460 (385); buccal capsule 50–60 (53) long by 23–43 (38) wide; esophagus 1.04–1.75 (1.61) mm long; nerve ring 535–740 (655), deirids 210–300 (265), excretory pore 690–920 (800) from anterior end; spicules 1.16–1.45 (1.33) mm. Dorsal and lateral lobes of bursa about equal in length; ventral lobes shorter. Ventral rays apposed, reaching margin of bursa; externolateral ray divergent, shorter, almost reaching margin of bursa; mediolateral and posterolateral rays apposed, reaching margin of bursa; externodorsal ray arising close to lateral trunk, not reaching margin of bursa; dorsal ray long, slender at origin, dividing at midlength into 2 arcuate branches that reach margin of bursa; lateral branchlets short, arising soon after bifurcation, terminating in small ele-

Table 1. Comparative measurements (mm) of *Popovastrongylus wallabiae* from *Macropus rufogiseus* from Bathurst, New South Wales (Johnson and Mawson, 1939), and from Launceston and Pipers River, Tasmania.

	New South Wales		Tasmania	
	Male	Female	Male (n = 5)	Female (n = 3)
Length	8.4	11.4	6.0–9.5 (7.7)	10–10 (10.3)
Width	—	—	0.22–0.40 (0.33)	0.48–0.52 (0.49)
Buccal capsule:				
Width × depth	0.025–0.045	—	0.025–0.03 (0.025) × 0.048–0.06 (0.05)	0.025–0.003 (0.028) × 0.05 (0.05)
Esophagus	0.80	—	1.00–1.09 (1.04)	1.02–1.08 (1.04)
Deirids	0.25	—	0.23–0.33 (0.27)	0.28–0.32 (0.29)
Nerve ring	—	—	0.40–0.48 (0.44)	0.45–0.54 (0.49)
Excretory pore (from anterior end)	—	—	0.48–0.64 (0.56)	0.61–0.64 (0.62)
Spicules	0.80	—	0.84–0.97 (0.92)	—
Vulva to posterior	—	0.80	—	0.75–0.89 (0.83)
Tail	—	0.30	—	0.48–0.66 (0.62)
Vagina	—	—	—	0.37–0.40 (0.39)
Eggs	—	0.13 × 0.07	—	0.135 × 0.07

vations on internal surface of bursa. Anterior lip of genital cone small, conical, with single apical papilla; posterior lip smaller, with 2 bilobed appendages. Spicules elongate, alate, anterior extremities irregularly knobbed; distal tips blunt and straight, alae finely striated, extending to spicule tips. Gubernaculum absent.

FEMALES (measurements of 10 specimens): Length 8–12 (10) mm; width 375–595 (470); buccal capsule 50–65 (62) long by 40–65 (50) wide; esophagus 1.60–1.99 (1.79) mm long; nerve ring 605–825 (700); deirids 230–380 (295), excretory pore 730–1090 (895) from anterior end; tail 370–490 (425); vulva to posterior end 635–890 (760); vagina 335–470 (415); eggs 112–138 (120) by 43–66 (54). Tail long, slender, tapering to conical tip; vulva immediately anterior to anus; vagina short, broad ovejector with vestibule longer than sphincters that are longer than infundibula; eggs ellipsoidal.

Taxonomic Summary

TYPE SPECIMENS: Holotype male SAMA AHC 31310; allotype female SAMA AHC 31311; paratypes 5 male, 10 female SAMA AHC 19844.

TYPE HOST: *Thylogale billardierii* (Desmarest, 1822).

TYPE LOCALITY: Launceston, Tasmania.

SITE OF INFECTION: Stomach.

SPECIMENS STUDIED: Types from *T. billardierii*, from Tasmania; 10 male, 14 female from Launceston, collectors D. Obendorf, I. Beveridge, 20 February 1990, 18 November 1990, SAMA AHC 19844, AHC 19791, AHC 26578; 3 male, 4 female from Piper's River, collector D. Obendorf, 26 January 1983, SAMA AHC 16373, AHC 31312; 1 female from Georgetown, collector D. Obendorf, 28 June 1982, SAMA AHC 16398; 2 female from Golconda, collector I. Beveridge, April 1977, SAMA AHC 13925.

ETYMOLOGY: The name of the new species refers to its type locality.

Remarks

Popovastrongylus tasmanientsis sp. n. is readily distinguished from its congeners by its oval mouth opening and buccal capsule, in having the intestinal wall extending anteriorly to surround the esophageal bulb, and spicules longer than 1150 μm. *Popovastrongylus pearsoni*, which also occurs in *M. rufogriseus* from Tasmania (Mawson, 1977), has a distinct shelf-like projec-

tion in a circular buccal capsule. The nerve ring is in the mid-esophageal position in *P. tasmaniensis* but is posterior and surrounds the isthmus in *P. pearsoni. Popovastrongylus wallabiae,* the other species occuring in Tasmania (Mawson, 1977), has a quadrangular mouth opening and a buccal capsule circular in cross-section. The submedian papillae of *P. wallabiae* are short, and the setae are not easily seen at low magnifications, while *P. tasmaniensis* has prominent papillae and setae. The dorsal lobe of the bursa of *P. tasmaniensis* is shorter (about the same length as the lateral lobes), not longer than the lateral lobes as in *P. wallabiae.* Additional characters that distinguish *P. tasmaniensis* from *P. irma* are not having the base of the buccal capsule thickened by an outer sclerotized ring and having the nerve ring in a mid-esophageal, not posterior, position surrounding the isthmus. The shape of the posterior end of the female of *P. irma,* constricted between vulva and anus and markedly swollen in the vaginal region, is unique to *P. irma.*

Popovastrongylus thylogale, also occurring in pademelons, can be further distinguished from *P. tasmaniensis* by an annular thickening around the middle of the buccal capsule, the posterior position of the nerve ring, and the dorsal lobe longer than the lateral lobes of the bursa.

Popovastrongylus tasmaniensis differs further from *P. macropodis* in having a relatively thinner inflation of the lining of the buccal capsule, which is expanded anteriorly but does not almost occlude the lumen as in *P. macropodis.*

Popovastrongylus pluteus sp. n. (Figs. 22–31)

Description

GENERAL DESCRIPTION: Small worms; body covered with numerous fine transverse striations; mouth opening circular, surrounded by elevated, finely striated collar indented on external margin; cephalic collar present, posterior to lateral collar bearing 2 amphids and 4 cephalic papillae each with 2 setae. Buccal capsule cylindrical, circular in cross-section, longer than wide; walls sclerotized, refractile, thickened in posterior part; inner lining thick, transparent, folded in mid-region to produce irregular shelf-like projection, almost occluding lumen. Esophageal corpus long, isthmus not distinct, bulb ovoid.

MALES (measurements of 2 specimens): Length 5, 6 mm; width 240, 290; buccal capsule 33, 46 long by 26, 26 wide; esophagus 0.985, 1.01 mm long; nerve ring to anterior end 402, 402; deirids to anterior end 135, excretory pore to anterior end 470, 455. Spicules 950. Dorsal and lateral lobes of bursa about equal in length, ventral lobes shorter. Ventral rays apposed, reaching margin of bursa; externolateral ray divergent, almost reaching margin of bursa; mediolateral and posterolateral rays apposed, reaching margin of bursa; externodorsal ray arising close to lateral trunk, almost reaching margin of bursa; dorsal ray dividing at midlength into 2 arcuate branches that reach margin of bursa, lateral branchlets short, arising close to bifurcation; terminating in small elevations on internal surface of bursa. Anterior lip of genital cone large and conical, with single apical papilla; posterior lip smaller, with 2 bilobed appendages. Spicules elongate, alate, tips not seen. Gubernaculum absent.

FEMALES (measurements of 10 specimens): Length 7–8 (7.4) mm; width 255–375 (305); buccal capsule 35–50 (40) long by 22–30 (26) wide; esophagus 1.02–1.14 (1.08) mm long; nerve ring 345–390 (370), deirids 105–140 (125), excretory pore 400–475 (435) from anterior end; tail 470–585 (530) long, vulva to posterior end 705–885 (790); vagina 400–595 (505); eggs 95–105 (100) by 42–52 (45). Tail long, slender with conical tip; vulva immediately anterior to anus; vagina short, broad at anterior end, ovejector with vestibule and sphincters about same length, infundibula shorter; eggs ellipsoidal.

Taxonomic Summary

TYPE SPECIMENS: Holotype male SAMA AHC 31253, allotype female AHC 31324, paratypes 3 female, 1 male AHC 14546.

TYPE HOST: *Macropus robustus* Gould, 1841.

TYPE LOCALITY: Rivertree, New South Wales.

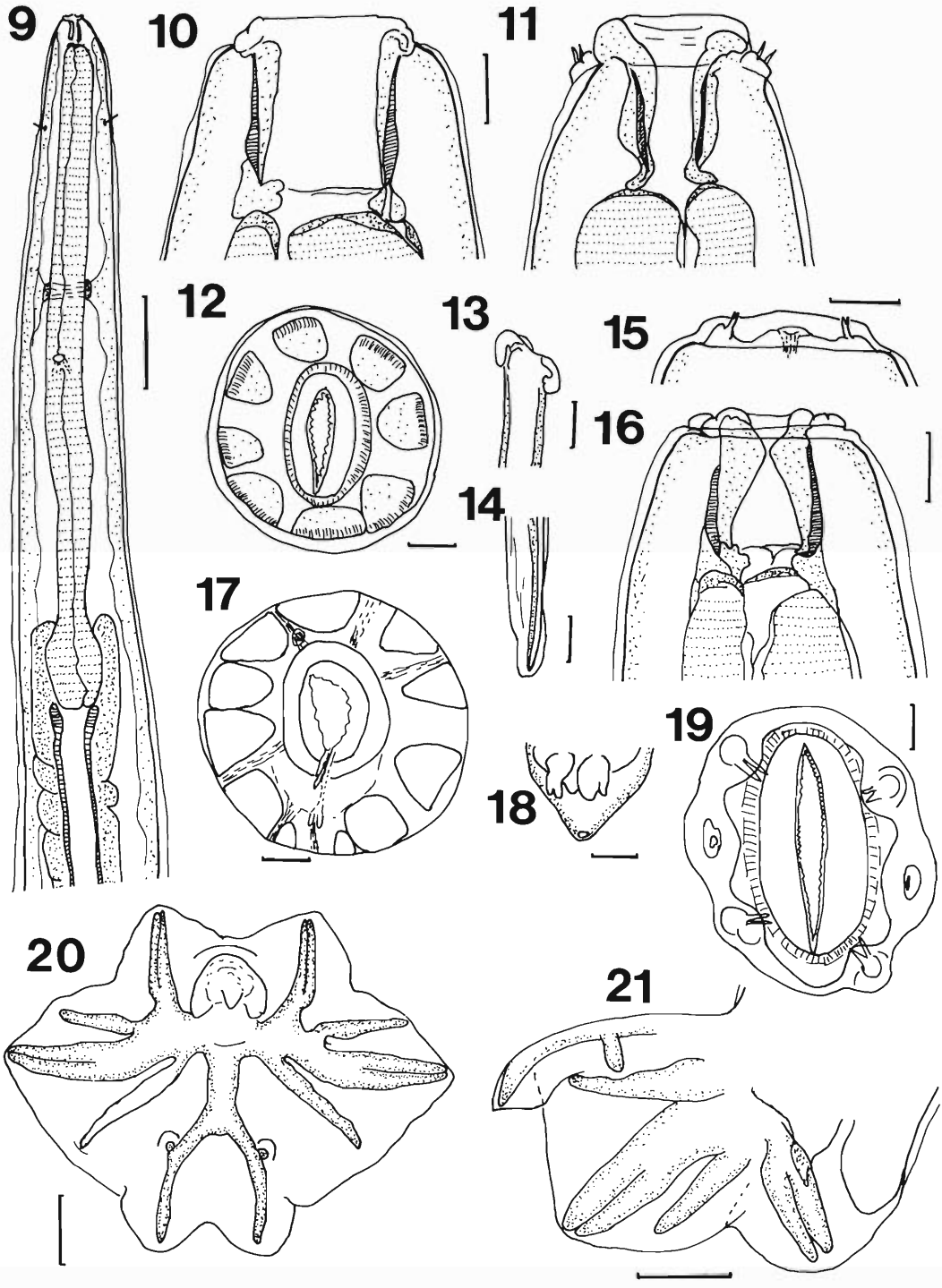
SITE OF INFECTION: Stomach.

SPECIMENS STUDIED: Types from *M. robustus,* New South Wales; 17 female from Rivertree, 28 August 1975, SAMA AHC 31252.

ETYMOLOGY: The specific name refers to the shelf-like projection in the buccal capsule.

Remarks

Popovastrongylus pluteus sp. n. is 1 of 2 species with a shelf-like projection in the buccal capsule, the other being *P. pearsoni. Popova-*



strongylus pluteus differs from *P. pearsoni* in the shape of the mouth opening, circular not quadrangular in apical view; the buccal capsule circular in cross section rather than slightly oval; and the amphids not on extremely prominent lateral projections. The nerve ring and excretory pore of *P. pearsoni* are more posterior (Mawson, 1977, Fig. 31, p. 57), with the nerve ring surrounding the junction of the isthmus and corpus of the esophagus, than in *P. pluteus,* which has the nerve ring and excretory pore in the mid-esophageal region. The branchlets of the dorsal ray of *P. pluteus* arise closer to its bifurcation than do those of *P. pearsoni.*

Popovastrongylus pluteus is similar in general features to *P. wallabiae* but differs in the features of the cephalic end, particularly in the form of the inner lining of the buccal capsule. *Popovastrongylus wallabiae* does not have an internal shelf, and the shape of the mouth opening is circular, not quadrangular. The deirids, nerve ring, and excretory pore of *P. pluteus,* although located in the mid-region of the esophagus, are each more anterior than their counterparts on *P. wallabiae* (135, 402, and 463 compared with 270, 440, and 560, respectively, for males). The dorsal lobe of the bursa of *P. pluteus* is about the same length as the lateral lobes, but in *P. wallabiae* it is longer. The vagina of *P. wallabiae* (370–400) is shorter and its eggs (95–105 by 42–52) are smaller (135 by 70) than in *P. pluteus.*

Popovastrongylus pluteus can be distinguished from *P. macropodis,* which also occurs in *M. robustus,* by the presence of a shelf in the buccal capsule and in having a circular, not triangular mouth opening. The buccal capsule is more slender than that of *P. macopodis* (40 × 26 compared with 37 × 30), and the inner lining is not as inflated as that of *P. macropodis.* The vagina is longer (400–595) in *P. pluteus,* compared with 360–400 in *P. macropodis.*

Discussion

This study has increased the number of known hosts of *Popovastrongylus* to include *T. billardierii* and extended the known distribution of the genus to include New South Wales. Of the 2 new species, *P. tasmaniensis* has been found only in *T. billardierii,* the Tasmanian pademelon, from Tasmanian localities, and *P. pluteus* only in *M. robustus,* the common wallaroo from New South Wales.

Of the previously known species, *P. macropodis* is found in wallaroos as well as the other large kangaroos, *M. rufus* and *M. giganteus,* the red and eastern grey kangaroos, but only in northern Queensland (Beveridge, 1986; Arundel et al., 1979, 1990). *Popovastrongylus thylogale,* also found in pademelons, has a distribution limited to *T. stigmatica,* the red-legged pademelon, and *T. thetis,* the red-necked pademelon in Queensland. There is a single report of an accidental infection of *P. thylogale* from a captive agile wallaby, *Macropus agilis* (Gould, 1842) (Spratt et al., 1991). *Popovastrongylus thylogale* does not, however, occur in free-ranging agile wallabies (Speare et al., 1983). Other northern hosts of this parasite are *P. persephone,* the Proserpine rock-wallaby, a host that harbors several nematode species that normally occur in pademelons and are not normally found in other species of rock-wallaby (Begg et al., 1995; Beveridge, 1986), and *T. brunii,* the dusky pademelon, found only in Papua New Guinea. This latter occurrence emphasizes the northern distribution of *P. thylogale* as it has been found in neither the red-legged pademelon nor the red-necked pademelon in New South Wales (Beveridge, 1986; Smales, 1997).

Popovastrongylus wallabiae occurs only in the red-necked wallaby, *M. rufogriseus,* and is the most widely distributed species of the genus, being found in southern Queensland and Tasmania (Mawson, 1977). It has not been reported

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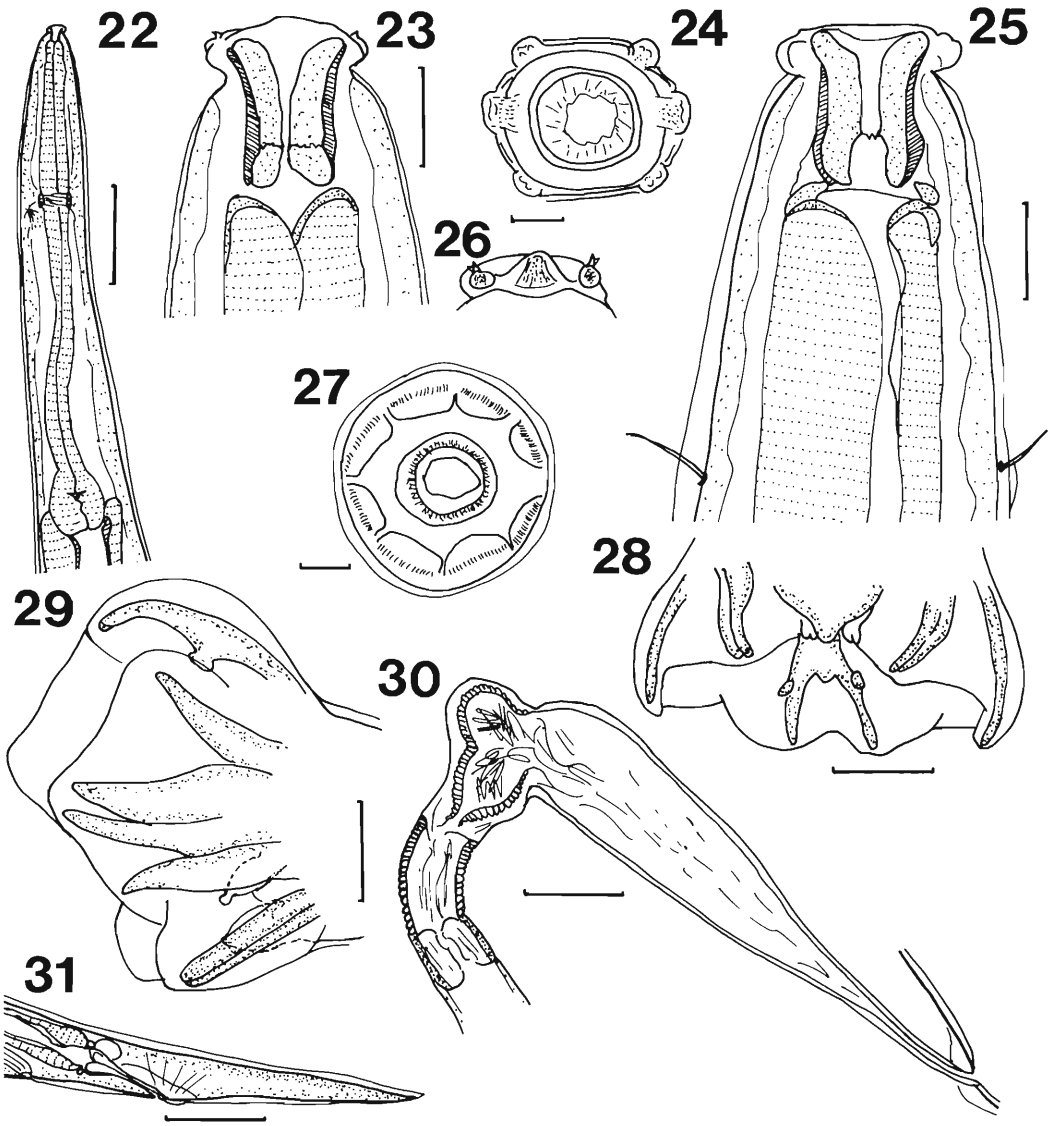
Figures 9–21. ***Popovastrongylus tasmaniensis*** **sp. n. from** ***Thylogale billardierii.*** **9. Anterior end (ventral view). 10. Cephalic end, optical section (lateral view). 11. Cephalic end, optical section (ventral view). 12. Buccal capsule, transverse optical section, at mid-level. 13. Spicule, anterior end (lateral view). 14. Spicule tip (lateral view). 15. Cephalic collar (lateral view). 16. Cephalic end (dorsal view). 17. Buccal capsule, transverse optical section at posterior level. 18. Genital cone (dorsal view). 19. Mouth opening (en face view). 20. Bursa (apical view). 21. Bursa (lateral view). Scale bars: Figure 9 = 200 μm; Figures 10, 11, 13–16, 18 = 25 μm; Figures 12, 17, 19 = 10 μm; Figures 20, 21 = 50 μm.**



Figures 22–31. ***Popovastrongylus pluteus*** **sp. n. from** ***Macropus robustus.*** **22. Anterior end (lateral view). 23. Cephalic end, optical section (lateral view). 24. Mouth opening (en face view). 25. Cephalic end, optical section (ventral view). 26. Cephalic collar (lateral view). 27. Buccal capsule, transverse optical section at level of shelf. 28. Bursa (ventral view). 29. Bursa (lateral view). 30. Ovejector (lateral view). 31. Female posterior end (lateral view). Scale bars: Figures 22, 31 = 200 μm; Figures 23, 25, 26 = 25 μm; Figures 24, 27 = 10 μm; Figures 28, 29 = 50 μm; Figure 30 = 100 μm.**

in red-necked wallabies from New South Wales, Victoria, or South Australia. This may be because of either a lack of sampling effort (the parasite being present but not detected) or a disjunct distribution of the parasite. *Popovastrongylus pearsoni* also occurs in red-necked wallabies from the same localities in Tasmania as *P. wallabiae* and has only been found on continental Australia in a common wallaroo in the Tidbinbilla Nature Reserve in the Australian Capital Territory. Since the macropod population on the reserve includes red-necked wallabies, tammars, and species of rock-wallaby, it is reasonable to suppose that this record was an accidental infection. It does, however, occur in 3 other hosts, the tammar and western grey kan-

garoo from Kangaroo Island (Smales and Mawson, 1978; Beveridge, 1986) and *Petrogale lateralis* Gould, 1842, the black-footed wallaby from Pearson Island (Johnston and Mawson, 1940; Mawson, 1971; Beveridge, 1986), both southern localities offshore from South Australia.

Popovastrongylus irma is found only in the western brush wallaby in southwestern Australia (Mawson, 1977), but in contrast to *P. pearsoni* it has not been reported from sympatric hosts in the region.

In general, species of *Popovastrongylus* infect mainly pademelons and the small wallabies, *M. irma, M. eugenii,* and *M. rufogriseus.* The genus is not common in the larger kangaroos from northern Queensland, with only 1 or 2 nematodes present in each individual host (Beveridge, 1986) and does not normally occur in kangaroos in the southern states (Beveridge and Arundel, 1979; Arundel et al., 1979, 1990). Neither is the genus found in *Wallabia bicolor,* the swamp wallaby (Beveridge et al., 1985), nor the macropodid genera *Hypsiprymnodon, Aepyprymnus, Onychogalea, Lagorchestes,* or *Dendrolagus* (Beveridge et al., 1992).

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Literature Cited

Arundel, J. H., I. Beveridge, and P. J. Presidente. 1979. Parasites and pathological findings in enclosed and free-ranging populations of *Macropus rufus* (Desmarest) (Marsupialia) at Menindee, New South Wales. Australian Wildlife Research 6:361–379.

———, **K. J. Dempster, K. E. Harrigan, and R. Black.** 1990. Epidemiological observations on the helminth parasites of *Macropus giganteus* Shaw in Victoria. Australian Wildlife Research 17:39–51.

Begg, M., I. Beveridge, N. B. Chilton, P. M. Johnson, and M. G. O'Callaghan. 1995. Parasites of the Proserpine rock wallaby, *Petrogale persephone* (Marsupialia: Macropodidae). Australian Mammalogy 18:45–53.

Beveridge, I. 1986. New species and new records of *Popovastrongylus* Mawson, 1977 (Nematoda: Cloacininae) from Australian marsupials. Bulletin du Muséum National d'Histoire Naturelle, 4th Series 8:257–265.

———. 1987. The systematic status of Australian Strongyloidea. Bulletin du Muséum National d'Histoire Naturelle, 4th Series 9:107–126.

———, **and J. H. Arundel.** 1979. Helminth parasites of grey kangaroos, *Macropus giganteus* Shaw and *M. fuliginosus* (Desmarest) in Eastern Australia. Australian Wildlife Research 6:69–77.

———, **P. J. A. Presidente, and R. Speare.** 1985. Parasites and associated pathology of the swamp wallaby, *Wallabia bicolor* (Marsupialia). Journal of Wildlife Diseases 21:377–385.

———, **R. Speare, P. M. Johnson, and D. M. Spratt.** 1992. Helminth parasite communities of macropodoid marsupials of the genera *Hypsiprymnodon, Aepyprymnus, Thylogale, Onychogalea, Lagorchestes* and *Dendrolagus* from Queensland. Wildlife Research 19:359–376.

Johnston, T. H., and P. M. Mawson. 1939. Strongylate nematodes from marsupials in New South Wales. Proceedings of the Linnean Society of New South Wales 64:513–536.

———, **and** ———. 1940. Nematodes from South Australia marsupials. Transactions of the Royal Society of South Australia 64:95–100.

Mawson, P. M. 1971. Pearson Island Expedition 1969. 8. Helminths. Transactions of the Royal Society of South Australia 95:169–183.

———. 1977. Revision of the genus *Macropostrongylus* and descriptions of three new genera: *Popovastrongylus, Dorcopsinema* and *Arundelia* (Nematoda: Trichonematidae). Transactions of the Royal Society of South Australia 101:51–62.

Smales, L. R. 1997. The status of *Cyclostrongylus medioannulatus* Johnston and Mawson, 1940. Transactions of the Royal Society of South Australia 121:165.

———, **and P. M. Mawson.** 1978. Nematode parasites of the Kangaroo Island Wallaby, *Macropus eugenii* (Desmarest). 1. Seasonal and geographic distribution. Transactions of the Royal Society of South Australia 102:9–16.

Speare, R., I. Beveridge, P. M. Johnson, and L. A. Corner. 1983. Parasites of the agile wallaby *Macropus agilis* (Marsupialia). Australian Wildlife Research 10:89–96.

Spratt, D. M., I. Beveridge, and E. L. Walter. 1991. A catalogue of Australasian monotremes and marsupials and their recorded helminth parasites. Records of the South Australian Museum, Monograph Series 1:1–105.

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67(1), 2000 pp. 60–65

Angiostoma onychodactyla sp. n. (Nematoda: Angiostomatidae) and Other Intestinal Helminths of the Japanese Clawed Salamander, *Onychodactylus japonicus* (Caudata: Hynobiidae), from Japan

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ABSTRACT: *Angiostoma onychodactyla* sp. n. from the intestines of the Japanese clawed salamander, *Onychodactylus japonicus,* is described and illustrated. *Angiostoma onychodactyla* is most similar to *Angiostoma plethodontis* in that lateral alae are absent, and there is a bulb without valves. The major difference between these 2 species is in the number and position of the caudal papillae. In addition, this sample of *O. japonicus* harbored 3 species of trematodes, *Cephalouterina leoi, Mesocoelium brevicaecum,* and *Pseudopolystoma dendriticum,* 1 species of nematode, *Parapharyngodon japonicus,* and 1 acanthocephalan species (cystacanth stage).

KEY WORDS: *Angiostoma onychodactyla* sp. n., Angiostomatidae, Japanese clawed salamander, *Onychodactylus japonicus,* Hynobiidae, Japan.

Onychodactylus japonicus (Houttuyn, 1782), the Japanese clawed salamander, is restricted to forested mountainous areas of Honshu and Shikoku Islands, Japan (Kuzmin, 1995). Previously reported helminths of *Onychodactylus japonicus* include the monogenetic trematode *Pseudopolystoma dendriticum* (Ozaki, 1948), the digenetic trematodes *Cephalouterina leoi* Uchida, Uchida, and Kamei, 1986, and *Mesocoelium brevicaecum* Ochi, 1930, the cestode *Cylindrotaenia* sp. (=*Baerietta* sp., larvae only), and the nematodes *Amphibiocapillaria tritonispunctati* (Diesing, 1851) (=*Capillaria filiformis* (Linstow, 1881)), *Parapharyngodon japonicus* Bursey and Goldberg, 1999; *Pseudoxyascaris japonicus* Uchida and Itagaki, 1979, *Pharyngodon* sp., and *Rhabditis* sp. (Wilkie, 1930; Pearse, 1932; Ozaki, 1948; Uchida and Itagaki, 1979; Uchida et al., 1986; Bursey and Goldberg, 1999).

Further study of the sample of *Onychodactylus japonicus* examined by Bursey and Goldberg (1999) revealed 43 females and 17 males of an undescribed species of *Angiostoma.* To our knowledge, there are no reports of species of *Angiostoma* from Japanese salamanders, although Wilkie (1930) reported unidentified rhabditids from *Hynobius retardatus* Dunn, 1923, the Hokkaido salamander, and *O. japonicus* collected in Yumoto, Fukushima Prefecture. The purpose of this paper is to describe a new species of nematode, *Angiostoma onychodactyla,* from the salamander *Onychodactylus japonicus* from Japan and to provide a current parasite list for this host.

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Material and Methods

Sixty-eight *Onychodactylus japonicus* were examined (collection data given in Bursey and Goldberg, 1999). All had been captured by hand and fixed in neutral buffered 10% formalin, then preserved in 70% alcohol. The body cavity was opened by a longitudinal incision from vent to throat, and the gastrointestinal tract was removed and opened longitudinally. Nematodes were placed in undiluted glycerol, allowed to clear, and examined under a light microscope. Trematodes were stained in hematoxylin and mounted in balsam for study. Measurements are given in micrometers.

Results

In addition to the previously described *Parapharyngodon japonicus* and *Angiostoma onychodactyla* described below, 3 species of trematodes (*Cephalouterina leoi, Mesocoelium brevicaecum, Pseudopolystoma dendriticum*) and 1 species of acanthocephalan (unidentified cystacanth) were also found. Forty (59%) of 68 salamanders were infected with helminths. Prevalence, mean intensity, and mean abundance for each helminth species are presented in Table 1.

Angiostoma onychodactyla sp. n. (Figs. 1–9)

Description

GENERAL: Transparent nematodes lacking lateral alae. Cuticle thin, nonstriated. Sexual di-

Table 1. Prevalence, intensity, and abundance of helminths collected from 68 *Onychodactylus japonicus*.

Helminth	Prevalence (%)	Mean intensity ± SD	Mean abundance ± SD	Site
Trematoda				
Cephalouterina leoi	3	1	0.03 ± 0.17	Small intestine
Mesocoelium brevicaecum	5	5.0 ± 2.0	0.22 ± 1.09	Small intestine
Pseudopolystoma dendriticum	1	1	0.02 ± 0.12	Bladder
Nematoda				
Angiostoma onychodactyla sp. n.	24	3.8 ± 3.8	0.88 ± 2.41	Small intestine
Parapharyngodon japonicus	38	4.8 ± 5.6	1.82 ± 4.13	Large intestine
Acanthocephala				
Unidentified cystacanths*	1	1	0.02 ± 0.12	Coelom

* New host record.

morphism not prominent. Oral opening with 3 lips. Esophagus with corpus, isthmus, and pseudobulb, nerve ring at level of anterior isthmus. Excretory pore anterior to the esophagointestinal junction.

MALE (holotype and 9 paratypes; mean and range): Length 3.36 (2.60–3.90) mm. Maximum width 103 (89–128). Buccal cavity 9 (6–11) deep. Length of esophagus 297 (251–319), corpus 160 (145–175), isthmus 76 (66–88), bulb 60 (55–68). Nerve ring 192 (188–285) and excretory pore 236 (188–285) from anterior end. Spicules equal, 128 (120–143), well chitinized, arcuate. Gubernaculum well chitinized, 44 (37–48). Testis single and reflexed. Caudal alae well-developed, supported by 8 pairs of postcloacal pedunculate papillae that do not reach the ala edge. Tail spike extends approximately 20 beyond bursa. Subventral cloacal sensilla absent. Phasmids lateral, immediately posterior to terminal pair of postcloacal papillae.

FEMALE (allotype and 9 paratypes; mean and range): Length 4.21 (3.25–5.07) mm. Width at level of vulva 117 (89–153). Buccal cavity 9 (6–11) deep. Esophagus 301 (274–342), corpus 162 (149–170), isthmus 78 (68–86), bulb 60 (57–63). Nerve ring 202 (171–274), excretory pore 228 (200–268) from anterior end, respectively. Vulva 2.06 (1.53–2.40) mm from anterior end, slightly pre-equatorial. Tail elongated, 189 (171–239). Amphidelphic; uteri divergent; anterior uterus directed anteriorly, posterior uterus directed posteriorly; ovaries reflexed. Uteri containing numerous elliptical eggs, 56 (51–58) × 48 (46–57), larvae absent.

Taxonomic Summary

TYPE HOST: *Onychodactylus japonicus* (Houttuyn, 1782), Japanese clawed salamander.

TYPE LOCALITY: Hineomata, Fukushima Prefecture, Honshu, Japan, 37°01′N, 139°23′E.

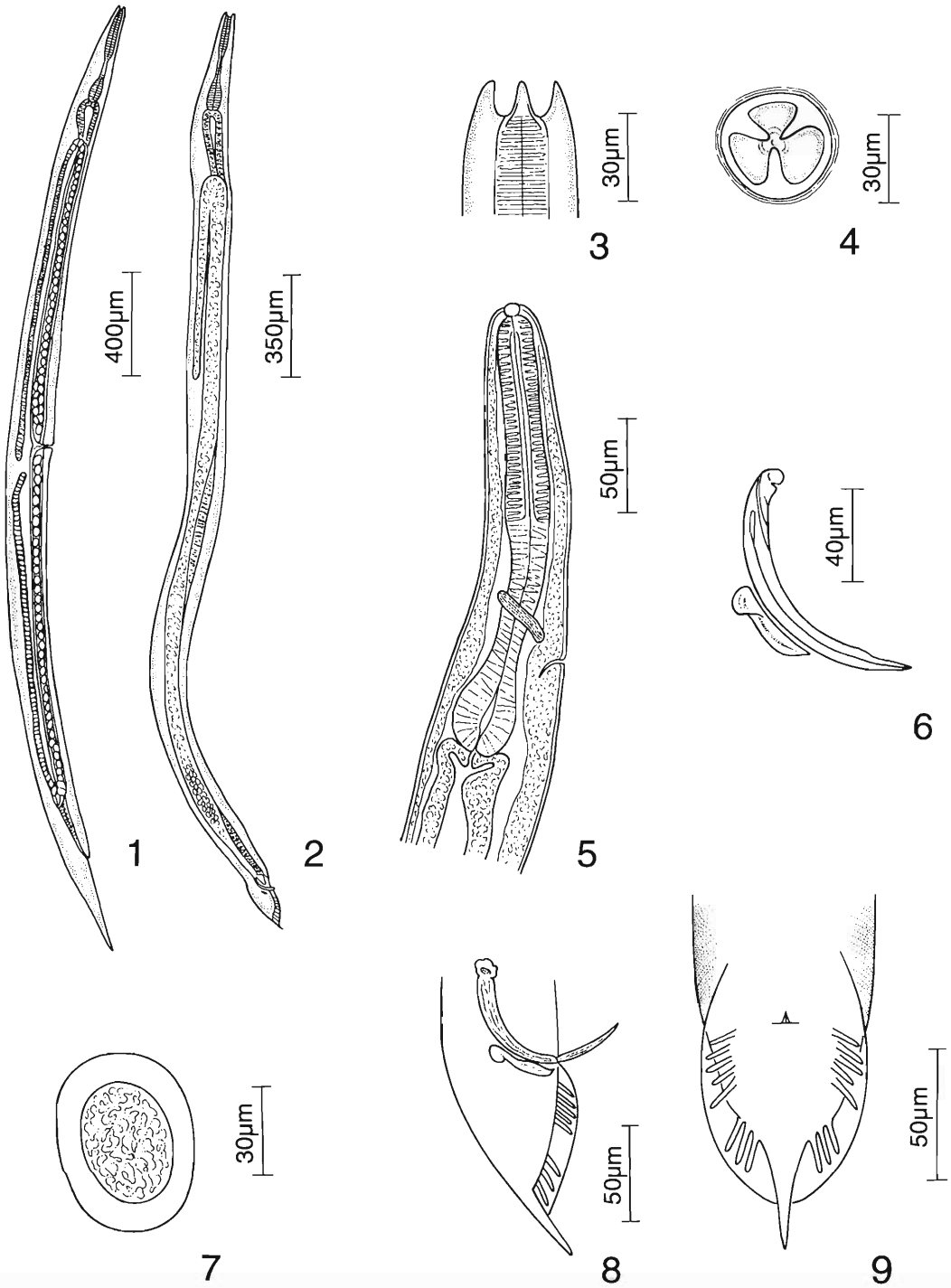
SITE OF INFECTION: Small intestine.

TYPE SPECIMENS: Holotype: male, United States National Parasite Collection (USNPC), Beltsville, Maryland, USNPC 88645; allotype, female, USNPC 88646; paratypes (9 males, 9 females) USNPC 88647.

ETYMOLOGY: The new species is named in reference to the genus of the host.

Remarks

The genus *Angiostoma* now consists of 10 species in the monogeneric family Angiostomatidae (order Rhabditida); 8 species infect terrestrial gastropods and 2 species are from salamanders. The type species, *Angiostoma limacis* Dujardin, 1845, has been collected from arionid gastropods in western Europe (Morand and Spiridonov, 1989). Six additional species are known from terrestrial gastropods in the Palaearctic Realm: *Angiostoma asamati* Spiridonov, 1985, from a gigantolimacid (Spiridonov, 1985); *Angiostoma aspersae* Morand, 1986, from helicids (Morand, 1986); *Angiostoma dentifera* (Mengert, 1953) from limacids (Morand and Spiridonov, 1989); *Angiostoma kimmeriensis* Korol and Spiridonov, 1991, from a zonitid (Korol and Spiridonov, 1991); *Angiostoma spiridonovi* Morand, 1992, and *Angiostoma stammeri* (Mengert, 1953) from limacids (Mengert, 1953; Morand, 1992). *Angiostoma schizoglossae* Morand and Barker, 1995, was described from a specimen taken from a rhytidid gastropod endemic to New Zealand, Australian Realm (Morand and Barker, 1995). *Angiostoma plethodontis* Chitwood, 1933, was described from the northern redback salamander, *Plethodon cinereus* (Green,



Figures 1–9. ***Angiostoma onychodactyla*** **sp. n. 1. Female, entire, lateral view. 2. Male, entire, lateral view. 3. Female, anterior end. 4. Female, en face view of anterior end. 5. Male, esophageal region. 6. Spicule, gubernaculum. 7. Egg. 8. Male, posterior end, lateral view. 9. Male, posterior end, ventral view.**

Table 2. Parasite list for *Onychodactylus japonicus*.

Helminth	Prevalence	Reference
Trematoda		
Pseudopolystoma dendriticum	Not given	Ozaki, 1948
	Not given	Uchida and Itagaki, 1979
	1% (1/68)	This study
Mesocoelium brevicaecum	Not given	Uchida et al., 1986
	4% (3/68)	This study
Cephalouterina leoi	Not given	Uchida et al., 1986
	3% (2/68)	This study
Cestoda		
Cylindrotaenia sp. (immature)	Not given	Uchida et al., 1986
Nematoda		
Amphibiocapillaria tritonispunctati	5% (1/20)	Pearse, 1932
Angiostoma onychodactyla sp. n.	24% (16/68)	This study
Parapharyngodon japonicus	38% (26/68)	Bursey and Goldberg, 1999
Pseudoxyascaris japonicus	Not given	Uchida and Itagaki, 1979
Rhabditis sp.	Not given	Wilkie, 1930
Unidentified nematode	Not given	Uchida et al., 1986
Unidentified oxyurids	Not given	Wilkie, 1930
	45% (9/20)	Pearse, 1932
Acanthocephala		
Unidentified cystacanths	1% (1/68)	This study

1818), a Nearctic salamander (Chitwood, 1933). *Angiostoma onychodactyla* is the second species to be described from salamanders, albeit a Palaearctic salamander.

Discussion

A key to the known species of *Angiostoma* was published by Morand and Barker (1995). Of these 8 species, *Angiostoma onychodactyla* is more similar to *A. limacis* and *A. plethodontis* in that lateral alae are absent, and there is a bulb without valves. In *A. limacis,* the tip of the tail has denticles, while in *A. onychodactyla* and *A. plethodontis,* the tail is elongated and without denticles. The major difference between *A. onychodactyla* and *A. plethodontis* is in the number and position of the caudal papillae, *A. onychodactyla* with 8 pairs (all postcloacal) compared with *A. plethodontis* with 9 pairs (2 precloacal pairs and 7 postcloacal). Other differences include length of spicules (128 in *A. onychodactyla* compared with 60) and length of gubernaculum (44 compared with 25). Adamson (1986) suggested that salamander hosts acquired infection by ingesting parasitized molluscs, but more work will be required to test this hypothesis.

Onychodactylus japonicus also harbored 3 species of trematodes: 2 individuals of *Cephalouterina leoi,* 12 of *Mesocoelium brevicaecum,* and 1 of *Pseudopolystoma dendriticum*; 1 species of nematode, 124 individuals of *Parapharyngodon japonicus*; and 1 cystacanth of an unidentified species of acanthocephalan. These species have been previously reported from *O. japonicus.*

Cephalouterina leoi was described from 3 specimens found by Uchida et al. (1986) during examination of the small intestines of 900 *O. japonicus.* This is the second report of *C. leoi*; the only known host is *O. japonicus. Mesocoelium brevicaecum,* originally described by Goto and Ozaki (1929a) from the intestine of the Japanese common toad, *Bufo japonicus* Schlegel, 1838, is often found in the small intestine of other Japanese amphibians, namely, the Kajika frog, *Buergeria buergeri* (Temminck and Schlegel, 1838), the wrinkled frog, *Rana rugosa* Temminck and Schlegel, 1838, the Mitsjama salamander, *Hynobius nebulosus* (Schlegel, 1838), the Stejneger's oriental salamander, *H. stejnegeri* Dunn, 1923, and the Japanese newt, *Triturus pyrrhogaster* (Boie, 1826) (Goto and Ozaki, 1929a, b). Nasir and Díaz (1971) referred all Japanese species of *Mesocoelium* to *M. brevicaecum.* Pearse (1932) was the first to report *M. brevicaecum* from *O. japonicus* and this is the second report of *M. brevicaecum* in this host. *Pseudopolystoma dendriticum* was originally de-

scribed as *Polystoma dendriticum* by Ozaki (1948) from individuals taken from the urinary bladder of *O. japonicus*. Yamaguti (1963) revised the taxonomy. Uchida and Itagaki (1979) reported it from the same host. This is the third report of *P. dendriticum*; the only known host is *O. japonicus*.

In addition to these trematodes, 111 females and 13 males of *Parapharyngodon japonicus* were harbored by 26 (38%) *O. japonicus*, the only known host. To our knowledge, there are no other reports of *Parapharyngodon* from Japanese salamanders; however, Hasegawa (1988) reported an unidentified but different species of *Parapharyngodon* from a lizard, the Japanese ateuchosaurus, *Ateuchosaurus pellopleurus* (Hallowell, 1860), from Okinawa, Japan.

The single acanthocephalan was too immature to identify. Van Cleave (1925) described *Acanthocephalus nanus* from the intestine of *Triturus* (=*Diemictylus*) *pyrrhogaster* and *Rana rugosa* from Japan and Pearse (1932) reported *A. nanus* as well as unidentified encysted acanthocephalans from *T. pyrrhogaster* and the giant salamander, *Megalobatrachus japonicus* (Temminck, 1837), collected near Tokyo. This is the first report of acanthocephalan cystacanths in *O. japonicus*.

All helminths were deposited in the United States National Parasite Collection, Beltsville, Maryland: *Cephalouterina leoi*, USNPC 88648; *Mesocoelium brevicaecum*, USNPC 88649; *Pseudopolystoma dendriticum*, USNPC 88650; *Parapharyngodon japonicus*, USNPC 88651; acanthocephalan cystacanth, USNPC 88652. A list of the known parasites of *O. japonicus* is given in Table 2. More work will be required to determine the distribution patterns and the variety of hosts of the helminths found in this study.

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Literature Cited

Adamson, M. L. 1986. Modes of transmission and evolution of life histories in zooparasitic nematodes. Canadian Journal of Zoology 64:1375–1384.

Bursey, C. R., and S. R. Goldberg. 1999. *Parapharyngodon japonicus* sp. n. (Nematoda: Pharyngodonidae) from the Japanese clawed salamander, *Onychodactylus japonicus* (Caudata: Hynobiidae) from Japan. Journal of the Helminthological Society of Washington 66:180–186.

