Fisheries
American Fisheries Society • www.fisheries.org
VOL 38 NO 6
JUNE 2013

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03632415(2013)38(6)

Fisheries

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262 A colony of the federally threatened Tulotoma attached to the underside of a small boulder from lower Choccolocco Creek, Talladega County, Alabama. Inset shows a large colony on the underside of a boulder from the lower Coosa River, Elmore County, Alabama. Photo Credit: Paul Johnson.

263 Pleated Juga *Juga plicifera* from the Willamette River near Corvallis, Benton County, Oregon. The Pleated Juga is distributed in the Pacific Northwest from California to British Columbia, Canada. Photo Credit: Thomas Tarpley, ADCNR.

Cover: The federally endangered Plicate Rocksnail, *Leptoxis plicata*, historically occupied more than 700 km of riverine habitat throughout the Black Warrior River Basin in Alabama. Habitat loss through dams, channelization, and water quality problems limited the gastropod's distribution to approximately 34 km of the Locust Fork, north of Birmingham. More information about recovery efforts for this species can be found at fl.biology.usgs.gov/afs_snail/index_draft.html.


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The American Fisheries Society (AFS), founded in 1870, is the oldest and largest professional society representing fisheries scientists. The AFS promotes scientific research and enlightened management of aquatic resources for optimum use and enjoyment by the public. It also encourages comprehensive education of fisheries scientists and continuing on-the-job training.

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Fees include $19 for *Fisheries* subscription.

Nonmember and library subscription rates are $174.

Fisheries (ISSN 0363-2415) is published monthly by the American Fisheries Society; 5410 Grosvenor Lane, Suite 110; Bethesda, MD 20814-2199 Periodicals postage paid at Bethesda, Maryland, and at an additional mailing office. A copy of Fisheries Guide for Authors is available from the editor or the AFS website, www.fisheries.org. If requesting from the managing editor, please enclose a stamped, self-addressed envelope with your request.

Postmaster: Send address changes to Fisheries, American Fisheries Society; 5410 Grosvenor Lane, Suite 110; Bethesda, MD 20814-2199.

Fisheries is printed on 10% post-consumer recycled paper with soy-based printing inks.

Scientific Meetings Are Essential

John Boreman, President

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One of the many positive characteristics of my first graduate advisor at Cornell was his determination to ensure that all of his students had the opportunity to attend scientific meetings sponsored by the American Fisheries Society (AFS) and other professional societies. I remember writing thank-you notes to John Olin, a Cornell trustee, who regularly donated money to our department to support graduate student travel. These meetings were important to my fellow grad students and me because they provided an opportunity to see presentations of cutting-edge work in the fisheries field and interact with grad students and scientists outside our immediate circle. They also provided us with a means of meeting potential employers. On the way to the meetings my major advisor would quiz us on the key fisheries professionals who would likely be attending, what their current research interests were, and how our work could be tied to those interests. By doing this, we were able to strike up an intelligent conversation with them about their work and probe their thoughts about how it might help us in ours. I was able to interact with such notables as Bill Ricker, Ray Beverton, Ken Carlander, Stanislas Snieszko, Carl Hubbs, George Spangler, F. E. J. Frye, and Tom Waters—all fisheries "heroes" in my book.

A philosophy instilled in me as a graduate student, and one that I tried to pass on to my own students, is that scientists should never stop being students. They should maintain a passion for learning that carries them through their professional careers. Learning how other scientists are addressing the same problems you are facing, what techniques they have discovered to facilitate their data analyses that could help you with yours, and even how they present their findings in a coherent and efficient manner are all benefits of attending scientific meetings.

Graduate (and undergraduate) students are now offered a variety of opportunities to qualify for funding support to enable them to attend AFS meetings. It seems that almost every AFS section, chapter, and division has