Chitwood, B. G. 1933. On some nematodes of the superfamily Rhabditoidea and their status as parasites of reptiles and amphibians. Journal of the Washington Academy of Science 23:508–520.

Goto, S., and Y. Ozaki. 1929a. Brief notes on new trematodes I. Japanese Journal of Zoology 2:213–217.

———, **and** ———. 1929b. Brief notes on new trematodes III. Japanese Journal of Zoology 3:73–82.

Hasegawa, H. 1988. *Parapharyngodon* sp. (Nematoda: Pharyngodonidae) collected from the lizard, *Ateuchosaurus pellopleurus* (Sauria: Scincidae) on Okinawajima Island, Japan. Akamata 5:11–14.

Korol, E. N., and S. E. Spiridonov. 1991. *Angiostoma kimmeriensis* sp. n. and *Agfa tauricus* sp. n.—parasitic Rhabditida (Nematoda) from Grimean terrestrial molluscs. Helminthologica 28:179–182.

Kuzmin, S. L. 1995. The Clawed Salamanders of Asia. Genus *Onychodactylus*. Biology, Distribution, and Conservation. Westarp Wissenschaften, Magdeburg, Germany, 108 pp.

Mengert, H. 1953. Nematoden und Schnecken. Zeitschrift für Morphologie und Ökologie der Tiere 41:311–349.

Morand, S. 1986. *Angiostoma aspersae* n. sp. (Nematoda, Angiostomatidae) parasite de *Helix aspersa* Müller (Gastropoda, Helicidae). Bulletin du Museum National d'Histoire Naturelle, Paris 8:111–115.

———. 1992. *Angiostoma spiridonovi* sp. n. (Nematoda: Angiostomatidae) from *Limax flavus* (Gastropoda: Limacidae). Journal of the Helminthological Society of Washington 59:212–217.

———, **and G. M. Barker.** 1995. *Angiostoma schizoglossae* n. sp. (Nematoda: Angiostomatidae) from the New Zealand endemic slug *Schizoglossa novoseelandica* (Gastropoda: Rhytididae). Journal of Parasitology 81:94–98.

———, **and S. Spiridonov.** 1989. Redescription de trois espèces d'Angiostomatidae (Nematoda, Rhabditida), parasites de gastropodes pulmonés stylommatophores, et description du cycle évolutif de deux d'entre elles. Bulletin du Museum National d'Histoire Naturelle, Paris 11:367–385.

Nasir, P., and M. T. Díaz. 1971. A redescription of *Mesocoelium monas* (Rudolphi, 1819) Freitas, 1958, and specific determination in genus *Mesocoelium* Odhner, 1910 (Trematoda, Digenea). Rivista di Parassitologia 32:149–158.

Ozaki, Y. 1948. A new trematode, *Polystoma dendriticum* from the urinary bladder of *Onychodactylus japonicus* in Shikoku. Biosphaera 2:33–37.

Pearse, A. S. 1932. Parasites of Japanese salamanders. Ecology 13:135–152.

Spiridonov, S. E. 1985. *Angiostoma asamati* sp. n. (Angiostomatidae: Rhabditida)—new species of nematodes from slugs (Mollusca). Helminthologia 22:253–261.

Uchida, A., and H. Itagaki. 1979. Studies on the am-

phibian helminths in Japan. VI. *Pseudoxyascaris japonicus* n. g. and n. sp. (Oxyascarididae; Nematoda) and *Pseudopolystoma dendriticum* (Monogenea; Trematoda) from a salamander. Japanese Journal of Parasitology 28:43–50.

———, **K. Uchida, and A. Kamei.** 1986. Studies on the amphibian helminth in Japan. IX. A new digenetic trematode, *Cephalouterina leoi* n. sp., from salamanders, *Onychodactylus japonicus* and the new host record of the digenetic trematode, *Mesocoelium brevicaecum*. Bulletin of the Azabu University of Veterinary Medicine 7:97–101.

Van Cleave, H. J. 1925. Acanthocephala from Japan. Parasitology 17:149–156.

Wilkie, J. S. 1930. Some parasitic nematodes from Japanese Amphibia. Annals and Magazine of Natural History, Series 10, 6:606–614.

Yamaguti, S. 1963. Systema Helminthum. Volume IV. Monogenea and Aspidocotylea. Interscience Publishers, New York. 699 pp.

2000 Meeting Schedule of the Helminthological Society of Washington

January 19, 2000	Smithsonian Institution, National Museum of Natural History, Washington, DC, 7:30 pm (Contact person: Bill Moser, 202-357-2473).
March 22, 2000	Johns Hopkins Montgomery County Center (Provisional), Rockville, MD, 7:30 pm (Contact person: Tom Simpson (JHU), 410-366-8814, or Louis Miller (NIH), 301-496-2183).
May 6, 2000	Joint Meeting with the New Jersey Society for Parasitology, at the New Bolton Center, University of Pennsylvania, Kennett Square, PA, 2:00 pm (Contact person: Jay Farrell, 215-898-8561).
October, 2000	Date, time, and place to be announced.
November, 2000	Anniversary Dinner Meeting. Date, time, and place to be announced

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Carolinensis tuffi sp. n. (Nematoda: Trichostrongylina: Heligmosomoidea) from the White-Ankled Mouse, *Peromyscus pectoralis* Osgood (Rodentia: Cricetidae) from Texas, U.S.A.

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ABSTRACT: *Carolinensis tuffi* sp. n. (Nematoda: Trichostrongylina: Heligmosomoidea) from the small intestine of the white-ankled mouse, *Peromyscus pectoralis* Osgood (Rodentia: Cricetidae), from Texas, U.S.A., is described and illustrated. The new species is closest to *Carolinensis carolinensis* in its synlophe and to *Carolinensis dalrymplei* and *Carolinensis kinsellai* in the pattern of the caudal bursa. *Carolinensis romerolagi* (Gibbons and Kumar, 1980) is transferred to the genus *Paraheligmonella* and becomes *Paraheligmonella romerolagi* (Gibbons and Kumar, 1980) comb. n.

KEY WORDS: Nematoda, Trichostrongylina, Heligmosomoidea, *Carolinensis tuffi* sp. n., *Peromyscus pectoralis,* white-ankled mouse, Rodentia, Cricetidae, Texas, U.S.A.

The Nippostrongylinae, parasites of rodents of the families Arvicolidae and Cricetidae, may have arisen in the Palearctic Region (with the genus *Carolinensis* (Travassos, 1937)) and may have evolved from North America (with the genus *Hassalstrongylus* Durette-Desset, 1971) to South America (with the genus *Stilestrongylus* Freitas, Lent, and Almeida, 1937) (Durette-Desset, 1971, 1985). In the small intestine of a Nearctic cricetid, the white-ankled mouse *Peromyscus pectoralis* Osgood, 1904, we found a new species described below of particular interest, since it is a morphologic intermediary between the genera *Carolinensis* and *Hassalstrongylus.*

Materials and Methods

Hosts were live-trapped in Sherman traps by one of us (A.S.) in 1995 and 1996 under Texas Parks and Wildlife Permit SPR-0890–234, killed, and the whole carcasses were frozen for later examination. Nematodes were fixed and stored in 70% ethanol with 5% glycerine, studied in temporary wet mounts in water, and, when necessary, cleared in lactophenol. En face views and sections were mounted and studied in lactophenol. Measurements are given in micrometers unless otherwise stated; those relating to the holotype and allotype are in parentheses.

The nomenclature used for the family group is that of Durette-Desset and Chabaud (1993). The synlophe was studied following the method of Durette-Desset (1985), and the nomenclature used for the study of the caudal bursa is that of Durette-Desset and Chabaud (1981). Type specimens were deposited in the Helminthological Collections of the Muséum National d'Histoire Naturelle, Paris, France (MNHN). Voucher specimens from the type locality were also deposited in the United States National Parasite Collection, Beltsville, Maryland, accession No. 88849.

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Results

Carolinensis tuffi sp. n. (Figs. 1–8)

Description

Small nematodes, coiled to varying degrees along ventral side. Position of excretory pore in relation to length of esophagus very variable, mainly within second third of esophagus between 45% and 71% in male, 42% to 66% in female. Deirids, when visible, at same level but not visible in all specimens.

Head (based on 2 specimens): Cephalic vesicle present; buccal aperture triangular; 4 externolabial papillae, 2 amphids and 4 cephalic papillae; dorsoesophageal gland visible (Fig. 2).

Synlophe (studied in transverse sections of body in 1 male and 1 female): In both sexes, cuticle surface bears continuous ridges with chitinous reinforcement, beginning at different levels between cephalic vesicle and nerve ring and ending immediately anterior to caudal bursa in male, at vulvar level in female (Fig. 3). Number of ridges 20 in male and 19 in female at mid body. Axis of orientation of ridges passing though ventral right and dorsal left quadrant, inclined about 60° on sagittal axis in ventral left

quadrant and 70° in male, 75° in female in dorsal left quadrant. Ridges of equivalent size, except ventral right ones and ventral ridge adjacent to left ridge, all of which are smaller (Figs. 5, 6).

MALES (based on 5 specimens): Length 3.75–6.3 mm (6.4 mm) and width 90–100 (100) at mid body. Cephalic vesicle 50×40–60×40 (60×45). Nerve ring 120–170 (160), excretory pore 160–250 (230) from cephalic apex, respectively. Esophagus 350–380 (380) long (Fig. 1). Caudal bursa subsymmetric, very elongated laterally, of type 2–2–1 (Fig. 8). Rays 2 and 3 longer than ray 4; rays 4 and 5 divergent at their extremities; ray 8 arising perpendicularly at the root of the dorsal ray and not reaching edge of bursa; dorsal ray divided into 2 branches in distal third, each branch divided again into 2 unequal branches; externals (ray 9) being longer than the internal (ray 10), and almost reaching edge of caudal bursa. Spicules very thin, alate, 380–500 (460) long, ending in a sharp tip (Fig. 8). Gubernaculum absent. Genital cone triangular in ventral view, bearing a small papilla 0 on its ventral lip (Fig. 7). Papilla 7 not observed. Presence of membrane situated between genital cone and dorsal ray (Fig. 8).

FEMALES (based on 10 specimens): Length 7.3–8.8 mm (7.3 mm) and width 110–150 (150) at mid body. Cephalic vesicle 50×40–80×60 (60×50). Nerve ring 130–220 (160), excretory pore 175–320 (210) from cephalic apex, respectively. Esophagus 420–480 (450) long. Monodelphic, vulva at 110–160 (100) from caudal extremity. Vagina vera 30–50 (40) long. Vestibule 80–110 (70) long, with median constriction and posterior diverticulum, sphincter 40×30–50×40 (50×40). Infundibulum with proximal section curving in and out (twisting) 90–130 (120) (Fig. 4). Uterus 1.2–1.8 mm (1.7 mm) long, containing 30–36 (25) eggs. Eggs 65×40–80×60 (85×50) at morula stage. In 1 female, eggs in distal section of uterus embryonated. Tail conical, with round tip (Fig. 4).

Taxonomic Summary

TYPE HOST: White-ankled mouse, *Peromyscus pectoralis* Osgood, 1904.

TYPE LOCALITY: Colorado Bend State Park, San Saba County, Texas (31°05′N, 98°30′W), U.S.A.

SITE: Small intestine.

PREVALENCE AND INTENSITY: 57 of 189 hosts (30%) infected with 11.2 ± 3.3 SD nematodes; range, 1–175.

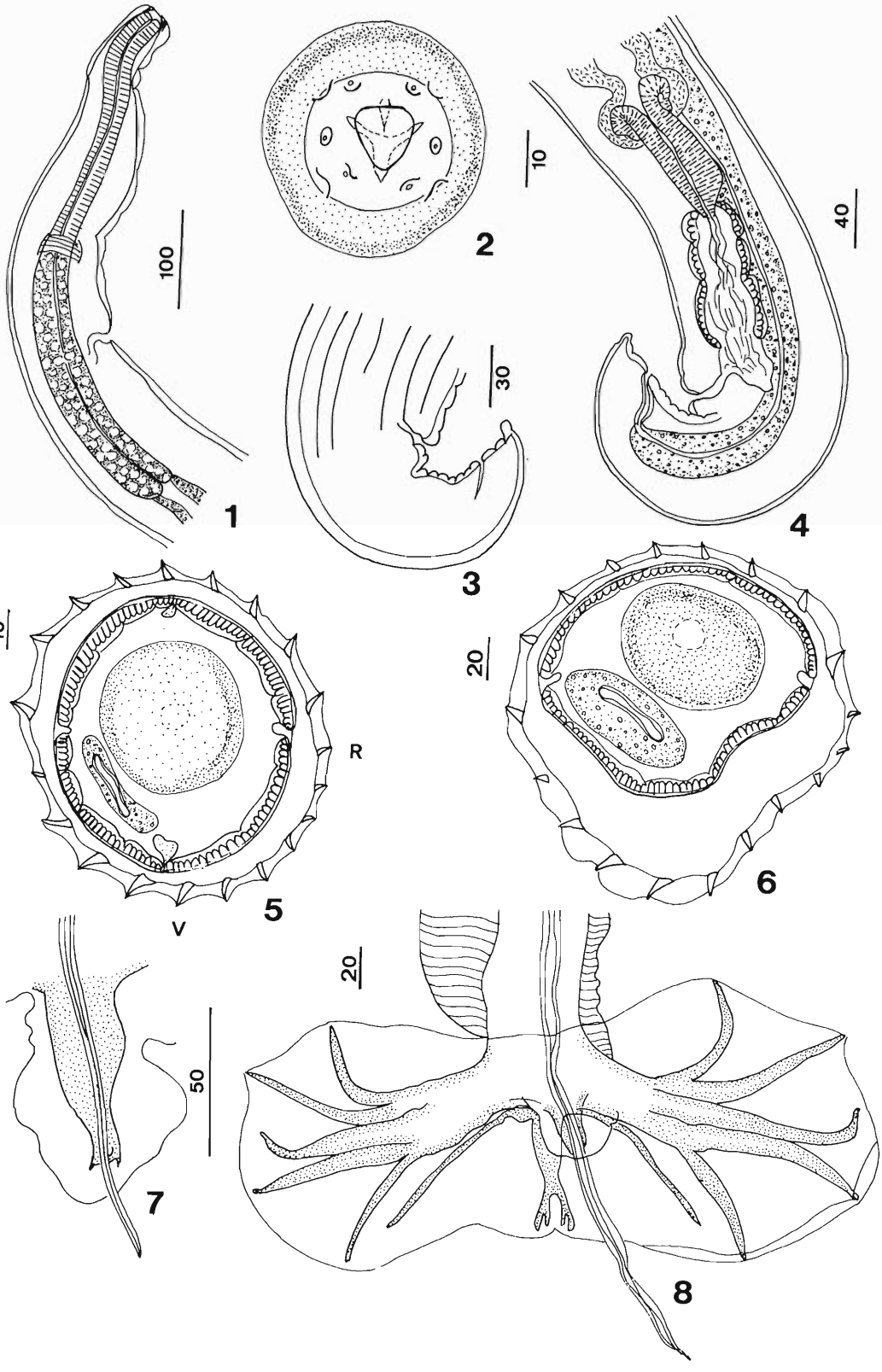
DEPOSITED SPECIMENS: Holotype male and allotype female MNHN 447 KXa; paratypes (4 males, 9 females) MNHN 447 KXb.

ETYMOLOGY: The species is named in honor of Dr. Donald W. Tuff of Southwest Texas State University.

Discussion

The specimens from *P. pectoralis* possess the main characters of the subgenus *Carolinensis* (Heligmonellidae: Nippostrongylinae), which was raised to the level of genus by Durette-Desset (1983): the caudal bursa is of type 2–2–1, the genital cone is poorly developed, and the left cuticular dilatation is absent. Species of this genus are mainly parasitic in Holarctic Arvicolidae and Cricetidae. Among the species described, the above specimens closely resemble *Carolinensis carolinensis* (Dikmans, 1935), a parasite of *Peromyscus maniculatus* (Wagner, 1845) and *Microtus ochrogaster* (Wagner, 1842) in the United States in the characters of the synlophe; the left lateral ridges are no more developed than the other ridges, the inclination of the axis of orientation is the same, and the number of cuticular ridges is relatively high. The new species differs by the pattern of the caudal bursa, the shape of the tips of the spicules, and 19–20 ridges versus 16 in *C. carolinensis* (Durette-Desset, 1974). It closely resembles *Carolinensis dalrymplei* (Dikmans, 1935), a parasite of *Ondatra zibethica* (Linnaeus, 1766) and *Microtus pennsylvanicus* (Ord, 1815) in the United States and *Carolinensis kinsellai* (Durette-Desset, 1969), a parasite of *Neofiber alleni* True, 1884, in the United States, in the pattern of the caudal bursa and, as in *C. kinsellai,* by the presence of a small ventral ridge adjacent to the left ridge. It differs from these species by the arising of ray 8 at the root of the dorsal ray, by the presence of a membrane between the genital cone and the dorsal ray, by the proximal twisting of the infundibulum, and by the high number of cuticular ridges (13 in *C. kinsellai,* not known in *C. dalrymplei*) (Durette-Desset, 1969).

According to Durette-Desset (1971), the genera *Carolinensis, Hassalstrongylus,* and *Stilestrongylus* belong to the same evolutionary line. The line may have arisen in the Palearctic Region and may have evolved from North America to South America with the following elements: (1)



increase in the number of cuticular ridges, (2) rotation of the axis of orientation, and (3) lengthening of the genital cone. This line was divided into 3 genera: *Carolinensis* in the Palearctic Region, *Hassalstrongylus* in the Nearctic Region, and *Stilestrongylus* in the Neotropical Region. But evolution being gradual, the generic separations are necessarily arbitrary, and the geographic localities overlap in the Americas. *Carolinensis* is also present in North America, and *Hassalstrongylus* is present in South America.

The phyletic position of the new species is interesting since it possesses some characteristics of the genus *Hassalstrongylus*: a high number of cuticular ridges (13–16 in *Carolinensis* vs 19–25 in *Hassalstrongylus*), disappearance of the gradient of size of the cuticular ridges, which tends to an equalization of their size and the appearance of a new symmetry in relation to the axis of orientation, and a relatively developed genital cone. According to Durette-Desset (1974), *Hassalstrongylus musculi* (Dikmans, 1935) was an example of an intermediary species between the genera *Hassalstrongylus* and *Stilestrongylus. Carolinensis tuffi* seems to be an intermediary between the genera *Carolinensis* and *Hassalstrongylus.*

The species *Boreostrongylus romerolagi* Gibbons and Kumar (1980) was described from a Mexican lagomorph, *Romerolagus diazi* (Ferrari Arez, 1893), and was automatically transferred to the genus *Carolinensis,* since *Boreostrongylus* was considered a synonym of *Carolinensis* by Durette-Desset (1983). However, this species is very different from the other species of the genus and can be classified in the genus *Paraheligmonella* Durette-Desset, 1971, particularly because of its synlophe: the left and right ridges are hypertrophied; a lateromedial gradient in the size of the ridges is present; and the axis of orientation is inclined 45° to the sagittal axis (Gibbons and Kumar, 1980). We thus propose a new combination: *Paraheligmonella romerolagi* (Gibbons and Kumar, 1980) comb. n. (=*Boreostrongylus romerolagi* Gibbons and Kumar, 1980; = *Carolinensis romerolagi* (Gibbons and Kumar, 1980), Durette-Desset, 1982).

Acknowledgments

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Literature Cited

Dikmans, G. 1935. New nematodes of the genus *Longistriata* in rodents. Journal of Parasitology 25: 72–81.

Durette-Desset, M. C. 1969. Étude du système des arêtes cuticulaires de trois Nématodes Héligmosomes: *Longistriata kinsellai* n.sp., *L. seurati* Travassos et Darriba, 1929, *L. hokkaidensis* Chabaud, Rausch, et Desset, 1963, parasites de Rongeurs. Annales de Parasitologie Humaine et Comparée 44:617–624.

———. 1971. Essai de classification des Nématodes Héligmosomes. Corrélations avec la paléobiogéographie des hôtes. Mémoires du Muséum National d'Histoire Naturelle, Nouvelle série, Série A, Zoologie 69:1–126

———. 1974. Nippostrongylinae (Nematoda: Heligmosomidae) néarctiques. Annales de Parasitologie Humaine et Comparée 49:435–450.

———. 1983. Keys to Genera of the Super-Family Trichostrongyloidea. CIH Keys to the Nematode Parasites of Vertebrates, No. 10, R.C. Anderson, and A.G. Chabaud, eds. Commonwealth Agricultural Bureau, Farnham Royal, Buckinghamshire, England, 1–86.

———. 1985. Trichostrongyloid nematodes and their vertebrate hosts. Reconstruction of the phylogeny of a parasitic group. Advances in Parasitology 24: 239–306.

———, **and A. G. Chabaud.** 1981. Nouvel essai de

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Figures 1–8. ***Carolinensis tuffi*** **sp. n. in** ***Peromyscus pectoralis*** **from Texas, drawings based on paratypes. 1. Male, anterior extremity, right lateral view. 2. Female, head, apical view. 3. Female tail, disappearance of cuticular ridges. 4. Female, posterior extremity, left lateral view. 5. Male synlophe at mid body. 6. Female synlophe at mid body. 7. Male, genital cone and membrane, ventral view. 8. Male, caudal bursa, ventral view. V = ventral side; R = right side. Scales in micrometers.**

classification des Nématodes Trichostrongyloidea. Annales de Parasitologie Humaine et Comparée 56:297–312.

———, **and** ———. 1993. Note sur la Nomenclature des Strongylida au-dessus du groupe famille. Annales de Parasitologie Humaine et Comparée 68: 111–112.

Gibbons, L., and V. Kumar. 1980. *Boreostrongylus romerolagi* n.sp. (Nematoda: Heligmonellidae) from a Mexican volcano rabbit, *Romerolagus diazi*. Systematic Parasitology 1:117–122.

Travassos, L. 1937. Revisão da família Trichostrongylidae Leiper, 1912. Monographias do Instituto Oswaldo Cruz 1:1–512.

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An Unusual Case of Anisakiasis in California, U.S.A.

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ABSTRACT. In the first case of its kind, anisakiasis is documented in a 44-yr-old California male whose neck was penetrated transesophageally by 1 third-stage larva of *Pseudoterranova decipiens*. The larva subsequently emerged from the neck region through an ulcerating sore. The larva showed some evidence of development and is described. The clinical history of the patient is reviewed. The patient subsequently died of causes unrelated to the anisakiasis infection.

KEY WORDS: anisakiasis, *Pseudoterranova decipiens*, third-stage larva, human infection, morphology, case history, California, U.S.A.

Of the many genera of ascaroid (Anisakidae) nematodes causing anisakiasis in vertebrates (Myers, 1975), only 2 species cause human infections in North America (McKerrow and Deardorff, 1988; U.S. Food and Drug Administration/Center for Food Safety and Applied Nutrition [FDA/CFSAN], 1992). The cod worm, *Pseudoterranova decipiens* (Krabbe, 1878) Gibson and Colin, 1981, infects marine mammals, most importantly seals, in the North Atlantic and North and South Pacific; the herring worm, *Anisakis simplex* (Rudolphi, 1809) Baylis, 1920, infects marine mammals, particularly whales in the eastern Pacific and elsewhere in the world (see Myers [1959, 1975] and Gibson [1983] for synonymies). Despite the high incidence of worms of the genus *Anisakis* in fishes (Myers, 1979) and whales in the western Pacific, human cases in North America involving larvae of *Anisakis* are rare. The higher incidence of human infection with larvae of the genus *Pseudoterranova* (Lichtenfels and Brancato, 1976; Kliks, 1983), particularly in the northern Atlantic coast, appears to be related to the large seal populations there (Myers, 1976).

Two patterns of disease describe the clinical symptomology of anisakiasis in North America. The asymptomatic luminal condition described for larvae of *Pseudoterranova* does not involve tissue penetration, and worms are expelled by coughing, vomiting, or defecating. In infections with larvae of *Anisakis*, however, penetration of the gut wall is reported by many observers (Kates et al., 1973; Lichtenfels and Brancato, 1976; Myers, 1976; Margolis, 1977; Ishikura et al., 1993). These cases are easily misdiagnosed as appendicitis, Crohn's disease, gastric ulcer, or gastrointestinal cancer (McKerrow and Deardorff, 1988; FDA/CFSAN, 1992; Alonso et al., 1997). Documentation of our present case, however, demonstrates that larvae of *Pseudoterranova* can be as invasive as has traditionally been described in infections with *Anisakis* in Holland and Japan (Oshima, 1972, 1987; Yoshimura et al., 1979; Ishikura et al., 1993).

In North America, the public health impact of anisakiasis is limited to consumers of such foods as sushi and sashimi. Approximately 50 cases were documented in the United States up to 1988, and fewer than 10 cases have been documented annually (FDA/CFSAN, 1992) since the first North American cases in the United States were documented in the early 1970's (Little and Most, 1973; Pinkus et al., 1975). The first confirmed human case of anisakiasis was reported in Holland in 1960 (Van Thiel et al., 1960), and Holland remains the most important anisakiasis-endemic region of the world. By 1990, 292 of the 559 European cases were reported in Holland, whereas 12,586 cases were reported in Japan. The Japanese cases included 11,629 gastric, 567 intestinal, and 45 extragastrointestinal cases of infection by *Anisakis* and only 335 cases of gastric infection by *Pseudoterranova* (Ishikura et al., 1993). This report documents worm morphology and the clinical

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picture and compares our results with related findings in other cases.

Materials and Methods

One 70% ethanol-preserved nematode extracted by J.B. and W.D. from the neck of a 44-yr-old Venice, California, caucasian male patient on 12 April 1998 was received by O.M.A. on 18 April 1998 for identification. The worm was first examined externally, then stained in Mayer's acid carmine overnight, destained in 4% HCl in 70% ethanol, dehydrated in ascending concentrations of ethanol, cleared in graded terpineol–100% ethanol, and prepared as a whole mount in Canada balsam. Figures were made with the aid of photoprojection. All measurements are in millimeters unless otherwise indicated. Width measurements refer to maximum width. The specimen is deposited in the U.S. National Parasite Collection (USNPC), Beltsville, Maryland.

Results

The patient (T.S.) was a 44-yr-old caucasian male with amyotrophic lateral sclerosis (ALS), a degenerative neuromuscular disorder. He was completely healthy until 1992, when he was diagnosed (spinal tap) with Lyme disease after suffering flu-like symptoms and joint pains. He received 2 courses of antibiotics, including ampicillin, bioxin, doxycycline, and vancomycin, before he felt cured. In 1994, he was further diagnosed with ALS at 2 major medical centers in South Carolina and New York and was told that he had 6 mo to live. Muscle biopsies showed cell death consistent with ALS. The patient sought help at The Natural Medicine Center in 1996 as his condition continued to deteriorate. All his laboratory tests were normal (including explorations of possible neurotoxins) except for a spinal tap that showed Lyme disease in the central nervous system. He was treated with heavy doses of antibiotics but without favorable results.

During April 1998, he received a series of colonic irrigations, during which time "parasites" were claimed to have been observed. These presumed "parasites" were not collected by the junior authors nor observed by O.M.A. However, a worm was actually extracted from a neck sore and sent to O.M.A. for identification. Upon external examination, the worm was initially identified as an anisakid nematode. After processing (see above), the identity of the nematode was determined to be a third-stage larva of *P. decipiens*. At that time, T.S. indicated on the Requisition Form that he was experiencing "severe weakness, neuromuscular damage, severe weight loss, loss of balance, and speech impairment." He also indicated no travel history but a history of diet often including sushi and sashimi over the previous few years. After the worm diagnosis, T.S. was treated with pharmaceutical anti-parasitic medications (albendazole and praziquantel) in large doses. Subsequently, his symptoms of malaise disappeared and his red blood cell count dramatically increased to 4.30 from a pretreatment low of 2.74 on 11 March 1998. After repeat treatment with praziquantel, T.S. felt better though his muscular condition remained unchanged. For the past few years, T.S. had always felt ill and experienced loss of function. He recently weighed 105 lb, down from a pre-illness weight of 160 lb. Before his death in April 1999, T.S. was unable to use his hands and could barely ambulate, with help. He could talk only with difficulty and breathed adequately but not enough to blow his nose.

Description of the third-stage larva of *Pseudoterranova decipiens* (Figs. 1–3)

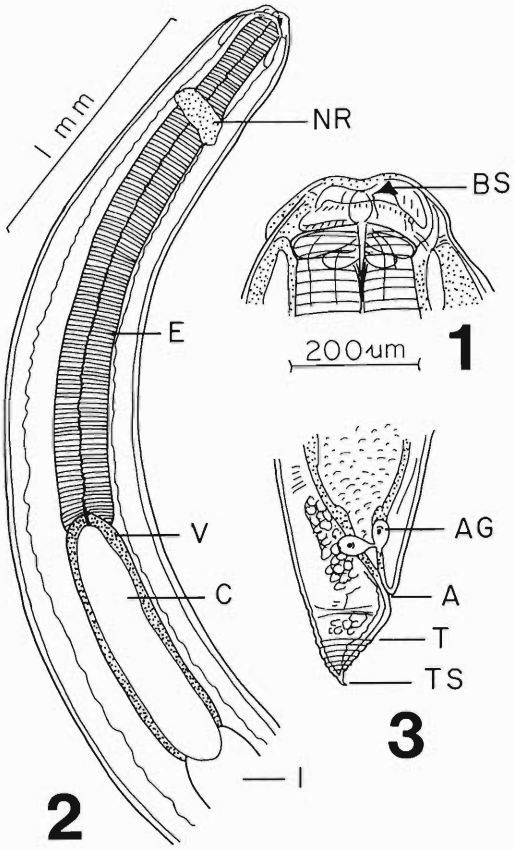
Body 42.12 long by 0.85 wide near middle. Cuticle wrinkled at regular intervals, about 0.025 thick but thinner toward both ends. One dorsal, 2 subventral large fleshy lips each with 2 rounded lobes, anterior dentigenous ridges, and large papilla (Fig. 1). Prominent boring tooth (spine) anteriorly. Excretory pore ventral at base of lips. Nerve ring prominent, 0.42 from anterior tip. Esophagus 2.03 long by 0.24 wide at base. Cecum extends anteriorly and about as long as ventriculus, 1.02 long by 0.18 wide (Fig. 2). Ventricular appendage and alae absent. Reproductive structures not observed. Tail (anus to posterior end) 0.16 long. Anal glands prominent, each with a single darkly stained nucleus. Conically shaped fine-pointed mucron (caudal spine) 0.025 long (Fig. 3). Evidence of development (molting) noted as the larva appeared trapped in the cuticle of the previous stage at various points.

Taxonomic summary

HOST: *Homo sapiens* Linnaeus, 1758.
LOCALITY: California, U.S.A.
SITE OF INFECTION: Neck.
SPECIMEN DEPOSITED: USNPC No. 88504.

Remarks

The specimen was identified as *P. decipens* primarily because it possessed a cecum and no



Figures 1–3. *Pseudoterranova decipiens* third-stage larva extracted from a neck lesion of a patient in California. 1. Anterior tip of body showing details of lips and boring spine (BS). 2. Anterior portion of body showing anteriorly projecting intestinal cecum (C) overlapping the ventriculus (V), esophagus (E), intestine (I), and nerve ring (NR). 3. Posterior end of worm showing anal glands (AG), anus (A), tail (T), and tail spine (TS). Scale bar (200 µm) applies to Figs. 1 and 3.

ventricular appendage and its excretory pore opened at the base of the lips. The cecum was about as long as the ventriculus. Additional significant features include prominent boring and caudal spines and the structure of lips as well as measurements of the described organs and trunk (above).

Discussion

The morphological features of the described larva were similar to those reported for other larvae of *P. decipiens* (reported as *Phocanema*) recovered from North American patients by Kates et al. (1973), Kliks (1983), Little and Most (1973), and Lichtenfels and Brancato (1976). There are minor differences in measurements, and none of the above authors reported anal glands; Kates et al. (1973) did not observe a boring tooth; Little and Most (1973) noted a female reproductive system but no caudal spine; Lichtenfels and Brancato (1976) noted cervical papillae and excretory gland about one-third the body length. Clearly, the morphological variations in human anisakids need to be documented to ascertain their identity and to determine the correct correlations with geographic distribution and histopathologic changes.

In contrast with the considerably greater prevalence of human infections with the highly invasive *A. simplex* in Europe and Japan compared with the noninvasive *P. decipiens*, most anisakiasis cases in North America involve *P. decipiens* (Kates et al., 1973; Jackson, 1975; Lichtenfels and Brancato, 1976; Myers, 1976; Margolis, 1977; Deardorff et al., 1986; Oshima, 1987; Ishikura et al., 1993; Alonso et al., 1997). The pattern of differential pathogenicity has been documented in Holland (Yoshimura et al., 1979), the United States (Jackson, 1975; Lichtenfels and Brancato, 1976; McKerrow and Deardorff, 1988), and Japan (Oshima, 1972, 1987; Ishikura et al., 1993) and was reviewed by Margolis (1977). Findings from our study, however, disagree with the above "pathogenic capacity" picture (Margolis, 1977) and document severe invasiveness of the larva of *P. decipiens*. Evidence of the invasiveness of the larvae of *Pseudoterranova* is extremely rare. When such cases occur (Little and MacPhail, 1972), as is usual in larvae of *Anisakis* (Hayasaka et al., 1971; Pinkus et al., 1975; Yoshimura et al., 1979; Deardorff et al., 1986; Oshima, 1987; McKerrow and Deardorff, 1988; Ishikura et al., 1993), the larvae usually invade the gut wall lower at gastric or intestinal sites. In a very rare case of invasive *Pseudoterranova*, the larva was recovered from the abdominal cavity of a male from Massachusetts (Little and MacPhail, 1972). All other cases of human infection with *Pseudoterranova* in North America were detected when larvae were eliminated by the mouth (Lichtenfels and Brancato, 1976; McKerrow and Deardorff, 1988). A larva was identified as "Anisakis" from histological sections in the tonsils of a

6-yr-old Indian girl from Oman, Arabian Peninsula (Bhargava et al., 1996), and extragastrointestinal "anisakidosis" has been reported in the mucous membrane of pharynx and esophagus in Japan (Ishikura et al., 1993); the identity of these worms was not elaborated further.

We attribute the upper gastrointestinal invasiveness of the larva of *P. decipiens* through the unusual esophageal site to the immune depression of the patient or weakness from ALS. The continued penetration of the worm through the neck tissue and the exiting through the neck sore represent an extreme case of invasiveness that, to the best of our knowledge, has not been previously reported in larvae of either *Anisakis* or *Pseudoterranova*.

We believe that the state of the patient could have been related to 1 or more of the following 3 factors: ALS, Lyme disease, or anisakid(s). Symptoms of anisakiasis may persist after worm death because some lesions have been found upon surgical removal that contain only nematode remnants. Stenosis of the pyloric sphincter was observed in a case where exploratory laparotomy had revealed a worm that was not removed (FDA/CSFAN, 1992). Simultaneous multiple infections with as many as 10 anisakid worms have been reported in Japan (Ishikura et al., 1993). Although acute necrotizing eosinophilic granulomatous inflammation involving the intestine has been documented in cases of invasive anisakiasis, hypersensitivity, sensitization, and a chronic form of the disease lasting about 2 yr have also been documented (Pinkus et al., 1975; Alonso et al., 1997).

Literature Cited

Alonso, A., A. Daschner, and A. Moreno-Ancillo. 1997. Anaphylaxis with *Anisakis simplex* in the gastric mucosa. New England Journal of Medicine 337:350–352.

Bhargava, D., R. Raman, M. Z. El Azzouni, K. Bhargava, and B. Bhusnurmath. 1996. Anisakiasis of the tonsils. Journal of Laryngology and Otology 110:387–388.

Deardorff, T. L., T. Fukumura, and R. B. Raybourne. 1986. Invasive anisakiasis. A case report from Hawaii. Gastroenterology 90:1047–1050.

Gibson, D. 1983. The systematics of ascaridoid nematodes: a current assessment. Pages 321–338 *in* A. F. Stone, H. M. Platt, and L. Khalil, eds. Nematode Systematics Association. Special Vol. 22. Academic Press, London.

Hayasaka, H., H. Ishikura, and T. Takayama. 1971. Acute regional illeitis due to *Anisakis* larvae. International Surgery 55:8–14.

Ishikura, H., K. Kikuchi, K. Nagasawa, T. Ooiwa, H. Takamiya, N. Sato, and K. Sugane. 1993. Anisakidae and anisakidosis. Progress in Clinical Parasitology 3:43–102.

Jackson, G. J. 1975. The "new disease" status of human anisakiasis and North American cases: a review. Journal of Milk and Food Technology 38: 769–773.

Kates, S., K. A. Wright, and R. Wright. 1973. A case of human infection with the cod nematode *Pseudoterranova* sp. American Journal of Tropical Medicine and Hygiene 22:606–608.

Kliks, M. M. 1983. Anisakiasis in the western United States. Four new case reports from California. American Journal of Tropical Medicine and Hygiene 32:526–532.

Lichtenfels, J. R., and F. P. Brancato. 1976. Anisakid larva from the throat of an Alaskan Eskimo. American Journal of Tropical Medicine and Hygiene 25:691–693.

Little, M. D., and J. C. MacPhail. 1972. Large nematode larva from the abdominal cavity of a man in Massachusetts. American Journal of Tropical Medicine and Hygiene 21:948–950.

———, **and H. Most.** 1973. Anisakid larva from the throat of a woman in New York. American Journal of Tropical Medicine and Hygiene 22:609–612.

Margolis, L. 1977. Public health aspects of "codworm" infections: a review. Journal of Fisheries Research Board of Canada 34:887–898.

McKerrow, J. H., and T. L. Deardorff. 1988. Anisakiasis: revenge of the sushi parasite (letter). New England Journal of Medicine 319:1228–1229.

Myers, B. J. 1959. *Phocanema,* a new genus for the anisakid nematode of seals. Canadian Journal of Zoology 37:459–465.

———. 1975. The nematodes that cause anisakiasis. Journal of Milk and Food Technology 38:774–782.

———. 1976. Research then and now on the Anisakidae nematodes. Transactions of the American Microscopical Society 95:137–142.

———. 1979. Anisakine nematodes in fresh commercial fish from waters along the Washington, Oregon and California coasts. Journal of Food Protection 42:380–384.

Oshima T. 1972. *Anisakis* and anisakiasis in Japan and adjacent area(s). Pages 301–393 *in* K. Morishita, Y. Komiya, and H. Matsubayashi, eds. Progress of Medical Parasitology in Japan. Vol. 4. Meguro Parasitological Museum, Tokyo.

———. 1987. Anisakiasis—is the sushi bar guilty. Parasitology Today 3:44–48.

Pinkus, G. S., C. Coolidge, and M. D. Little. 1975. Intestinal anisakiasis. First case report from North America. American Journal of Medicine 59:114–120.

U.S. Food and Drug Administration/Center for Food Safety and Applied Nutrition. 1992. Bad bug book. *Anisakis simplex* and related worms.

Pages 1–3 *in* Foodborne Pathogenic Microorganisms and Natural Toxins Handbook. World Wide Web, http://vm.cfsan.fda.gov/~mow/chap25.html

Van Theil, P. H., F. C. Kuipers, and R. T. Roskam. 1960. A nematode parasitic to herring causing abdominal syndromes in man. Tropical and Geographical Medicine 12:97–115.

Yoshimura, H., N. Akao, K. Kondo, and Y. Ohnishi. 1979. Clinicopathological studies on larval anisakiasis, with special reference to the report of extra-gastrointestinal anisakiasis. Japanese Journal of Parasitology 28:347–354.

Book Available

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Neotropical Monogenoidea. 36. Dactylogyrids from the Gills of *Rhamdia guatemalensis* (Siluriformes: Pimelodidae) from Cenotes of the Yucatan Peninsula, Mexico, with Proposal of *Ameloblastella* gen. n. and *Aphanoblastella* gen. n. (Dactylogyridae: Ancyrocephalinae)

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ABSTRACT: *Ameloblastella* gen. n. and *Aphanoblastella* gen. n. are proposed for dactylogyrids from the gills of pimelodid catfishes (Siluriformes) in the Neotropical Biogeographical Region. Species of *Ameloblastella* and *Aphanoblastella* are characterized on the bases of gonadal position, hook shank morphology, presence/absence of eyes and eye granules, and morphology of the male reproductive system. Two species of *Urocleidoides* (sensu lato) and 1 of *Vancleaveus* are transferred to *Ameloblastella* as *A. chavarriai* (Price, 1938) comb. n. (type species), *A. mamaevi* (Kritsky and Thatcher, 1976) comb. n., and *A. platensis* (Suriano and Incorvaia, 1995) comb. n., respectively. Three species of *Urocleidoides* (sensu lato) from pimelodid catfishes are transferred to *Aphanoblastella* as *A. travassosi* (Price, 1938) comb. n. (type species), *A. robustus* (Mizelle and Kritsky, 1969) comb. n., and *A. mastigatus* (Suriano, 1986) comb. n.

KEY WORDS: Monogenoidea, Dactylogyridae, *Ameloblastella, Aphanoblastella, Ameloblastella chavarriai* comb. n., *Ameloblastella mamaevi* comb. n., *Ameloblastella platensis* comb. n., *Aphanoblastella travassosi* comb. n., *Aphanoblastella mastigatus* comb. n., *Aphanoblastella robustus* comb. n., catfish, Siluriformes, Pimelodidae, *Rhamdia guatemalensis,* cenotes, Mexico.

Price (1938) described the dactylogyrids, *Cleidodiscus chavarriai* and *Cleidodiscus travassosi,* from the gills of the pimelodid catfish, *Rhamdia rogersi* (Regan, 1907) (a junior synonym of *Rhamdia laticauda* (Kner, 1857) according to Silfvergrip, 1996) in Costa Rica. Molnar et al. (1974) transferred the 2 species to *Urocleidoides* Mizelle and Price, 1964, based on the emended diagnosis of the genus provided by Mizelle et al. (1968). The diagnosis in Mizelle et al. (1968) greatly expanded the generic bounds of *Urocleidoides,* and within 5 years, species of the genus had been described from fishes of 5 orders: Atheriniformes, Characiformes, Gymnotiformes, Perciformes, and Siluriformes. Kritsky et al. (1986) restricted *Urocleidoides* to species having a sinistral vaginal sclerite, transferred several species from the genus to *Gussevia* Kohn and Paperna, 1964, and considered 22 described species incertae sedis, the latter group including *U. chavarriai* and *U. travassosi.* The purpose of this investigation was to determine the generic placement of several previously described species of *Urocleidoides* sensu lato described from pimelodid catfishes in the Neotropical Biogeographical Region. The study is based on new collections of *U. chavarriai* and *U. travassosi* on *Rhamdia guatemalensis* (Günther, 1864) (a junior synonym of *Rhamdia quelen* (Quoy and Gaimard, 1824) according to Silfvergrip, 1996) from cenotes (sinkholes) in the Yucatan Peninsula, Mexico.

Materials and Methods

Fish hosts, *R. guatemalensis,* were collected by hook and line or casting nets from cenotes on the Yucatan Peninsula, Mexico, during 1993–1998 (see Scholz et al., 1995). Gill baskets were removed from fish, placed in petri dishes with tap water, and examined with a dissection microscope. Methods of collection, preservation, mounting, and illustration of helminths were those described by Kritsky et al. (1986), except that

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the gill baskets of some hosts were fixed in hot (≈90°C) 4% formalin according to methods presented by Scholz and Hanzelová (1998); specimens fixed in hot formalin had extended or relaxed peduncles, while those fixed in ambient formalin (≈30°C) had contracted peduncles. Measurements, all in micrometers, were made with a filar micrometer according to procedures of Mizelle and Klucka (1953), except that length of the male copulatory organ is an approximation of total length obtained by using a calibrated Minerva curvimeter on camera lucida drawings; average measurements are followed by ranges and the number (*n*) of specimens measured in parentheses; unstained flattened specimens mounted in Hoyer's or Malmberg's media were used to obtain measurements of hooks, anchors, and the copulatory complex; all other measurements were obtained from unflattened specimens stained with Gomori's trichrome or Mayer's carmine and mounted in Canada balsam. Voucher specimens of helminths collected during this study were deposited in the United States National Parasite Collection (USNPC), Beltsville, Maryland, U.S.A., and the helminth collections of the University of Nebraska State Museum (HWML), Lincoln, Nebraska, U.S.A.; the Instituto de Biología, Universidad Nacional Autónoma de México (UNAM), Mexico City, Mexico; the Parasitology Laboratory, Center for Investigation and Advanced Studies of the National Polytechnic Institute (CINVESTAV-IPN) (CHCM), Merida, Mexico, and the Institute of Parasitology, Academy of Sciences of the Czech Republic (IPCAS), České Budějovice, Czech Republic, as indicated in the following redescriptions. For comparative purposes, the following specimens were examined: 2 voucher specimens of *U. chavarriai* (USNPC 73178) and 3 voucher specimens of *U. travassosi* (USNPC 73179), both lots deposited by Molnar et al. (1974); holotype and 9 paratypes of *Urocleidoides robustus* Mizelle and Kritsky, 1969 (USNPC 71009, 73565, HWML 22941); and a voucher specimen of *Philocorydoras* sp. (probably =*U. margolisi* Molnar, Hanek, and Fernando, 1974) (USNPC 88965).

Results

Class Monogenoidea Bychowsky, 1937
Order Dactylogyridea Bychowsky, 1937
Dactylogyridae Bychowsky, 1933
Ameloblastella gen. n.

DIAGNOSIS: Body fusiform, slightly flattened dorsoventrally, comprising cephalic region, trunk, peduncle, haptor. Tegument smooth. Two terminal, 2 bilateral cephalic lobes; 3 bilateral pairs of head organs; cephalic glands unicellular, lateral or posterolateral to pharynx. Eyes absent; accessory eye granules subspherical. Mouth subterminal, midventral, anterior to pharynx; pharynx muscular, glandular; esophagus present; 2 intestinal ceca confluent posterior to gonads, lacking diverticula. Genital pore midventral near level of intestinal bifurcation. Gonads intercecal, overlapping; testis dorsal to germarium. Vas deferens looping left intestinal cecum; seminal vesicle a dilation of vas deferens. Copulatory complex comprising basally articulated male copulatory organ, accessory piece. Male copulatory organ tubular, coiled; coil with counterclockwise rings (see Kritsky et al., 1985). Accessory piece with complex distal region serving as guide for male copulatory organ, articulation piece extending within rings to base of male copulatory organ. Seminal receptacle pregermarial; vaginal aperture sinistral; vitellaria coextensive with intestine. Haptor globose to subhexagonal, armed with dorsal, ventral anchor/bar complexes, 7 pairs of similar hooks; hook distribution ancyrocephaline (Mizelle, 1936; see Mizelle and Price, 1963). Ventral bar with posteromedial projection. Hook with shank comprising 2 subunits; proximal subunit expanded. Parasites of the gills of neotropical pimelodid catfishes (Siluriformes).

TYPE SPECIES: *Ameloblastella chavarriai* (Price, 1938) comb. n. (=*Cleidodiscus chavarriai* Price, 1938) from *R. rogersi* (Regan), *R. guatemalensis* (Günther), *Rhamdia sebae* (Valenciennes, 1840), and *R. quelen* (Quoy and Gaimard) in Costa Rica, Mexico, and Trinidad, respectively.

OTHER SPECIES: *Ameloblastella mamaevi* (Kritsky and Thatcher, 1976) comb. n. (=*Urocleidoides mamaevi* Kritsky and Thatcher, 1976) from *Cephalosilurus zungaro* (Humboldt, 1833) in Colombia, South America. *Ameloblastella platensis* (Suriano and Incorvaia, 1995) comb. n. (=*Vancleaveus platensis* Suriano and Incorvaia, 1995) from *Pimelodus clarias maculatus* (Lacépède, 1803) in Argentina, South America.

ETYMOLOGY: The generic name is from Greek (*amel/o* = neglected + *blast/o* = germ, branch) appended to the diminutive ending (*-ella*), and refers to the long period before recognition of generic placement of its members.

REMARKS: *Ameloblastella* gen. n. is primarily characterized by dactylogyrids with 1) overlapping gonads (testis dorsal to germarium); 2) subspherical accessory eye granules; 3) a basally articulated male copulatory organ and accessory piece; 4) a coiled male copulatory organ with counterclockwise rings; 5) a ventral bar with a medial process; 6) a seminal vesicle formed by a simple dilation of the vas deferens; 7) inflated hook shanks, each composed of 2 subunits

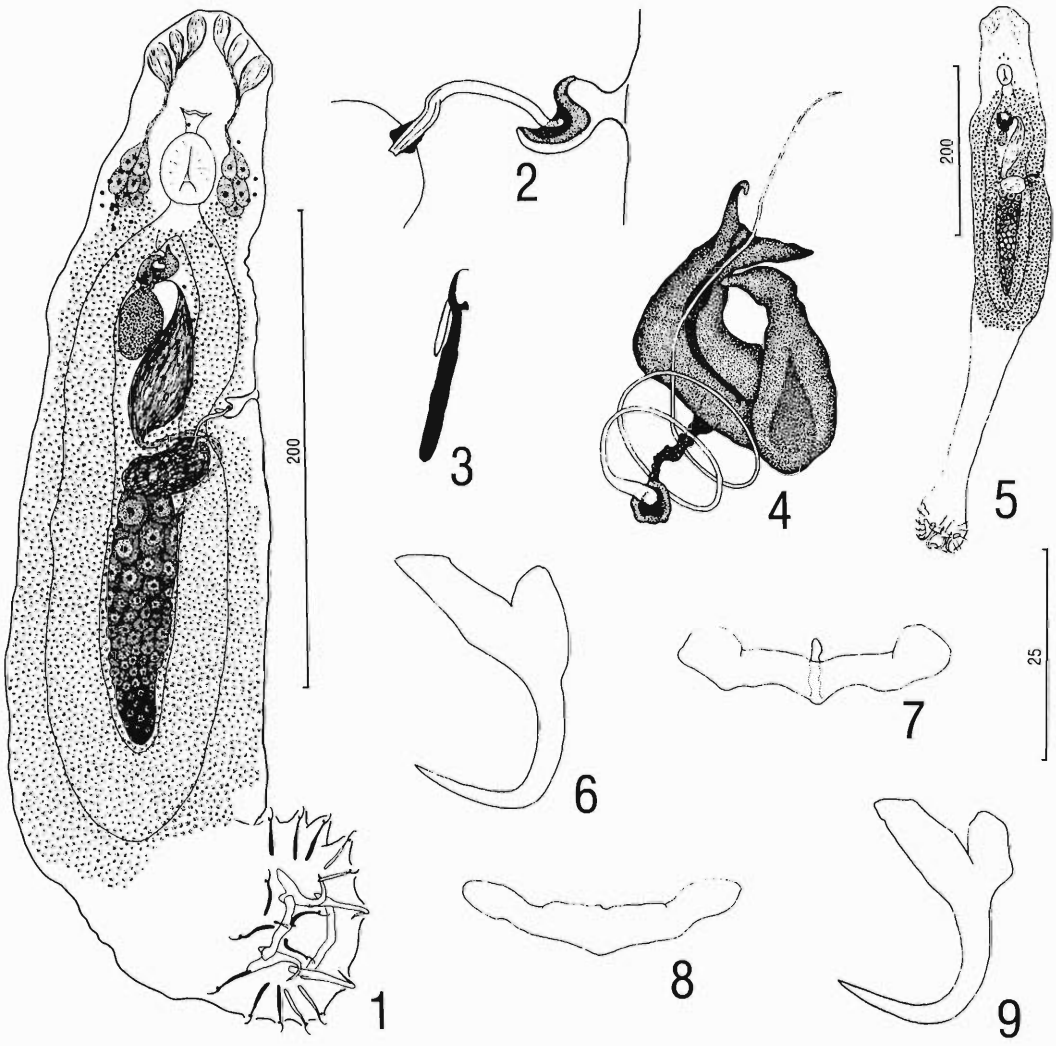
(proximal subunit expanded); 8) a sinistral vaginal aperture; and 9) absence of eyes. Of dactylogyrid genera with members infecting freshwater siluriforms in the Neotropics, characters defining *Ameloblastella* suggest a relationship with *Vancleaveus* Kritsky, Thatcher, and Boeger, 1986, and *Philocorydoras* Suriano, 1986. Members of these 3 genera share the characteristics of possessing overlapping gonads (testis dorsal to germarium), a ventral bar with a medial process, hook shanks comprised of 2 subunits (proximal subunit expanded), subspherical eye granules, and a dilation of the vas deferens to form the seminal vesicle. *Ameloblastella* differs from both *Vancleaveus* and *Philocorydoras* by the position of the vaginal aperture (ventral in *Vancleaveus* and *Philocorydoras*). It further differs from *Philocorydoras* by lacking eyes and having a coiled male copulatory organ (male copulatory organ an arced tube in *Philocorydoras*); and from *Vancleaveus* by the absence of a basal fold on the superficial root of the dorsal anchor and an expanded distal subunit of the hook shank (both present in *Vancleaveus*) (see Kritsky et al., 1986; Suriano, 1986b).

Of the 22 species of *Urocleidoides* considered incertae sedis by Kritsky et al. (1986), 2 of them are transferred to *Ameloblastella* as *A. chavarriai* (Price, 1938) comb. n. and *A. mamaevi* (Kritsky and Thatcher, 1976) comb. n. While *A. chavarriai* is the type species of *Ameloblastella* and defines the genus, *A. mamaevi* possesses all diagnostic features of *Ameloblastella* (see Kritsky and Thatcher, 1976).

Suriano and Incorvaia (1995) described *Vancleaveus platensis* from the gills of the pimelodid, *P. c. maculatus*. This helminth is not a member of *Vancleaveus* because of the absence of basal folds on the superficial root of the dorsal anchor and the presence of a sinistral vaginal aperture (vaginal pore ventral in *Vancleaveus* spp.). The original description of the species indicates that it possesses all of the diagnostic features of *Ameloblastella,* except for the presence of a nonarticulated male copulatory organ and accessory piece. However, specimens of this species in the senior author's collection and collected from *Pimelodus clarias* (Lacépède) from the Rio de la Plata, Argentina, show a delicate articulation process attaching the base of the male copulatory organ to the accessory piece. The latter finding supports the transfer of *V. platensis* to *Ameloblastella*.

Ameloblastella chavarriai (Price, 1938) comb. n. (Figs. 1–9)

REDESCRIPTION: Body 596 (408–742; n = 30) long; greatest width 113 (92–134; n = 33) usually in posterior trunk. Cephalic lobes poorly to moderately developed. Accessory eye granules in cephalic, anterior trunk regions. Pharynx ovate, 27 (24–30; n = 33) in greatest width; esophagus short to moderately long. Peduncle contracted, broad (ambient temperature formalin fixation) or elongate, narrow (hot formalin fixation); haptor subhexagonal, 84 (69–108; n = 28) wide, 72 (60–88; n = 29) long. Ventral anchor 33 (30–36; n = 22) long, with elongate superficial root, short deep root, slightly curved shaft, straight point; base 19 (16–21; n = 16) wide. Dorsal anchor 27 (23–31; n = 16) long, with well-developed roots, slightly curved shaft, elongate straight point; deep root protruding posteriorly from anchor base; anchor base 18 (17–20; n = 10) wide. Ventral bar 33 (29–37; n = 22) long, yoke-shaped, with posteromedial process usually bent anteriorly dorsal to bar. Dorsal bar 30 (26–34; n = 20) long, broadly V-shaped, with slightly enlarged ends. Hook with protruding truncate thumb, delicate point; hook 24 (20–27; n = 36) long; filamentous hooklet (FH) loop extending to level of union of shank subunits. Male copulatory organ 141 (128–158; n = 8) long, a coil of about 2.5 rings; diameter of proximal ring 17 (15–20; n = 20); base of male copulatory organ with small sclerotized plate. Accessory piece 32 (27–38; n = 26) long, 25 (22–32; n = 22) wide, comprising 2 subunits; dextral subunit terminally acute, subtriangular, with expanded lateral margins; sinistral subunit L-shaped with flared termination, serving as guide for male copulatory organ. Testis elongate, fusiform (lateral margins indistinct); seminal vesicle large, fusiform, lying diagonally in median field of anterior trunk posterior to male copulatory organ; prostatic reservoir lying to right of seminal vesicle and body midline, posterior to male copulatory organ. Germarium an inverted elongate cone, 119 (84–166; n = 20) long, 35 (27–53; n = 21) wide; oviduct, ootype not observed; uterus delicate, infrequently with single egg; vagina a sclerotized tube; vaginal aperture on sclerotized papilla lying in small indentation of body margin; seminal receptacle



Figures 1–9. ***Ameloblastella chavarriai*** **(Price, 1938) comb. n. 1. Whole mount (composite, ventral). 2. Vagina. 3. Hook. 4. Copulatory complex (ventral). 5. Whole mount (fixed in hot formalin). 6. Ventral anchor. 7. Ventral bar. 8. Dorsal bar. 9. Dorsal anchor. All figures are to the 25-μm scale except Figures 1 and 5 (200-μm scales).**

large, subovate. Egg 63 (n = 1) long, 30 (n = 1) wide, ovate, with short proximal filament.

SYNONYMS: *Cleidodiscus chavarriai* Price, 1938; *Urocleidoides chavarriai* (Price, 1938) Molnar, Hanek, and Fernando, 1974.

HOST AND LOCALITY: Gills of *Rhamdia guatemalensis* (Günther); Ixin-há Cenote, Yucatan, Mexico (20°37′14″N; 89°06′40″W) (11 July 1994; 11 May 1997; 26 October 1998).

PREVIOUS RECORDS: *Rhamdia rogersi* (Regan) (type host), San Pedro Montes de Oca, Costa Rica (Price, 1938); *R. quelen* (Quoy and Gaimard) and *R. sebae* (Valenciennes), Cumuto River near Coryal, Trinidad (Molnar et al., 1974).

SPECIMENS STUDIED: 57 vouchers, USNPC 88963, HWML 15015, UNAM 3710, IPCAS M-354, CHCM 313; 2 vouchers deposited by Molnar et al. (1974), USNPC 73178.

REMARKS: *Ameloblastella chavarriai* (Price, 1938) comb. n. is the type species of the genus. Although the morphometrics of present specimens differ from those reported in the original description by Price (1938), our specimens pos-

sess the diagnostic morphological features of the species. Measurements of the body (247 μm long, 80 μm wide), haptor (45 μm long, 70 μm wide), and pharynx (20 μm wide) reported by Price (1938) are noticeably smaller than those presented herein, suggesting that the type specimens were strongly contracted as a result of fixation. We do not consider these differences sufficient to warrant description of a new species for the helminths in our collection, since the method of fixation greatly influences the morphometrics of soft body parts. Fixation of our material in hot formalin resulted in extended specimens (Fig. 5), while ambient temperature formalin fixation (the method most likely used by Price, 1938) resulted in contracted specimens with a body length of about half that of those fixed in hot formalin. Haptoral sclerites and the copulatory complex in our specimens correspond morphometrically to respective values provided by Price (1938). Measurements by Molnar et al. (1974) generally fall within the ranges of the combined measurements of Price (1938) and those presented herein, respectively.

Although numerous collections of *R. guatemalensis* were made from cenotes throughout the Yucatan Peninsula (see Scholz et al., 1995), *A. chavarriai* was found only in Ixin-há Cenote. This suggests an apparent limited distribution of the species in the Yucatan Peninsula. However, individual collections from Ixin-há Cenote conducted at different periods of 1997–1998 showed intensity levels of *A. chavarriai* relative to the other dactylogyrid species (*Aphanoblastella travossosi*) on this host to vary from being predominant (≥95%) to nearly insignificant (≤5%). The possibility, therefore, exists that the observed limited distribution of the parasite in the Yucatan Peninsula could be a result of sampling error.

Aphanoblastella gen. n.

DIAGNOSIS: Body fusiform, comprising cephalic region, trunk, peduncle, haptor. Tegument smooth. Two terminal, 2 bilateral cephalic lobes; 3 bilateral pairs of head organs; cephalic glands unicellular, lateral or posterolateral to pharynx. Four eyes (2 pairs); granules subspherical. Mouth subterminal, midventral; pharynx muscular, glandular; esophagus present or absent; intestinal ceca 2, confluent posterior to gonads, lacking diverticula. Genital pore midventral near level of intestinal bifurcation. Gonads intercecal, tandem; testis postgermarial. Vas deferens looping left intestinal cecum; seminal vesicle a dilation of vas deferens. Copulatory complex comprising non-articulated male copulatory organ, accessory piece. Male copulatory organ tubular, coiled; coil with counterclockwise rings. Accessory piece simple, distally serving as guide for male copulatory organ. Seminal receptacle pregermarial; vaginal aperture sinistral, nonsclerotized; vitellaria coextensive with intestine. Haptor globose, armed with dorsal, ventral anchor/bar complexes, 7 pairs of similar hooks; hook distribution ancyrocephaline. Ventral bar with posteromedial projection. Hook with nondilated shank comprising 1 subunit. Parasites of the gills of neotropical pimelodid fishes (Siluriformes).

TYPE SPECIES: *Aphanoblastella travassosi* (Price, 1938) comb. n. (=*Cleidodiscus travassosi* Price, 1938) from *R. rogersi* (Regan), *R. guatemalensis* (Günther), *R. sebae* (Valenciennes), and *R. quelen* (Quoy and Gaimard) in Costa Rica, Mexico, and Trinidad, respectively; from *Pimelodella laticeps* Eigenmann in Argentina.

OTHER SPECIES: *Aphanoblastella mastigatus* (Suriano, 1986) comb. n. (=*Urocleidoides mastigatus* Suriano, 1986) from *Rhamdia sapo* (Valenciennes, 1840) in Argentina. *Aphanoblastella robustus* (Mizelle and Kritsky, 1969) comb. n. (=*Urocleidoides robustus* Mizelle and Kritsky, 1969) from *Rhamdia* sp. in the Amazon River system of Brazil (original host specimen collected from an aquarium in the United States).

ETYMOLOGY: The generic name is from Greek (*aphan/o* = invisible, secret + *blast/o* = germ, branch) appended to the diminutive ending (*-ella*) and refers to prior difficulty in assigning its member species to an appropriate generic taxon.

REMARKS: *Aphanoblastella* gen. n. is primarily characterized by dactylogyrids with 1) tandem gonads (testis posterior to germarium); 2) 2 pairs of eyes comprising subspherical granules; 3) a nonarticulated male copulatory organ and accessory piece; 4) a coiled male copulatory organ with counterclockwise rings; 5) a ventral bar with medial process; 6) a seminal vesicle formed by a simple dilation of the vas deferens; 7) uniform, nondilated hook shanks each comprising 1 subunit; 8) a sinistral vaginal pore; and 9) a nonsclerotized vaginal tube. *Aphanoblastella* resembles *Cosmetocleithrum* Kritsky, Thatch-

er, and Boeger, 1986, and *Amphocleithrium* Price and Gonzalez-Romero, 1969, by having tandem gonads, nondilated hook shanks each with 1 subunit, counterclockwise rings in the male copulatory organ, a seminal vesicle formed by a simple dilation of the vas deferens, a sinistral vaginal pore, and a nonsclerotized vagina. It differs from *Cosmetocleithrum* by lacking 2 submedial projections on the dorsal bar and by having well-developed eyes (Kritsky et al., 1986). *Aphanoblastella* differs from *Amphocleithrium* by possessing 2 pairs of eyes and a nonarticulated male copulatory organ and accessory piece (Suriano and Incorvaia, 1995).

The internal anatomy of species of *Aphanoblastella* is also identical to that of *Demidospermus* Suriano, 1983, as emended by Kritsky and Gutiérrez (1998). *Aphanoblastella* differs from *Demidospermus* spp. by possessing short ventral bars with a medial process (ventral bars elongate, V- or W-shaped, lacking a medial process in *Demidospermus*). Thumbs of hook pairs 5 and 6 are modified in species of *Demidospermus* (see Kritsky and Gutiérrez, 1998), whereas all hooks are similar and unmodified in *Aphanoblastella* spp.

Three previously described species of *Urocleidoides* (sensu lato) from *Rhamdia* spp., *U. travassosi* (Price, 1938), *U. robustus* Mizelle and Kritsky, 1969, and *U. mastigatus* Suriano, 1986, are transferred to *Aphanoblastella* as *A. travassosi* (Price, 1938) comb. n., *A. robustus* (Mizelle and Kritsky, 1969) comb. n., and *A. mastigatus* (Suriano, 1986) comb. n., respectively. *Aphanoblastella travassosi* comb. n. is the type species and therefore defines the new genus. *Aphanoblastella mastigatus* comb. n. also is clearly a member of the genus based on the original description (compare figures and description in Suriano, 1986a). *Aphanoblastella mastigatus* appears to be the sister species of *A. travassosi.*

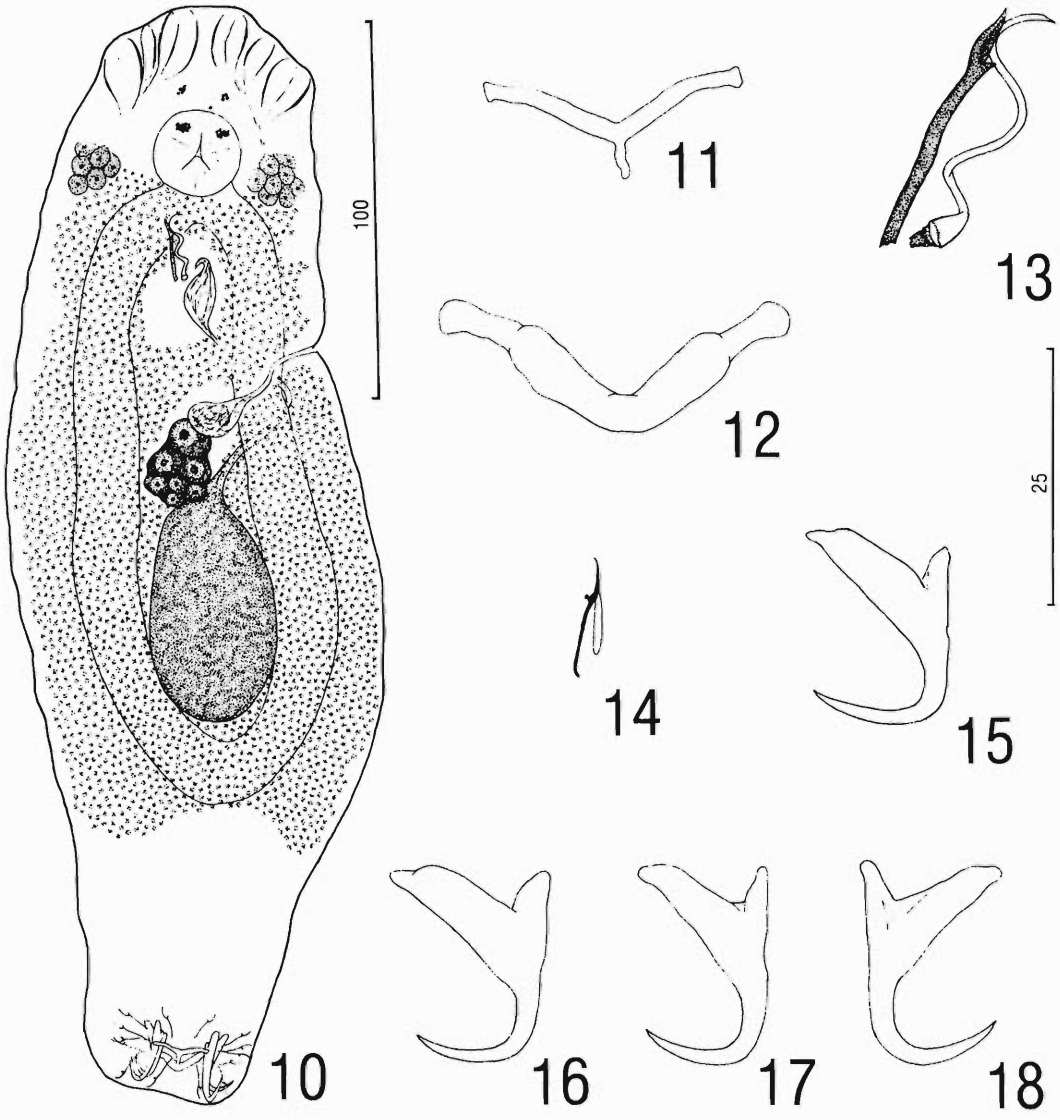
The original description of *U. robustus* by Mizelle and Kritsky (1969) was based on unstained and cleared specimens mounted in Gray and Wess' medium. Mizelle and Kritsky (1969) described the gonads to be tandem or overlapping, suggesting that the germarium is anterior to the testis with which it may overlap; the type specimens of *U. robustus* available to us were not useful in confirming gonadal position. Thus, our transfer of this species to *Aphanoblastella* is based on the original statements by Mizelle and Kritsky (1969) concerning gonadal position and on the similar position and morphology of sclerotized haptoral and copulatory structures to those of *A. travassosi.*

Aphanoblastella travassosi (Price, 1938) comb. n. (Figs. 10–18)

REDESCRIPTION: Body 282 (204–364; n = 34) long; greatest width 104 (77–127; n = 32) in posterior trunk. Cephalic lobes poorly to moderately developed. Eyes equidistant, posterior pair larger; accessory granules usually uncommon in cephalic region. Pharynx subspherical, 28 (21–33; n = 23) in diameter; esophagus short. Peduncle broad; haptor 55 (45–63; n = 31) wide, 40 (33–50; n = 32) long. Ventral anchor 22 (21–24; n = 13) long, with elongate superficial root, short deep root, straight shaft, curved elongate point; base 16 (14–17; n = 11) wide, variable. Dorsal anchor 24 (21–27; n = 11) long, with well-developed roots, straight shaft, elongate curved point; base 16 (14–18; n = 12) wide. Ventral bar 32 (29–37; n = 10) long, delicate, broadly V-shaped, with posteromedial process directed posteriorly; dorsal bar 37 (31–44; n = 9) long, broadly V-shaped, with narrowed bulbous ends. Hook 13 (12–14; n = 23) long, with protruding thumb, delicate point, fine shank; FH loop about two-thirds shank length. Male copulatory organ 41 (38–45; n = 5) long, a coil of about 2 rings, base of male copulatory organ with small sclerotized plate; diameter of proximal ring 5 (4–6; n = 6). Accessory piece 31 (28–36; n = 4) long, rod-shaped, with broad terminal acute tip. Testis ovate, 51 (40–59; n = 19) long, 35 (25–46; n = 18) wide; seminal vesicle indistinct, fusiform, lying to left of male copulatory organ; prostatic reservoir not observed. Germarium 28 (20–44; n = 23) long, 22 (18–25; n = 23) wide, pyriform, comprising comparatively few cells; oviduct, ootype not observed; uterus delicate; vagina a diagonal tube extending to left body margin, vaginal aperture simple; seminal receptacle small.

SYNONYMS: *Cleidodiscus travassosi* Price, 1938; *Urocleidoides travassosi* (Price, 1938) Molnar, Hanek, and Fernando, 1974.

HOST AND LOCALITY: Gills of *Rhamdia guatemalensis* (Günther); Ixin-há Cenote, Yucatan, Mexico (20°37′14″N; 89°06′40″W) (13 June



Figures 10–18. ***Aphanoblastella travassosi*** **(Price, 1938) comb. n. 10. Whole mount (composite, ventral). 11. Ventral bar. 12. Dorsal bar. 13. Copulatory complex (ventral). 14. Hook. 15. Dorsal anchor. 16–18. Ventral anchors. All drawings are to the 25-μm scale except Figure 10 (100-μm scale).**

1994; 11 July 1994; 11 May 1997; 26 October 1998).

OTHER RECORDS (specimens not used in this study): *R. guatemalensis*: Hubiku Cenote (20°49′79″N; 88°01′21″W) (18 April 1994); cenote in village of Hunucmá (21°00′03″N; 89°52′06″W) (8 November 1993); Scan-Yui Cenote (20°40′20″N; 88°32′51″W) (25 January 1994); Tixkanka Cenote (21°14′55″N; 88°58′45″W) (23 May 1994); Xcanganchén Cenote (20°36′43″N; 89°05′32″W) (16 November 1993); Homún Cenote (20°44′19″N; 89°17′49″W) (3 November 1993); Xmucuy Cenote (20°33′63″N; 88°59′50″W) (16 November 1993) (Mendoza-Franco et al., 1999).

PREVIOUS RECORDS: *Rhamdia rogersi* (Regan), type host, San Pedro Montes de Oca, Costa Rica (Price, 1938); *R. quelen* (Quoy and Gaimard) and *R. sebae* (Valenciennes), Cumuto River near Coryal, Trinidad (Molnar et al.,

1974); *P. laticeps* Eigenmann, Laguna de Chascomús, Buenos Aires, Argentina (Suriano, 1986a).

SPECIMENS STUDIED: 49 vouchers, USNPC 88964, HWML 15016, UNAM 3711, IPCAS M-353, CHCM 314; 3 vouchers deposited by Molnar et al. (1974), USNPC 73179.

REMARKS: *Aphanoblastella travassosi* is widely distributed in cenotes of the Yucatan Peninsula. All available specimens were slightly to strongly contracted as a result of fixation in ambient 4% formalin; however, our measurements correspond fairly closely to respective values reported by Price (1938), Molnar et al. (1974), and Suriano (1986a). Price (1938) did not describe or draw the accessory piece of the copulatory complex of this species but indicated that one was present; the drawing of the copulatory complex by Molnar et al. (1974) corresponds to our Figure 13, while that provided by Suriano (1986a) is apparently distorted.

Discussion

Of the 22 species of *Urocleidoides* from the Neotropical Region that were considered incertae sedis by Kritsky et al. (1986), 17 remain to be reassigned at the generic level: *Urocleidoides affinis* Mizelle, Kritsky, and Crane, 1968, from Characidae (Characiformes); *Urocleidoides amazonensis* Mizelle and Kritsky, 1969, from Pimelodidae (Siluriformes); *Urocleidoides carapus* Mizelle, Kritsky, and Crane, 1968, from Gymnotidae (Gymnotiformes); *Urocleidoides catus* Mizelle and Kritsky, 1969, from Pimelodidae (Siluriformes); *Urocleidoides corydori* Molnar, Hanek, and Fernando, 1974, from Callichthyidae (Siluriformes); *Urocleidoides costaricensis* (Price and Bussing, 1967) Kritsky and Leiby, 1972, from Characidae (Characiformes); *Urocleidoides gymnotus* Mizelle, Kritsky, and Crane, 1968, from Gymnotidae (Gymnotiformes); *Urocleidoides heteroancistrium* (Price and Bussing, 1968) Kritsky and Leiby, 1972, from Characidae (Characiformes); *Urocleidoides kabatai* Molnar, Hanek, and Fernando, 1974, from Characidae (Characiformes); *Urocleidoides lebedevi* Kritsky and Thatcher, 1976, from Pimelodidae (Siluriformes); *Urocleidoides margolisi* Molnar, Hanek, and Fernando, 1974, from Callichthyidae (Siluriformes); *Urocleidoides megorchis* Mizelle and Kritsky, 1969, from Pimelodidae (Siluriformes); *Urocleidoides microstomus* Mizelle, Kritsky, and Crane, 1968, from Characidae (Characiformes); *Urocleidoides sictus* Mizelle, Kritsky, and Crane, 1968, from Characidae (Characiformes); *Urocleidoides strombicirrus* (Price and Bussing, 1967) Kritsky and Thatcher, 1974, from Characidae (Characiformes); *Urocleidoides trinidadensis* Molnar, Hanek, and Fernando, 1974, from Characidae (Characiformes); and *Urocleidoides virescens* Mizelle, Kritsky, and Crane, 1968, from Gymnotidae (Gymnotiformes). Previously, Kritsky et al. (1989) transferred *Urocleidoides variabilis* Mizelle and Kritsky, 1969, a parasite of neotropical Cichlidae (Perciformes), to *Sciadicleithrum* Kritsky, Thatcher, and Boeger, 1989. In the present study, 5 species of *Urocleidoides* (sensu lato) from pimelodids (1 described subsequent to the emended diagnosis of *Urocleidoides* by Kritsky et al., 1986) are reassigned to *Ameloblastella* gen. n. or *Aphanoblastella* gen. n.

Six of the *Urocleidoides* spp. remaining as incertae sedis occur on siluriform catfishes in the Neotropical Biogeographical Region. Based on the comparative morphology of the copulatory complexes of *U. corydori, U. margolisi,* and *Philocorydoras platensis* Suriano, 1986, the former 2 species should probably be transferred to *Philocorydoras* (see Suriano, 1986b; Molnar et al., 1974). We have not formally made this transfer because details of the internal anatomy, particularly those of the reproductive systems, are lacking.

A new genus for *U. amazonensis, U. catus, U. megorchis,* and perhaps *U. lebedevi* is probably justified based in part on presence of modified hook pairs 5 and 6 (slender hook shank, degenerate thumb, and straight point). While these species lack other generic characters of *Demidospermus* (as emended by Kritsky and Gutiérrez, 1998) and therefore cannot be accommodated in it, species of *Demidospermus* also have modified hook pairs 5 and 6, suggesting a phylogenetic relationship between these species and *Demidospermus* spp. Mizelle and Kritsky's (1969) use of "gut normal" in their descriptions of *U. amazonensis, U. catus,* and *U. megorchis* is presumed to mean that the gut consists of a bifurcated esophagus and 2 ceca confluent posterior to the gonads. However, *U. lebedevi* was reported to have 2 blind ceca posterior to the gonads (Kritsky and Thatcher, 1976). Features of the digestive system in all of these species must be verified before formal proposals for generic placement can be made.

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Literature Cited

Kritsky, D. C., W. A. Boeger, and V. E. Thatcher. 1985. Neotropical Monogenea. 7. Parasites of the pirarucu, *Arapaima gigas* (Cuvier), with descriptions of two new species and redescription of *Dawestrema cycloancistrium* Price and Nowlin, 1967 (Dactylogyridae: Ancyrocephalinae). Proceedings of the Biological Society of Washington 98:321–331.

———, **and P. A. Gutiérrez.** 1998. Neotropical Monogenoidea. 34. Species of *Demidospermus* (Dactylogyridae, Ancyrocephalinae) from the gills of pimelodids (Teleostei, Siluriformes) in Argentina. Journal of the Helminthological Society of Washington 65:147–159.

———, **and V.E. Thatcher.** 1976. New monogenetic trematodes from freshwater fishes of western Colombia with the proposal of *Anacanthoroides* gen. n. (Dactylogyridae). Proceedings of the Helminthological Society of Washington 43:129–134.

———, ———, **and W. A. Boeger.** 1986. Neotropical Monogenea. 8. Revision of *Urocleidoides* (Dactylogyridae, Ancyrocephalinae). Proceedings of the Helminthological Society of Washington 53:1–37.

———, ———, **and** ———. 1989. Neotropical Monogenea. 15. Dactylogyrids from the gills of Brazilian Cichlidae with proposal of *Sciadicleithrum* gen. n. (Dactylogyridae). Proceedings of the Helminthological Society of Washington 56:128–140.

Mendoza-Franco, E. F., T. Scholz, C. Vivas-Rodríguez, and J. Vargas-Vázquez. 1999. Monogeneans of freshwater fishes from cenotes (=sinkholes) of the Yucatan Peninsula, Mexico. Folia Parasitologica 46:267–273.

Mizelle, J. D. 1936. New species of trematodes from the gills of Illinois fishes. American Midland Naturalist 17:785–806.

———, **and A. R. Klucka.** 1953. Studies on monogenetic trematodes. XIV. Dactylogyridae from Wisconsin fishes. American Midland Naturalist 49:720–733.

———, **and D. C. Kritsky.** 1969. Studies on monogenetic trematodes. XXXIX. Exotic species of Monopisthocotylea with the proposal of *Archidiplectanum* gen. n. and *Longihaptor* gen. n. American Midland Naturalist 81:370–386.

———, ———, **and J. W. Crane.** 1968. Studies on monogenetic trematodes. XXXVIII. Ancyrocephalinae from South America with the proposal of *Jainus* gen. n. American Midland Naturalist 80: 186–198.

———, **and C. E. Price.** 1963. Additional haptoral hooks in the genus *Dactylogyrus*. Journal of Parasitology 49:1028–1029.

Molnar, K., G. Hanek, and C. H. Fernando. 1974. Ancyrocephalids (Monogenea) from freshwater fishes of Trinidad. Journal of Parasitology 60: 914–920.

Price, E. W. 1938. The monogenetic trematodes of Latin America. Livro Jubilar Professor Travassos, Rio de Janeiro, Brazil 3:407–413.

Scholz, T., and V. Hanzelová. 1998. Tapeworms of the Genus *Proteocephalus* Weinland, 1858 (Cestoda: Proteocephalidae), Parasites of Fishes in Europe. Academia, Publishing House of the Academy of Sciences of the Czech Republic, Prague. No. 2, 1998, 119 pp.

———, **J. Vargas-Vásquez, F. Moravec, C. Vivas-Rodríquez, and E. Mendoza-Franco.** 1995. Cenotes (sinkholes) of the Yucatan Peninsula, Mexico, as a habitat of adult trematodes of fish. Folia Parasitologica 42:173–192.

Silfvergrip, A. M. C. 1996. A systematic revision of the neotropical catfish genus *Rhamdia* (Teleostei, Pimelodidae). Stockholm University, Stockholm, Sweden. 156 pp. + 8 plts.

Suriano, D. M. 1986a. El genero *Urocleidoides* Mizelle y Price, 1964 (Monogenea: Ancyrocephalidae). Anatomia y posición sistematica. *Urocleidoides mastigatus* sp. nov. y *U. travassosi* (Price, 1934) Molnar, Hanek y Fernando, 1974 parasitas de *Rhamdia sapo* (Valenciennes, 1840) Eigenmann y Eigenmann, 1888 y *Pimelodella laticeps* Eigenmann, 1917 (Pisces: Siluriformes) de la Laguna de Chascomus, Republica Argentina. Physis (Buenos Aires), Sec. B, 44 (107):73–80.

———. 1986b. *Philocorydoras platensis* gen. n. et sp. n. (Monogenea: Ancyrocephalidae) from *Corydoras paleatus* (Jenyns) (Pisces: Callichthyidae) in Laguna Chascomús-República Argentina. Helminthologia 23:249–256.

———, **and I. S. Incorvaia.** 1995. Ancyrocephalid (Monogenea) parasites from siluriform fishes from the Paranean-Platean ichthyogeographical province in Argentina. Acta Parasitologica 40:113–124.

Comp. Parasitol.
67(1), 2000 pp. 85–91

Species of *Sciadicleithrum* (Dactylogyridae: Ancyrocephalinae) of Cichlid Fishes from Southeastern Mexico and Guatemala: New Morphological Data and Host and Geographical Records

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ABSTRACT: A survey of species of *Sciadicleithrum* (Monogenea: Dactylogyridae) from the gills of cichlid fishes from the Yucatan Peninsula of Mexico and neighboring regions is provided. *Sciadicleithrum mexicanum* Kritsky, Vidal-Martínez, and Rodríguez-Canul is reported from *Cichlasoma urophthalmus* (type host), *Cichlasoma aureum*, and *Petenia splendida* (new host records) from Mexico, and *Cichlasoma trimaculatum* from Guatemala (new host and geographical record); *Sciadicleithrum bravohollisae* Kritsky, Vidal-Martínez, and Rodríguez-Canul from *Cichlasoma geddesi*, *Cichlasoma lentiginosum*, *Cichlasoma managuense*, *Cichlasoma salvini*, and *Cichlasoma* sp. (all new host records); *Sciadicleithrum splendidae* Kritsky, Vidal-Martínez, and Rodríguez-Canul from *Cichlasoma friedrichstahli* and *C. managuense* (new host records); and *Sciadicleithrum meekii* Mendoza-Franco, Scholz, and Vidal-Martínez from *Cichlasoma callolepis*, *Cichlasoma helleri*, and *C. managuense* (new host records) from Mexico. Data on morphological and biometrical variability of individual species from different hosts are provided. Species of *Sciadicleithrum* from Mexico and Guatemala exhibit wide host specificity. The present records expand distributional areas of all of the species of *Sciadicleithrum* studied.

KEY WORDS: *Sciadicleithrum*, Monogenea, Dactylogyridae, Cichlidae, host specificity, zoogeography, Mexico, Guatemala.

Sciadicleithrum was proposed by Kritsky et al. (1989) to accommodate 9 species of Ancyrocephalinae (Monogenea: Dactylogyridae) from cichlid fishes from South America. Since then, 4 other species have been described from cichlids of the Peninsula of Yucatan, Mexico, namely *Sciadicleithrum mexicanum* Kritsky, Vidal-Martínez, and Rodríguez-Canul, 1994, from *Cichlasoma urophthalmus* (Günther, 1862); *Sciadicleithrum bravohollisae* Kritsky, Vidal-Martínez, and Rodríguez-Canul, 1994, from *Cichlasoma pearsei* (Hubbs, 1936), *Cichlasoma synspilum* Hubbs, 1935, and *Petenia splendida* Günther, 1862; *Sciadicleithrum splendidae* Kritsky, Vidal-Martínez, and Rodríguez-Canul, 1994, from *Petenia splendida*; and *Sciadicleithrum meekii* Mendoza-Franco, Scholz, and Vidal-Martínez, 1997, from *Cichlasoma meeki* (Brind, 1918) (Kritsky et al., 1994; Mendoza-Franco et al., 1997).

During a recent helminthological study on cichlid fishes from southeastern Mexico, monogeneans assigned to *Sciadicleithrum* were found on the gills of several species of Cichlidae, including hosts not reported previously. This new material allowed us to supplement the original descriptions (sometimes based on a limited number of specimens) by morphological and biometrical data on intraspecific variability of individual species from different fish hosts. Numerous new host and geographical records also made it possible to evaluate the specificity of species of *Sciadicleithrum* from the Yucatan Peninsula.

Material and Methods

Cichlids were collected by line and hook, throw nets, or electrofishing from the following localities in southeastern Mexico and Guatemala: Mexico. State of Chiapas: Cedros River (16°45′21″N; 91°09′30″W) and Lacanjá River (16°46′21″N; 91°04′21″W). State of Tabasco: Santa Anita Lagoon (18°22′15″N; 92°53′10″W); El Yucateco Lagoon (18°11′33″N; 94°00′35″W); Paraíso River (18°25′35″N; 93°12′00″W); Ilusiones Lagoon (17°59′46″N; 92°56′17″W); Horizonte Lagoon (18°14′57″N; 92°49′59″W); Yumká Lake (18°00′37″N; 92°48′12″W); Puyacatengo River (17°34′58″N; 92°53′22″W). State of Campeche: Palizada River (18°17′16″N; 91°56′52″W); Silvituc La-

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goon (18°37′50″N; 90°16′50″W); La Pera Lagoon (18°16′57″N; 91°56′21″W); Términos Lagoon, Santa Gertrudis station (18°26′51″N; 91°48′59″W); Rancho II station (18°20′30″N; 91°42′30″W); El Viento station (18°26′01″N; 91°49′48″W). State of Yucatán: Dzaptún Cenote (20°51′19″N; 90°14′09″W); Chaamac Cenote (20°51′53″N; 90°09′18″W); Petentuche Cenote (21°33′90″N; 88°04′44″W); Dzonot Cervera Cenote (21°22′36″N; 88°49′59″W); Ojo de Agua (= water spring), Celestún Lagoon (20°52′37″N; 90°21′18″W). State of Quintana Roo: Mahahual (18°58′17″N; 87°57′30″W); Cenote Azul (Bacalar) (18°38′11″N; 88°24′46″W); Raudales Lagoon (18°42′27″N; 88°15′22″W); Hondo River (18°17′N; 88°38′W); Valle Hermoso Lagoon (19°10′N; 88°31′W); Rancho Don Milo (18°37′43″N; 88°01′15″W). Guatemala: Champerico River, Department of Retalhuleu, near the border with El Salvador (14°20′N; 91°54′W).

Sampling dates and parameters of infection are provided for each species in the results section. Fishes were transported alive to the laboratory and dissected using standard parasitological procedures. Monogeneans found on the gills were isolated and fixed with a glycerin–ammonium picrate mixture and then remounted in Canada balsam (Ergens, 1969). However, in most cases this technique was modified using Berland's solution before applying a glycerin–ammonium picrate mixture (Berland, 1961). A worm was placed in a small droplet of water on a slide, a small amount of Berland's solution was added using a fine brush, and the worm was covered with a coverslip. Excessive solution was removed with filter paper. Each corner of the coverslip was sealed with Du-Noyer sealant (Ergens and Gelnar, 1992) and glycerin-ammonium picrate solution was added to the edge of the coverslip. Processed worms were remounted in Canada balsam according to Ergens (1969).

In addition, some worms, fixed in 4% formalin, were stained with Gomori's trichrome to study the morphology of internal organs; others were mounted unstained in glycerin jelly. All measurements are given in micrometers; the mean is followed by the range and number of specimens measured in parentheses. The number of fishes infected of the total examined is followed by the mean and the minimum and maximum in parentheses. Drawings were made with the aid of an Olympus drawing tube.

Specimens were deposited in the National Helminthological Collection of Mexico, Institute of Biology, National Autonomous University of Mexico (UNAM), Mexico (CNHE); the United States National Parasite Collection, Beltsville, Maryland, U.S.A. (USNPC); the Institute of Parasitology, Academy of Sciences of the Czech Republic, České Budějovice, Czech Republic (IPCAS); the Natural History Museum, London, United Kingdom (BMNH); and the Laboratory of Parasitology, Center for Research and Advanced Studies of the National Polytechnic Institute (CINVESTAV-IPN), Mérida, Yucatan, Mexico (CHCM).

Results

Sciadicleithrum bravohollisae Kritsky, Vidal-Martínez, and Rodríguez-Canul, 1994

MEASUREMENTS (based on 10 specimens from *Cichlasoma salvini* (Günther 1862)): Haptor 142 (120–190; $n = 5$) wide, 77 (62–105; $n = 6$) long. Pharynx spherical, 47 (39–65; $n = 4$) in diameter. Ventral hamuli 29 (27–32; $n = 18$) long; base width 17 (16–18; $n = 18$). Dorsal hamuli 33 (31–34; $n = 15$) long; base width 15 (13–16; $n = 15$). Ventral bar 35 (29–45; $n = 7$) long; dorsal bar 36 (31–44; $n = 6$) long. Hooks 15 (13–15; $n = 20$) long. Male copulatory organ 31 (25–36; $n = 4$) long. Accessory piece 20 (18–22; $n = 6$) long.

HOSTS, LOCALITIES, SAMPLING DATES, AND PARAMETERS OF INFECTION: *Cichlasoma geddesi* Regan, 1905, Horizonte Lagoon (17 November 1998), 1 fish infected of 1 examined; mean intensity of infection 12 specimens; *Cichlasoma lentiginosum* (Steindachner, 1864), Lacanjá River (21 May 1998), 2/3, 3 (minimum intensity 3, maximum intensity 4); *Cichlasoma managuense* (Günther, 1869), Santa Gertrudis (17 March 1998), 1/1, 2; *C. pearsei,* Santa Gertrudis (17 March 1998), 1/7, 2; Lacanjá River (21 May 1998), 2/3, (3–6); Palizada River (15 June 1998), 1/1, 4; *C. salvini,* Dzaptún Cenote (21 August 1996; 1 October 1997), 3/4, 2 (3–5), 5/5 (4–5); Lacanjá River (21 May 1998), 1/1, 2; Ilusiones Lagoon (16 November 1998), 5/10, 2 (1–3); Horizonte Lagoon (17 November 1998), 1/1, 19; Yumká Lake (18 November 1998), 1/3, 1; Puyacatengo River (19 November 1998), 2/10, 3 (1–6); *C. synspilum,* Cenote Azul (2 March 1998), 1/1, 6; Rancho II (17 March 1998), 1/6, 1; Raudales Lagoon (8 May 1998), 2/16, 14 (8–21); Palizada River (15 June 1998), 1/6, 5; *Cichlasoma* sp., Paraíso River (20 March 1998), 1/1, 5.

SPECIMENS DEPOSITED: Voucher specimens from *C. pearsei* and *C. synspilum* in CHCM (Nos. 215 and 214), from *C. salvini* in CNHE (Nos. 3132 and 3133), IPCAS (No. M-348), USNPC (Nos. 88946 and 88950), and CHCM (Nos. 229 and 231).

REMARKS: Specimens found in *C. salvini* do not differ from those of *S. bravohollisae,* as described from *C. pearsei, C. synspilum,* and *P. splendida* by Kritsky et al. (1994). The present material enabled us to add some new data on the morphology of the copulatory complex and the vaginal aperture. Kritsky et al. (1994) reported the length of the accessory piece to be 31–45 and 26–37 in specimens from *C. pearsei* and *C. synspilum,* respectively. The present specimens have the accessory piece considerably shorter, measuring only 18–22.

The shape of the vaginal aperture is similar to

Table 1. Measurements (in micrometers; mean with range in parentheses; *n* = number of measurements) of *Sciadicleithrum mexicanum* Kritsky, Vidal-Martínez, and Rodríguez-Canul, 1994, from 4 species of cichlid fishes from the Yucatan Peninsula of Mexico and Guatemala.

	*Cichlasoma urophthalmus**	*n*	*Cichlasoma trimaculatum*†	*n*	*Cichlasoma aureum*	*n*	*Petenia splendida*	*n*
Body length	320 (245–398)	24	302 (249–319)	4	—	0	—	0
Pharynx width	18 (15–20)	20	22 (19–26)	3	25 (16–32)	11	28	1
Ventral hamuli length	33 (29–35)	22	33 (32–35)	14	30 (29–32)	22	32 (31–33)	10
Ventral hamuli width	16 (15–17)	17	15 (14–16)	14	14 (13–15)	22	15 (14–17)	10
Dorsal hamuli length	39 (35–41)	19	39 (35–41)	12	36 (35–37)	18	37 (34–39)	10
Dorsal hamuli width	14 (13–16)	16	16 (14–19)	11	13 (13–15)	18	12 (11–15)	10
Ventral bar length	34 (30–37)	21	38 (31–44)	7	38 (32–43)	11	36 (35–40)	5
Dorsal bar length	31 (29–33)	21	34 (32–37)	7	34 (29–39)	11	34 (31–42)	5
Hooks	15 (14–17)	71	15 (15–16)	8	15 (14–17)	31	17 (15–18)	12
MCO‡ length	62 (53–68)	17	42 (35–45)	4	39 (32–44)	11	45 (42–51)	5
Accessory piece	45 (37–52)	12	—	0	48 (42–52)	11	47	1

* Original descriptions of *S. mexicanum* by Kritsky et al. (1994).

† Champerico River, Guatemala.

‡ Male copulatory organ.

the original description in the presence of 2 opposing funnel-shaped distal sclerites. Comparison of specimens from *C. salvini* and *C. synspilum* showed that this structure was smaller (length 10–11) and more delicate in the former fish host than in worms from *C. synspilum* (length 19–23). Measurements of the haptor of the currently studied specimens are also noticeably greater than those of this species reported from *C. pearsei, C. synspilum,* and *P. splendida* (Kritsky et al., 1994). It is possible that the specimens studied are more strongly extended, as a result of fixation with Berland's solution and a glycerin–ammonium picrate mixture, since the method of fixation greatly influences size in soft body parts. Specimens found on *C. geddesi, C. lentiginosum, C. managuense, C. salvini,* and *Cichlasoma* sp. represent new host records and expand the distributional area of *S. bravohollisae* to the Mexican states of Campeche, Chiapas, and Tabasco.

Sciadicleithrum meekii Mendoza-Franco, Scholz, and Vidal-Martínez, 1997

MEASUREMENTS (based on 8 specimens from *C. meeki*): Haptor 74 (63–80; *n* = 3) wide. Pharynx spherical, 15 (13–15; *n* = 5) in diameter. Ventral hamuli 19 (16–23; *n* = 4) long; base width 12 (11–13; *n* = 4). Dorsal hamuli 33 (32–34; *n* = 2) long; base width 11. Ventral bar 19 (16–23; *n* = 4) long; dorsal bar 29 (27–30; *n* = 4) long. Hooks 13 (12–13; *n* = 14) long.

HOSTS, LOCALITIES, SAMPLING DATES, AND PARAMETERS OF INFECTION: *Cichlasoma callolepis* (Regan, 1904), Lacanjá River (19 May 1998), 1/3, 9; *Cichlasoma helleri* (Steindachner, 1864), Ilusiones Lagoon (16 November), 2/10, 2 (1–3); Horizonte Lagoon (17 November 1998), 1/10, 1; Yumká Lake (18 November 1998), 1/10, 3; *C. meeki,* Mahahual (8 December 1997), 2/10 (2–12); Mahahual (2 March 1998), 2/2, 4 (3–5); Chaamac Cenote (16 April 1998), 2/6, 7 (2–12); *C. managuense,* El Viento (17 March 1998), 1/1, 2; Santa Anita Lagoon (24 April 1998), 1/1, 3; Hondo River (11 May 1998), 1/4, 3.

SPECIMENS DEPOSITED: Voucher specimens from *C. helleri* in CNHE (No. 3722), CHCM (No. 227) and USNPC (No. 88947).

REMARKS: Findings of *S. meekii,* originally described from *C. meeki,* in 3 other cichlid species demonstrate that this parasite is not restricted to the type host, *C. meeki.*

Sciadicleithrum mexicanum Kritsky, Vidal-Martínez, and Rodríguez-Canul, 1994

MEASUREMENTS: Measurements of 28 specimens studied from different hosts are given in Table 1.

HOSTS, LOCALITIES, SAMPLING DATES, AND PARAMETERS OF INFECTION: *Cichlasoma aureum* (Günther, 1862), Petentuche Cenote (10 October 1997), 2/2, 54 (32–76); Ojo de Agua in Celestún Lagoon (15 August 1997), 1/2, 13; Chaamac Cenote (16 April 1998), 1/1, 75; Dzonot Cervera Cenote (15 April 1998), 1/1, 5; *Cichlasoma friedrichstahli* (Heckel, 1840), Dzonot Cervera

Cenote (15 April 1998), 1/1, 20; *Cichlasoma octofasciatum* (Regan, 1903), Cedros River (19 May 1998), 4/4, 4 (1–7); *Cichlasoma trimaculatum* (Günther, 1869), Champerico River (16 December 1995), 3/3, 34 (14–37); *C. urophthalmus,* El Yucateco Lagoon (30 January 1998), 7/8, 65 (2–284); Cenote Azul (2 March 1998), 1/3; 4, Mahahual (2 March 1998), 3/3, 16 (4–16); Dzonot Cervera Cenote (15 April 1998), 1/3, 1; Rancho Don Milo (8 May 1998), 1/3, 12; La Pera Lagoon (15 June 1998), 1/8, 50; Petentuche Cenote (10 October 1997), 1/1, 52; *P. splendida,* Dzaptún Cenote (21 August 1996), 1/1, 18; Silvituc Lagoon (15 July 1997), 1/1, 7; Valle Hermoso Lagoon (2 March 1998), 1/1, 15; Palizada River (15 June 1998), 1/2, 12; Santa Anita Lagoon (24 April 1998), 1/1.

SPECIMENS DEPOSITED: Voucher specimens from *C. aureum* and *C. friedrichstahli* in USNPC (Nos. 88943 and 88945); from *C. trimaculatum* in CNHE (No. 3136), USNPC (No. 88944), BMNH (No. 1999.7.13.25), and CHCM (Nos. 224 and 225); from *P. splendida* in CNHE (Nos. 3135 and 3136), IPCAS (No. M-343), USNPC (No. 87303), and CHCM (No. 220).

REMARKS: The morphology and measurements of the specimens found in the different hosts correspond well to the description of *S. mexicanum* from *C. urophthalmus* by Kritsky et al. (1994). *Cichlasoma aureum, C. trimaculatum,* and *P. splendida* represent new host records. The finding of *S. mexicanum* in Guatemala is the first record of this parasite in Central America. The present data, together with those of Mendoza-Franco et al. (1999), who reported *S. mexicanum* from *C. friedrichstahli, C. octofasciatum,* and *C. synspilum,* demonstrate a wide host specificity of *S. mexicanum.*

Sciadicleithrum splendidae Kritsky, Vidal-Martínez, and Rodríguez-Canul, 1994 (Figs. 1–11)

MEASUREMENTS: Measurements of 41 specimens studied from different hosts and localities are given in Table 2.

HOSTS, LOCALITIES, SAMPLING DATES, AND PARAMETERS OF INFECTION: *C. friedrichstahli,* Dzaptún Cenote (1 August 1997), 1/1, 9; Mahahual (2 March 1998), 2/2, 9 (7–11); Cedros River (19 May 1998), 8/15, 10 (1–18); *C. managuense,* Santa Gertrudis (17 March 1998), 1/1, 4.

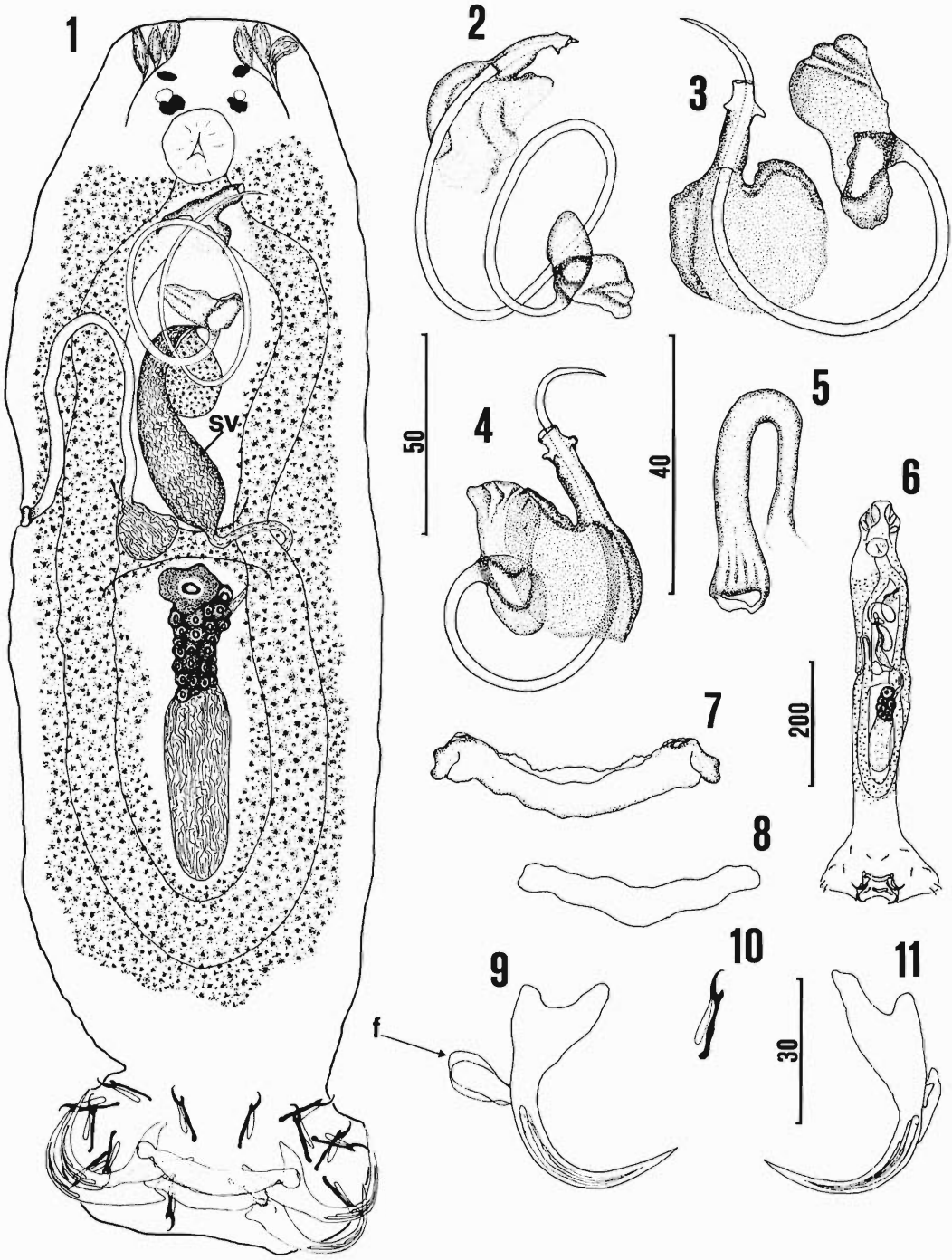
SPECIMENS DEPOSITED: Voucher specimens from *C. friedrichstahli* in CNHE (Nos. 3720 and 3721), CHCM (No. 218), USNPC (Nos. 88948 and 88949), and BMNH (No. 1999.7.13.26).

REMARKS: Both species of *Cichlasoma* studied are new hosts of *S. splendidae.* Specimens obtained from *C. friedrichstahli* closely resemble in their morphology those of *S. splendidae* from *P. splendida* previously described by Kritsky et al. (1994). All possess hamuli relatively similar in size and shape, the base of the copulatory organ with bilobed proximal branch, and a vagina comprising a sclerotized tube looping anteriorly on the dextromedial half of the trunk. However, there are slight differences between the present material and that of *S. splendidae* in the number of coils of the male copulatory organ (1.5 rings in the specimens studied versus more than 2 in *S. splendidae*) and the size of the accessory piece, 38 (30–45) in worms from *C. friedrichstahli* and 22 in specimens from the type host. Similarly to *S. bravohollisae,* the differences in the measurements of sclerotized and soft body parts of specimens from *C. friedrichstahli* might be related to the size of the worms and the method of fixation (see Fig. 6). A similar phenomenon has previously been observed among individual specimens of *Sciadicleithrum umbilicum* Kritsky, Thatcher, and Boeger, 1989, from South America (Kritsky et al., 1989).

The original description of *S. splendidae* was based on only 2 specimens (Kritsky et al., 1994). The additional material from this study evidences that the shape and number of coils of the male copulatory organ vary among specimens from the same hosts (Figs. 2–4) and that this species possesses a seminal vesicle (see Fig. 1) that lacks a thickened wall, as is present in congeneric species of *Sciadicleithrum* from Yucatan (Kritsky et al., 1994; Mendoza-Franco et al., 1997). As for *S. bravohollisae, S. splendidae* occurs in members of 2 genera of cichlid fishes, *Cichlasoma* and *Petenia.*

Discussion

Species of *Sciadicleithrum* were first reported from cichlids from South America (Kritsky et al., 1989) and subsequently from cichlid fishes from southeastern Mexico (Kritsky et al., 1994; Mendoza-Franco et al., 1997, 1999). The present study confirmed previous observations by the latter authors that the fauna of monogeneans assigned to *Sciadicleithrum* from the Yucatan Peninsula of Mexico and neighboring areas is depauperate in the number of species.



Figures 1–11. ***Sciadicleithrum splendidae*****. 1. Total view (ventral). 2–4. Copulatory complexes (2, 3. ventral view; 4. dorsal view). 5. Vagina. 6. Whole mount (fixed with Berland's solution and a glycerin–ammonium picrate mixture). 7. Ventral bar. 8. Dorsal bar. 9. Ventral hamulus. 10. Hook. 11. Dorsal hamulus. Scale bars = 50 μm (Fig. 1), 30 μm (Figs. 2, 7–11), 40 μm (Figs. 3–5), and 200 μm (Fig. 6). sv = seminal vesicle; f = filament.**

Table 2. Measurements (in micrometers; mean with range in parentheses; *n* = number of structures measured) of *Sciadicleithrum splendidae* Kritsky, Vidal-Martínez, and Rodríguez-Canul, 1994, from 3 species of cichlid fishes from 4 localities (Chiapas, Tabasco, Yucatán, and Quintana Roo) from southeastern Mexico.

	*Petenia splendida**	*n*	*Cichlasoma friedrichstahli*†	*n*	*Cichlasoma friedrichstah-li*‡	*n*	*Cichlasoma friedrichstah-li*§	*n*	*Cichlasoma motaguense*‖	*n*
Body length	250	1	621 (406–690)	8	—	0	—	0	—	0
Pharynx width	20	1	35 (24–43)	8	19 (17–24)	2	19 (17–20)	8	21 (18–24)	4
Ventral hamuli length	32	1	38 (36–38)	16	31 (29–34)	22	29 (22–31)	6	31 (30–32)	3
Ventral hamuli width	17	1	16 (15–18)	16	14 (12–16)	20	12 (12–13)	4	16	1
Dorsal hamuli length	40	1	42 (39–44)	16	37 (33–39)	16	36 (36–37)	4	38 (33–41)	4
Dorsal hamuli width	14	1	12 (10–14)	15	12 (12–14)	10	13	2	14 (14–15)	2
Ventral bar length	39	2	50 (42–53)	8	38 (36–43)	15	41 (40–41)	3	43 (40–46)	3
Dorsal bar length	37	2	41 (37–48)	8	30 (26–35)	14	32 (30–33)	3	32 (31–32)	2
Hooks	(15–16)	6	15 (15–16)	8	15 (15–16)	26	16 (15–17)	13	16 (15–17)	6
MCO¶ length	31 (27–35)	2	44 (42–50)	9	40 (31–49)	20	36 (29–39)	7	38 (27–51)	4
Accessory piece length	22	1	38 (30–45)	8	34 (29–36)	12	29 (20–39)	6	32 (24–47)	3

* Original descriptions of *S. splendidae* by Kritsky et al. (1994).
† Dzaptún Cenote, Yucatán.
‡ Cedros River, Chiapas.
§ Mahahual, Quintana Roo.
‖ Santa Gertrudis, Tabasco.
¶ Male copulatory organ.

In addition, this study expanded the spectrum of fish hosts of individual *Sciadicleithrum* taxa found in southeastern Mexico and Guatemala by 13 new host records. Each of the 4 species of *Sciadicleithrum* found in this area exhibits relatively wide host specificity in the same family, since they occur in as many as 8 cichlid species (*S. bravohollisae* in *C. geddesi, C. lentiginosum, C. managuense, C. pearsei, C. salvini, C. synspilum, P. splendida,* and *Cichlasoma* sp.).

Three species of *Sciadicleithrum* found in Yucatan occur even in members of 2 closely related genera, *Cichlasoma* and *Petenia.* It is also noteworthy that both *P. splendida* and *C. managuense* from southeastern Mexico harbor as many as 3 species of *Sciadicleithrum,* namely, *S. bravohollisae, S. mexicanum,* and *S. splendidae*; and *S. bravohollisae, S. splendidae,* and *S. meekii,* respectively. In the Terminos Lagoon (stations El Viento and Santa Gertrudis), 3 species of *Sciadicleithrum* (*S. bravohollisae, S. meekii,* and *S. splendidae*) occurred, and all these species are found on *C. managuense.* It can be assumed that horizontal transmission of these monogeneans to this cichlid occurred in this locality, with *C. helleri* probably serving as a source of *S. meeki,* and *C. pearsei* of *S. bravohollisae. Sciadicleithrum splendidae* was found only on *C. managuense* at this locality and may represent the original host of *S. splendidae.*

This study has also provided new data on intraspecific variability of *Sciadicleithrum* species from a wide spectrum of fish hosts and geographical regions. It is obvious that the knowledge of intraspecific morphological and biometrical variation is necessary to prevent descriptions of new taxa based only on slight morphological differences. This is important if the original descriptions were based on limited numbers of specimens, as in the case of *S. splendidae* (Kritsky et al., 1994).

A species of *Sciadicleithrum, S. mexicanum,* is reported from Guatemala for the first time in this study. However, the occurrence of other *Sciadicleithrum* taxa in Guatemala is highly probable, and we suggest that more studies on the helminth parasites of freshwater in that country and Central America in general be carried out. Investigations into fish helminths, including gill monogeneans, are therefore needed for a better understanding of the evolution of these parasites and their hosts in the transient area between the Nearctic and Neotropical zoogeographical regions.

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Literature Cited

Berland, B. 1961. Use of glacial acetic acid for killing parasitic nematodes for collection purposes. Nature, London 191:1320–1321.

Ergens, R. 1969. The suitability of ammonium picrate-glycerin in preparing slides of lower Monogenoidea. Folia Parasitologica 16:320.

———, **and M. Gelnar.** 1992. Monogenea and other ectoparasitic metazoans. Pages 4–32 *in* Methods of Investigating Metazoan Parasites. Training course on fish parasites. České Budějovice, March 10–23. Institute of Parasitology, Czechoslovak Academy of Sciences.

Kritsky, D. C., V. E. Thatcher, and W. A. Boeger. 1989. Neotropical Monogenea. 15. Dactylogyrids from the gills of Brazilian Cichlidae with proposal of *Sciadicleithrum* gen. n. (Dactylogyridae). Proceedings of the Helminthological Society of Washington 56:128–140.

———, **V. M. Vidal-Martínez, and R. Rodríguez-Canul.** 1994. Neotropical Monogenoidea. 19. Dactylogyridae of cichlids (Perciformes) from the Yucatan Peninsula, with descriptions of three new species of *Sciadicleithrum* Kritsky, Thatcher and Boeger, 1989. Journal of the Helminthological Society of Washington 61:26–33.

Mendoza-Franco, E. F., T. Scholz, and V. M. Vidal-Martínez. 1997. *Sciadicleithrum meekii* sp. n. (Monogenea: Ancyrocephalinae) from the gills of *Cichlasoma meeki* (Pisces: Cichlidae) from cenotes (=sinkholes) of the Yucatan Peninsula, Mexico. Folia Parasitologica 44:205–208.

———, ———, **C. Vivas-Rodríguez, and J. Vargas-Vázquez.** 1999. Monogeneans of freshwater fishes from cenotes (=sinkholes) in the Yucatan Peninsula, Mexico. Folia Parasitologica 46:267–273.

Meeting Announcement

The **Third Seminar on Food- and Water-Borne Parasitic Zoonoses in the 21st Century** will be held December 6–8, 2000 at the Royal River Hotel in Bangkok, Thailand. The meeting is jointly organized by the Faculty of Tropical Medicine of Mahidol University, the Parasitology and Tropical Medicine Association of Thailand, the TROPMED Alumni Association, and the SEAMEO TROPMED Network. For further information contact: Dr. Suvanee Supavej, Secretary of the 3rd FBPZ Organizing Committee, Faculty of Tropical Medicine, Mahidol University, 420/6 Rajvithi Road, Bangkok 10400, Thailand; phone (622) 2460321 or (662) 2469000-13, Fax (662) 2468340 or (662) 2469006, e-mail: tmssp@mahidol.ac.th.

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Digenean Fauna of Amphibians from Central Mexico: Nearctic and Neotropical Influences

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ABSTRACT: Specimens from 20 amphibian species from central Mexico were examined for helminths. We found 21 digenean species; 4 of them are recorded for the first time in Mexico. Twenty-two new host and 21 new locality reports are added. Previous reports of these helminth species are summarized, and biogeographical aspects of hosts and parasites are discussed.

KEY WORDS: Digenea, taxonomy, amphibians, Mexico, biogeography.

Mexico possesses one of the highest amphibian species richnesses in the world, with 285 species recorded so far, and an unusual level of endemism (60.7%) (Flores-Villela, 1993, 1998). In spite of this richness and the importance of this group of vertebrates in ecosystems, only 10% of the species in Mexico have been studied for helminth parasites.

We recently conducted a study of helminth parasites of amphibians in selected aquatic ecosystems in Mexico. We surveyed helminths of frogs, toads, and salamanders from several lakes of the Mexican plateau (Mesa Central), a tropical rain forest (Los Tuxtlas, Veracruz State), and a tropical dry deciduous and semideciduous forest (Chamela, Jalisco State). In this paper, we present a list of the digenetic trematodes of 20 species of amphibians that we analyzed during the last 3 yr. We also provide information about previous records of each helminth species in Mexico and discuss biogeographical aspects of parasites and hosts.

Materials and Methods

Between July 1996 and May 1998, we examined 647 specimens of amphibians belonging to 20 species and 8 genera (Table 1). We sampled in 12 localities: 8 from the Mesa Central, 1 from the Pacific coast, and 3 from the coast of the Gulf of Mexico (Map 1). However, digeneans were found only in frogs and salamanders of the following localities: Ciénaga de Lerma, Estado de México (19°17′N, 99°30′W); Lago de Chapala, Jalisco (20°17′N, 103°11′W); Lago de Cuitzeo, Michoacán (19°53′N, 100°50′W); Lago de Pátzcuaro, Michoacán (19°30′N, 101°36′W); Lago de Zacapu, Michoacán (19°49′N, 101°47′W); Manantiales de Cointzio, Michoacán (19°35′N, 101°14′W); Los Tuxtlas, Veracruz (Laguna El Zacatal, Laguna Escondida, and Los Tuxtlas Field Station; 20°37′N, 98°12′W); Estero Chamela, Jalisco (19°30′N, 105°6′W).

Hosts were collected by hand or with seine nets and were kept alive before parasitological analysis, which was carried out within 24 hr after capture. Hosts were killed with an overdose of anesthetic (sodium pentobarbitol) and examined by standard procedures.

Digeneans were relaxed with hot tap water, fixed in Bouin's fluid for 8 hr under coverglass pressure, and then placed in vials containing 70% alcohol; later, they were stained with Mayer's paracarmin, Delafield's hematoxylin, or Gomori's trichrome and mounted in permanent slides with Canada balsam. Drawings were made with the aid of a drawing tube. Voucher specimens of collected worms were deposited at the Colección Nacional de Helmintos (CNHE), Biology Institute, Mexico City.

Hosts were fixed following standard procedures (Simmons, 1985) and deposited at the Colección Nacional de Anfibios y Reptiles (CNAR), Biology Institute, Universidad Nacional Autónoma de México (UNAM), and in the Colección Herpetológica del Museo de Zoología (Faculty of Sciences, UNAM).

Results

We identified 21 digenean species (Figs. 1–20) of 11 genera and 10 families collected in 10 of the 20 species of frogs and salamanders analyzed (Table 2). Four of these represent new records in Mexico, *Catadiscus rodriguezi, Glypthelmins parva, Glypthelmins* sp., and *Fibricola* sp. metacercariae. We also add 22 new host records and 21 new locality records. The frog *Rana brownorum,* the salamanders *Ambystoma dumerilii, Ambystoma mexicanum,* and *Ambystoma tigrinum,* the toads *Bufo marinus* and *Bufo valliceps,* the hylids *Hyla arenicolor, Hyla eximia,* and *Pachymedusa dachnicolor,* and the leptodactylid *Eleutherodactylus rhodopis* were free from digenean infections.

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Table 1. Hosts, localities, and numbers of hosts examined in Mexico.

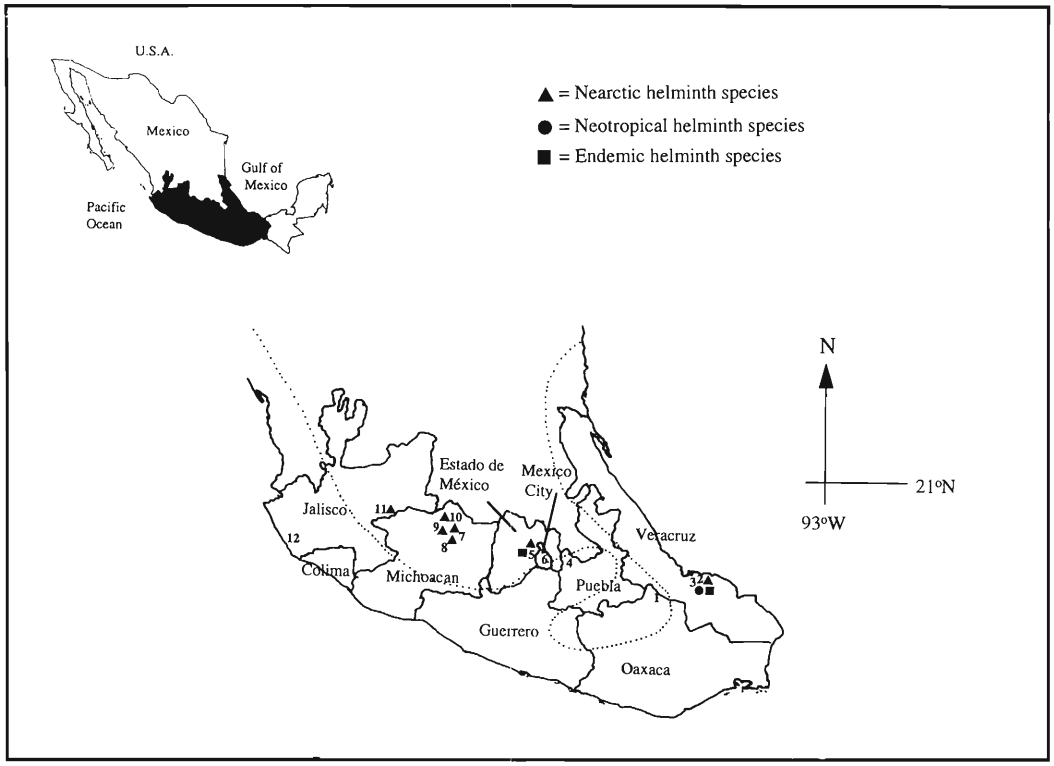
Host	Locality		Sample size
Anura			
Bufo marinus Linnaeus, 1758	Presa Miguel de la Madrid, Tuxtepec, Oaxaca		18
	Chamela, Jalisco		1
Bufo valliceps Weigmann, 1833	Laguna Escondida, Los Tuxtlas, Veracruz		4
Eleutherodactylus rhodopis Cope, 1867	Laguna El Zacatal, Los Tuxtlas, Veracruz		1
Hyla arenicolor Cope, 1886	Manantiales de Cointzio, Michoacán		11
Hyla eximia Baird, 1854	Manantiales de Cointzio, Michoacán		19
Leptodactylus melanonotus Hallowell, 1861	Laguna Escondida, Los Tuxtlas, Veracruz		4
Pachymedusa dachnicolor Cope, 1864	Chamela, Jalisco		2
Rana brownorum Sanders, 1973	Laguna El Zacatal, Los Tuxtlas, Veracruz		14
Rana dunni Zweifel, 1957	Lago de Pátzcuaro, Michoacán		74
	Lago de Zacapu, Michoacán		18
Rana forreri Boulenger, 1883	Estero Chamela, Chamela, Jalisco		12
Rana megapoda Taylor, 1942	Manantiales de Cointzio, Michoacán		27
	Lago de Chapala, Jalisco		4
Rana montezumae Baird, 1854	Ciénaga de Lerma, Estado de México		46
Rana neovolcanica Hillis and Frost, 1985	Lago de Cuitzeo, Michoacán		84
	Manantiales de Cointzio, Michoacán		41
Rana vaillanti Brocchi, 1877	Laguna Escondida, Los Tuxtlas, Veracruz		31
Smilisca baudinii Duméril and Bibron, 1841	Laguna Escondida, Los Tuxtlas, Veracruz		5
		Total	416
Urodela			
Ambystoma andersoni Krebs and Brandon, 1984	Lago de Zacapu, Michoacán		48
Ambystoma dumerilii Duges, 1870	Lago de Pátzcuaro, Michoacán		89
Ambystoma lermaensis (adults) Taylor, 1940	Ciénaga de Lerma, Estado de México		16
Ambystoma lermaensis (larvae)	Ciénaga de Lerma, Estado de México		42
Ambystoma mexicanum Shaw, 1789	Lago de Xochimilco, Mexico City		34
Ambystoma tigrinum Green, 1825	Lago La Mina Preciosa, Puebla		2
		Total	231

Discussion

Three clearly distinguishable groups are in this list, not considering *Fibricola* sp. and *Haematoloechus* sp. (Map 1). The first group is composed of species with nearctic distribution, such as *Cephalogonimus americanus, Glypthelmins californiensis, Glypthelmins quieta, Gorgoderina attenuata, Megalodiscus americanus, Halipegus occidualis, Haematoloechus complexus, Haematoloechus coloradensis, Haematoloechus longiplexus,* and *Haematoloechus medioplexus,* which have been previously recorded in Mexico and other parts of North America (see Brooks [1984] and references therein). The second group of species, including *C. rodriguezi, Glypthelmins facioi, G. parva, Loxogenes* (*Langeronia*) *macrocirra,* and *Mesocoelium monas,* has been recorded in South and Central America (Prudhoe and Bray, 1982). Finally, the third group is composed of endemic species: *Haematoloechus illimis, Haematoloechus pulcher,* and *Glypthelmins* sp.

The digenean fauna of the endemic amphibians (*A. andersoni, A. lermaense, A. dumerilii, R. montezumae, R. dunni, R. neovolcanica,* and *R. megapoda*) in the Transverse Volcanic Axis clearly has a Nearctic origin because none of the neotropical species of digeneans was found in this region, and the trematode fauna is formed of nearctic and endemic species of digeneans. On the other hand, the digenean fauna of the nonendemic host species *Leptodactylus melanonotus, Rana vaillanti,* and *Smilisca baudinii,* all collected in the Los Tuxtlas region in the tropical lowlands of the Gulf of Mexico, has a strong neotropical influence. Five of the 9 species reported from that region show a neotropical distribution.

In both cases, the parasite fauna reflects the biogeographic and phylogenetic links of the hosts. The endemic species of frogs in the Transverse Volcanic Axis, which represents the boundary between the nearctic and neotropical biogeographic zones, are members of the "*Rana*



Map 1. Map of Mexico showing collecting sites and limits of nearctic and neotropical regions (dotted line). 1 = Presa Miguel de la Madrid, Tuxtepec, Oaxaca; 2 = Laguna Escondida, Los Tuxtlas, Veracruz; 3 = Laguna El Zacatal, Los Tuxtlas, Veracruz; 4 = Lago La Mina Preciosa, Puebla; 5 = Ciénaga de Lerma, Estado de México; 6 = Lago de Xochimilco, Mexico City; 7 = Manantiales de Cointzio, Michoacán; 8 = Lago de Pátzcuaro, Michoacán; 9 = Lago de Zacapu, Michoacán; 10 = Lago de Cuitzeo, Michoacán; 11 = Lago de Chapala, Jalisco; 12 = Chamela, Jalisco.

pipiens complex," widely distributed from central Mexico to Canada (Hillis et al., 1983). Apparently, this group of frogs harbors a relatively homogeneous digenean fauna throughout its range to the volcanic axis. Little is known about the parasitic fauna of this group of frogs in the lowlands of Mexico. In some cases, as in the genus *Haematoloechus,* where an enormous diversity of species has been recorded, several speciation events have occurred in the endemic amphibians of this biogeographical area. This is the case for *H. pulcher,* probably derived from *H. complexus,* and *Haematoloechus illimis,* whose sister taxon is not clearly distinguished

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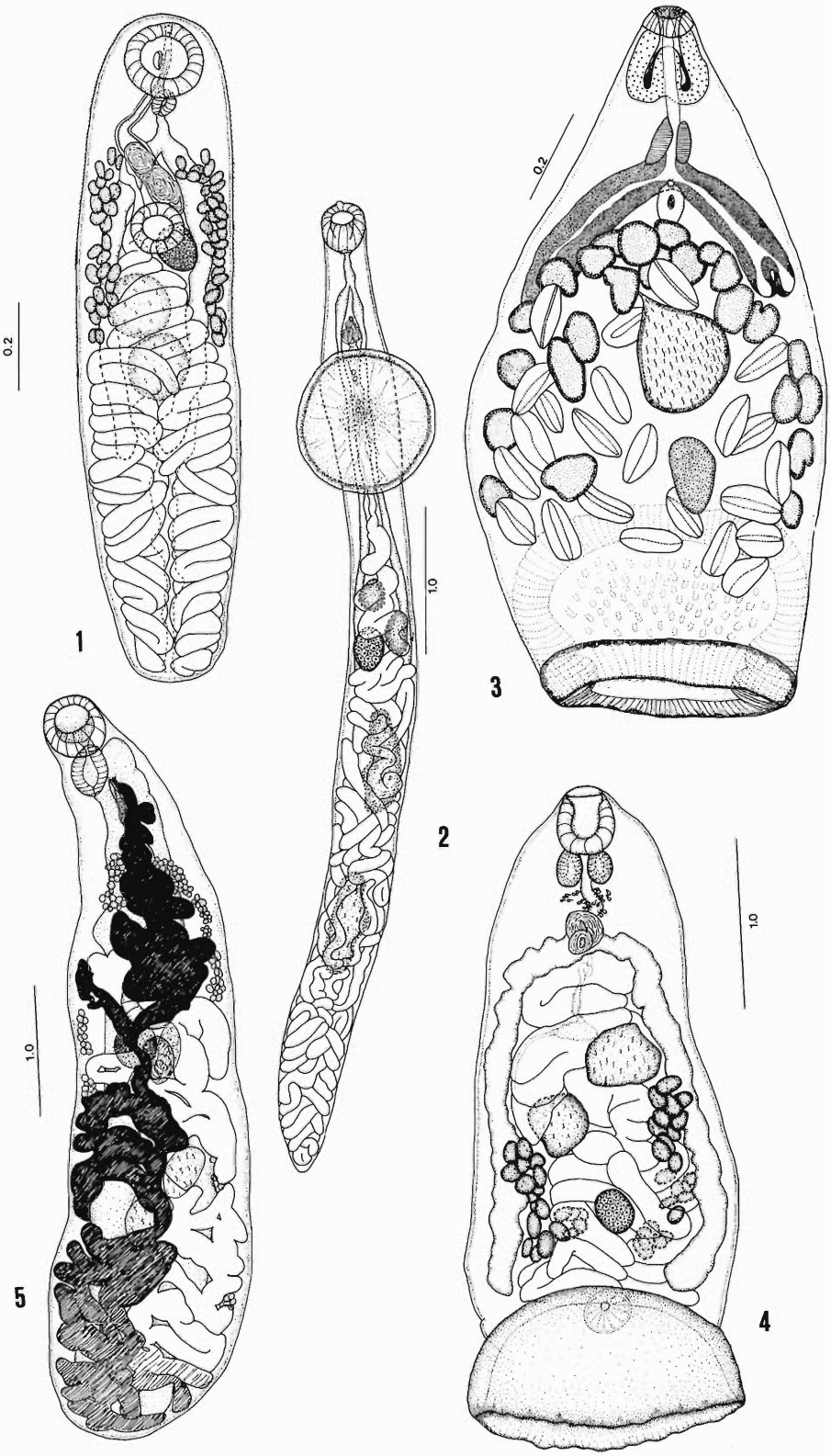
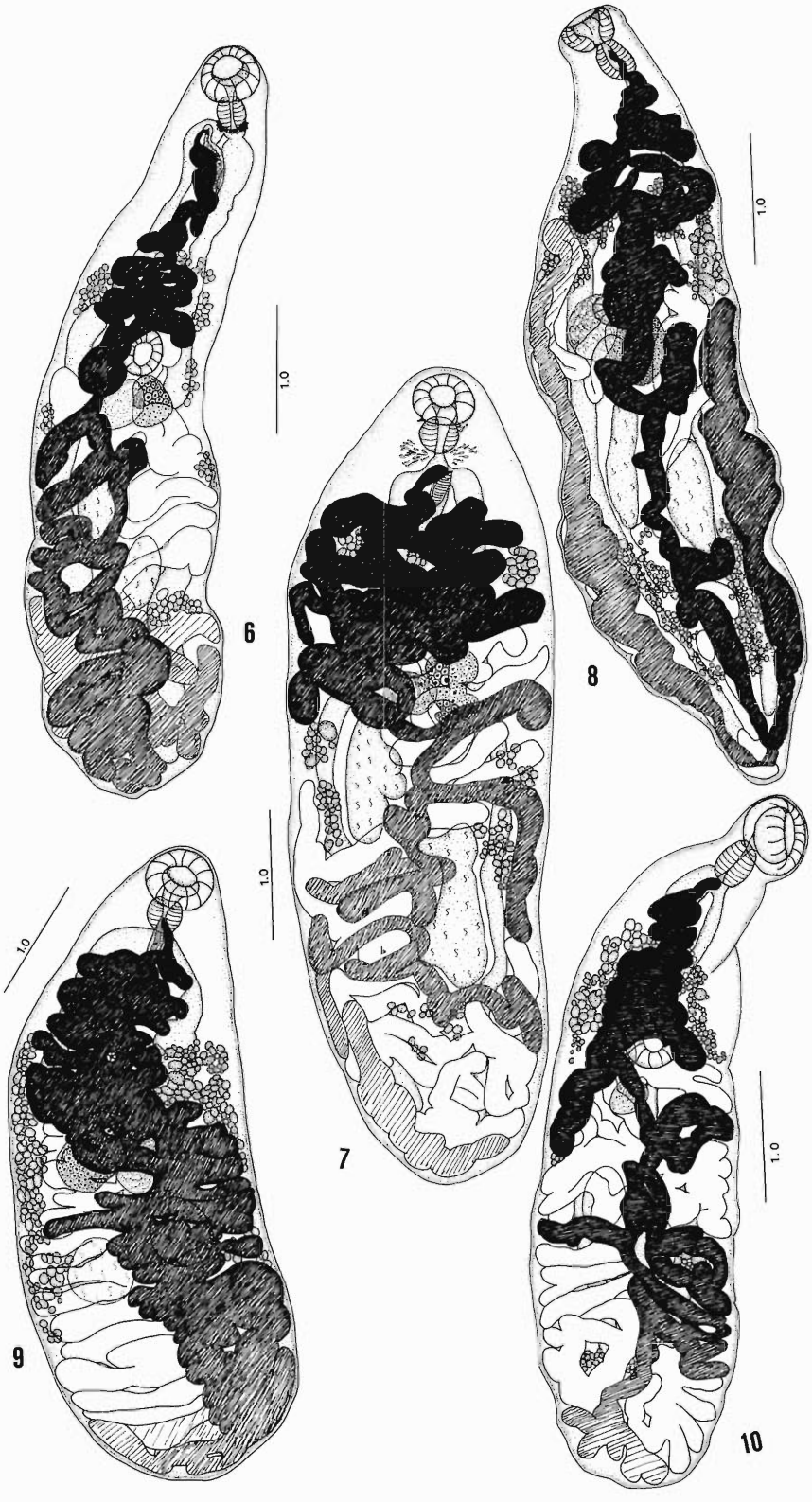
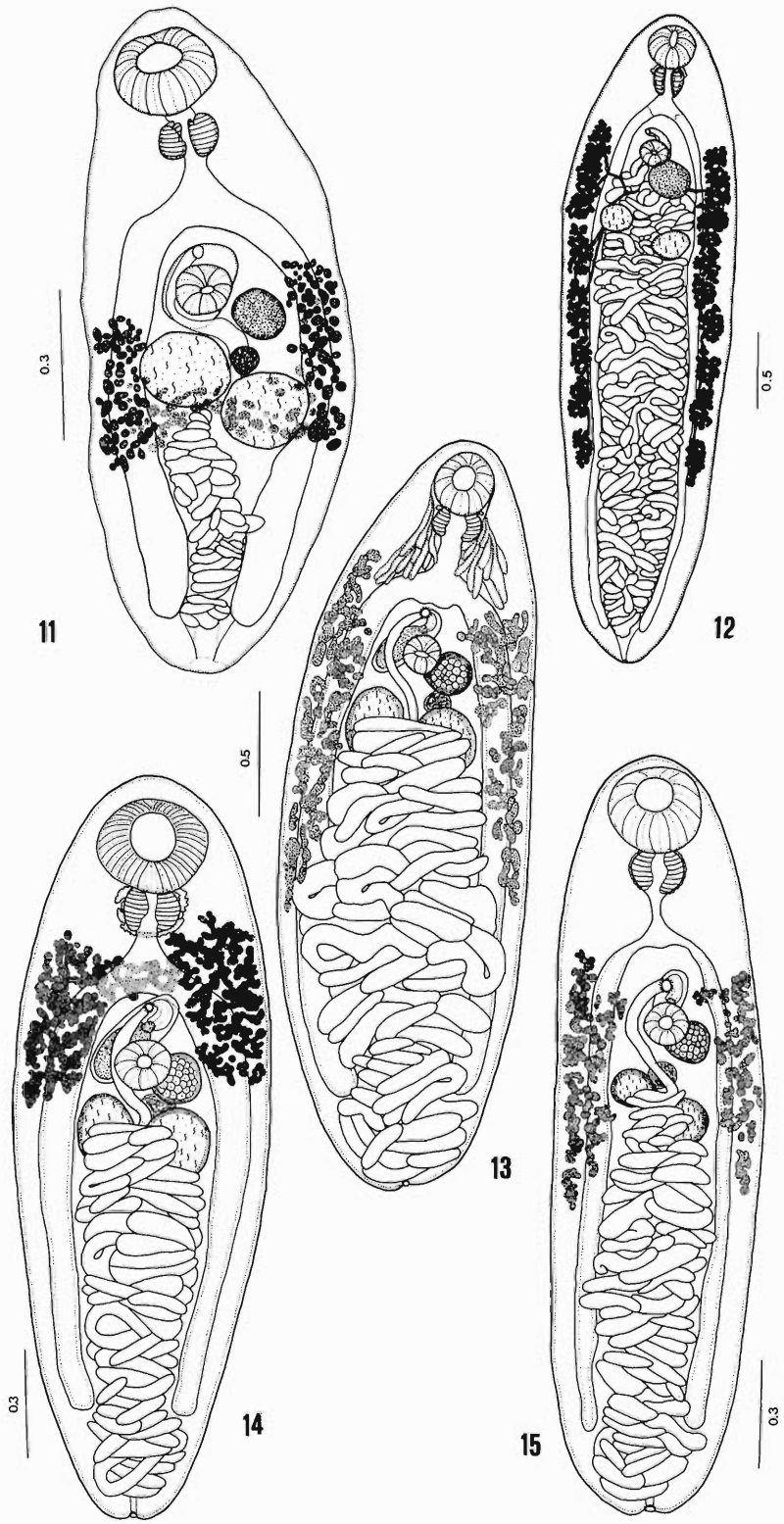
Figures 1–5. Ventral views. 1. *Cephalogonimus americanus* (Stafford, 1902) Stafford, 1905. 2. *Gorgoderina attenuata* (Stafford, 1902) Stafford, 1905. 3. *Megalodiscus americanus* Chandler, 1923. 4. *Catadiscus rodriguezi* Caballero, 1955. 5. *Haematoloechus coloradensis* (Cort, 1915) Ingles, 1932. Scales in millimeters.

Figures 6–10. Ventral views. 6. *Haematoloechus complexus* (Seely, 1906) Krull, 1933. 7. *Haematoloechus illimis* Caballero, 1942. 8. *Haematoloechus longiplexus* Stafford, 1902. 9. *Haematoloechus medioplexus* Stafford, 1902. 10. *Haematoloechus pulcher* Bravo, 1943. Scales in millimeters.

Figures 11–15. Ventral views. 11. *Glypthelmins parva* Travassos, 1934. 12. *Glypthelmins* sp. 13. *Glypthelmins californiensis* (Cort, 1919) Miller, 1930. 14. *Glypthelmins quieta* (Stafford, 1900) Stafford, 1905. 15. *Glypthelmins facioi* Brenes, Arroyo, Jiménez, and Delgado, 1959. Scales in millimeters.







(León-Règagnon et al., 1999). Species of *Haematoloechus* apparently have experienced a diversification in frogs and salamanders in central Mexico, representing the group with the highest species richness (6) in our samples. *Glypthelmins,* parasitic mainly in frogs of the new world, also shows high species richness. However, the presence at least of 4 species is mainly the result of independent host capture events, either from the neotropical or nearctic zones.

The distribution of the nonendemic frogs from the lowlands ranges from Ecuador and Colombia to Veracruz, Mexico (*R. vaillanti*), to Sonora, northwestern Mexico (*L. melanonotus*), and Texas (*S. baudinii*). Their digenean fauna in Veracruz is composed of a combination of neotropical and nearctic species. Only 2 digenean species, *Glypthelmins* sp. and *M. monas,* were recovered from *L. melanonotus* and *S. baudinii,* respectively. The former probably represents an undescribed species, and *M. monas* has been reported in numerous host genera in South America and Africa (Prudhoe and Bray, 1982). Seven digenean species were collected from *R. vaillanti,* and 3 of them show a neotropical distribution: *C. rodriguezi* in Panama (Caballero, 1955) and *G. parva* in Brazil (Prudhoe and Bray, 1982), both described from *Leptodactylus ocellatus,* and *G. facioi* in Costa Rica (Brenes et al., 1959) and Veracruz, Mexico (Razo-Mendivil et al., 1999), from *Rana palmipes* Spix, 1824, and *R. vaillanti,* respectively. The presence of these neotropical digeneans in Los Tuxtlas reflects the geographic distribution of the host genus *Leptodactylus* and the "*Rana palmipes* complex" (Frost, 1985; Hillis and De Sá, 1988). One species is endemic, *L.* (*L.*) *macrocirra,* and the 3 remaining species parasitizing *R. vaillanti* have a nearctic distribution; 2 of these species (*G. attenuata* and *C. americanus*) are also present in the endemic frogs of the Transverse Volcanic Axis, and the third species (*H. medioplexus*) has been recorded in several species of frogs from the United States and Canada, most commonly in members of the "*Rana pipiens* complex." The 3 species have a low host specificity and have been able to colonize several host groups, thus expanding their distribution range.

Apparently, a mixture of neotropical and nearctic species of parasites is taking effect in the lowlands of the Gulf of Mexico, with a series of very interesting phenomena of colonization of new hosts and habitats. Little is known about the amphibian parasite fauna of the tropical lowlands of the Pacific slope of Mexico or the southeastern part of the country. Those areas will undoubtedly be a source of extensive phylogenetic and biogeographic information on parasites and hosts in the future.

Contemporary ecological conditions are also important determinants of the parasitic fauna of a host species. Several authors have demonstrated a marked correlation between the relative amount of time spent in association with aquatic habitats and the number of species of platyhelminths hosted by frogs (Brandt, 1936; Prokopic and Krivanec, 1975; Brooks, 1976, 1984; Guillén, 1992). Our data clearly demonstrate that frogs and salamanders harbor the richer digenean fauna compared with the less water-dependent hylids or toads, where digeneans were almost or absolutely absent (the small sample size in leptodactylids precludes any discussion about their helminth fauna). Within frogs and salamanders, diet is the factor that most determines the richness of the digenean communities. Frogs become infected when they prey upon insects or copepods (which is the case in species of *Haematoloechus* and *Halipegus,* respectively), when they swallow their own skin bearing encysted metacercariae during ecdysis, or when they feed upon infected tadpoles (species of *Catadiscus, Cephalogonimus, Glypthelmins, Gorgoderina,* and *Megalodiscus*) (Yamaguti, 1975; Prudhoe and Bray, 1982). Salamanders of the genus *Ambystoma* Tschudi, 1838, hosted fewer digenean species than frogs. García-Altamirano et al. (1993) reported that *A. dumerilii* in Lake Pátzcuaro feeds mainly on crayfish and fish. As evidenced by the presence of *C. americanus, G. attenuata,* and *Haematoloechus* spp. in salamanders of our samples, it is possible that they oc-

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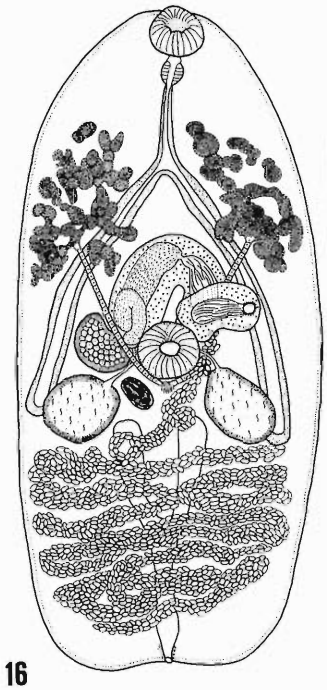
Figures 16–20. Ventral views. 16. *Loxogenes* (*Langeronia*) *macrocirra* (Caballero y Bravo, 1949) Yamaguti, 1971. 17. *Mesocoelium monas* (Rudolphi, 1819) Teixeira de Freitas, 1958. 18. *Halipegus occidualis* Stafford, 1905. 19. *Fibricola* sp. (metacercaria). 20. *Ochetosoma* sp. (metacercaria). Scales in millimeters.



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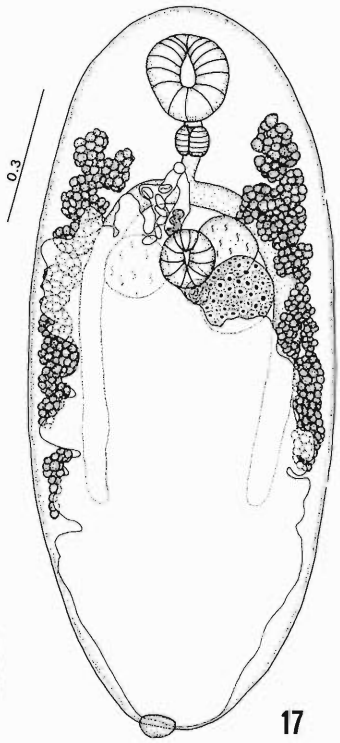

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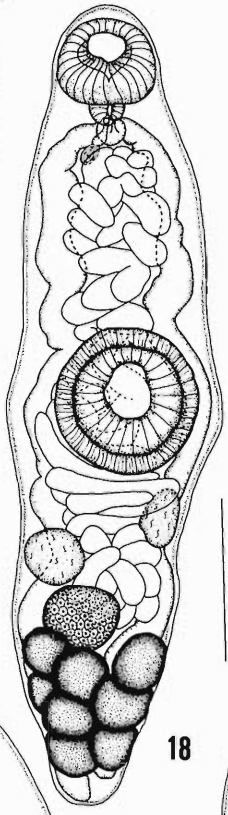

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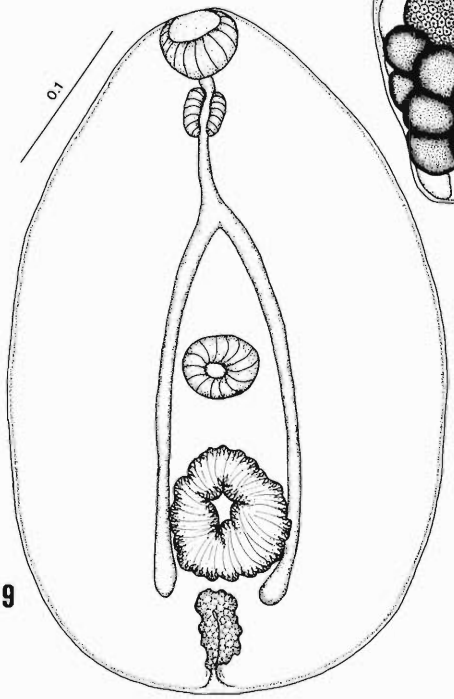

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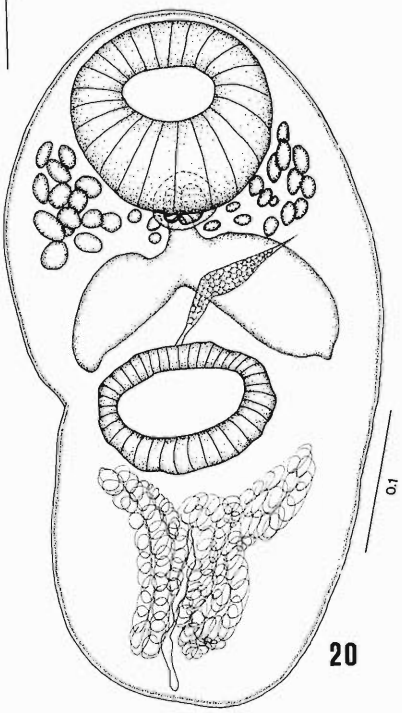

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Table 2. Digenetic trematodes of some amphibians in Mexico.

Helminth	Infection site	Host	Locality* (CNHE accession no.)	Previous records
Family Cephalogonimidae (Looss, 1899) Nicoll, 1914				
Cephalogonimus americanus (Stafford, 1902) Stafford, 1905 (Fig. 1)	Intestine	*Ambystoma andersoni*†	LZA† (3409, 3410)	
		Ambystoma dumerlii	LPA	García-Altamirano et al., 1993
		Ambystoma lermaensis†	CLE† (3411)	
		Rana berlandieri	EZA	Guillén, 1992
		Rana dunni	LZA (3357, 3358)	
			LPA (3408)	
			LPA	García-Altamirano et al., 1993; Pulido, 1994
		Rana montezumae	CLE (3359, 3360)	
			LXO	Caballero and Bravo-Hollis, 1940 *in* Lamothe et al., 1997
		Rana neovolcanica†	MCO† (3370)	
		Rana pipiens‡	LXO	Caballero and Bravo-Hollis, 1940 *in* Lamothe et al., 1997
		Rana vaillanti	LES (3425)	
			LES	Guillén, 1992
		Rhyacosiredon altamirani	SAL	Lamothe, 1964
		Bufo marinus	TUX, LES	Guillén, 1992
Family Gorgoderidae Looss, 1901				
Gorgoderina attenuata (Stafford, 1902) (Fig. 2)	Urinary bladder	*Ambystoma andersoni*†	LZA† (3412, 3413)	
		Ambystoma lermaensis†	CLE (3414, 3415)	
		Ambystoma tigrinum	LXO, CLE	Bravo-Hollis, 1943
		Rana dunni	LZA (3402), LPA (3405)	
			LPA	García-Altamirano et al., 1993; Pulido, 1994
		Rana megapoda†	MCO† (3403, 3404)	
		Rana montezumae	CLE (3401)	
			CLE	Caballero, 1942c
			Unspecified locality in central Mexico	Sokoloff and Caballero, 1933
			LXO, LTE	Iglesias, 1992 *in* Lamothe et al., 1997
		Rana neovolcanica†	MCO† (3403, 3404)	
		Rana pipiens‡	CLE	Caballero, 1942c
		Rana vaillanti	LES (3428)	
			LES	Guillén, 1992

Remarks: Pigulevsky (1953) stated that the material of Sokoloff and Caballero (1933) belonged to a new species, *G. skarbilovitschi* Pigulevsky, 1953, characterized by having lobed testes. We consider this difference to be a result of intraspecific variation.

Table 2. Continued.

Helminth	Infection site	Host	Locality* (CNHE accession no.)	Previous records
Family Paramphistomidae Fischoeder, 1901				
Megalodiscus americanus Chandler, 1923 (Fig. 3)	Intestine	*Rana dunni*†	LZA† (3353)	
		Rana megapoda†	LCU† (3351, 3352)	
			MCO† (3356)	
		Rana montezumae	CLE (3347–3350)	
			LXO, CLE	Bravo-Hollis, 1941
		Rana neovolcanica†	MCO (3354, 3355)	
		Rana pipiens‡	LMO	Martínez, 1969
Catadiscus rodriguezi Caballero, 1955 (Fig. 4)	Intestine	*Rana vaillanti*†	LES† (3308)	
Remarks: This species was originally described of *Leptodactylus pentadactylus* from Valle de Antón in Panama (Caballero, 1955). This is the first time it is recorded in Mexico.				
Family Haematoloechidae Odening, 1964				
Haematoloechus coloradensis (Cort, 1915) Ingles, 1931 (Fig. 5)	Lungs	*Rana dunni*	LZA† (3395, 3396)	
			LPA	García-Altamirano et al., 1993; Pulido, 1994; León-Règagnon et al., 1999
		Rana montezumae	CLE (3397)	
			CLE	Bravo-Hollis, 1945 *in* Lamothe et al., 1997; León-Règagnon et al., 1999
Remarks: Kennedy (1981) considered *H. coloradensis* to be a junior synonym of *H. complexus,* but León-Règagnon et al. (1999) demonstrated the validity of these species on the basis of molecular and morphological evidence.				
Haematoloechus complexus (Seely, 1906) Krull, 1933 (Fig. 6)	Lungs	*Ambystoma lermaensis*	CLE (3417)	
			CLE	León-Règagnon et al., 1999
			LXO, LTE	Iglesias, 1992 *in* Lamothe et al., 1997
		Rana megapoda†	MCO† (3380, 3379)	
		Rana montezumae	CLE (3374–3378)	
			CLE	León-Règagnon, 1992; León-Règagnon et al., 1999
		Rana neovolcanica†	MCO† (3380, 3379)	
		Rana pipiens‡	RPE, PLB	Martínez, 1969
Remarks: Originally recorded as *Ostiolum complexum* by Martínez (1969).				
Haematoloechus illimis Caballero, 1942 (Fig. 7)	Eustachian tubes, lungs	*Rana montezumae*	CLE (3381–3383)	
			CLE	Caballero, 1942a; León-Règagnon et al., 1999
Haematoloechus longiplexus Stafford, 1902 (Fig. 8)	Lungs	*Rana montezumae*	CLE (3394)	
			CLE	Caballero, 1941; León-Règagnon et al., 1999
		Rana pipiens‡	CLE	Caballero, 1941
			LTE	Iglesias, 1992 *in* Lamothe et al., 1997
Remarks: Originally recorded as *Haematoloechus macrorchis* Caballero, 1941, this species was declared junior synonym of *H. longiplexus* by León-Règagnon et al. (1999).				

Table 2. Continued.

Helminth	Infection site	Host	Locality* (CNHE accession no.)	Previous records
Haematoloechus medioplexus Stafford, 1902 (Fig. 9)	Lungs	*Rana berlandieri*	EZA	Guillén, 1992
		Rana montezumae	CLE, LXO	Caballero, 1941
		Rana pipiens‡	CLE, LXO	Caballero, 1941
		Rana vaillanti	LES (3424)	
			LES	Guillén, 1992
			LES	León-Règagnon et al., 1999
		Bufo valliceps	LES	Guillén, 1992
		Bufo marinus	TUX	Guillén, 1992
Haematoloechus pulcher Bravo-Hollis, 1943 (Fig. 10)	Lungs	*Ambystoma lermaensis*†	CLE (3418)	
		Ambystoma tigrinum	CLE	Bravo-Hollis, 1943
		Rana montezumae†	CLE (3398–3400)	
Haematoloechus sp.	Lungs	*Rana forreri*†	ECH†	
Remarks: Specimens belong to a different species from those mentioned above, but their poor preservation condition precluded specific identification. Not deposited in the CNHE.				
Family Macroderoididae Goodman, 1952				
Glypthelmins californiensis (Cort, 1919) Miller, 1930 (Fig. 13)	Intestine	*Rana dunni*	LPA (3280)	
			LZA† (3281, 3283, 3294)	
			LZA	Razo-Mendivil et al., 1999
			LPA	Pulido, 1994; Razo-Mendivil et al., 1999
		Rana megapoda	MCO	Razo-Mendivil et al., 1999
		Rana montezumae	CLE (3282)	
			CLE	Caballero, 1942b; Razo-Mendivil et al., 1999
			LXO	Caballero, 1941 *in* Lamothe et al., 1997
		Rana neovolcanica	MCO	Razo-Mendivil et al., 1999
Remarks: Records of this species by León-Règagnon (1992) and Guillén (1992) belong to *G. quieta* and *G. facioi,* respectively, in accordance with Razo-Mendivil et al. (1999).				
Glypthelmins quieta (Stafford, 1900) Stafford, 1905 (Fig. 14)	Intestine	*Rana dunni*	LPA (3273), LZA (3274)	
			LPA	Pulido, 1994; Razo-Mendivil et al., 1999
			LZA	Razo-Mendivil et al., 1999
		Rana megapoda	LCU† (3346), MCO (3416)	
			LCH† (3406)	
			MCO	Razo-Mendivil et al., 1999
		Rana montezumae	CLE (3271, 3275–3279)	
			CLE	León-Règagnon, 1992; Razo-Mendivil et al., 1999
			LXO, LTE	Iglesias, 1992 *in* Lamothe et al., 1997

Table 2. Continued.

Helminth	Infection site	Host	Locality* (CNHE accession no.)	Previous records
		Rana neovolcanica	MCO (3272)	
			MCO	Razo-Mendivil et al., 1999
Remarks: Specimens of León-Règagnon (1992) and 1 specimen of Pulido (1994) were orginally reported as *G. californiensis* and transferred to *G. quieta* by Razo-Mendivil et al. (1999).				
Glypthelmins facioi Brenes, Arroyo, Jiménez, and Delgado, 1959 (Fig. 15)	Intestine	*Rana berlandieri*	EZA	Guillén, 1992; Razo-Mendivil et al., 1999
		Rana vaillanti	LES (3285)	
			LES	Guillén, 1992; Razo-Mendivil et al., 1999
Remarks: Specimens of Guillén (1992) were originally reported as *G. californiensis* and transferred to *G. facioi* by Razo-Mendivil et al. (1999).				
Glypthelmis parva Travassos, 1934 (Fig. 11)	Intestine	*Rana vaillanti*†	LES† (3391)	
Remarks: This species was originally described in *Leptodactylus ocellatus* from Brazil (Travassos, 1924). This is the first time it is recorded in Mexico.				
Glypthelmins sp. (Fig. 12)	Intestine	*Leptodactylus melanonotus*†	LES† (3392)	
Remarks: These specimens represent a new species (Razo-Mendivil, unpubl. data)				
Family Lecithodendriidae Odhner, 1910				
Loxogenes (*Langeronia*) *macrocirra* (Caballero and Bravo-Hollis, 1949) Yamaguti, 1971 (Fig. 16)	Intestine	*Bufo marinus*	LCA, TUX	Guillén, 1992; Guillén, 1992 *in* Lamothe et al., 1997
		Rana berlandieri	EZA	Guillén, 1992; Guillén, 1992 *in* Lamothe et al., 1997
		Rana pipiens‡	PLB	Martínez, 1969
			Undetermined locality in Mexico	Caballero and Bravo-Hollis, 1949
		Rana vaillanti	LES (3307)	
			LES, TUX	Guillén, 1992; Guillén, 1992 *in* Lamothe et al., 1997
Remarks: Originally described as *Langeronia macrocirra* Caballero and Bravo-Hollis, 1949.				
Family Brachycoeliidae Johnston, 1912				
Mesocoelium monas (Rudolphi, 1819) Teixeira de Freitas, 1958 (Fig. 17)	Intestine	*Bufo marinus*	LCA	Guillén, 1992; Guillén, 1992 *in* Lamothe et al., 1997
			TUX	Guillén, 1992; Guillén, 1992 *in* Lamothe et al., 1997
		Smilisca baudini	LES† (3309)	
			EZA	Guillén, 1992; Guillén, 1992 *in* Lamothe et al., 1997

Table 2. Continued.

Helminth	Infection site	Host	Locality* (CNHE accession no.)	Previous records
		Rana neovolcanica	MCO (3272)	
Family Hemiuridae Looss, 1907				
Halipegus occidualis Stafford, 1905 (Fig. 18)	Eustachian tubes	*Rana montezumae*	CLE (3361, 3362)	
			LXO	Caballero, 1941; Caballero, 1947; Iglesias, 1992 *in* Lamothe et al., 1997
			CLE	Caballero, 1941; Iglesias, 1992 *in* Lamothe et al., 1997
			LTE	Iglesias, 1992 *in* Lamothe et al., 1997
		Rana pipiens‡	CLE	Caballero, 1941
Remarks: Caballero (1941) described *H. lermensis,* declared a junior synonym of *H. occidualis* by Rankin (1944). Caballero (1947) reported *Halipegus amhertensis* Rankin, 1944, in *Rana montezumae* from LXO, but after reexamination of specimens, McAlpine and Burt (1998) considered them to be *H. occidualis.*				
Family Diplostomidae Poirier, 1886				
Fibricola sp. (metacercariae) (Fig. 19)	Urinary bladder	*Rana montezumae*†	CLE† (3365, 3369)	
Remarks: Identification of this material is based on its comparison with the description of *Fibricola texensis* Chandler, 1942, metacercariae (Chandler, 1942), and the description of *F. caballeroi* Zerecero, 1943, in mammals from Mexico City (Zerecero, 1943).				
Family Plagiorchiidae (Lühe, 1901) Ward, 1917				
Ochetosoma sp. (metacercariae) (Fig. 20)	Intestine wall and liver	*Rana montezumae*†	CLE (3363, 3371)	
		Rana dunni	LZA† (3364)	Pulido, 1994
		Rana megapoda†	MCO† (3372, 3373)	
		Rana neovolcanica†	MCO† (3372, 3373)	
		Ambystoma dumerilii	LPA	García-Altamirano et al., 1993
	From fishes	*Goodea atripinnis*	LPA	Pérez-Ponce de León et al., 1996
		Neoophorus diazi	LPA	Pérez-Ponce de León et al., 1996

* CLE = Ciénaga de Lerma; ECH = Estero Chamela; EZA = Laguna El Zacatal; LCA = Lago de Catemaco, Veracruz; LCH = Lago de Chapala; LCU = Lago de Cuitzeo; LES = Laguna Escondida; LMO = Laguna Montford, Nuevo León; LPA = Lago de Pátzcuaro; LTE = Lago de Texcoco, Estado de México; LZA = Lago de Zacapu; MCO = Manantiales de Cointzio; PLB = Presa La Boca, Nuevo León; RPE = Río Pesquería, Nuevo León; SAL = Salazar, Estado de México; TUX = Los Tuxtlas Field Station, Veracruz.

† First host or locality record.

‡ Host record made before the species of the "*Rana pipiens* complex" were differentiated. The geographic range of *R. pipiens* Schreber, 1782 does not extend into Mexico (Hillis et al., 1983; Frost, 1985; Flores-Villela, 1993).

casionally prey on tadpoles and insect larvae also.

Only 30 (10.5%) of the total number of species of amphibians reported in Mexico (285) (Flores-Villela, 1998) have been surveyed for helminth parasites so far. Seventy-three helminth species have been recorded. Interestingly enough, 25 of the 73 (34%) are endemic species found only in Mexico (see Baker [1987] and Lamothe et al. [1997]). However, our knowledge about helminth parasites of amphibians in Mexico is still far from complete. Parasitic organisms are becoming an important part of the body of knowledge about the natural history of their hosts, and this information can be easily used as a powerful and predictive tool to support biodiversity studies and conservation initiatives, as has been shown by Hoberg (1996, 1997). We plan to continue collecting data on the parasite fauna of amphibians in Mexico and, in this way, contribute to the understanding of their biology and their role in biodiversification and as monitors of climatic change.

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Literature Cited

Baker, M. R. 1987. Synopsis of the Nematoda parasitic in amphibians and reptiles. Memorial University of Newfoundland Occasional Papers in Biology 11:1–325.

Brandt, B. B. 1936. Parasites of certain North Carolina Salientia. Ecological Monographs 6:491–532.

Bravo-Hollis, M. 1941. Revisión de los géneros *Diplodiscus* Diesing, 1836 y *Megalodiscus* Chandler, 1923 (Trematoda: Paramphistomoidea). II. Anales del Instituto de Biología, Universidad Nacional Autónoma de México 12:643–661.

———. 1943. Estudio sistemático de los trematodos parásitos de los "ajolotes" de México I. Anales del Instituto de Biología, Universidad Nacional Autónoma de México 14:141–159.

Brenes, M. R., G. S. Arroyo, O. Jiménez-Quiroz, and E. Delgado-Flores. 1959. Algunos trematodos de *Rana pipiens*. Descripción de *Glypthelmins facioi* n. sp. Revista de Biología Tropical 72:191–197.

Brooks, R. D. 1976. Parasites of amphibians of the Great Plains. Part 2. Platyhelminths of amphibians in Nebraska. Bulletin of the University of Nebraska State Museum 10:65–92.

———. 1984. Platyhelminthes. Pages 247–258 *in* G. L. Hoff, F. L. Frye, and E. R. Jacobson, eds. Diseases of Amphibians and Reptiles. Plenum Publishing Corporation, New York.

Caballero, C. E. 1941. Trematodos de la Ciénaga de Lerma, Méx. Anales del Instituto de Biología, Universidad Nacional Autónoma de México 12: 623–641.

———. 1942a. Trematodos de las ranas de la Ciénaga de Lerma, Estado de México. II. Descripción de una nueva especie de *Haematoloechus*. Revista Brasileira de Biologia 2:155–158.

———. 1942b. Trematodos de las ranas de la Ciénaga de Lerma, Estado de México. III. Redescripción de una forma norteamericana de *Haematoloechus* y algunas consideraciones sobre *Glypthelmins californiensis* (Cort, 1919). Anales del Instituto de Biología, Universidad Nacional Autónoma de México 13:71–79.

———. 1942c. Trematodos de las ranas de la Ciénaga de Lerma, Estado de México. IV. Anales del Instituto de Biología, Universidad Nacional Autónoma de México 13:635–640.

———. 1947. Sexta contribución al conocimiento de la parasitología de *Rana montezumae*. Redescripción de *Halipegus amherstensis* Rankin, 1944. Anales del Instituto de Biología, Universidad Nacional Autónoma de México 18:473–477.

———. 1955. Helmintos de la República de Panamá XIII. Una nueva especie de *Catadiscus* Cohn, 1904 (Trematoda: Digenea). Revista Iberica de Parasitología Tomo Extra:23–26.

———, **and M. Bravo-Hollis.** 1949. Description d'un nouveau genre de Pleurogeninae (Trematoda: Lecithodendriidae) de grenouilles du Mexique *Langeronia macrocirra* n.g. n.sp. Annales de Parasitologie Humaine et Comparée 24:193–199.

Chandler, A. C. 1942. The morphology and life cycle of a new strigeid, *Fibricola texensis*, parasitic in raccoons. Transactions of the American Microscopical Society 56:156–167.

Flores-Villela, O. 1993. Herpetofauna Mexicana. Carnegie Museum of Natural History, Pittsburgh, Special Publication 17:1–73.

———. 1998. Herpetofauna de México: distribución y endemismo. Pages 251–278 *in* T. Ramamoorthy, R. Bye, A. Lot, and J. Fa, eds. Diversidad Biológica de México. Orígenes y Distribución. Instituto de Biología, Universidad Nacional Autónoma de México, México, D.F.

Frost, D. R. 1985. Amphibian Species of the World, a Taxonomic and Geographical Reference. Asso-

ciation of Systematics Collections, Lawrence, Kansas. 732 pp.

García-Altamirano, I., G. Pérez-Ponce de León, and L. García-Prieto. 1993. Contribución al conocimiento de la comunidad de helmintos de dos especies de anfibios endémicos del lago de Pátzcuaro, Michoacán: *Rana dunni* y *Ambystoma dumerilii*. Cuadernos Mexicanos de Zoología 1:73–80.

Guillén, H. S. 1992. Comunidades de helmintos de algunos anuros de "Los Tuxtlas", Veracruz. M.Sc. Thesis, Faculty of Sciences, Universidad Nacional Autónoma de México, México, D.F. 90 pp.

Hillis, D. M., and R. De Sá. 1988. Phylogeny and taxonomy of the *Rana palmipes* group (Salientia: Ranidae). Herpetological Monographs 2:1–26.

———, J. S. Frost, and D. A. Wright. 1983. Phylogeny and biogeography of the *Rana pipiens* complex: a biochemical evaluation. Systematic Zoology 32:132–143.

Hoberg, P. E. 1996. Faunal diversity among avian parasite assemblages: the interaction of history, ecology, and biogeography in marine systems. Bulletin of the Scandinavian Society of Parasitologists 6:65–89.

———. 1997. Phylogeny and historical reconstruction: host–parasite systems as keystones in biogeography and ecology. Pages 243–261 *in* M. Reaka-Kudla, D. O. Wilson, and E. O. Wilson, eds. Biodiversity II. Joseph Henry Press, Washington, D.C.

Kennedy, M. J. 1981. A revision of species of *Haematoloechus* Looss, 1899 (Trematoda: Haematoloechidae) from Canada and the United States. Canadian Journal of Zoology 59:1836–1846.

Lamothe, A. R. 1964. Tremátodos de los anfibios de México III. Redescripción de *Cephalogonimus americanus* Stafford, 1902, clave para las especies del género y registro de un nuevo hospedero. Anales del Instituto de Biología, Universidad Nacional Autónoma de México 35:115–121.

———, L. García-Prieto, D. Osorio-Sarabia, and G. Pérez-Ponce de León. 1997. Catálogo de la Colección Nacional de Helmintos. Publicación Especial. Instituto de Biología, Universidad Nacional Autónoma de México–Comisión Nacional para el Conocimiento y Uso de la Biodiversidad, México, D.F. 211 pp.

León-Règagnon, V. 1992. Fauna helmintológica de algunos vertebrados acuáticos de la Ciénaga de Lerma, Estado de México. Anales del Instituto de Biología, Universidad Nacional Autónoma de México 63:151–153.

———, D. R. Brooks, and G. Pérez-Ponce de León. 1999. Differentiation of Mexican species of *Haematoloechus* Looss, 1899 (Digenea: Plagiorchiformes) combining molecular and morphological evidence. Journal of Parasitology 85:935–946.

Martínez, V. J. M. 1969. Parásitos de algunos anfibios colectados en diferentes áreas de los municipios de Escobedo, Pesquería y Santiago, Nuevo León, México. B.Sc. Thesis, Faculty of Biological Sciences, Universidad Autónoma de Nuevo León, México. 51 pp.

McAlpine, F. D., and M. D. B. Burt. 1998. Taxonomic status of *Halipegus* spp. (Digenea: Derogenidae) parasitic in the mouth and eustachian tubes of North American and Mexican amphibians. Journal of the Helminthological Society of Washington 65:10–15.

Pérez-Ponce de León, G., L. García-Prieto, D. Osorio-Sarabia, and V. León-Règagnon. 1996. Helmintos Parásitos de Peces de Aguas Continentales de México. Serie Listados Faunísticos de México. VI. Instituto de Biología, Universidad Nacional Autónoma de México. México, D.F. 100 pp.

Pigulevsky, S. V. 1953. Family Gorgoderidae Looss, 1901 (Subfamily Phyllodistomatinae Pigulevsky, 1952 and Plesiochorinae Pigulevsky, 1952). Pages 253–615 *in* K. I. Skrjabin, ed. Trematodes of Animals and Man. Vol. VIII. Izdatel'stvo Akademii Nauk S.S.S.R., Moscow. (In Russian.)

Prokopic, J., and K. Krivanec. 1975. Helminths of amphibians, their interaction and host–parasite relationships. Acta Scientarium Naturalium Brno 9: 1–48.

Prudhoe, S., and R. A. Bray. 1982. Platyhelminth Parasites of the Amphibia. Oxford University Press, Oxford, England. 217 pp.

Pulido, F. G. 1994. Helmintos de *Rana dunni* especie endémica del Lago de Pátzcuaro, Michoacán, México. Anales del Instituto de Biología, Universidad Nacional Autónoma de México 65:205–207.

Rankin, J. S. 1944. A review of the trematode genus *Halipegus* Looss, 1899, with an account of the life history of *H. amherstensis* n. sp. Transactions of the American Microscopical Society 63:149–164.

Razo-Mendivil, U., J. P. Laclette, and G. Pérez-Ponce de León. 1999. New host and locality records for three species of *Glypthelmins* (Digenea: Macroderoididae) in anurans of Mexico. Journal of the Helminthological Society of Washington 66:197–201.

Simmons, J. E. 1985. Herpetological Collecting and Collections Management. Society for the Study of Amphibians and Reptiles Herpetological Circular 16. The University of Texas, Tyler, Texas. 57 pp.

Sokoloff, D., and E. C. Caballero. 1933. Primera contribución al conocimiento de los parasitos de *Rana montezumae*. Anales del Instituto de Biología, Universidad Nacional Autónoma de México 4:15–21.

Travassos, L. 1924. Contribuções para o conhecimento da fauna helmintológica dos batráquios do Brasil. Sciência Médica 2:746–748.

Yamaguti, S. 1975. A Synoptical Review of Life Histories of Digenetic Trematodes of Vertebrates with Special Reference to the Morphology of Their Larval Forms. Keigaku Publishing Co., Tokyo, Japan. 590 pp.

Zerecero, M. C. 1943. Algunos Tremátodos de las ratas domésticas de la Ciudad de México. Anales del Instituto de Biología, Universidad Nacional Autónoma de México 14:507–526.

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Research Note

New Host and Distribution Record of *Gordius difficilis* (Nematomorpha: Gordioidea) from a Vivid Metallic Ground Beetle, *Chlaenius prasinus* (Coleoptera: Carabidae) from Western Nebraska, U.S.A.

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ABSTRACT: *Gordius difficilis* (Montgomery, 1898) Smith, 1994 is recorded from a creek in a juniper forest in western Nebraska. Subsequent pitfall data shows *Chlaenius prasinus* Dejean, 1826 to be the definitive host. This represents the first report of *G. difficilis* from the American Midwest, of *C. prasinus* as a host of nematomorphs, and as a host for *G. difficilis.*

KEY WORDS: *Gordius difficilis, Chlaenius prasinus,* vivid metallic ground beetles, Nematomorpha, Nebraska, U.S.A.

Nematomorphs are a well-recognized, widely distributed but poorly studied phylum (Chandler, 1985). Sometimes referred to as horsehair or gordian worms, freshwater nematomorphs are obligate parasites as larvae but free-living as adults.

Gordius aquaticus difficilis Montgomery, 1898 was originally described from a single male specimen. Although Montgomery (1898) recognized 5 structural differences between *G. aquaticus difficilis* and *Gordius aquaticus robustus* Montgomery, 1898, he assigned *G. aquaticus difficilis* as a subspecies rather than a distinct species. Based on this early description, Miralles (1975) synonymized *G. aquaticus difficilis* with *G. robustus* Leidy, 1851, but Chandler (1985) synonymized *G. aquaticus difficilis* with *Gordius paraensis* Camerano, 1892.

More recently, Smith (1994) used scanning electron microscopy to show that *G. aquaticus difficilis* is distinct enough to warrant considering it as a separate species, *G. difficilis.* This determination was based on the presence of a parabolic line of hairlike structures anterior to the cloacal opening, as well as the presence of distinct areoles in the midbody of the female.

In mid-June 1998, *G. difficilis* was found in White Gate Creek, Keith County, Nebraska (41°12′20.5″N, 101°39′86.3″W). This site consists of a first-order, spring-fed creek surrounded by juniper trees (*Juniperus scopulorum* Sargent, 1897) and various deciduous vegetation. The creek has a sandy bottom and often contains algal blooms because of the use of the creek by cattle. Nineteen free-living individuals were collected from the creek between late June and late July, 10 males ranging in size from 68–307 mm and 9 females ranging in size from 89–208 mm. Individuals were often found entangled in the algae or attached to sticks or rocks on the banks of the creek.

In late June 1998, 4 lines of 10 pitfall traps were set adjacent to White Gate Creek. Of 6 trapped *Chlaenius prasinus* Dejean, 1826, 2 were infected with 3 worms each. None of the other invertebrates trapped contained nematomorphs. One host contained 2 female worms and 1 male worm; the other host contained 3 female worms. The males ranged in size from 103–297 mm, and the females ranged in size from 185–203 mm. The hosts were void of gonads, fatbodies, and intestines but appeared to behave normally.

Worms were killed in 70% EtOH and brought up to 100% glycerine prior to examination. All specimens were temporarily mounted in glycerine for observation. Worms were as described by Montgomery (1898) and Smith (1994). Briefly, the male posterior is bifurcated with a subterminal ventral cloacal opening (Fig. 1). A line of hairlike structures curves around the anterior end of the cloacal opening. Posterior to the cloaca is a postcloacal crescent, extending about one-fourth the length of the lobed ends. Females have entire posteriors. Cuticular areoles are more prominent in females compared with males when viewed with light microscopy.

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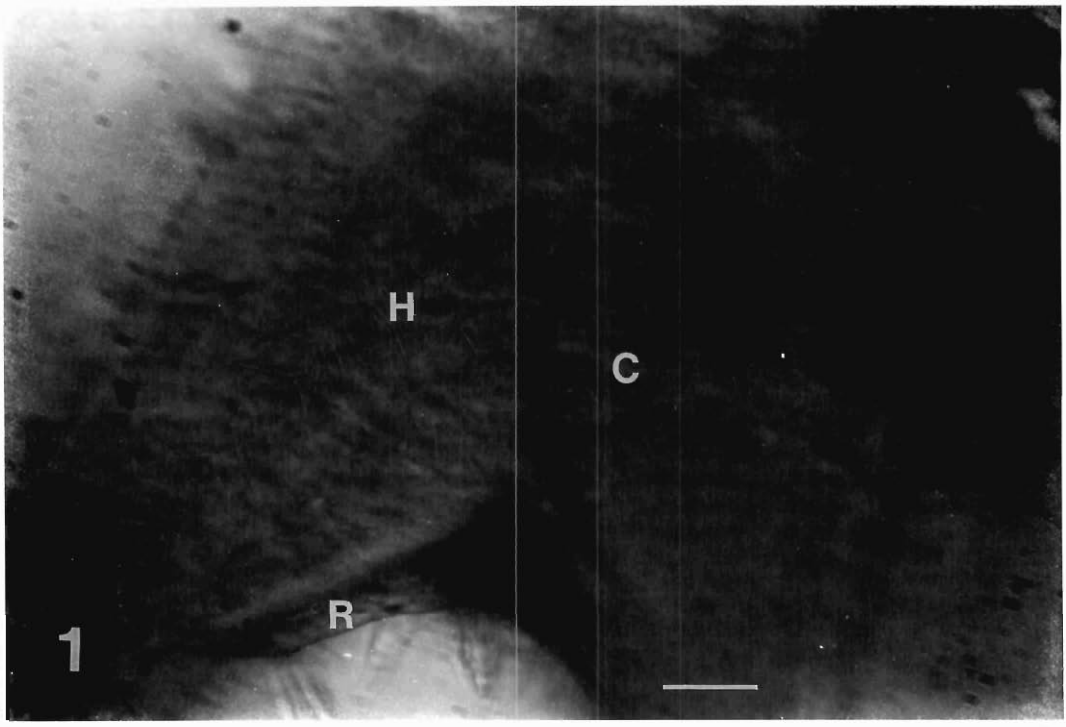

Figure 1. ***Gordius difficilis,*** **posterior end of a male. Scale bar = 35 μm. Note cloacal opening (C), precloacal line of hairlike structure (H), and postcloacal ridge (R).**

Gordians have been recorded from one individual of *Chlaenius sericeus* Forster, 1771, but the worm could be identified only as *Gordius* sp. because of the immaturity of the specimen (Leffler, 1984). The only other record of the genus *Chlaenius* as a host for a "nematoid parasite" was from *Chlaenius tomentosus* Say, 1830. In that report, insect parts and the worm were located in the stomach of the beetle (Forbes, 1880). However, nematomorphs are usually found outside the host's gut. Thus, it is likely that the worm was ingested while inside another insect and was not a parasite of the beetle.

Gordius difficilis has only been reported from Roan Mountain, western North Carolina (Montgomery, 1898) and from Franklin County, Massachusetts (Smith, 1994). This report extends the known range of *G. difficilis* and for the first time provides information of a host for this species.

We would like to thank Myrna Gainsforth for providing access to White Gate Creek and the Cedar Point Biological Station for providing facilities. This project was partially funded by the Center for Great Plains Studies Research grants-in-aid for graduate students (University of Nebraska–Lincoln, fall 1998).

Literature Cited

Chandler, C. M. 1985. Horsehair worms (Nematomorpha, Gordioidea) from Tennessee, with a review of taxonomy and distribution in the United States. Journal of the Tennessee Academy of Sciences 60:59–62.

Forbes, S. A. 1880. Notes on insectivorous Coleoptera. Bulletin of the Illinois Laboratory of Natural History 1:167–176.

Leffler, S. R. 1984. Record of a horsehair worm (Nematomorpha: Gordiidae) parasitizing *Chlaenius sericeus* Forster (Coleoptera: Carabidae) in Washington. Coleopterists Bulletin 38:130.

Miralles, D. A. B. 1975. Nuevo aporte al conocimiento de la Gordiofauna Argentina. Neotropica 21: 99–103.

Montgomery, T. H. J. 1898. The Gordiacea of certain American collections with particular reference to the North American fauna. Bulletin of the Museum of Comparative Zoology, Harvard 32:23–59.

Smith, D. G. 1994. A reevaluation of *Gordius aquaticus difficilis* Montgomery 1898 (Nematomorpha, Gordioidea, Gordiidae). Proceedings of the Academy of Natural Sciences of Philadelphia 145:29–34.

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Research Note

Intestinal Helminths of Seven Species of Agamid Lizards from Australia

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ABSTRACT: The intestinal tracts of 243 lizards representing 7 species of Agamidae from Australia (*Ctenophorus caudicinctus, Ctenophorus fordi, Ctenophorus isolepis, Ctenophorus reticulatus, Ctenophorus scutulatus, Lophognathus longirostris,* and *Pogona minor*) were examined for helminths. One cestode species, *Oochoristica piankai,* and 8 nematode species, *Abbreviata anomala, Kreisiella chrysocampa, Kreisiella lesueurii, Maxvachonia brygooi, Parapharyngodon fitzroyi, Skrjabinoptera gallardi, Skrjabinoptera goldmanae,* and *Wanaristrongylus ctenoti,* were found. Larvae of *Abbreviata* sp. were also present. Twelve new host records are reported.

KEY WORDS: Sauria, lizards, Agamidae, survey, Cestoda, *Oochoristica piankai,* Nematoda, *Abbreviata anomala, Kreisiella chrysocampa, Kreisiella lesueurii, Maxvachonia brygooi, Parapharyngodon fitzroyi, Skrjabinoptera gallardi, Skrjabinoptera goldmanae, Wanaristrongylus ctenoti, Abbreviata* sp., Australia.

The family Agamidae is well represented in Australia and about 60 species are known (Cogger, 1992). Helminth records exist for 17 species (Baker, 1987; Jones, 1995a; Bursey et al., 1996). The purpose of this paper is to present the initial report of helminths harbored by *Ctenophorus caudicinctus* (Günther, 1875) (the ring-tailed dragon), *Ctenophorus fordi* (Storr, 1965) (the mallee dragon), and *Ctenophorus scutulatus* (Stirling and Zietz, 1893) (the lozenge-marked dragon), and additional helminth data for 4 previously examined species: *Ctenophorus isolepis* (Fischer, 1881) (the military dragon), *Ctenophorus reticulatus* (Gray, 1845) (the western netted dragon), *Lophognathus longirostris* Boulenger, 1883 (the Australian water dragon), and *Pogona minor* (Sternfeld, 1919) (the dwarf bearded dragon). In addition, patterns of infection for helminths of Australian agamids, were examined.

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The 7 species examined in this study range through much of Australia but overlap in Western Australia (Table 1). *Ctenophorus caudicinctus* is known from western Queensland through the Northern Territory and northern South Australia to most of Western Australia; *C. fordi* is widely distributed through southeastern Western Australia and southern South Australia with outlying populations in western Victoria and western New South Wales; *C. isolepis* is found in Western Australia, Northern Territory, northern South Australia, and western Queensland; *C. reticulatus* occurs throughout most of the southern half of Western Australia and northern South Australia; *C. scutulatus* is known from southern Western Australia and northwestern South Australia; *L. longirostris* occurs from the coast of Western Australia through central Australia to western Queensland; *P. minor* ranges from the central coast of Western Australia through central Australia and South Australia (Cogger, 1992).

Two hundred forty-three lizards were borrowed from the herpetology collection of the Natural History Museum of Los Angeles County (LACM), Los Angeles, California, U.S.A., and examined for intestinal helminths. These specimens had been collected in 1966–1968 for a series of ecological studies by Eric R. Pianka (The University of Texas at Austin, U.S.A.). The stomach of each lizard had been removed, examined for food contents, and deposited in the Western Australian Museum, Perth, Western Australia; the carcasses with livers and intact intestines were deposited in LACM. Numbers of individuals, mean snout-vent length (SVL), year of collection, and museum accession number of host species are as follows: *Ctenophorus caudicinctus* (N = 25, SVL = 63 mm ± 4 SD, range = 55–71 mm), Collected 1968, Western Austra-

Table 1. Prevalence (%), mean intensity ± SD (x ± SD), and range (r) for intestinal helminths from Australian agamid lizards.

Helminth	Host								
	Ctenophorus caudicinctus			*Ctenophorus fordi*			*Ctenophorus isolepis*		
	%	x ± SD	r	%	x ± SD	r	%	x ± SD	r
Cestoda									
Oochoristica piankai	—	—	—	4*	1.0	—	18*	2.3 ± 1.6	1–8
Nematoda									
Abbreviata anomala	—	—	—	—	—	—	—	—	—
Kreisiella chrysocampa	—	—	—	—	—	—	4	2.0 ± 1.4	1–4
Kreisiella lesueurii	—	—	—	23*	2.2 ± 1.2	1–4	—	—	—
Maxvachonia brygooi	—	—	—	—	—	—	2*	1.0	—
Parapharyngodon fitzroyi	—	—	—	—	—	—	—	—	—
Skrjabinoptera gallardi	20*	1.0	—	—	—	—	—	—	—
Skrjabinoptera goldmanae	—	—	—	—	—	—	—	—	—
Wanaristrongylus ctenoti	—	—	—	—	—	—	1	2.0	—
Abbreviata sp. (larvae)	—	—	—	—	—	—	3*	1.0	—

lia. LACM 55115, 55117–55119, 55123–55125, 55127, 55128, 55130–55133, 55139, 55141–55143, 55145, 55152, 55154, 55156, 55163, 55164, 55166, 55167; *Ctenophorus fordi* (N = 26, SVL = 50 mm ± 3 SD, range = 46–58 mm). Collected 1967, Western Australia. LACM 59240, 59245, 59246, 59251, 59259, 59262, 59268, 59271, 59272, 59274, 59275, 59279, 59290, 59296, 59299, 59300, 59304, 59306–59308, 59312, 59316, 59319, 59321–59322, 59324; *Ctenophorus isolepis* (N = 127, SVL = 55 mm ± 5 SD, range = 35–67 mm). Collected 1967–1968, Western Australia. LACM 54575–54599, 54650–54676, 54678–54699, 54775–54799, 54825–54848, 54850–54853. Collected 1966–1967, Northern Territory. LACM 54694–54699; *Ctenophorus reticulatus* (N = 5, SVL = 76 mm ± 5 SD, range = 72–83 mm). Collected 1967, Western Australia. LACM 55051, 55054, 55055, 55062, 55063; *Ctenophorus scutulatus* (N = 25, SVL = 87 mm ± 11 SD, range = 71–107 mm). Collected 1966–1967, Western Australia. LACM 54933–54936, 54940, 54942, 54946, 54949, 54952, 54956–54958, 54960, 54962, 54963, 54970, 54975, 54982, 54993, 54996, 54998, 54999, 55004, 55005, 55012; *Lophognathus longirostris* (N = 10, SVL = 74 mm ± 19 SD, range = 48–98 mm). Collected 1966–1968, Western Australia. LACM 55334, 55335, 55342, 55345, 55354, 55355, 55357, 55366, 55373, 55377; *Pogona minor* (N = 25, SVL = 111 mm ± 11 SD, range = 88–129 mm). Collected 1967, Western Australia. LACM 54854–54857, 54859, 54862, 54864–54866, 54868, 54869, 54872, 54873, 54875–54880, 54882, 54884, 54890, 54892, 54896, 54899.

The intestines, body cavity, and liver of each lizard were examined for adult helminths and helminth larvae (such as cystacanths, pleurocercoids, and tetrathyridia) using a dissecting microscope. Stomachs from these specimens were unavailable for our examination; however, Jones (1987) reported helminths from stomachs of *C. isolepis, L. longirostris,* and *P. minor* from the Pianka Collection in the Western Australian Museum, which are listed in Table 2. Helminths were placed on a glass slide in a drop of undiluted glycerol for study under a compound microscope. Nematodes were identified from these preparations; selected cestodes were stained with hematoxylin and mounted in balsam for identification. Nematodes in vials of 70% ethanol, and permanent stained mounts of cestodes were deposited in the United States National Parasite Collection (USNPC), Beltsville, Maryland.

One species of Cestoda, *Oochoristica piankai* Bursey, Goldberg, and Woolery, 1996 (USNPC 88548, 88550, 88555) and 8 species of Nematoda, *Abbreviata anomala* Jones, 1986 (USNPC 88559), *Kreisiella chrysocampa* Jones, 1985 (USNPC 88551, 88560), *Kreisiella lesueurii* Jones, 1986 (USNPC 88549, 88561), *Maxvachonia brygooi* Mawson, 1972 (USNPC 88552, 88556, 88562), *Parapharyngodon fitzroyi* Jones, 1992 (USNPC 88563), *Skrjabinoptera goldmanae* Mawson, 1970 (USNPC 88557, 88564), *Skrjabinoptera gallardi* (Johnston and Mawson,

Table 1. Extended.

Host											
Ctenophorus reticulatus			*Ctenophorus scutulatus*			*Lophognathus longirostris*			*Pogona minor*		
%	x ± SD	r	%	x ± SD	r	%	x ± SD	r	%	x ± SD	r
60*	7.0 ± 3.5	5–11	—	—	—	—	—	—	—	—	—
—	—	—	—	—	—	10*	1.0	—	—	—	—
—	—	—	—	—	—	10	1.0	—	—	—	—
—	—	—	—	—	—	—	—	—	12	2.0 ± 1.0	1–3
—	—	—	48*	4.1 ± 4.1	1–13	—	—	—	28	6.7 ± 5.6	1–13
—	—	—	—	—	—	—	—	—	20*	1.2 ± 0.5	1–2
—	—	—	—	—	—	—	—	—	—	—	—
—	—	—	28*	1.1 ± 0.4	1–2	—	—	—	12	1.0	—
—	—	—	—	—	—	—	—	—	—	—	—
—	—	—	4*	1.0	—	—	—	—	4	1.0	—

* New host record.

1942) (USNPC 88547), and *Wanaristrongylus ctenoti* Jones, 1987 (USNPC 88553), were found. Larvae of *Abbreviata* sp. (USNPC 88554, 88558, 88565) were also present. All of these helminths were found in the lumen of the intestines, with the exception of larval *Abbreviata* sp. which were found in cysts in the intestinal wall. No cystacanths, pleurocercoids, or tetrathyridia were found in the body cavity or attached to the viscera.

Prevalences, mean intensity ± SD, and range are presented in Table 1. None of the helminths found in this study is host specific. Recorded helminths of agamid lizards from Australia are listed in Table 2. Of these, *Pseudothamugadia physignathi* Lopez-Neyra, 1956, *Oswaldofilaria innisfailensis* (Mackerras, 1962), *Oswaldofilaria pflugfelderi* (Frank, 1964) and *Oswaldofilaria samfordensis* Manzanell, 1982, all filarioids, have been found to infect a single host species and, surprisingly, the same host species, *Physignathus lesueurii* (Gray, 1831) (the eastern water dragon). *Abbreviata anomala, Abbreviata pilbarensis* Jones, 1986, *Oswaldofilaria chlamydosauri* (Breinl, 1913), *S. gallardi, Strongyluris paronai* (Stossich, 1902), and *Wanaristrongylus pogonae* Jones, 1987 are known only from agamid hosts. The remaining helminths have been reported from agamids as well as other lizard families: *O. piankai* from Gekkonidae; *Abbreviata antarctica* (Linstow, 1899), Scincidae, Varanidae; *Abbreviata confusa* (Johnston and Mawson, 1942), Varanidae, as well as several snake species; *Abbreviata tumidocapitis* Jones, 1983, Gekkonidae, Varanidae; *K. chrysocampa,* Scincidae; *K. lesueurii,* Scincidae; *M. brygooi,* Scincidae, Varanidae; *P. fitzroyi,* Scincidae; *Parapharyngodon kartana* (Johnston and Mawson, 1941), Gekkonidae, Scincidae; *Physalopteroides filicauda* Jones, 1985, Gekkonidae, Pygopodidae, Scincidae, Varanidae; *Pseudorictularia dipsarilis* (Irwin-Smith, 1922), Scincidae; *S. goldmanae,* Gekkonidae, Scincidae, Varanidae; *W. ctenoti,* Gekkonidae, Scincidae, Varanidae.

Physalopterid larvae are widely distributed in Australia and have been reported from agamid, gekkonid, scincid, and varanid lizards as well as several species of snakes (Jones, 1995b). This is the first report of larvae of *Abbreviata* sp. from *C. isolepis* and *C. scutulatus;* however, Jones (1995b) reported physalopteran larvae from *C. isolepis.* Currently, species of *Physaloptera* are not known to occur in Australian reptiles. *Physaloptera gallardi* Johnston and Mawson, 1942, from *Pogona barbata* (Cuvier, 1829) (the bearded dragon) was reassigned to *Skrjabinoptera* by Chabaud (1956), and *Physaloptera bancrofti* Irwin-Smith, 1922 from *Phyllurus platurus* (White, 1790) (the southern leaf-tailed gecko) was reassigned to *Abbreviata* by Schulz (1927). Physalopterid larvae found in Australian lizards are most likely species of either *Abbreviata* or *Skrjabinoptera.*

The data presented here suggest that Australian agamid lizards are infected by helminth generalists. Bush et al. (1997) presented a hier-

Table 2. Helminths of agamid lizards from Australia.

Lizard species	Helminth species								
	Oochoristica piankai	*Abbreviata anomala*	*Abbreviata antarctica*	*Abbreviata confusa*	*Abbreviata pilbarensis*	*Abbreviata tumidocapitis*	*Kreisiella chrysocampa*	*Kreisiella lesueurii*	*Maxvachonia brygooi*
Amphibolurus muricatus (White, 1790)	—	—	X†	—	—	—	—	—	—
Chlamydosaurus kingii Gray, 1825	—	X‡	—	X‡	—	—	—	—	X‡
Ctenophorus caudicinctus (Günther, 1875)	—	—	—	—	—	—	—	—	—
Ctenophorus decresii (Duméril and Bibron, 1837)	—	—	—	—	—	—	—	—	X‖
Ctenophorus fionni (Proctor, 1923)	—	—	—	—	—	—	—	—	—
Ctenophorus fordi (Storr, 1965)	X§	—	—	—	—	—	—	X§	—
Ctenophorus isolepis (Fischer, 1881)	X§	—	—	—	—	X#	X§	—	X§
Ctenophorus maculatus (Gray, 1831)	—	—	—	—	—	—	—	—	X‖
Ctenophorus nuchalis (de Vis, 1884)	—	—	—	—	—	—	—	—	—
Ctenophorus reticulatus (Gray, 1845)	X§	—	—	—	—	—	—	—	—
Ctenophorus scutulatus (Stirling and Zietz, 1893)	—	—	—	—	—	—	—	—	X§
Lophognathus longirostris Boulenger, 1883	—	X§	—	—	—	—	X§	—	—
Moloch horridus Gray, 1841	X**	—	—	—	—	—	—	—	—
Physignathus lesueurii (Gray, 1831)	—	—	—	—	—	—	—	—	—
Pogona barbata (Cuvier, 1829)	—	—	—	—	—	—	—	—	X‖
Pogona microlepidota (Glauert, 1952)	—	—	X§§	—	—	—	—	—	X§§
Pogona minima (Loveridge, 1933)	—	—	—	—	—	—	—	X§§	—
Pogona minor (Sternfeld, 1919)	—	X§§	X§§	—	X§§	—	—	X§	X§
Pogona mitchelli (Badham, 1976)	—	X§§	—	—	X§§	—	—	X§§	X§§
Pogona nullarbor (Badham, 1976)	—	—	X§§	—	—	—	—	X§§	—

* Johnston and Mawson, 1942.
† Johnston and Mawson, 1943.
‡ Jones, 1994.
§ This paper.
‖ Mawson, 1972.
¶ Mawson, 1971.
# Jones, 1995a.
** Bursey et al., 1996.
†† Mackerras, 1962.
‡‡ Manzanell, 1982.
§§ Jones, 1986.
‖‖ Jones, 1987.

Table 2. Extended.

Helminth species													
Oswaldofilaria chlamydosauri	*Oswaldofilaria innisfailensis*	*Oswaldofilaria pflugfelderi*	*Oswaldofilaria samfordensis*	*Parapharyngodon fitzroyi*	*Parapharyngodon kartana*	*Physaloptéroides filicauda*	*Pseudoricturlaria dipsarilis*	*Pseudothamagudia physignathi*	*Skrjabinoptera gallardi*	*Skrjabinoptera goldmanae*	*Strongyluris paronai*	*Wanaris strongylus crenoti*	*Wanaris strongylus pogonae*
X†	—	—	—	—	—	—	—	—	X†	—	X†	—	—
X†	—	—	—	—	—	X‡	—	—	—	X‡	X‡	—	—
—	—	—	—	—	—	—	—	—	X§	—	—	—	—
—	—	—	—	—	—	—	—	—	—	—	—	—	—
—	—	—	—	—	X¶	—	—	—	—	—	—	—	—
—	—	—	—	—	—	—	—	—	—	—	—	—	—
—	—	—	—	—	—	X#	—	—	—	X#	—	X§	—
—	—	—	—	—	—	—	—	—	—	—	—	—	—
—	—	—	—	—	—	X#	—	—	—	X#	—	—	—
—	—	—	—	—	—	—	—	—	—	—	—	—	—
—	—	—	—	—	—	—	—	—	—	X§	—	—	—
—	—	—	—	—	—	X#	—	—	—	—	—	—	—
—	—	—	—	—	—	—	—	—	—	—	—	—	—
—	X††	X‡‡	X‡‡	—	—	—	—	X†	—	—	—	—	—
X†	—	—	—	—	—	—	—	—	X*	—	X†	—	—
—	—	—	—	—	—	X§§	X§§	—	—	—	X§§	—	—
—	—	—	—	—	—	—	—	—	—	—	X§§	—	—
—	—	—	—	X§	—	X§§	—	—	—	X§	X§§	—	—
—	—	—	—	—	—	X§§	X§§	—	—	X§§	X§§	—	X‖‖
—	—	—	—	—	—	X§§	—	—	—	—	—	—	—

archy of parasite community terms, including infracommunity (helminths in a single host), component community (helminths of a host species), and supracommunity (helminths in sympatric hosts). Table 2 represents the contribution of agamid lizards to the Australian helminth supracommunity. Mean number of helminth species harbored per host species was 3.50 ± 0.58 SE (range, 1–9). Aho (1990) compiled distributional patterns for lizards in general and reported the mean total number of helminth species per host species to be 2.06 ± 0.13 SE (range, 1–4). Whether this greater number of helminth species per host species is a local Australian phenomenon or the result of insufficient data on the helminths of this diverse fauna must await further studies of Australian lizard helminths.

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Literature Cited

Aho, J. M. 1990. Helminth communities of amphibians and reptiles: comparative approaches to understanding patterns and processes. Pages 157–195 *in* G. W. Esch, A. O. Bush, and J. M. Aho, eds. Parasite Communities: Patterns and Processes. Chapman and Hall, London.

Baker, M. R. 1987. Synopsis of the Nematoda parasitic in amphibians and reptiles. Occasional Papers in Biology, Memorial University of Newfoundland 11:1–325.

Bursey, C. R., S. R. Goldberg, and D. N. Woolery. 1996. *Oochoristica piankai* sp. n. (Cestoda: Linstowiidae) and other helminths of *Moloch horridus* (Sauria: Agamidae) from Australia. Journal of the Helminthological Society of Washington 63: 215–221.

Bush, A.O., K. D. Lafferty, J. M. Lotz, and A. W. Shostak. 1997. Parasitology meets ecology on its own terms: Margolis et al. revisited. Journal of Parasitology 83:575–583.

Chabaud, A. G. 1956. Essai de révision des physaloptères parasites de reptiles. Annales de Parasitologie Humaine et Comparée 31:29–52.

Cogger, H. G. 1992. Reptiles and Amphibians of Australia, 5th ed., Reed Books, Chatswood, New South Wales, Australia. 775 pp.

Johnston, T. H., and P. M. Mawson. 1942. The Gallard collection of parasitic nematodes in the Australian Museum. Records of the Australian Museum 21:110–115.

———, **and** ———. 1943. Remarks on some nematodes from Australian reptiles. Transactions of the Royal Society of South Australia 67:183–186.

Jones, H. I. 1986. Gastrointestinal nematodes in the lizard genus *Pogona* Storr (Agamidae) in Western Australia. Australian Journal of Zoology 34:689–705.

———. 1987. *Wanaristrongylus* gen. n. (Nematoda: Trichostrongyloidea) from Australian lizards, with descriptions of three new species. Proceedings of the Helminthological Society of Washington 54: 40–47.

———. 1994. Gastrointestinal nematodes of the frill-neck lizard, *Chlamydosaurus kingii* (Agamidae), with particular reference to *Skrjabinoptera goldmanae* (Spirurida: Physalopteridae). Australian Journal of Zoology 42:371–377.

———. 1995a. Gastric nematode communities in lizards from the Great Victoria Desert, and an hypothesis for their evolution. Australian Journal of Zoology 43:141–164.

———. 1995b. Pathology associated with physalopterid larvae (Nematoda: Spirurida) in the gastric tissues of Australian reptiles. Journal of Wildlife Diseases 31:299–306.

Mackerras, M. J. 1962. Filarial parasites (Nematoda: Filarioidea) of Australian animals. Australian Journal of Zoology 10:400–457.

Manzanell, R. 1982. *Oswaldofilaria* spp. (Filarioidea, Nematoda) in Australian agamid lizards with a description of a new species and a redescription of *O. chlamydosauri* (Breinl). Annales de Parasitologie Humaine et Comparée 57:127–143.

Mawson, P. M. 1971. Pearson Island Expedition 1969.—8. Helminths. Transactions of the Royal Society of South Australia 95:169–183.

———. 1972. The nematode genus *Maxvachonia* (Oxyurata: Cosmocercidae) in Australian reptiles and frogs. Transactions of the Royal Society of South Australia 96:101–108.

Schulz, R. E. 1927. Die Familie Physalopteridae Leiper, 1908 (Nematodes) und die Prinzipien ihrer Klassifikation. Pages 287–312 *in* Sammlung Helmintologischer Arbeiten Prof. K. I. Skrjabin Gewidmet. Moscow.

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Research Note

Descriptions of Cystacanths of *Mediorhynchus orientalis* and *Mediorhynchus wardi* (Acanthocephala: Gigantorhynchidae)

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ABSTRACT: Cystacanths of *Mediorhynchus orientalis* Belopol'skaya and 1 cystacanth of *Mediorhynchus wardi* Schmidt and Canaris were collected from opportunistically infected Surinam cockroaches, *Pycnoscelis surinamensis* (Linnaeus), at the National Aviary in Pittsburgh, Pennsylvania, U.S.A. Morphological measurements and descriptions of cystacanths of *M. orientalis* and *M. wardi* are provided for the first time.

KEY WORDS: *Mediorhynchus orientalis, Mediorhynchus wardi,* Acanthocephala, Gigantorhynchidae, cystacanths, Surinam cockroach, aviary, description, Pennsylvania, U.S.A.

Mediorhynchus orientalis Belopol'skaya, 1953 was originally described from juvenile specimens collected from a little ringed plover, *Charadrius dubius curonicus* Gmelin, 1789, in Russia. Schmidt and Kuntz (1977) subsequently redescribed the species from numerous adults and juveniles collected from 10 species of passeriform birds and a Pacific golden plover, *Pluvialis fulva* (Gmelin, 1789); (as *Charadrius dominicus fulvus*), from Taiwan, Borneo, and Hawaii. *Mediorhynchus wardi* Schmidt and Canaris, 1967 was described from numerous adult specimens collected from 4 species of passeriform birds in Njoro, Kenya.

Forty-five species of *Mediorhynchus* were recorded as valid in Amin's (1985) list of Acanthocephala. Two additional species not included in Amin's list were apparently described during its preparation (George and Nadakal, 1984). Five of these have the cystacanths described. Cystacanths of *Mediorhynchus petrochenkoi* Gvosdev and Soboleva, 1966, were described by Lisitsina and Tkach (1994); of *Mediorhynchus centurorum* Nickol, 1969 by Nickol (1977); and of *Mediorhynchus grandis* Van Cleave, 1916 by Moore (1962). Brief descriptions of the cystacanth stage of *Mediorhynchus papillosus* Van Cleave, 1916, were provided by Ivashkin and Shmitova (1969) and Gafurov (1975), and of *Mediorhynchus micracanthus* (Rudolphi, 1819) Meyer, 1933 by Rizhikov and Dizer (1954).

Cystacanths of *Mediorhynchus orientalis* were reported from opportunistically infected Surinam cockroaches, *Pycnoscelis surinamensis* (Linnaeus, 1758) (Blaberidae) at the National Aviary in Pittsburgh, Pennsylvania, U.S.A. (Bolette, 1990). These cockroaches occurred within a free-flight exhibit that housed a variety of birds originating from various geographical localities. This method of housing most likely contributed to the accidental introduction of *M. orientalis* into the enclosure. The following description of *M. orientalis* cystacanths is based on 9 everted specimens, 1 male and 8 females, collected from the coelomic cavities of infected *P. surinamensis* from the previous report (Bolette, 1990). Additionally, while reexamining the cystacanths previously recovered from Surinam cockroaches (Bolette, 1990), 1 specimen was determined to be *M. wardi.* The following description of *M. wardi* is based on this single everted female cystacanth. This report represents the first description of *M. orientalis* and *M. wardi* cystacanths.

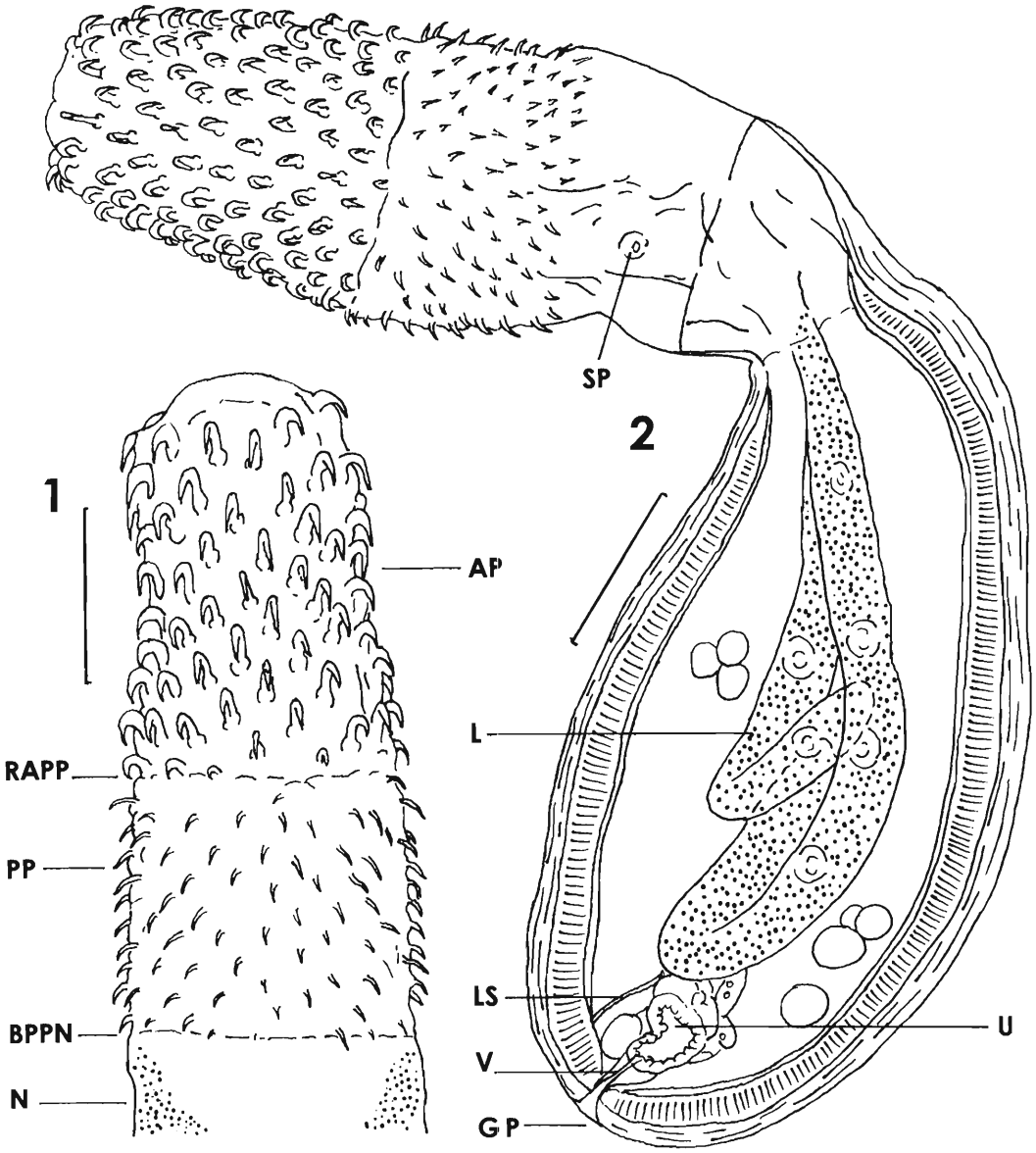
The cockroaches were killed with ethyl acetate. Cystacanths were mechanically excysted, placed in refrigerated tap water to elicit proboscis evagination, killed and preserved in AFA fixative, and later transferred to 70% ethyl alcohol. Selected specimens were stained in borax-carmine, dehydrated in ascending concentrations of ethyl alcohol, cleared in ascending concentrations of xylene, and mounted in Permount™ (Fisher Scientific, Fairlawn, New Jersey, U.S.A.). Voucher specimens were deposited in the United States National Parasite Collection, Beltsville, Maryland (USNPC No. 88032). Measurements are in μm unless stated otherwise, with means in parentheses. Trunk measurements do not include the neck. Hook and spine measurements were determined in complete profile.

Mediorhynchus orientalis Belopol'skaya, 1953

GENERAL (CYSTACANTH): Trunk short, oblong, slightly tapered at distal end. Proboscis truncate, conical (Fig. 1). Proboscis armature similar in both sexes. 19–24 (usually 20–21) nearly longitudinal rows of 4–6 (usually 5) hooks each. Anterior 2–3 hooks 37.5–50.0 (47.3); middle 2–3 hooks 37.5–45.0 (40.7); posterior 1–2 hooks 20.0–40.0 (29.2). Rootless spines arranged in 34–38 rows of 3–6 (usually 4–5) spines each; 27.5–45.0 (36.75) long. Lemnisci long, slender, usually folded, and partly retained in neck region and anterior part of trunk.

FEMALE CYSTACANTHS (based on 8 specimens): Trunk 1.49–1.89 (1.66) mm long, 0.48–0.73 (0.58) mm wide at widest point. Proboscis 637–726 (695) long. Anterior proboscis 398–458 (428) long, 308–338 (318) wide at base. Posterior proboscis 199–348 (272) long, 289–398 (350) wide at base. Neck 131–192 (158) long, 364–455 (391) wide at base. Sensory pit 27.5–30.0 (24.0) long, 27.5–45.0 (35.0) wide, located 22.5–140 (87.0) distal to posteriormost spine.

MALE CYSTACANTH (1 specimen): Trunk 1.59 mm long, 0.62 mm wide at widest point. Proboscis 662 long. Anterior proboscis 384 long, 283 wide at base. Posterior proboscis 278 long, 318 wide at base. Neck 167 long, 333 wide at base. Sensory pit 22.5 long, 30.0 wide, located 17.5 distal to posteriormost spine.



Figures 1, 2. Everted cystacanths of *Mediorhynchus* recovered from Surinam cockroaches, *Pycnoscelis surinamensis*. 1. Proboscis of *M. orientalis*; bar = 200 μm. AP, anterior proboscis; BPPN, borderline between posterior proboscis and neck; N, neck; PP, posterior proboscis; RAPP, ridge between anterior and posterior proboscis. 2. Female *M. wardi*; bar = 250 μm. GP, gonopore; L, lemniscus; LS, ligament strand; SP, sensory pit, U, uterus; V, vagina.

Mediorhynchus wardi Schmidt and Canaris, 1967

FEMALE CYSTACANTH (1 specimen): Trunk short, subglobose, rounded at distal end (Fig. 2), 1.89 mm long, 0.63 mm wide at widest point. Proboscis truncate, conical, 718 long. Anterior proboscis 468 long, 402 wide at base. Posterior proboscis 250 long, 409 wide at base. 25 nearly longitudinal rows of 7–8 hooks each. Anterior 2–3 hooks 32.5–37.5 (36.3); middle 2–3 hooks 27.5–30.0 (29.6); posterior 2 hooks 22.5–30.0 (25.6). Rootless spines arranged in 40 rows of

4–5 spines each: 25.0–35.0 (31.4). Neck 202 long, 343 wide at base. Sensory pit 25.0 long, 32.5 wide, located 125 distal to most posterior spine. Lemnisci long, slender, partly folded, extended far into trunk.

The proboscis armature arrangement of *M. orientalis* cystacanths is identical to that of adults of this species as redescribed by Schmidt and Kuntz (1977). However, the proboscis armature and neck measurements of the cystacanths examined differed from those of adults in the following respects. The lengths of the middle 2–3 and posterior 1–2 hooks of cystacanths were 37.5–45.0 and 20.0–40.0, respectively, while those listed for adults were 34–42 and 30–44 (Schmidt and Kuntz, 1977). The neck length and width of female cystacanths and the neck width of the male measured 131–192 by 364–455 and 333, respectively; the measurements of adult females and males were reported as 216–240 by 530–600 and 500–535, respectively. The proboscis of the male cystacanth was slightly longer at 662, while the proboscides of adult males were 500–600 long.

The proboscis hook and spine length measurements of the cystacanth of *M. wardi* differed slightly from those given for adults of this species by Schmidt and Canaris (1967). Cystacanth hook and spine length measurements ranged from 22.5–37.5 and 25.0–35.0, respectively, while those listed for adult *M. wardi* were 31.0–36.0 and 21.0–28.0. The proboscis width of the cystacanth was slightly narrower than in adults; anterior and posterior proboscis width for the cystacanth measured 402 and 409, respectively, while the corresponding measurements reported for adults were 425 and 515–545. Neck length of the cystacanth was slightly longer than adults, measuring 202, while the reported adult neck length was 165. Additionally, the cystacanth did not show any evidence of an anterior trunk swelling, as described for adults. However, because the armature arrangement and all other proboscis measurements are consistent with those described by Schmidt and Canaris (1967) for adult *M. wardi*, the single female specimen was assigned to this species.

Literature Cited

Amin, O. M. 1985. Classification. Pages 27–72 *in* D. W. T. Crompton and B. B. Nickol, eds. Biology of the Acanthocephala. Cambridge University Press, Cambridge, United Kingdom.

Bolette, D. P. 1990. Intermediate host of *Mediorhynchus orientalis* (Acanthocephala: Gigantorhynchidae). Journal of Parasitology 76:575–577.

Gafurov, A. K. 1975. New intermediate hosts of the acanthocephalan *Mediorhynchus papillosus* Van Cleave, 1916. Zoologicheskii Sbornik 2:103–104. (In Russian).

George, P. V., and A. M. Nadakal. 1984. Three new species of Acanthocephala (Gigantorhynchidea) from birds of Kerala. Acta Parasitologica Polonica 29:97–106.

Ivashkin, V. M., and G. Y. Shmitova. 1969. The life cycle of *Mediorhynchus papillosus*. Trudy Gel'mintologicheskoi Laboratorii 20:62–63.

Lisitsina, O. I., and V. V. Tkach. 1994. Morphology of cystacanths of some acanthocephalans from aquatic and terrestrial intermediate hosts in the Ukraine. Helminthologia 31:83–90.

Moore, D. V. 1962. Morphology, life history, and development of the acanthocephalan *Mediorhynchus grandis* Van Cleave, 1916. Journal of Parasitology 48:76–86.

Nickol, B. B. 1977. Life history and host specificity of *Mediorhynchus centurorum* Nickol 1969 (Acanthocephala: Gigantorhynchidae). Journal of Parasitology 63:104–111.

Rizhikov, K. M., and Y. B. Dizer. 1954. Biology of *Macracanthorhynchus catulinus* and *Mediorhynchus micracanthus*. Doklady Akademii Nauk SSSR 95:1367–1369.

Schmidt, G. D., and A. G. Canaris. 1967. Acanthocephala from Kenya with descriptions of two new species. Journal of Parasitology 53:634–637.

———, **and R. E. Kuntz.** 1977. Revision of *Mediorhynchus* Van Cleave 1916 (Acanthocephala) with a key to species. Journal of Parasitology 63:500–507.

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Research Note

Gastrointestinal Helminths of Four Lizard Species from Moorea, French Polynesia

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ABSTRACT: The gastrointestinal tracts of 82 lizards comprising 2 gekkonids, *Gehyra oceanica* (N = 20) and *Lepidodactylus lugubris* (N = 31), and 2 scincids, *Cryptoblepharus poecilopleurus* (N = 4) and *Emoia cyanura* (N = 27), from Moorea, French Polynesia, were examined for helminths. One species of cestode, *Cylindrotaenia decidua,* 5 species of nematodes, *Maxvachonia chabaudi, Pharyngodon oceanicus, Spauligodon gehyrae, Skrjabinoptera* sp. (larvae), and an unidentified oxyurid were found. Eleven new host records and 11 new locality records are reported.

KEY WORDS: lizard, *Cryptoblepharus poecilopleurus, Emoia cyanura, Gehyra oceanica, Lepidodactylus lugubris,* Cestoda, Nematoda, Moorea, French Polynesia.

Eight species of lizards—the snake-eyed skink, *Cryptoblepharus poecilopleurus* (Wiegmann, 1834); the moth skink, *Lipinia noctua* (Lesson, 1830); the copper-tailed skink, *Emoia cyanura* (Lesson, 1830); the stump-toed gecko, *Gehyra mutilata* (Wiegmann, 1834); the oceanic gecko, *Gehyra oceanica* (Lesson, 1830); the Indo-Pacific gecko, *Hemidactylus garnotii* Duméril and Bibron, 1836; the Indo-Pacific tree gecko, *Hemiphyllodactylus typus* Bleeker, 1860; and the mourning gecko, *Lepidodactylus lugubris* (Duméril and Bibron, 1836)—occur on Moorea, French Polynesia (Ineich and Blanc, 1988). These species are widely distributed in the Pacific Islands (Burt and Burt, 1932). Helminths have been reported from *G. oceanica, H. garnotii,* and *L. lugubris* (Table 1), but to our knowledge, there are no published reports of helminths from the other 5 lizard species. The purpose of this note is to report helminths for *C. poecilopleurus, E. cyanura, G. oceanica,* and *L. lugubris* from Moorea, French Polynesia, and to list 11 new host and 11 new locality records for these helminths.

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Of the 8 species of lizards on Moorea, 82 individuals representing 4 species were collected by hand by one of us (S.R.G.) in April 1992: 4 *C. poecilopleurus,* 7 *E. cyanura,* 3 *G. oceanica* at Marae Titiroa, 2 km below Belvédere Viewpoint, ca. 457 m elevation, Opunohu Valley (17°33′S, 149°50′W); 20 *E. cyanura,* 12 *G. oceanica,* 11 *L. lugubris* at the Richard B. Gump South Pacific Biological Research Station, ca. 60 m elevation, ca. 3 km west of Paopao (17°31′S, 149°49′W); 5 *G. oceanica,* 20 *L. lugubris* at Paopao, ca. 20 m elevation (17°31′S, 149°51′W). These were the only lizard species observed at the time of collection.

Lizards were fixed in 10% formalin for 24 hours and preserved in 70% ethanol. The abdominal cavity was opened, and the esophagus, stomach, and small and large intestines were removed, slit longitudinally, and examined under a dissecting microscope. All lizards were deposited in the herpetology collection of the Natural History Museum of Los Angeles County (LACM), Los Angeles, California, U.S.A.: *C. poecilopleurus:* LACM 141065–141068; *E. cyanura:* LACM 141038–141064; *G. oceanica:* LACM 141009–141028; *L. lugubris:* LACM 140976–141006.

Each nematode was cleared on a glass slide in undiluted glycerol. Cestodes were stained with hematoxylin and mounted in balsam. Identifications were made from these preparations with use of a compound microscope. Number of helminths, prevalence, mean intensity, and range of infection are given in Table 2. Terminology is in accordance with Bush et al. (1997).

One species of cestode, *Cylindrotaenia decidua* (Ainsworth, 1985), and 5 species of nematodes, *Maxvachonia chabaudi* Mawson, 1972; *Pharyngodon oceanicus* Bursey and Goldberg, 1999; *Spauligodon gehyrae* Bursey and Gold-

Table 1. Previous helminth records for *Gehyra oceanica, Hemidactylus garnotii,* and *Lepidodactylus lugubris.*

Host Helminth	Locality	Reference
Gehyra oceanica (Lesson, 1830)		
Oochoristica javaensis Kennedy, Killick, and Beverley-Burton, 1982	Guam	Goldberg et al., 1998
Pharyngodon oceanicus Bursey and Goldberg, 1999	Moorea, Rarotonga, Tahiti	Bursey and Goldberg, 1999
Spauligodon gehyrae Bursey and Goldberg, 1996	Federated States of Micronesia, Fiji, Guam, Marquesas, Moorea, Rota, Tuamotu	Bursey and Goldberg, 1996a
	Guam, Rota	Goldberg et al., 1998
Hemidactylus garnotii Duméril and Bibron, 1836		
Platynosomum fastosum Kossack, 1910	Island of Oahu, Hawaii	Loo, 1971
	Island of Oahu, Hawaii	Brown et al., 1995
Skrjabinodon dossae (Caballero, 1968)	Leyte, Luzon	Schmidt and Kuntz, 1972
Unidentified oxyurids	Island of Oahu, Hawaii	Brown et al., 1995
Lepidodactylus lugubris (Duméril and Bibron, 1836)	Guam	Dailey et al., 1998
Allopharynx macallisteri Dailey, Goldberg, and Bursey, 1998	Rota	Goldberg et al., 1998
Cylindrotaenia allisonae (Schmidt, 1980)	Islands of Hawaii, Oahu	Goldberg and Bursey, 1997
	Rota	Goldberg et al., 1998
Pharyngodon lepidodactylus Bursey and Goldberg, 1996	Islands of Hawaii, Oahu	Bursey and Goldberg, 1996b
	Islands of Hawaii, Oahu	Goldberg and Bursey, 1997
	Guam, Rota	Goldberg et al., 1998
Skrjabinelazia machidai Hasegawa, 1984	Island of Oahu, Hawaii	Goldberg and Bursey, 1997
Unidentified oxyurids	Island of Oahu, Hawaii	Brown et al., 1995
Raillietiella frenatus Ali, Riley, and Self, 1981	Island of Oahu, Hawaii	Brown et al., 1995
	Island of Oahu, Hawaii	Goldberg and Bursey, 1997

Table 2. Number of helminths (*N*), prevalence as percentage (P), mean intensity (xI ± SD), and range (r) of infections for helminths from *Cryptoblepharus poecilopleurus*, *Emoia cyanura*, *Gehyra oceanica*, and *Lepidodactylus lugubris* collected in Moorea, French Polynesia.

Host	*Cryptoblepharus poecilopleurus* (*n* = 4)				*Emoia cyanura* (*n* = 27)				*Gehyra oceanica* (*n* = 20)				*Lepidodactylus lugubris* (*n* = 31)			
	N	P	xI ± SD	r	*N*	P	xI ± SD	r	*N*	P	xI ± SD	r	*N*	P	xI ± SD	r
Cestoda																
Cylindrotaenia decidua	2*	50	1.0 ± 0.0	—	5*	19	1.0 ± 0.0	—	1*	5	1.0	—	6*	10	2.0 ± 1.7	1–4
Nematoda																
Maxvachonia chabaudi	1*	25	1.0	—	42*	22	7.0 ± 4.2	3–15	1*	5	1.0	—	—	—	—	—
Pharyngodon oceanicus	—	—	—	—	—	—	—	—	17	15	5.7 ± 8.1	1–15	—	—	—	—
Spauligodon gehyrae	—	—	—	—	—	—	—	—	362	50	36.2 ± 63.6	1–213	—	—	—	—
Skrjabinoptera sp. (larvae)	2*	25	2.0	—	8*	26	1.1 ± 0.4	1–2	—	—	—	—	4*	6	2.0 ± 0.0	—
Unidentified oxyurids	3*	25	1.0	—	—	—	—	—	—	—	—	—	—	—	—	—

* New host record.

berg, 1996, *Skrjabinoptera* sp. (larvae only), and an unidentified oxyurid (3 immature females only), were found. Helminths were site specific: *C. decidua* (small intestine); *M. chabaudi* (intestinal tract); *P. oceanicus* (large intestine); *S. gehyrae* (intestinal tract); *Skrjabinoptera* sp. (stomach); and unidentified oxyurids (large intestine).

Selected specimens were placed in vials of 70% ethanol and deposited in the United States National Parasite Collection, Beltsville, Maryland: from *C. poecilopleurus*: *C. decidua,* USNPC 88665; *M. chabaudi,* USNPC 88666; *Skrjabinoptera* sp., USNPC 88667; undetermined oxyurids, USNPC 88668. From *E. cyanura*: *C. decidua,* USNPC 88669; *M. chabaudi,* USNPC 88670; *Skrjabinoptera* sp., USNPC 88671. From *G. oceanica*: *C. decidua,* USNPC 88672; *M. chabaudi,* USNPC 88673; *P. oceanicus,* USNPC 88674; *S. gehyrae,* USNPC 88675. From *L. lugubris*: *C. decidua,* USNPC 88676; *Skrjabinoptera* sp., USNPC 88677.

Cylindrotaenia decidua was originally described from specimens taken from the dark ground skink, *Oligosoma* (=*Leiolopisma) nigriplantare maccani* (Peters, 1879) collected in Wellington, New Zealand, as *Baerietta decidua* by Ainsworth (1985) but was subsequently reassigned to *Cylindrotaenia* by M. K. Jones (1987). This is the second published report of *C. decidua. Cryptoblepharus poecilopleurus, E. cyanura, G. oceanica,* and *L. lugubris* are new host records; Moorea is a new locality record.

Maxvachonia chabaudi has previously been reported only from Australian reptiles in 1 species of gecko, 11 species of skinks, 1 species of varanid, and 1 species of snake (Mawson, 1972; H. I. Jones, 1988; Goldberg and Bursey, 1995; Goldberg et al., 1999). *Cryptoblepharus poecilopleurus, E. cyanura,* and *G. oceanica* are new host records; Moorea is a new locality record.

Pharyngodon oceanicus was described from specimens taken from *G. oceanica* collected on Rarotonga, Cook Islands; additional localities include Moorea (lizards from current study) and Tahiti, Society Islands (Bursey and Goldberg, 1999). Currently, *G. oceanica* is the only known host.

Spauligodon gehyrae was described from specimens found in *G. oceanica* collected on Guam, Mariana Islands; additional localities are in Table 1. This is the second report of *S. ge-*

hyrae from a collection of lizards. *Gehyra oceanica* is the only known host.

Cryptoblepharus poecilopleurus, E. cyanura, and *L. lugubris* are new host records for larvae of *Skrjabinoptera* sp.; Moorea is a new locality record. Oxyurids have not been previously reported in *C. poecilopleurus;* however, once identified, this species would be a new host and locality record. Identification of oxyurids requires male specimens, thus, a description cannot be done at this time.

Further examinations of lizards from additional localities will be needed before the helminth fauna of Pacific Island lizards can be known.

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Literature Cited

Ainsworth, R. 1985. *Baerietta decidua* n. sp. (Cestoda: Nematotaeniidae) from the New Zealand skink *Leiolopisma nigriplantare maccani* Hardy, 1977. New Zealand Journal of Zoology 12:131–135.

Brown, S. G., S. Kwan, and S. Shero. 1995. The parasitic theory of sexual reproduction: parasitism in unisexual and bisexual geckos. Proceedings of the Royal Society of London B 260:317–320.

Bursey, C. R., and S. R. Goldberg. 1996a. *Spauligodon gehyrae* n. sp. (Nematoda: Pharyngodonidae) from *Gehyra oceanica* (Sauria: Gekkonidae) from Guam, Mariana Islands, Micronesia. Journal of Parasitology 82:962–964.

———, **and** ———. 1996b. *Pharyngodon lepidodactylus* sp. n. (Nematoda: Pharyngodonidae) from the mourning gecko, *Lepidodactylus lugubris* (Lacertilia: Gekkonidae), from Hawaii. Journal of the Helminthological Society of Washington 63: 51–55.

———, **and** ———. 1999. *Pharyngodon oceanicus* sp. n. (Nematoda: Pharyngodonidae) from the oceanic gecko, *Gehyra oceanica* (Sauria: Gekkonidae) of the Pacific Islands. Journal of the Helminthological Society of Washington 66:37–40.

Burt, C. E., and M. D. Burt. 1932. Herpetological results of the Whitney South Sea Expedition. VI. Pacific Island amphibians and reptiles in the collection of the American Museum of Natural History. Bulletin of the American Museum of Natural History 63:461–597.

Bush, A. O., K. D. Lafferty, J. M. Lotz, and A. W. Shostak. 1997. Parasitology meets ecology on its own terms: Margolis et al. revisited. Journal of Parasitology 83:575–583.

Dailey, M. D., S. R. Goldberg, and C. R. Bursey. 1998. *Allopharynx macallisteri* sp. n. (Trematoda: Plagiorchiidae) from the mourning gecko, *Lepidodactylus lugubris,* from Guam, Mariana Islands, Micronesia, with a key to the species of the genus *Allopharynx.* Journal of the Helminthological Society of Washington 65:16–20.

Goldberg, S. R., and C. R. Bursey. 1995. Gastrointestinal nematodes of two Australian skinks, *Ctenotus regius* and *Ctenotus schomburgkii.* Journal of the Helminthological Society of Washington 62: 237–238.

———, **and** ———. 1997. New helminth records for the mourning gecko, *Lepidodactylus lugubris* (Gekkonidae) from Hawaii. Bishop Museum Occasional Papers. Records of the Hawaii Biological Survey for 1996. Part 2: Notes, 49:54–56.

———, ———, **and H. Cheam.** 1998. Gastrointestinal helminths of four gekkonid lizards, *Gehyra mutilata, Gehyra oceanica, Hemidactylus frenatus* and *Lepidodactylus lugubris* from the Mariana Islands, Micronesia. Journal of Parasitology 84: 1295–1298.

———, ———, **and S. Hernandez.** 1999. Nematodes of two skinks, *Ctenotus leonhardii* and *Ctenotus quattuordecimlineatus* (Sauria: Scincidae), from Western Australia. Journal of the Helminthological Society of Washington 66:89–92.

Ineich, I., and C. P. Blanc. 1988. Distribution des reptiles terrestres en Polynésie oriental. Atoll Research Bulletin 318:1–75.

Jones, H. I. 1988. Nematodes from nine species of *Varanus* (Reptilia) from tropical northern Australia, with particular reference to the genus *Abbreviata* (Physalopteridae). Australian Journal of Zoology 36:691–708.

Jones, M. K. 1987. A taxonomic revision of the Nematotaeniidae Lühe, 1910 (Cestoda: Cyclophyllidea). Systematic Parasitology 10:165–245.

Loo, B. 1971. The gecko as a new intermediate host for the cat liver fluke, *Platynosomum fastosum,* in Hawaii. Honors thesis, University of Hawaii, 34 pp.

Mawson, P. M. 1972. The nematode genus *Maxvachonia* (Oxyurata: Cosmocercidae) in Australian reptiles and frogs. Transactions of the Royal Society of South Australia 96:101–108.

Schmidt, G. D., and R. E. Kuntz. 1972. Nematode parasites of Oceanica. XIX. Report on a collection from Philippine reptiles. Transactions of the American Microscopical Society 91:63–66.

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Research Note

New Records of Endohelminths of the Alligator Snapping Turtle (*Macroclemys temminckii*) from Arkansas and Louisiana, U.S.A.

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ABSTRACT: Viscera were collected from alligator snapping turtles, *Macroclemys temminckii* (Harlan), caught by commercial trappers in Arkansas and Louisiana. A total of 1,708 parasites were recovered from 44 turtles. Endohelminths identified were 4 species of nematodes (*Brevimulticaecum tenuicolle* Rudolphi, *Falcaustra chelydrae* Harwood, *Falcaustra wardi* Mackin, and *Serpinema trispinosus* Leidy) and 3 species of acanthocephalans (*Neoechinorhynchus chrysemydis* Cable and Hopp, *Neoechinorhynchus emydis* Leidy, and *Neoechinorhynchus pseudemydis* Cable and Hopp). All but *F. chelydrae* are new records for *Macroclemys temminckii*.

KEY WORDS: acanthocephalan, alligator snapping turtle, endohelminth, *Macroclemys temminckii*, nematode, parasite, Arkansas, Louisiana, U.S.A.

The alligator snapping turtle, *Macroclemys temminckii* Harlan, 1835, is a large freshwater chelydrid found along the Gulf Coastal Plains and the Mississippi River Valley, U.S.A. (Lovich, 1993). Although some endohelminths are known to be harbored by this turtle, this study documents several endohelminths not previously reported. The most recent parasite work of *M. temminckii* was conducted by McAllister et al. (1995). In their report, 2 alligator snapping turtles were found to harbor 3 different forms of hemogregarines and the nematode *Falcaustra chelydrae* Harwood, 1932. Additional parasites recovered from *M. temminckii* include the trematode *Lophotaspis interiora* Ward and Hopkins, 1931, and a new species of *Eimeria* Upton et al., 1992. Here, we provide further details on the variation of the endohelminth fauna of *M. temminckii*.

Alligator snapping turtles were caught by commercial trappers in southeastern Arkansas and Louisiana, U.S.A., in the spring and summer of 1993 and 1994. Turtles were generally caught in hoop nets or on baited hooks. Often a number of turtles were delivered to a processor in Louisiana and held in a storage tank for several days until there was a sufficient quantity to process. Viscera were collected and frozen for later analysis. Samples were collected as part of another study on the food habits of *M. temminckii* (Elsey, unpubl.). Viscera were thawed, and stomachs and intestinal tracts were examined for endohelminths. If present, grossly visible parasites were counted and preserved in 70% ethanol for later identification. When required, nematodes were cleared using lactophenol. Temporary mounts of the specimens were made using glycerin jelly. Once identified, the nematodes were returned to 70% ethanol. The acanthocephalans were stained with Semichon's acetocarmine for 24 hours and destained with acid alcohol. Destaining was arrested using 0.1% sodium bicarbonate. Specimens were dehydrated in ethanol, cleared using methyl salicylate, and mounted in Kleermount™. Identifications of nematodes were made using descriptions provided by Baker (1979, 1986) and Sprent (1979). Use of ecological terms follow suggestions of Margolis et al. (1982).

Seven species of helminths were recovered from 44 alligator snapping turtles (Table 1). The parasites include 4 species of nematodes and 3 species of 1 genus of acanthocephalan. In this study, *F. chelydrae* was the only endohelminth found that has been previously documented as a parasite of this turtle. To our knowledge, this is the first record of the nematodes *Brevimulticaecum tenuicolle* Rudolphi, 1819, *Falcaustra wardi* Mackin, 1936, *Serpinema trispinosus* Leidy, 1852, and the acanthocephalans *Neoechinorhynchus chrysemydis* Cable and Hopp, 1954, *Neoechinorhynchus emydis* Leidy, 1851, and *Neoechinorhynchus pseudemydis* Cable and

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Table 1. Parasites recovered from *Macroclemys temminckii* in southeastern Arkansas and Louisiana.

Parasite	Prevalence*	Mean intensity ± SD†	Range	Abundance ± SD‡
Acanthocephala	**9%**	**26.0 ± 44.1**	**1–51**	**2.4 ± 13.9**
Neoechinorhynchus chrysemydis (USNPC 88658)	2%	21.0	—	0.5 ± 3.2
Neoechinorhynchus emydis (USNPC 88659)	2%	51.0	—	1.2 ± 7.7
Neoechinorhynchus pseudemydis (USNPC 88660)	9%	8.0 ± 8.3	1–20	0.7 ± 3.2
Nematoda	**98%**	**37.3 ± 56.4**	**1–319**	**36.5 ± 56.0**
Brevimulticaecum tenuicolle (USNPC 88661)	14%	5.2 ± 9.7	1–25	1.0 ± 4.1
Falcaustra chelydrae (USNPC 88663)	84%	41.4 ± 59.5	1–319	34.8 ± 56.6
Falcaustra wardi (USNPC 88662)	2%	1.0	—	0.0 ± 0.2
Serpinema trispinosus (USNPC 88664)	16%	5.9 ± 5.8	1–14	0.9 ± 3.1

* Prevalence = number of individuals of a host species infected with a particular parasite species ÷ number of hosts examined.

† Abundance = total number of individuals of a particular parasite species in a sample of hosts ÷ total number of individuals of the host species in the sample.

‡ Mean intensity = total number of individuals of a particular parasite species in a sample of a host species ÷ number of infected individuals of the host species in the sample.

Hopp, 1954, from the alligator snapping turtle. Individual turtles harbored up to 4 species of parasites. Thirty-five turtles (79.5%) contained 1 species, 7 turtles (15.9%) had 2 species, and 2 (4.6%) had 4 species. A total of 1,708 parasite specimens were identified. Infected hosts held from 1 to 319 parasites.

Species of *Falcaustra* are commonly reported parasites of aquatic turtles (Conboy et al., 1993). In this study, *F. chelydrae* accounted for 89.6% (1,531) of the total parasites identified and was harbored by 84.1% (37) of the turtles studied. *Falcaustra wardi* accounted for less than 1.0% (1) of the total number of parasites and was detected in only 1 turtle (2.3%).

Serpinema trispinosus is another nematode common in aquatic turtles (Conboy et al., 1993). However, this is the first account of *M. temminckii* harboring this parasite. *Serpinema trispinosus* accounted for 2.4% (41) of the total number of parasites recovered and was found in 15.9% (7) of the turtles.

Brevimulticaecum tenuicolle has been found only in the American alligator, *Alligator mississippiensis* Daudin, 1803 (Sprent, 1979). This nematode can be differentiated from other species based on lobulated, teat-shaped ventricular appendices (Sprent, 1979). In this study 1.8% (31) of the total parasites were *B. tenuicolle*. Of the turtles studied, 13.6% (6) harbored this parasite. This is the first record of this species of helminth in the alligator snapping turtle.

Acanthocephalans of the genus *Neoechinorhynchus* are common endohelminths of aquatic turtles (Petrochenko, 1971). Prior to this report, none has been observed in *M. temminckii*. Species of *Neoechinorhynchus* represented 6.1% (104) of the parasites in this study (1.2% *N. chrysemydis*, 3.0% *N. emydis*, and 1.9% *N. pseudemydis*), and parasitized 9.1% (4) of the turtles.

In summary, this research added 6 new species to the helminth fauna of the alligator snapping turtle. Future natural history and endohelminth surveys of *M. temminckii* could contribute to a better overall understanding of the parasitic life cycle, parasite diversity, and host-parasite relationship.

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Literature Cited

Baker, M. R. 1979. *Serpinema* species (Nematoda: Camallanidae) from turtles of North America and Europe. Canadian Journal of Zoology 57:934–939.

———. 1986. *Falcaustra* species (Nematoda: Kathlaniidae) parasitic in turtles and frogs in Ontario. Canadian Journal of Zoology 64:228–237.

Conboy, G. A., J. R. Laursen, G. A. Averbeck, and B. E. Stromberg. 1993. Diagnostic guide to some of the helminth parasites of aquatic turtles. The Compendium 15:1217–1224.

Lovich, J. E. 1993. *Macroclemys, M. temminckii.* Pages 562.1–562.4 *in* C. H. Ernst, ed. Catalogue of American Amphibians and Reptiles. Society for the Study of Amphibians and Reptiles, New York.

Margolis, L., G. W. Esch, J. C. Holmes, A. M. Kuaris, and G. A Schad. 1982. The use of ecological terms in parasitology (report of an *ad hoc* committee of the American Society of Parasitologists). Journal of Parasitology 68:131–133.

McAllister, C. T., S. J. Upton, and S. E. Trauth. 1995. Hemogregarines (Apicomplexa) and *Falcaustra chelydrae* (Nematoda) in an alligator snapping turtle, *Macroclemys temminckii* (Reptilia: Testudines), from Arkansas. Journal of the Helminthological Society of Washington 62:74–77.

Petrochenko, V. I. 1971. Acanthocephala of Domestic and Wild Animals. Keter Press Binding: Winer Bindery Ltd., Jerusalem, Israel. 465 pp.

Sprent, J. F. A. 1979. Ascaridoid nematodes of amphibians and reptiles: *Multicaecum* and *Brevimulticaecum.* Journal of Helminthology 53:91–116.

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Research Note

Parasites of Eastern Indigo Snakes (*Drymarchon corais couperi*) from Florida, U.S.A.

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ABSTRACT: Nineteen species of parasites (2 trematodes, 3 cestodes, 10 nematodes, 2 acanthocephalans, 1 pentastomid, and 1 tick) were identified from 21 eastern indigo snakes (*Drymarchon corais couperi* Holbrook, 1842) collected in Florida, U.S.A., between 1967 and 1999. For the 12 indigo snakes from which quantitative data were obtained, the most prevalent parasites were the nematodes *Kalicephalus inermis coronellae* Ortlepp, 1923, and *Kalicephalus appendiculatus* Molin, 1861, each occurring in 10 snakes, and cystacanths of *Macracanthorhynchus ingens* (Listow, 1879), which were present in all 12 snakes. The tick *Amblyomma dissimile* Koch, 1844, infested indigo snakes from Brevard County. Twelve new host records are presented.

KEY WORDS: eastern indigo snake, *Drymarchon corais couperi,* parasites, trematodes, cestodes, nematodes, acanthocephalans, pentastomids, cystacanths, Florida, U.S.A.

The eastern indigo snake (*Drymarchon corais couperi* Holbrook, 1842) occurs throughout Florida and much of southern Georgia, U.S.A., although the populations located in Georgia and the Florida panhandle may be very localized (Moler, 1992). It was first protected by the state of Florida in 1972 (Florida Game and Fresh Water Fish Commission, 1972) and was federally listed as threatened in 1978 (U.S. Fish and Wildlife Service, 1978). This project was undertaken to identify the possible impact parasites have on the threatened indigo snake in Florida.

Nine road-killed indigo snakes were necropsied at the Archbold Biological Station (ABS), Highlands County, Florida, between 1967 and

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1987, and from these only a sample of parasites that were seen grossly was collected. Twelve additional eastern indigo snakes were quantitatively examined for parasites between 1992 and 1999. Snakes were collected as roadkills in the following counties in Florida: Alachua (n = 1), Brevard (n = 4), Charlotte (n = 1), Indian River (n = 1), Levy (n = 1), Monroe (n = 2), Okaloosa (n = 1), and Osceola (n = 1). Most snakes were frozen until necropsy, when they were examined following the methods of Kinsella and Forrester (1972). Because of small sample size and the lack of comparable sampling techniques, no statistical analysis was attempted. All indigo snake specimens were deposited in the Florida Museum of Natural History, Gainesville, Florida. Snakes were collected under state and federal collection and salvage permits. Cestodes and trematodes were preserved in Roudabush's AFA and nematodes in 70% ethanol with glycerin. Cestodes and trematodes were stained with either Harris' hematoxylin or Semichon's acetocarmine and mounted in neutral Canada balsam. Nematodes were cleared and mounted in lactophenol. Tissues for histological examination were fixed in 10% neutral buffered formalin, routinely processed, paraffin-embedded, sectioned at 5 μm, and stained with hematoxylin and eosin. Terminology used follows Bush et al. (1997). Voucher specimens of helminths were deposited in the United States National Parasite Collection, Beltsville, Maryland (accession numbers 88619–88632, 88642–88643), and ticks were deposited in the National Tick Collection, Statesboro, Georgia, U.S.A. (accession numbers RML122786, RML122787).

A total of 17 species of helminths (2 trematodes, 3 cestodes, 10 nematodes, 2 acanthocephalans), 1 pentastomid, and 1 tick was collected from the 21 indigo snakes (Table 1). All helminths, except for the 3 species of *Kalicephalus,* are new host records.

From the 9 ABS snakes, the following were identified: *Kalicephalus rectiphilus, Kiricephalus coarctatus,* and cystacanths of *Macracanthorhynchus ingens.* These samples were not included in Table 1 and will not be discussed further, but are presented here as Highlands County records only.

Prevalences and intensities of parasites for the 12 quantitatively examined snakes are listed in Table 1. Three species of *Kalicephalus* (*K. inermis coronellae, K. appendiculatus,* and *K. rectiphilus*) were collected; 6 indigo snakes had all 3 species present, and the other 6 indigo snakes had 2 species. Schad (1962) reported that, as adults, *Kalicephalus* localize themselves in the gut without overlapping in their distribution in the host. This seems to be true for the 3 species of *Kalicephalus* in the indigo snakes we examined. There was some overlapping in distribution of the 3 species (Table 1), but this might have been because of postmortem migration or passive displacement of gut contents when the snakes were killed.

Cystacanths of *M. ingens* were encysted in the mesenteries, mainly on the serosal surface of the small intestine. In histological sections, the cystacanths were located predominantly within the expanded intestinal serosa, with fewer present in the muscular tunics, and were rarely found within the mucosal lamina propria. The intact cystacanths were surrounded by 1–3 layers of fibrous connective tissue with no discernible inflammatory response. Many of the cystacanths were degenerated as characterized by the loss of histological anatomic detail. In these cases, the celomic cavities of the cystacanths were replaced by necrotic cellular debris and fragments of mineralized debris. This accumulation of debris was surrounded by a rim of degenerated heterophils and macrophages, which in turn was surrounded by 1–3 layers of fibrous connective tissue. Cystacanths present within the mucosal lamina propria had been replaced entirely by dense infiltrates of degenerated leucocytes surrounded by multiple layers of fibrous connective tissue. Inflammatory cells were not present outside the fibrous capsule surrounding the degenerated cystacanths. The presence of an inflammatory reaction and degenerated cystacanths was not reported by Goldberg et al. (1998) with the oligacanthorhynchid cystacanths in the longnose snakes (*Rhinocheilus lecontei* Baird & Girard, 1853) that they surveyed.

Elkins and Nickol (1983) reported 7 species of Louisiana snakes that were infected with cystacanths of *M. ingens.* They indicated also that snakes may be a significant epizootiological factor in the life cycle of *M. ingens.* The indigo snake should be considered a paratenic host for these acanthocephalans. They probably become infected with cystacanths by several routes. Being vertebrate generalists in their food habits, indigo snakes in Florida prey on several species of snakes, fishes, frogs, toads, lizards, small tur-

Table 1. Parasites from 12 eastern indigo snakes collected in Florida, U.S.A.

Species of parasite	Location in host*	Number of snakes infected	Intensity		Counties†
			Mean	Range	
Trematoda					
Ochetosoma kansense‡ (Crow, 1913)	ES, OC, ST	7	15	3–34	B, C, I, L, O
Ochetosoma elongatum‡ (Pratt, 1903)	BC, ES, SI, LI, LN	3	337	35–541	B
Cestoda					
Proteocephalus sp.‡	SI	1	2	—	B
Larval cestode (tetrathyridium)‡	ME	2	7	3–10	O
Larval cestode (sparganum)‡	ME	2	3	2–3	I, K
Nematoda					
Kalicephalus inermis coronellae Ortlepp, 1923	ES, ST	10	37	2–128	A, B, C, K, L, M, O
Kalicephalus appendiculatus Molin, 1861	ST, SI	10	27	5–50	B, C, I, L, M, O,
Kalicephalus rectiphilus Harwood, 1932	SI, LI	9	17	1–77	A, B, C, K, L, M, O
Physaloptera obtussima‡ Molin, 1860	ES	1	1	—	K
Terranova caballeroi‡ Barus and Coy Otero, 1966	ST	1	1	—	K
Strongyloides sp.	SI, LI	3	3	1–5	B, C, I
Eustrongylides larvae‡	ST	1	2	—	B
Gnathostoma larvae‡	ME	1	2	—	M
Physaloptera larvae‡	ST	6	16	1–80	A, B, I, M, O
Larval nematodes	SI	5	5	1–9	B, I, O
Acanthocephalan (cystacanths)					
Centrorhynchus spinosus‡ (Kaiser, 1893)	ME	3	18	3–30	B, K
Macracanthorhynchus ingens‡ (Listow, 1879)	ME	12	151	1–515	A, B, C, I, K, L, M, O
Pentastoma					
Kiricephalus coarctatus (Diesing, 1850)	BC, LN	8	3	1–7	B, C, I, K, L, M, O
Acari					
Amblyomma dissimile Koch, 1844	SK	3	7	2–10	B

* BC = body cavity; ES = esophagus; LI = large intestine; LN = lungs; ME = mesenteries; OC = oral cavity; SI = small intestine; SK = skin; ST = stomach.

† County where parasite was found: A = Alachua; B = Brevard; C = Charlotte; I = Indian River; K = Okaloosa; L = Levy; M = Monroe; O = Osceola.

‡ New host records.

tles, birds, and small mammals (Moler, 1992), which may also be paratenic hosts for *M. ingens*. Larval stages have been identified from Florida mice (*Podomys floridanus* (Chapman, 1889)) and cotton mice (*Peromyscus gossypinus* (Le Conte, 1853)) in Florida (Forrester, 1992). In Florida, adults of *M. ingens* have been reported mainly from raccoons (*Procyon lotor* (Linnaeus, 1758)) (Forrester, 1992) and black bears (*Ursus americanus floridanus* (Merriam, 1896)) (Conti et al., 1983). Cystacanths of *Centrorhynchus spinosus* also were encysted in the mesenteries. They encysted mainly on the serosal surface of the small intestine, intermixed with the cystacanths of *M. ingens,* but in much lower intensities (Table 1). The definitive hosts for *C. spinosus* are several species of birds, primarily owls (Nickol, 1983). In Florida we have unpublished records of them in barred owls, *Strix varia* Barton, 1799, eastern screech-owls, *Otus asio* Linnaeus, 1758, and great horned owls, *Bubo virginianus* (Gmelin, 1788). Raccoons and raptors in Florida could acquire these acanthocephalan infections from indigo snakes if these

snakes are part of their diet. The indigo snake has not been reported as a food item of black bears in Florida (Maehr and DeFazio, 1985). The encysted cystacanths did not seem to have any obvious detrimental effect on any of the indigo snakes necropsied. One road-killed female indigo snake with 515 *M. ingens* encysted in mesenteries around the small intestines had a large amount of visceral fat present and 11 eggs in utero.

Only 1 indigo snake (Okaloosa County) was infected with a single *Terranova caballeroi.* This ascarid is a common parasite of water snakes (*Nerodia* spp.) and cottonmouths (*Agkistrodon piscivorus* Lacépède, 1789) in the southeastern United States (Fontenot and Font, 1996).

Fourth-stage larvae of a species of *Eustrongylides* were found in the stomach wall of 1 snake from Brevard County. These were most likely the larvae of *Eustrongylides ignotus,* adults of which are parasitic in birds, most commonly Ciconiiformes (Spalding et al., 1993). The most important intermediate host for *E. ignotis* in Florida is the small mosquitofish (*Gambusia holbrooki* Girard, 1859), with some amphibians and reptiles serving as paratenic hosts (Coyner, 1998). This would be considered an accidental infection of a snake with a bird parasite.

In this study, *Amblyomma dissimile* infested indigo snakes only from Merritt Island in Brevard County. The ticks seemed to aggregate to a small localized area of about 5 cm in diameter. The skin in the areas of tick attachment was swollen, with some of the scales malformed. Histologically, the areas of tick attachment were marked by a pustular dermatitis that was acute, multifocal, and severe, with intralesional bacterial and fungal colonization. At the junctions between numerous scales were multifocal, locally extensive subcorneal pustules that contained degenerate heterophils intermixed with numerous gram-positive bacterial cocci. At several of the scale junctions the subcorneal aggregate of degenerate heterophils extended through the epidermis into the dermis. Durden et al. (1993) reported *A. dissimile* from an eastern indigo snake and a cotton mouse (*P. gossypinus*) from Merritt Island in 1990. Most indigo snakes seen on Merritt Island by one of us (P.E.M.) have been infested with *A. dissimile,* and Durden et al. (1993) suggested that a viable population of this tick species occurs there. *Amblyomma dissimile* has been reported infesting these additional hosts in Florida: pygmy rattlesnake (*Sistrurus miliarius* Linnaeus, 1766), yellow rat snake (*Elaphe obsoleta quadrivittata* Holbrook, 1836), Florida kingsnake (*Lampropeltis getula floridana* Blanchard, 1919), common kingsnake (*Lampropeltis getula* Linnaeus, 1766), eastern diamond rattlesnake (*Crotalus adamanteus* Palisot de Beauvois, 1799), pine snake (*Pituophis melanoleucus* Daudin, 1803), cottonmouth (*A. piscivorus*), gopher tortoise (*Gopherus polyphemus* Daudin, 1802), and giant toad (*Bufo marinus* Linnaeus, 1855), and reported in the following counties: Broward, Collier, Dade, Indian River, Lee, Martin, Palm Beach, and St. Lucie (Bequaert, 1932; Bequaert, 1945; Wilson and Kale, 1972; unpublished computer and manual searches of the data records of the Florida State Collection of Arthropods, Gainesville, Florida, U.S.A., and the National Tick Collection, Statesboro, Georgia, U.S.A., 1999). From these records *A. dissimile* seems to be well established in southern peninsular Florida.

Because most of the indigo snakes we examined were in good flesh and had deposits of visceral fat and several of the females had a normal number of eggs in utero, it is our assessment that the general health of the snakes did not seem to be compromised by the parasite intensities we report here. The attachment sites of *A. dissimile* may allow a pathway for secondary bacterial infections to infiltrate to deeper tissues. However, in the indigo snakes we examined, the bacterial infections were very localized.

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Literature Cited

Bequaert, J. 1932. *Amblyomma dissimile* Koch, a tick indigenous to the United States (Acarina: Ixodidae). Psyche 39:45–47.

———. 1945. Further records of the snake tick, *Amblyomma dissimile* Koch, in Florida. Bulletin of the Brooklyn Entomological Society 40:129.

Bush, A. O., K. D. Lafferty, J. M. Lotz, and A. W. Shostak. 1997. Parasitology meets ecology on its own terms: Margolis et al. revisited. Journal of Parasitology 83:575–583.

Conti, J. A., D. J. Forrester, and J. R. Brady. 1983. Helminths of black bears in Florida. Proceedings of the Helminthological Society of Washington 50:252–256.

Coyner, D. F. 1998. The epizootiology and transmission of *Eustrongylides ignotus* (Dioctophymatoidea) in intermediate hosts in Florida. Ph.D. Dissertation, University of Florida, Gainesville. 245 pp.

Durden, L. A., J. S. H. Klompen, and J. E. Keirans. 1993. Parasitic arthropods of sympatric opossums, cotton rats, and cotton mice from Merritt Island, Florida. Journal of Parasitology 79:283–286.

Elkins, C. A., and B. B. Nickol. 1983. The epizootiology of *Macracanthorhynchus ingens* in Louisiana. Journal of Parasitology 69:951–956.

Fontenot, L. W., and W. F. Font. 1996. Helminth parasites of four species of aquatic snakes from two habitats in southeastern Louisiana. Journal of the Helminthological Society of Washington 63: 66–75.

Forrester, D. J. 1992. Parasites and Diseases of Wild Mammals in Florida. University Press of Florida, Gainesville. 459 pp.

Florida Game and Fresh Water Fish Commission. 1972. Wildlife Code of the State of Florida. Chapter 16–E, Rules 16–E–3.00 and 16–E–4.02, pages 86–92. Effective date July 1972. Florida Game and Fresh Water Fish Commission, Tallahassee, Florida.

Goldberg, S. R., C. H. Bursey, and H. J. Holshuh. 1998. Prevalence and distribution of cystacanths of an oligacanthorhynchid acanthocephalan from the longnose snake, *Rhinocheilus lecontei* (Colubridae), in southwestern North America. Journal of the Helminthological Society of Washington 65:262–265.

Kinsella, J. M., and D. J. Forrester. 1972. Helminths of the Florida duck, *Anas platyrhynchos fulvigula.* Proceedings of the Helminthological Society of Washington 39:173–176.

Maehr, D. S., and J. T. DeFazio, Jr. 1985. Foods of black bears in Florida. Florida Field Naturalist 13: 8–12.

Moler, P. E. 1992. Eastern indigo snake. Pages 181–186 *in* P. E. Moler, ed. Rare and Endangered Biota of Florida. Volume III: Amphibians and Reptiles. University Press of Florida, Gainesville.

Nickol, B. B. 1983. *Centrorhynchus kuntzi* from the USA with description of the male and redescription of *C. spinosus* (Acanthocephala: Centrorhynchidae). Journal of Parasitology 69:221–225.

Schad, G. A. 1962. Studies on the genus *Kalicephalus* (Nematoda: Diaphanocephalidae). II. A taxonomic revision of the genus *Kalicephalus* Molin, 1861. Canadian Journal of Zoology 40:1035–1165.

Spalding, M. G., G. T. Bancroft, and D. J. Forrester. 1993. The epizootiology of eustrongylidosis in wading birds. Journal of Wildlife Diseases 29: 237–249.

U.S. Fish and Wildlife Service. 1978. Part 17. Endangered and threatened wildlife and plants. Listing of the eastern indigo snake as a threatened species. Federal Register 43:4026–4028.

Wilson, N., and H. W. Kale III. 1972. Ticks collected from Indian River County, Florida (Acari: Metastigmata: Ixodidae). Florida Entomologist 55:53–57.

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Research Note

Helminths of Two Sympatric Toad Species, *Bufo marinus* (Linnaeus) and *Bufo marmoreus* Wiegmann, 1833 (Anura: Bufonidae) from Chamela, Jalisco, Mexico

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ABSTRACT: Helminths of sympatric *Bufo marinus* (Linnaeus) (N = 49) and *Bufo marmoreus* Wiegmann (N = 19) from the Pacific coast of Jalisco, Mexico, are reported. *Bufo marinus* harbored *Ochoterenella digiticauda* Caballero y Caballero, *Rhabdias fuelleborni* Travassos, *Physaloptera* sp. (larvae), an unidentified species of nematode, and cystacanths of *Centrorhynchus* sp. *Bufo marinus* is a new host and Jalisco a new locality record for *R. fuelleborni* and *Physaloptera* sp. *Bufo marmoreus* harbored *Aplectana incerta* Caballero y Caballero, *R. fuelleborni, Physocephalus* sp. (larvae), and cystacanths of *Centrorhynchus* sp. *Bufo marmoreus* is a new host record for each of these helminths.

KEY WORDS: *Bufo marinus, Bufo marmoreus,* nematodes, *Aplectana incerta, Ochoterenella digiticauda, Rhabdias fuelleborni, Physaloptera* sp., *Physocephalus* sp., *Centrorhynchus* sp., cystacanth, Jalisco, Mexico.

Twenty-five species of *Bufo* have been reported from various regions of Mexico; 8 species are endemic (Flores-Villela, 1993). During September 1995, individuals of 2 species, *Bufo marinus* (Linnaeus, 1758) and *Bufo marmoreus* Wiegmann, 1833, from the Pacific coast of Jalisco State, Mexico, became available for examination for parasites. The cane toad, *B. marinus,* originally ranged from southern Texas to central Brazil but now has worldwide distribution (Zug and Zug, 1979). The marbled toad, *B. marmoreus,* is endemic to Mexico, occurring from the Transverse Volcanic Axis, Sierra Madre del Sur, and highlands of northern Oaxaca State eastward to the Gulf of Mexico coastal plain and Yucatan Peninsula, westward to the Pacific coast, and south to the Rio Balsas basin and the central depression of Chiapas State (Flores-Villela, 1993). There are several reports of helminths from *B. marinus* (Caballero y Caballero, 1949, 1954; Kloss, 1971; Goldberg and Bursey, 1992; Goldberg et al., 1995; Barton, 1997; Linzey et al., 1998), but to our knowlege there are no reports of helminths from *B. marmoreus.* The purpose of this note is to report helminths of *B. marinus* and *B. marmoreus* from Jalisco, Mexico.

Forty-nine *Bufo marinus* (mean snout-vent length, SVL = 129 mm ± 30 mm SD; range, 75–190 mm) and 19 *B. marmoreus* (SVL = 76 mm ± 5 mm SD; range, 65–83 mm) were examined. The toads had been collected by hand from Emiliano Zapata Village (19°24′N, 104°59′W) about 30 km south of the Chamela Biological Station, Instituto de Biología, Universidad Nacional Autónoma de México (IBUNAM), Jalisco, Mexico, and were deposited in the Colección Nacional de Anfibios y Reptiles, IBUNAM. The toads were killed by freezing, the body cavity was opened by a longitudinal incision from vent to throat, and the gastrointestinal tract was excised by cutting across the esophagus and the rectum. Stomachs and intestines were opened longitudinally and examined under a stereomicroscope. Helminths were removed and counted. Acanthocephalans and nematodes were fixed using 4% saline-formalin. Acanthocephalans were stained with Meyer's paracarmine, dehydrated in a graded ethanol series, cleared in methyl salicylate, and mounted in Canada balsam. Nematodes were dehydrated to 70% ethanol, cleared in glycerol, and exam-

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Table 1. Number, prevalence, mean intensity, range, and abundance for helminths collected from *Bufo marinus* and *Bufo marmoreus* from Chamela, Jalisco, Mexico.

Toad species Helminth species	Number of helminths	Site	Prevalence (%)	Mean intensity ± SD (range)	Mean abundance ± SD
Bufo marinus (N = 49)					
Ochoterenella digiticauda	24	Coelom	8	6.0 ± 6.0 (3–16)	0.5 ± 2.4
*Rhabdias fuelleborni**	145	Lungs	37	5.4 ± 1.2 (1–38)	3.0 ± 6.0
Physaloptera sp. (larvae)*	184	Stomach	31	12.3 ± 18.0 (1–59)	3.8 ± 11.3
Unidentified nematode	6	Intestine	6	2.0 ± 7.4 (1–4)	0.1 ± 0.6
Centrorhynchus sp. cystacanths	12	Coelom	22	1.1 ± 0.3 (1–2)	0.2 ± 0.5
Bufo marmoreus (N = 19)					
*Aplectana incerta**	848	Intestine	63	70.7 ± 42 (1–250)	4.47 ± 66.4
*Rhabdias fuelleborni**	17	Lung	16	5.7 ± 6.4 (2–13)	0.9 ± 13.0
Physocephalus sp. (encysted larvae)*	7	Coelom	5	7	0.36
Centrorhynchus sp. cystacanths*	1	Coelom	5	1	0.05

* New host record.

ined as temporary wet mounts. Voucher specimens were deposited in the Colección Nacional de Helmintos (CNHE), IBUNAM, *B. marinus*: *Ochoterenella digiticauda* Caballero y Caballero, 1944 (3775); *Rhabdias fuelleborni* Travassos, 1926 (3776); *Physaloptera* sp. (3774), *Centrorhynchus* sp. (3777); *B. marmoreus*: *Aplectana incerta* Caballero y Caballero, 1949 (3772), *R. fuelleborni* (3771), *Physocephalus* sp. (3773), *Centrorhynchus* sp. (3778). Terminology is in accordance with Bush et al. (1997).

Four species of nematodes and 1 species of acanthocephalan were found in *B. marinus*; 3 species of nematodes and 1 species of acanthocephalan were found in *B. marmoreus*. Numbers of parasites, prevalence, abundance, and sites of infection are given in Table 1. *Bufo marinus* harbored 371 helminths. *Rhabdias fuelleborni* had the highest prevalence (37%); *Physaloptera* sp. (larvae) had the greatest mean intensity (12.3). Mean number of helminth species per host was 1.0 ± 0.8 SD, mean intensity per host was 8.1 ± 15.3 SD. Twelve toads had no parasites, 23 were parasitized by 1 species, 13 had 2 or more helminth species. *Bufo marmoreus* harbored 873 helminths. The helminth species with highest prevalence (63%) and greatest mean intensity (45) was *A. incerta*. Mean number of helminth species per host was 0.9 ± 0.7 SD; mean intensity per host was 46.0 ± 67.0 SD. Six toads had no helminths, 9 were parasitized by 1 species, and 4 had 2 or more species. In *B. marinus*, species richness and mean abundance for the helminth fauna fell within the ranges reported by Aho (1990) for amphibians in general, i.e., a mean species richness per host individual of 0.98 ± 0.07 SE, and a mean abundance of 11.55 ± 1.86 SE. However, for *B. marmoreus*, mean abundance was much greater, in part because of the large number of individuals of *A. incerta* harbored by a few hosts.

The known helminth fauna for *B. marinus* in Mexico is presented in Table 2. This list includes 5 species of trematodes, 1 species of cestode, at least 13 species of nematodes, and 1 species of acanthocephalan. *Bufo marinus* is a new host and locality record for *Rhabdias fuelleborni* and *Physaloptera* sp. *Bufo marmoreus* is a new host and locality record for *A. incerta, R. fuelleborni, Physocephalus* sp., and cystacanths of *Centrorhynchus* sp.

None of the parasites found in this study was unique to *B. marinus* or *B. marmoreus*; all are shared with other amphibian or reptile species (Baker, 1987). However, 3 of these species, *A. incerta, O. digiticauda,* and *R. fuelleborni,* are typically found in toads. *Aplectana incerta* was originally described by Caballero y Caballero (1949) from *B. marinus* collected in Chiapas State, Mexico, and was subsequently reported from *Bufo debilis* Girard, 1854, *Bufo retiformis* Sanders and Smith, 1951, *Scaphiopus couchii* Baird, 1854, and *Spea multiplicata* Cope, 1863, from Arizona and New Mexico, U.S.A. (Goldberg and Bursey, 1991; Goldberg et al., 1995; Goldberg et al., 1996). *Ochoterenella digiticauda* is a common parasite of *B. marinus* in Costa Rica, Guatemala, Mexico, and Jamaica (Brenes and Bravo-Hollis, 1959; Wong and Bundy, 1985). *Rhabdias fuelleborni* is a neotropical spe-

Table 2. Published records of helminths from *Bufo marinus* from Mexico.

Species	Locality	Reference
Digenea		
Clinostomum attenuatum	Not given	Etges, 1991
Glypthelmins intermedia	Chiapas	Caballero y Caballero et al., 1944
	Oaxaca	Bravo-Hollis, 1948
Gorgoderina megalorchis	Oaxaca	Bravo-Hollis, 1948
Langeronia macrocirra	Veracruz	Guillén-Hernández, 1992
Mesocoelium monas	Veracruz	Guillén-Hernández, 1992
Cestoda		
Distoichometra bufonis	Nuevo León	Martínez, 1969
Nematoda		
*Aplectana hoffmani**	Puebla	Bravo-Hollis, 1943
Aplectana incerta	Chiapas	Caballero y Caballero, 1949, 1954
Aplectana itzocanensis	Veracruz	Caballero-Deloya, 1974
Aplectana sp.	Veracruz	Guillén-Hernández, 1992
Cruzia morleyi	Veracruz	Caballero-Deloya, 1974
Cosmocerca sp.	Veracruz	Guillén-Hernández, 1992
Ochoterenella caballeroi	Chiapas	Esslinger, 1987b
Ochoterenella chiapensis	Chiapas	Esslinger, 1988b
Ochoterenella digiticauda	Chiapas	Esslinger, 1987a
	Jalisco	This study
Ochoterenella figueroai	Chiapas	Esslinger, 1988a
Ochoterenella lamothei	Chiapas	Esslinger, 1988a
Ochoterenella nanolarvate	Chiapas	Esslinger, 1987b
Ochoterenella sp.	Veracruz	Guillén-Hernández, 1992
Oswaldocruzia subauricularis	Chiapas	Caballero y Caballero, 1949, 1954
Oswaldocruzia pipiens	Nuevo León	Martínez, 1969
Oswaldocruzia sp.	Veracruz	Guillén-Hernández, 1992
Rhabdias fuelleborni	Jalisco	This study
Rhabdias sphaerocephala†	Chiapas	Caballero y Caballero, 1949, 1954
	Veracruz	Caballero-Deloya, 1974
	Veracruz	Bravo-Hollis and Caballero y Caballero, 1940
	Nuevo León	Martínez, 1969
	Veracruz	Guillén-Hernández, 1992
Physaloptera sp. (larvae)	Jalisco	This study
Acanthocephala		
Centrorhynchus sp.	Veracruz	Guillén-Hernández, 1992
Centrorhynchus sp.	Jalisco	This study

* Junior homonym of *Aplectana incerta* per Baker (1985).
† Considered a Palaearactic species only by Baker (1987).

cies previously reported from *B. marinus* from Brazil, Costa Rica, Guatemala, and Bermuda (Brenes and Bravo-Hollis, 1959; Caballero y Caballero, 1954; Kloss, 1971; Goldberg et al., 1995; Linzey et al., 1998) as well as *Bufo arenarum* Hansel, 1867, *Bufo ictericus* Spix, 1824, *Bufo paracnemis* Lutz, 1925, and *Thoropa miliaris* (Spix, 1824), from Brazil, Uruguay, and Paraguay (Kloss, 1974; Masi-Pallares and Maciel, 1974).

The remaining helminths found in this study were juveniles of species requiring intermediate hosts to complete their life cycles. Larvae of *Physaloptera* sp. and *Physocephalus* sp. and cystacanths of *Centrorhynchus* sp. have frequently been reported from amphibians as well as from mammals, birds, and reptiles that habitually feed on insects (Goldberg et al., 1993). Larvae of *Physaloptera* sp. were found in the lumen of the stomach; larvae of *Physocephalus* sp. and the cystacanths were encysted in the peritoneum. The presence of larvae of *Physaloptera* sp. may reflect host diet preferences rather than host–parasite interactions, because encystment would be expected in paratenism. However, the number of cysts containing *Physocephalus* sp. and the cystacanths was too low to conclude that *B. marinus* is a paratenic host for these helminth species; rather, incidental infection is more likely.

Too few studies have been undertaken to draw conclusions about helminth communities in species of toads from Mexico. The data in Table 2 suggest that helminth species composition in *Bufo marinus* is variable from population to population. However, 3 features characterize these faunas: nematode species predominate; they are depauperate; and they are dominated by a single species.

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Literature Cited

Aho, J. M. 1990. Helminth communities of amphibians and reptiles: comparative approaches to understanding patterns and processes. Pages 157–195 *in* G. W. Esch, A. O. Bush, and J. M. Aho, eds. Parasite Communities: Patterns and Processes. Chapman and Hall, New York.

Baker, M. R. 1985. Redescription of *Aplectana itzocanensis* and *A. incerta* (Nematoda: Cosmocercidae) from amphibians. Transactions of the American Microscopical Society 104:272–277.

———. 1987. Synopsis of the Nematoda parasitic in amphibians and reptiles. Memorial University of Newfoundland, Occasional Papers in Biology 11: 1–325.

Barton, D. P. 1997. Introduced animals and their parasites: the cane toad, *Bufo marinus,* in Australia. Australian Journal of Ecology 22:316–324.

Bravo-Hollis, M. 1943. Dos nuevos nemátodos parásitos de anuros del sur de Puebla. Anales del Instituto de Biología, Universidad Nacional Autónoma de México 14:69–78.

———. 1948. Descripción de dos especies de tremátodos parásitos de *Bufo marinus* L. procedentes de Tuxtepec, Oaxaca. Anales del Instituto de Biología, Universidad Nacional Autónoma de México 19:153–161.

———, **and E. Caballero y Caballero.** 1940. Nemátodos parásitos de los batracios de México. IV. Anales del Instituto de Biología, Universidad Nacional Autónoma de México 11:239–247.

Brenes, R. R., and M. Bravo-Hollis. 1959. Helmintos de la República de Costa Rica VIII. Nematoda. 2. Algunos nemátodos de *Bufo marinus* (L) y algunas consideraciones sobre los géneros *Oxysomatium* y *Aplectana.* Revista de Biología Tropical 7: 35–55.

Bush, A. O., K. D. Lafferty, J. M. Lotz, and A. W. Shostak. 1997. Parasitology meets ecology on its own terms: Margolis et al. revisited. Journal of Parasitology 83:575–583.

Caballero y Caballero, E. 1949. Estudios helmintológicos de la región oncocercosa de México y de la República de Guatemala. Nematoda. Parte 5. Anales del Instituto de Biología, Universidad Nacional Autónoma de México 20:279–292.

———. 1954. Estudios helmintológicos de la región oncocercosa de México y de la República de Guatemala. Nematoda. Parte 8. Anales del Instituto de Biología, Universidad Nacional Autónoma de México 25:259–274.

———, **M. Bravo-Hollis, and M. C. Cerecero.** 1944. Estudios helmintológicos de la región oncocercosa de México y de la República de Guatemala. Trematoda. 1. Anales del Instituto de Biología, Universidad Nacional Autónoma de México 15:59–72.

Caballero-Deloya, J. 1974. Estudios helmintológicos de los animales silvestres de la Estación de Biología Tropical "Los Tuxtlas," Veracruz. Nematoda. 1. Algunos nemátodos parásitos de *Bufo horribilis* Weigmann, 1833. Anales del Instituto de Biología, Universidad Nacional Autónoma de México 45:45–50.

Esslinger, J. H. 1987a. Redescription of *Ochoterenella digiticauda* Caballero, 1944 (Nematoda: Filarioidea) from the toad *Bufo marinus,* with a redefinition of the genus *Ochoterenella* Caballero, 1944. Proceedings of the Helminthological Society of Washington 53:210–217.

———. 1987b. *Ochoterenella caballeroi* sp. n. and *O. nanolarvata* sp. n. (Nematoda: Filarioidea) from the toad *Bufo marinus.* Proceedings of the Helminthological Society of Washington 54:126–132.

———. 1988a. *Ochoterenella figueroai* sp. n. and *O. lamothei* sp. n. (Nematoda: Filarioidea) from the toad *Bufo marinus.* Proceedings of the Helminthological Society of Washington 55:146–154.

———. 1988b. *Ochoterenella chiapanaensis* sp. n. (Nematoda: Filarioidea) from the toad *Bufo marinus* in Mexico and Guatemala. Transactions of the American Microscopical Society 107:203–208.

Etges, J. F. 1991. *Clinostomum attenuatum* (Digenea) from the eye of *Bufo marinus.* Journal of Parasitology 77:634–635.

Flores-Villela, O. 1993. Herpetofauna Mexicana. Annotated list of the species of amphibians and reptiles of Mexico, recent taxonomic changes, and new species. Carnegie Museum of Natural History, Special Publication 17:1–73.

Goldberg, S. R., and C. R. Bursey. 1991. Helminths of three toads, *Bufo alvarius, Bufo cognatus* (Bufonidae) and *Scaphiopus couchi* (Pelobatidae) from southern Arizona. Journal of the Helminthological Society of Washington 58:142–146.

———, **and** ———. 1992. Helminths of the marine toad, *Bufo marinus* (Anura: Bufonidae) from American Samoa. Journal of the Helminthological Society of Washington 59:131–133.

———, ———, **and I. Ramos.** 1995. The component helminth community of three sympatric toad species, *Bufo cognatus, Bufo debilis* (Bufonidae), and *Spea multiplicata* (Pelobatidae) from New Mexico. Journal of the Helminthological Society of Washington 62:57–61.

———, ———, **B. K. Sullivan, and Q. A. Truong.** 1996. Helminths of the Sonoran green toad, *Bufo retiformis* (Bufonidae), from southern Arizona. Journal of the Helminthological Society of Washington 63:120–122.

———, ———, **and R. Tawil.** 1993. Gastrointestinal helminths of the western bush lizard, *Urosaurus graciosus graciosus* (Phrynosomatidae). Bulletin of the Southern California Academy of Science 92:43–51.

———, ———, **and** ———. 1995. Helminths of an introduced population of the giant toad, *Bufo marinus* (Anura: Bufonidae), from Bermuda. Journal of the Helminthological Society of Washington 62:64–67.

Guillén-Hernández, S. 1992. Comunidades de helmintos de algunos anuros de "Los Tuxtlas," Veracruz. Tesis de Maestría. Facultad de Ciencias, Universidad Nacional Autónoma de México. 90pp.

Kloss, G. R. 1971. Alguns *Rhabdias* (Nematoda) de *Bufo* no Brasil. Papéis Avulsos de Zoologia 24:1–52.

———. 1974. *Rhabdias* (Nematoda: Rhabditoidea) from the *marinus* group of *Bufo*. A study of sibling species. Arquivos de Zoologia 25:61–120.

Linzey, D. W., C. R. Bursey, and J. B. Linzey. 1998. Seasonal occurrence of helminths of the giant toad, *Bufo marinus* (Amphibia, Bufonidae), in Bermuda. Journal of the Helminthological Society of Washington 65:251–258.

Martínez, V. J. M. 1969. Parásitos de algunos anfibios colectados en diferentes áreas de los Municipios de Escobedo, Pesqueria y Santiago, Nuevo León, México. Tesis Licenciatura. Facultad de Ciencias Biológicas, Universidad Autónoma de Nuevo León, Monterrey. 51 pp.

Masi-Pallares, R., and S. Maciel. 1974. Helminthes en batracios del Paraguay (Parte 1), con descripción de una nueva especie, *Aplectana pudenda* (Oxyuridae: Cosmocercinae). Revista Paraguaya de Microbiología 9:55–60.

Wong, M. S., and D. A. P. Bundy. 1985. Population distribution of *Ochoterenella digiticauda* (Nematoda: Onchocercidae) and *Mesocoelium monas* (Digenea: Brachycoeliidae) in naturally infected *Bufo marinus* (Amphibia: Bufonidae) from Jamaica. Parasitology 90:457–461.

Zug, G. R., and P. B. Zug. 1979. The marine toad, *Bufo marinus:* a natural history resumé of native populations. Smithsonian Contributions to Zoology 284:1–58.

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Research Note

Endohelminths of the Ravine Salamander, *Plethodon richmondi*, from Southwestern West Virginia, U.S.A.

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ABSTRACT: Four species of endohelminths were found in 51 ravine salamanders, *Plethodon richmondi*, from southwestern West Virginia in February, March, April, October, and November 1996, and February 1997. The nematode *Angiostoma plethodontis* had the highest prevalence (29.4%), and the trematode *Brachycoelium storeriae* had the highest mean intensity (2.3). Larvae of *Batracholandros salamandrae* were present in both the small and large intestines of 5 hosts. An unidentified acanthocephalan cystacanth was found encapsulated in the mesentery of a single host. *Plethodon richmondi* represents new host records for *Angiostoma plethodontis* and *Brachycoelium storeriae*, and West Virginia is a new locality record for all of the helminth species identified.

KEY WORDS: *Angiostoma plethodontis, Batracholandros salamandrae, Brachycoelium storeriae, Plethodon richmondi,* ravine salamander, West Virginia, U.S.A.

The ravine salamander, *Plethodon richmondi* Netting and Mittleman, 1938, is a small, slender terrestrial plethodontid species inhabiting the wooded slopes of valleys and ravines from western Pennsylvania south to northeastern Tennessee and northwestern North Carolina and west to southeastern Indiana (Green and Pauley, 1987). In a parasite survey of plethodontid salamanders in Tennessee, Dunbar and Moore (1979) reported 2 species of helminths from *P. richmondi*; the tapeworm *Cylindrotaenia americana* Trowbridge and Hefley, 1934, and the nematode *Batracholandros salamandrae* (Schad, 1960) Petter and Quentin, 1976. Fifteen *P. rich-*

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Table 1. Prevalence and mean intensity of helminth parasites found in 51 *Plethodon richmondi* from southwestern West Virginia.

Parasite species	Prevalence*	Mean intensity ±1 SD (range)	Site of infection
Brachycoelium storeriae	10 (19.6)	2.3 ± 1.70 (1–7)	Small intestine
Angiostoma plethodontis	15 (29.4)	1.5 ± 0.64 (1–3)	Small intestine
Batracholandros salamandrae	5 (9.8)	1.2 ± 0.45 (1–2)	Small, large intestine
Acanthocephalan cystacanth (unidentified)	1 (2.0)	1.0 — (1)	Mesentery

* Number (%) infected.

mondi individuals were included in that survey, and prevalences were 6.7% and 20.0% for *C. americana* and *B. salamandrae,* respectively. These are the only reported helminths from this salamander host to date. Accordingly, this report presents new information on helminths of this plethodontid species, including prevalences and intensities of infection.

A total of 51 ravine salamanders (28 females and 23 males) was collected in Cabell and Wayne counties of West Virginia in February–April, October, and November 1996 and in February 1997. Salamanders were captured by hand in mature forests of beech, maple, and oak trees on cool rainy evenings. Hosts were placed in plastic bags with damp leaf litter and returned to the laboratory where they were maintained in a refrigerator at approximately 4°C. All salamanders were necropsied within 18 hr of capture. Immediately prior to necropsy, each salamander was measured for snout-vent length (SVL) to the nearest mm with vernier calipers, and weighed to the nearest 0.1 g on a Mettler Model BB300® electronic balance. Mean SVL of 50 mm (±SE = 1.5 mm) for females did not differ significantly from the mean SVL of 47 mm (±SE = 1.3 mm) for males ($t_{0.05,49} = 1.471$; $P > 0.05$). Mean weight of 1.53 g (±SE = 0.11 g) for females did not differ significantly from the mean of 1.28 g (±SE = 0.77) for males ($t_{0.05,49} = 1.852$; $P > 0.05$). Since neither mean snout-vent lengths nor total body weights for female versus male *P. richmondi* were statistically significant, data were pooled for both host sexes to determine the prevalences and mean intensities of infection for the various helminth species.

Salamanders were killed by decapitation. The sex of each individual was determined. At time of necropsy, the gastrointestinal tract was removed, and the small and large intestines were examined with a dissecting microscope for helminths. Nematodes were initially studied in temporary lactophenol mounts and then stored in 70% ethanol. Voucher specimens representing each helminth species were stained in Semichon's acetic carmine, dehydrated in an ethanol series, cleared in xylene, and mounted in Permount®. The terms *prevalence* and *mean intensity* follow the definitions of Bush et al. (1997).

In the study, 4 helminth species were found in *P. richmondi* individuals (Table 1). The trematode appears to be *Brachycoelium storeriae* Harwood, 1932, a diagnosis based, in part, on the morphological similarity of specimens in this study with the description provided by Cheng (1958), who argued convincingly for the separation of this trematode species from *Brachycoelium salamandrae* (Frolich, 1789). The diagnosis of *B. storeriae* from the terrestrial *P. richmondi* in West Virginia can be supported on an ecological basis as well. For example, Parker (1941) identified trematodes of this species from *Opheodrys aestivus* (Linnaeus, 1766) and *Ambystoma opacum* (Gravenhorst, 1807), both terrestrial hosts. Cheng (1958) also collected *B. storeriae* individuals from *Plethodon cinereus* (Green, 1818), another terrestrial host species. *Brachycoelium storeriae* has also been reported from *Pseudotriton ruber* (Sonnini, 1802) (Parker, 1941; Dunbar and Moore, 1979), a salamander species considered semiaquatic to semiterrestrial by the latter authors.

The nematode *Angiostoma plethodontis* Chitwood, 1933 found in the present study clearly conforms to its original description (Chitwood, 1933). A total of 22 *A. plethodontis* (13 females and 9 males) was collected from 15 *P. richmondi* (Table 1). This female:male ratio of 1.44:1.00 did not deviate significantly from the expected 1.00:1.00 ratio ($\chi^2 = 0.752$; df = 1; $0.5 > P > 0.1$).

The identification of *B. salamandrae* from *P. richmondi* may not be definitive, because all 6 individuals of this nematode species collected

were larvae. Still, we have concluded that the oxyuroid species found in this study is most likely *B. salamandrae* (Schad, 1960) Petter and Quentin, 1976 rather than *B. magnavulvaris* (Rankin, 1937) Petter and Quentin, 1976, because Dunbar and Moore (1979) argued that the latter species is not found in terrestrial hosts, such as *P. richmondi.*

Since only 1 acanthocephalan was found and it was in an encapsulated cystacanth stage, we offer no species or generic diagnoses.

This is the first report of *B. storeriae* and *A. plethodontis* from *P. richmondi,* and West Virginia is a new locality record for all helminth species collected. Voucher material is deposited in the United States National Parasite Collection, Beltsville, Maryland 20705, under accession numbers USNPC 88638 (*Angiostoma plethodontis* female and male); USNPC 88639 (*Batracholandros salamandrae*); USNPC 88640 (*Brachycoelium storeriae*); and USNPC 88641 (acanthocephalan cystacanth).

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Literature Cited

Bush, A. O., K. D. Lafferty, J. M. Lotz, and A. W. Shostak. 1997. Parasitology meets ecology on its own terms: Margolis et al. revisited. Journal of Parasitology 83:575–583.

Cheng, T. C. 1958. Studies on the trematode family Dicrocoeliidae. I. The genus *Brachycoelium* (Dujardin, 1845) and *Leptophallus* Lühe, 1909 (Brachycoeliinae). American Midland Naturalist 59: 67–81.

Chitwood, B. G. 1933. On some nematodes of the superfamily Rhabditoidea and their status as parasites of reptiles and amphibians. Journal of the Washington Academy of Sciences 23:508–520.

Dunbar, J. R., and J. D. Moore. 1979. Correlations of host specificity with host habitat in helminths parasitizing the plethodontids of Washington County, Tennessee. Journal of the Tennessee Academy of Science 54:106–109.

Green, N. B., and T. K. Pauley. 1987. Amphibians and Reptiles in West Virginia. University of Pittsburgh Press, Pittsburgh, Pennsylvania, 241 pp.

Parker, M. V. 1941. The trematode parasites from a collection of amphibians and reptiles. Report of the Reelfoot Lake Biological Station, 5. Journal of the Tennessee Academy of Science 16:27–44.

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Research Note

Abomasal Parasites in Southern Mule Deer (*Odocoileus hemionus fuliginatus*) from Coastal San Diego County, California, U.S.A.

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ABSTRACT: Trichostrongylid nematodes were collected from the abomasa of 15 (6.6%) of 227 southern mule deer (*Odocoileus hemionus fuliginatus*) from Camp Pendleton Marine Corps Base, San Diego County, California (U.S.A.). Three species of nematodes were found: *Haemonchus contortus, Teladorsagia circumcincta,* and *Nematodirus odocoilei.* Mean (±1 SD) intensity was 11.5 ± 24.6 nematodes per infected deer. All 15 infected deer were among the 184 animals shot during 2 controlled hunts in November 1990 and November 1991; no parasites were found in an additional 43 abomasa collected during 2 additional hunts in March 1991 and March 1992. This is the first known published report of abomasal nematodes from southern mule deer.

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Table 1. Abomasal parasites collected from 227 southern mule deer (*Odocoileus hemionus fulginatus*) at Camp Pendleton, San Diego County, California. All deer were collected in the November 1990 and 1991 hunts. No abomasal parasites were observed among the 43 deer sampled in the March 1991 and 1992 hunts.

Parasite species	No. deer infected	Prevalence (%)	No. parasites/deer			Total no. parasites collected
			Mean	SD	Range	
Haemonchus contortus	5	2.2	20.8	37.6	4–88	104
Teladorsagia circumcincta	2	0.9	12.0	0.0	12	24
Nematodirus odocoilei	5	2.2	5.6	3.6	4–12	28
Unknown*	4	1.8	4.0	0.0	4	16
Totals	15†	6.6	11.5	24.6	4–100	172

* Unidentifiable worm fragments were found in 4 additional deer.

† One deer was infected with both *H. contortus* and *T. circumcincta.*

KEY WORDS: southern mule deer, *Odocoileus hemionus fuliginatus,* abomasal nematodes, *Haemonchus contortus, Teladorsagia circumcincta, Nematodirus odocoilei,* California, U.S.A.

A number of studies have been published of abomasal parasites among mule deer (*Odocoileus hemionus* Rafinesque, 1817) of North America, and some controversy exists on the management value of using abomasal parasites as indicators of the physical condition and habitat relationships of these deer (Moore and Garner, 1980; Waid et al., 1985; Stubblefield et al., 1987). Among southern mule deer (*Odocoileus hemionus fuliginatus* Cowan, 1933) no published reports are known of abomasal parasites; on the basis of a single unpublished anonymous 1955 report of the California Department of Fish and Game, *Nematodirus filicollis* (Rudolphi, 1802) Ransom, 1907, was purportedly observed in 1 of 17 southern mule deer from Camp Pendleton Marine Corps Base, San Diego County, California (33°20′N, 117°20″W). Our objective was to identify the prevalence and intensity of abomasal parasites infecting the southern mule deer subspecies on Camp Pendleton.

Camp Pendleton comprises a 50,588-ha area, with riparian and oak woodlands, coastal sage scrub, grassland, and chaparral, in the northwestern corner of San Diego County. We collected 184 abomasa from 225 southern mule deer shot during 2 either-sex hunts in November 1990 and November 1991. An additional 43 abomasa were collected from 45 animals killed during 2 antlerless hunts in March 1991 and March 1992. At hunter-check stations, each abomasum was removed after ligation and frozen at −10°C prior to transportation to the laboratory.

Abomasa were thawed at 18–20°C. Abomasal contents first were rinsed through a 1.9-mm mesh to remove coarse material and then rinsed through a 150-μm mesh. Parasites and other material remaining on the 150-μm mesh were diluted with tap water and examined in 10-ml aliquots until 25% of the total volume for each abomasum was evaluated. From this 25% sample, the total number of each abomasal parasite species was estimated for each deer. Helminths collected were stored in 70% ethanol, mounted on slides in glycerin, and identified to species according to Skrjabin (1952), Durette-Desset (1974), and Levine (1980). Representative samples of all helminths were deposited into the U.S. National Parasite Collection, Beltsville, Maryland (Accession Numbers 84382–84387).

Of the 227 abomasa examined, 212 (93%) had no detectable helminths, 12 (5.2%) had an estimated total of 4 helminths, 2 (0.9%) had an estimated 12 helminths, and 1 (<1%) had an estimated 100 helminths. *Haemonchus contortus* (Rudolphi, 1803) and *Nematodirus odocoilei* (Becklund and Walker, 1967) had the highest prevalence among abomasal parasites at 3%, and *Teladorsagia circumcincta* (Stadelmann, 1894) Drozdz, 1965, had a prevalence of 1% (Table 1). Four of 78 males and 11 of 106 females taken in November hunts were infected; no helminths were found in the 43 (30 female and 13 male) antlerless deer evaluated from the March hunts.

The low prevalences and intensities of abomasal parasites among southern mule deer at Camp Pendleton severely limit their use as indicators of the relationships between the deer

and their habitats. These low parasite levels may have been influenced by several factors. A semiarid climate has been associated with low helminth prevalences (Waid et al., 1985; Stubblefield et al., 1987); at 3 weather stations on Camp Pendleton, annual rainfall ranged from 225 to 483 mm over the 2 yr of the study. Also, in an earlier study, Pious (1989) found that grasses comprised only 9% of the diet for deer at Camp Pendleton; low level of grass intake may reduce the likelihood of deer ingesting infective nematode larvae. Another factor is that during the unavoidable time lapse between killing the deer and collecting the abomasa, some abomasal parasites may have migrated out of the abomasum or some parasites (e.g., *Nematodirus odocoilei*) may have migrated into the abomasum. In addition, basing prevalence on the number of parasites found in only 25% of each abomasum could have resulted in overlooking very low intensities. Further, use of the 150-μm mesh for collecting parasites may have resulted in loss of small parasites, especially larvae. Finally, the frequent fires from incendiary devices on Camp Pendleton could serve to reduce the abundance of infective larvae on vegetation. Thus, the parasite prevalences and intensities we observed probably should be considered minimum values for this southern mule deer population.

The apparent absence of abomasal parasites from the 43 deer killed in the 2 March hunts is interesting. This phenomenon may be related to the development of a seasonal host immunity against intestinal parasites (Soulsby, 1966).

Haemonchus contortus and *T. circumcincta* both are common parasites of sheep. Camp Pendleton has had a history of grazing by sheep, cattle, and bison.

Although all of these parasites have been reported from other subspecies of mule deer, this is the first published report for the southern mule deer subspecies. The unpublished anonymous 1955 California Department of Fish and Game report of *N. filicollis* in 1 (6%) of 17 abomasa evaluated at Camp Pendleton reported a prevalence of abomasal parasites comparable with that found in our study (Table 1). Walker and Becklund (1970) noted that they examined many specimens of *N. filicollis* collected from deer and in every case reidentified them as *N. odocoilei*; thus, the original unpublished report probably also was of *N. odocoilei*. Finding parasite species characteristic of other mule deer subspecies (Walker and Becklund, 1970) among *O. hemionus fuliginatus* supports the notion that these abomasal parasites exercise little selectivity among mule deer subspecies. No clinical pathological lesions were associated with any of the infected deer in this study.

We greatly appreciate the assistance of Dr. Archie Mossman and Ms. Denise Bradley for help in several phases of this study and of Dr. John DeMartini and Dr. J. Ralph Lichtenfels for assistance in identifying the parasites.

Literature Cited

Durette-Desset, M. 1974. Keys to the genera of the superfamily Trichostrongyloidea. *In* R. C. Anderson and A. G. Chabaud, eds. Commonwealth Institute of Helminthology Number 10. Commonwealth Institute of Helminthology, The White House, St. Albans, England. 86 pp.

Levine, N. D. 1980. Nematode Parasites of Domestic Animals and of Man, 2nd ed. Burgess Publishing Company, Minneapolis, Minnesota. 477 pp.

Moore, G. M., and G. N. Garner. 1980. The relationship of abomasal parasite counts to physical condition of mule deer in southwestern Texas. Western Association of Fish and Wildlife Agencies 60:593–600.

Pious, M. 1989. Forage composition and physical condition of southern mule deer in San Diego County, California. M.S. Thesis, Humboldt State University, Arcata, California. 61 pp.

Skrjabin, K. I. 1952. Key to the Parasitic Nematodes. Volume III: Strongylata. Academy of the Sciences of the U.S.S.R. Translated from Russian for the National Science Foundation, Washington, D.C., and the U.S. Department of Agriculture by the Israel Program for Scientific Translations, The Office of Technical Services. 1961. U.S. Department of Commerce, Washington, D.C. 434 pp.

Soulsby, E. J. L. 1966. The mechanisms of immunity to gastro-intestinal nematodes. Pages 255–276 *in* E. J. L. Soulsby, ed. Biology of Parasites. Academic Press, New York. 354 pp.

Stubblefield, S. S., D. B. Pence, and R. J. Warren. 1987. Visceral helminth communities of sympatric mule deer and white-tailed deer from the Davis Mountains of Texas. Journal of Wildlife Diseases 23:113–120.

Waid, D. D., D. B. Pence, and R. J. Warren. 1985. Effects of season and physical condition on the gastrointestinal helminth community of white-tailed deer from the Texas Edwards Plateau. Journal of Wildlife Diseases 21:264–273.

Walker, M. L., and W. W. Becklund. 1970. Checklist of the internal and external parasites of deer, *Odocoileus hemionus* and *O. virginianus* in the United States and Canada. Index-Catalogue of Medical and Veterinary Zoology, Special Publication No. 1. U.S. Department of Agriculture, Washington, D.C. 45 pp.

Comp. Parasitol.
67(1), 2000 pp. 138–142

THE HELMINTHOLOGICAL SOCIETY OF WASHINGTON CONSTITUTION AND BY-LAWS

The name of the Society shall be the Helminthological Society of Washington.

The object of the Society shall be to provide for the association of persons interested in parasitology and related sciences for the presentation and discussion of items of interest pertaining to those sciences.

BY-LAWS

ARTICLE 1
Membership

Section 1. There shall be four classes of members, namely **regular**, **life**, **honorary**, and **emeritus**.

Section 2. Any person interested in parasitology or related sciences may be elected to **regular membership** in the Society. The privileges and responsibilities of regular members include eligibility to hold office, to vote, and to receive Society publications. Spouses of regular members may apply for election to regular membership with all the privileges and responsibilities except that they will not receive the Society publications and will pay annual dues at a reduced rate.

Section 3. Any person who has rendered conspicuous and continuous service as a member of the Society for a period of not less than 15 years, and has reached the age of retirement, may be elected to **life membership**. Life members shall have all of the privileges of regular members but shall be exempted from payment of dues. The number of life members shall not exceed five percent of the membership at the time of election.

Section 4. Any person who has attained eminent distinction in parasitology or related sciences may be elected to **honorary membership**. An honorary member shall have all the privileges of membership except voting, holding office, or having any interest in the real or personal property of the Society, and shall be exempted from the payment of dues. The number of honorary members shall not exceed 10 at any one time, and not more than one honorary member shall be elected in any one year.

Section 5. Any person who has been a member in good standing for not less than 10 years, and who has retired from active professional life, may upon application in writing to the Corresponding Secretary-Treasurer have the membership status changed to emeritus. An **emeritus member** shall be exempt from payment of dues and, with exception of receiving the Society's publications shall enjoy all privileges of membership. An emeritus member, upon payment of 75 percent of the current dues, may elect to receive the Society's publications.

Section 6. Candidates for election to regular membership may be sponsored and proposed only by members in good standing. The candidate shall submit a duly executed and signed application to the Recording Secretary, who in turn shall submit the application to the Executive Committee. The Executive Committee shall review the application, vote upon its acceptance, and report its actions at the next scheduled business meeting. Voting may be by voice or by ballot. In the event that there is no scheduled Executive Committee meeting within 60 days of receipt of the application, voting may be conducted by mail or by other expeditious means of communication. The Corresponding Secretary-Treasurer shall inform the applicant of the action of the Committee.

Section 7. Payment of dues shall be considered as evidence of acceptance of membership in the Society. Election to membership shall be void if the person elected does not pay dues within 3 months after the date of notification of election.

Section 8. Nominations for honorary and life membership, approved by the Executive Committee, shall be submitted to the membership for election at a regular meeting.

ARTICLE 2
Officers

Section 1. The officers of the Society shall be a President, a Vice-President, a Recording Secretary, a Corresponding Secretary-Treasurer, and such other officers as the Society may deem necessary. The four named officers shall also be the Directors of the Society. Only members in good standing and whose dues are not in arrears shall

be eligible for election to office. Terms of office shall be for 1 year.

Section 2. The President shall preside over all meetings, appoint all committees except the Executive Committee, and perform such other duties as may properly devolve upon a presiding officer. The President may appoint an Archivist, a Librarian, a Custodian of Back Issues, and an Assistant Corresponding Secretary-Treasurer, as needed.

Section 3. The Vice-President shall preside in the absence of the President and, when so acting, shall perform such duties as would otherwise devolve upon the President. The Vice-President shall serve as Program Officer.

Section 4. In the absence of both President and Vice-President, the member, among those present, who last held the office of President shall be the presiding officer. Under other circumstances, members may elect a presiding officer but business action taken shall be reviewed by the Executive Committee.

Section 5. The Recording Secretary shall record the proceedings of all meetings and shall present at each meeting a written report of the transactions of the preceding meeting, shall keep an accurate and complete record of the business transacted by the Society in its meetings, and shall notify the Corresponding Secretary-Treasurer of the election of new members. The Recording Secretary shall prepare for publication in the Society's publications an annual digest of scientific meetings and business transacted, including elections of officers and new members.

Section 6. The Corresponding Secretary-Treasurer shall be responsible for all funds, collections, payment of bills, and maintenance of financial records. At the beginning of each year, the Corresponding Secretary-Treasurer shall present to the Society an itemized statement of receipts and expenditures of the previous year; this statement shall be audited by at least two members of the Society.

ARTICLE 3
Executive Committee

Section 1. There shall be an Executive Committee that shall be the administrative body of the Society.

Section 2. The Executive Committee shall consist of 10 members in good standing as follows: the President, Vice-President, Recording Secretary, Corresponding Secretary-Treasurer, Editor, Immediate Past President, and four Members-at-Large. The Committee shall represent to the fullest practicable degree the varied scientific interests of the Society's membership and the local distribution of its members.

Section 3. The President shall serve as chairperson of the Executive Committee.

Section 4. Members-at-Large shall serve for a term of 2 years. Two Members-at Large shall be appointed each year in November by the President-elect for the prescribed term of 2 years.

Section 5. Vacancies occurring on the Executive Committee for any reason shall be filled by appointment by the President, and except as otherwise provided, the appointee shall serve for the remainder of the unexpired term.

Section 6. The Executive Committee shall carry out the provisions of the Constitution and By-Laws and shall make decisions on all matters of general and financial policy not otherwise set forth in the Constitution and By-Laws and shall report its action to the Society annually at the last regular meeting.

Section 7. The Executive Committee shall approve the selection of a depository for the current funds, direct the investment of the permanent funds, and act as the administrative body of the Society on all matters involving finance. It shall prepare and present to the Society at the beginning of each calendar year a budget based on the estimated receipts and expenditures of the coming year with such recommendations as may be desirable.

Section 8. With the presentation of the annual budget, the Executive Committee shall present to the Society, if feasible, the estimated cost for publication to be charged to contributors to the Society's publications for that year.

Section 9. Costs of publication, in excess of amounts borne by the Society, shall be borne by the authors in accordance with guidelines established by the Executive Committee.

Section 10. The Executive Committee shall pass on all applications for membership and on the reinstatement of delinquent members, except as otherwise provided, and shall report its actions to the Society.

ARTICLE 4
Nomination and Election of Officers

Section 1. The Executive Committee, acting as the Nominating Committee of the Society, shall prepare a slate of officers and present this

to the Society at the Anniversary meeting of each year. Independent nominations may be made in writing by any five members. In order to receive consideration, such nominations must be in the hands of the Recording Secretary at the time of the Anniversary meeting. The election will be held at the next regular meeting of the Society.

Section 2. The election of officers shall be held prior to the presentation of notes and papers at the last regular meeting of the calendar year. Voting may be either by voice or by ballot.

Section 3. The last order of business at the last meeting of the calendar year shall be the installation of officers, the naming of officers, and the naming of necessary appointees.

ARTICLE 5
Awards Committee

Section 1. There shall be an Awards Committee to select individuals for special commendation. The Committee shall consist of three members.

Section 2. Members shall serve for a term of 3 years with appointments staggered so that one new member is added each year. The senior member of the Committee shall serve as chairperson.

Section 3. The Awards Committee shall be charged with the duty of recommending candidates for the Anniversary Award, which may be given annually or less frequently at the discretion of the Committee.

Section 4. The recipient of the Anniversary Award shall be, or have been, a Society member who is honored for one or more achievements of the following nature: (a) outstanding contributions to parasitology or related sciences that bring honor and credit to the Society, (b) an exceptional paper read at a meeting of the Society or published in *Comparative Parasitology,* (c) outstanding service to the Society, and (d) other achievement or contribution of distinction that warrants highest and special recognition by the Society.

Section 5. The individual recommended for the Anniversary Award shall be subject to approval by the Executive Committee.

ARTICLE 6
Editorial Board

Section 1. There shall be an Editorial Board for the Society's publications, which shall include *Comparative Parasitology.*

Section 2. The Editorial Board shall consist of an Editor and other members in good standing, representing to the fullest practicable degree the varied scientific interests and the employment-group affiliations of the Society's membership.

Section 3. The Editor shall be elected by the Society, on the nomination by the Executive Committee, for a term of 3 to 5 years.

Section 4. Other members of the Editorial Board shall be appointed for terms of 3 years.

Section 5. The Editor, after consulting with the Editorial Board, shall appoint new members, formulate publication policies, and make all decisions with respect to format and content of the Society's publications. The Editor shall operate within financial limitations determined by the Executive Committee.

ARTICLE 7
Publications

The publications of the Society shall be issued at such times and in such form as the Society, through its Editorial Board, may determine.

ARTICLE 8
Meetings

Section 1. Meetings of the Society shall be held as often as deemed desirable by the Executive Committee.

Section 2. The November meeting of the Society shall be known as the Anniversary meeting, and the Anniversary Award, when made, ordinarily shall be presented at this meeting.

Section 3. Notice of the time and place of meetings shall be given by the Recording Secretary at least 10 days before the date of the meeting.

ARTICLE 9
Procedure

The rules contained in *Robert's Rules of Order,* Revised, shall govern the Society in all cases to which they are applicable and in which they are not inconsistent with the By-Laws or the special rules of order of the Society.

ARTICLE 10
Order of Business

Call to order.
Reading of minutes of the previous meeting.
Announcement of new members.
Reports of committees.
Unfinished business.

New business.
Presentation of notes and papers.
Installation of new officers.
Adjournment.

ARTICLE 11
Quorum

The members in attendance at any regular meeting shall constitute a quorum.

ARTICLE 12
Dues and Debts Owed to the Society

Section 1. Annual dues for regular and spouse members shall be fixed by the Executive Committee, subject to ratification by the Society. Spouse members shall pay dues at a reduced rate.

Section 2. The fiscal year for payment of dues and for all other business purposes shall be the same as the calendar year, that is, from 1 January to 31 December, and dues shall be payable on or before 1 January. The dues of a newly elected member paid prior to 1 July of the year of the new member's election shall be credited to that year; if paid after 1 July, they shall be credited either to the current fiscal year or to the following one, at the option of the new member. The dues shall include subscription to the Society's publications; only those members whose dues are paid shall receive the publication(s).

Section 3. All other obligations owed to the Society by members or nonmembers shall be due and payable 30 days after bills are rendered; the further extension of credit to those whose obligations are in arrears shall be a matter for decision by the Executive Committee.

ARTICLE 13
Suspension and Reinstatement

Any member whose dues are in arrears for 2 years shall be dropped from membership. Members who have been dropped for nonpayment of dues may be reinstated automatically upon payment of the dues in arrears and dues for the current year or may be otherwise reinstated by action of the Executive Committee.

ARTICLE 14
Provision for Dissolution of Funds

In the event the Society is disbanded, all monies shall be presented to the Trustees of the Brayton Howard Ransom Memorial Trust Fund to be used for such purposes as that continuing body may deem advisable.

ARTICLE 15
Amendments to the By-Laws

Any amendment to these By-Laws shall be presented in writing at a regular meeting. It shall not be acted upon until the following meeting. A two-thirds vote of the members in attendance shall be required for adoption.

ARTICLES OF INCORPORATION
OF
THE HELMINTHOLOGICAL SOCIETY OF WASHINGTON, INC.
(A Non-stock Corporation)

FIRST: I the undersigned, Charles A. Dukes, Jr., whose post office address in 300 Landover Mall West, Landover, Maryland 20785, being at least twenty-one years of age, do hereby form a corporation under and by virtue of the General Laws of the State of Maryland.

SECOND: The name of the corporation (which is hereinafter called the Corporation) is The Helminthological Society of Washington, Inc.

THIRD: The purposes for which the Corporation is formed are as follows:

(a) To provide for the association of persons interested in parasitology and related sciences for the presentation and discussion of items of interest pertaining to these sciences;

(b) To advance the science of parasitology, in both its fundamental and its economic aspects; to act as an agency for the exchange of information; to hold regular meetings and to promote and extend knowledge in all phases of parasitology;

(c) And to generally carry out any other business in connection therewith not contrary to the laws of the State of Maryland, and with all the powers conferred upon non-profit corporations which are contained in the General Laws of the State of Maryland.

FOURTH: The post office address of the principal office of the Corporation in this state is 9110 Drake Place, College Park, Maryland 20740. The name and post office address of the resident agent of the Corporation in this state is A. Morgan Golden, 9110 Drake Place, College Park, Maryland 20740. Said resident agent is a citizen of the State of Maryland and actually resides therein.

FIFTH: The Corporation is not authorized to issue capital stock.

SIXTH: The number of directors of the Corporation shall be four, which number may be increased or decreased pursuant to the By-Laws of the Corporation, but shall never be less than three; and the names of the directors who shall act until their successors are duly chosen and qualified are Nancy D. Pacheco, Louis S. Diamond, Sherman S. Hendrix, and Milford N. Lunde.

SEVENTH: The duration of the Corporation shall be perpetual.

IN WITNESS WHEREOF, I have signed these Articles of Incorporation on the 3rd day November, 1981. I acknowledge these Articles and this signature to by my act.

WITNESS:

[signed]	[signed]
Gary Greenwald	Charles A. Dukes, Jr.

APPLICATION FOR MEMBERSHIP
in the
HELMINTHOLOGICAL SOCIETY OF WASHINGTON

(Please Type or Print Legibly)

Name: _____

Mailing Address: _____

_____

_____

_____

Present Position and Name of Institution: _____

_____

Phone: _____ FAX: _____

E-Mail: _____

Highest Degree Earned and the Year Received: _____

Are You a Student? If so, for what degree and where? _____

_____

Fields of Interest: _____

_____

_____

If you are experienced in your field, would you consent to be a reviewer for manuscripts submitted for publication in the Society's journal, ***Comparative Parasitology***? If so, what specific subject area(s) do you feel most qualified to review?

_____

_____

_____ _____

Signature of Applicant Date

_____ _____

Signature of Sponsor (a member) Date

Mail the completed application along with a check (on a U.S. bank) or money order (in U.S. currency) for the first year's dues (US$25 for domestic active members and US$28 for foreign active members) to the Corresponding Secretary-Treasurer, Nancy D. Pacheco, 9708 DePaul Drive, Bethesda, MD, U.S.A. 20817

Helminthological Society of Washington Home Page: http://www.gettysburg.edu/~shendrix/helmsoc.html

THE HELMINTHOLOGICAL SOCIETY OF WASHINGTON

MISSION AND VISION STATEMENTS
May 7, 1999

THE MISSION

The **Helminthological Society of Washington,** the prototype scientific organization for parasitological research in North America, was founded in 1910 by a devoted group of parasitologists in Washington, D.C. Forging a niche in national and international parasitology over the past century, the **Society** focuses on comparative research, emphasizing taxonomy, systematics, ecology, biogeography, and faunal survey and inventory within a morphological and molecular foundation. Interdisciplinary and crosscutting, comparative parasitology links contemporary biodiversity studies with historical approaches to biogeography, ecology, and coevolution within a cohesive framework.

Through its 5 meetings in the Washington area annually, and via the peer reviewed ***Comparative Parasitology*** (continuing the ***Journal of the Helminthological Society of Washington*** in its 67th Volume), the **Society** actively supports and builds recognition for modern parasitological research. Taxonomic diversity represented in the pages of the ***Society's journal*** treats the rich helminth faunas in terrestrial and aquatic plants, invertebrates, and vertebrates, as well as parasitic protozoans and arthropods. Parasitology, among the most integrative of the biological sciences, provides data critical to elucidation of general patterns of global biodiversity.

THE VISION

The **Helminthological Society of Washington** celebrates a century of tradition and excellence in global parasitology, solving challenges and responding to opportunities for the future of society and the environment.

Members of the **Helminthological Society of Washington** contribute to understanding and protecting human health, agriculture, and the biosphere through comparative research emphasizing taxonomy, systematics, ecology, biogeography, and biodiversity assessment of all parasites. The **Society** projects the exceptional relevance of its programs to broader research and education in global biodiversity and conservation biology through the activities of its members and its journal, ***Comparative Parasitology.***

ANNIVERSARY AWARD RECIPIENTS

*Edna M. Buhrer	1960	*O. Wilford Olsen	1980
*Mildred A. Doss	1961	*Frank D. Enzie	1981
*Allen McIntosh	1962	Lloyd E. Rozeboom	1982
*Jesse R. Christie	1964	*Leon Jacobs	1983
*Gilbert F. Otto	1965	Harley G. Sheffield	1984
*George R. LaRue	1966	A. Morgan Golden	1985
*William W. Cort	1966	Louis S. Diamond	1986
*Gerard Dikmans	1967	*Everett L. Schiller	1987
*Benjamin Schwartz	1969	Milford N. Lunde	1988
*Willard H. Wright	1969	J. Ralph Lichtenfels	1989
*Aurel O. Foster	1970	A. James Haley	1990
*Carlton M. Herman	1971	*Francis G. Tromba	1991
*May Belle Chitwood	1972	Thomas K. Sawyer	1992
*Elvio H. Sadun	1973	Ralph P. Eckerlin	1993
E. J. Lawson Soulsby	1974	Willis A. Reid, Jr.	1994
David R. Lincicome	1975	Gerhard A. Schad	1995
Margaret A. Stirewalt	1975	Franklin A. Neva	1996
*Leo A. Jachowski, Jr.	1976	Burton Y. Endo	1997
*Horace W. Stunkard	1977	Sherman S. Hendrix	1998
*Kenneth C. Kates	1978	Frank W. Douvres	1999
*Everett E. Wehr	1979		

HONORARY MEMBERS

*George R. LaRue	1959	E. J. Lawson Soulsby	1990
*Vladimir S. Ershov	1962	Roy C. Anderson	1991
*Norman R. Stoll	1976	Louis Euzet	1992
*Horace W. Stunkard	1977	John C. Holmes	1993
*Justus F. Mueller	1978	Purnomo	1994
John F. A. Sprent	1979	Naftale Katz	1995
Bernard Bezubik	1980	*Robert Traub	1996
Hugh M. Gordon	1981	*Alan F. Bird	1997

CHARTER MEMBERS 1910

*W. E. Chambers	*Philip E. Garrison	*Maurice C. Hall	*Charles A. Pfender
*Nathan A. Cobb	*Joseph Goldberger	*Albert Hassall	*Brayton H. Ransom
*Howard Crawley	*Henry W. Graybill	*George F. Leonard	*Charles W. Stiles
*Winthrop D. Foster			

LIFE MEMBERS

*Maurice C. Hall	1931	*Everett E. Wehr	1977
*Albert Hassall	1931	Marion M. Farr	1979
*Charles W. Stiles	1931	*John T. Lucker, Jr.	1979
*Paul Bartsch	1937	George W. Luttermoser	1979
*Henry E. Ewing	1945	*John S. Andrews	1980
*William W. Cort	1952	*Leo A. Jachowski, Jr.	1981
*Gerard Dikmans	1953	*Kenneth C. Kates	1981
*Jesse R. Christie	1956	*Francis G. Tromba	1983
*Gotthold Steiner	1956	A. James Haley	1984
*Emmett W. Price	1956	*Leon Jacobs	1985
*Eloise B. Cram	1956	*Paul C. Beaver	1986
*Gerald Thorne	1961	*Raymond M. Cable	1986
*Allen McIntosh	1963	Harry Herlich	1987
*Edna M. Buhrer	1963	Glenn L. Hoffman	1988
*Benjamin G. Chitwood	1968	Robert E. Kuntz	1988
*Aurel O. Foster	1972	Raymond V. Rebois	1988
*Gilbert F. Otto	1972	Frank W. Douvres	1989
*Theodor von Brand	1975	A. Morgan Golden	1989
*May Belle Chitwood	1975	Thomas K. Sawyer	1989
*Carlton M. Herman	1975	*J. Allen Scott	1990
Lloyd E. Rozeboom	1975	Judith H. Shaw	1990
*Albert L. Taylor	1975	Milford N. Lunde	1991
David R. Lincicome	1976	*Everett L. Schiller	1991
Margaret A. Stirewalt	1976	Harley G. Sheffield	1991
*Willard H. Wright	1976	Louis S. Diamond	1994
*Benjamin Schwartz	1976	Mary Hanson Pritchard	1994
*Mildred A. Doss	1977		

*Deceased.

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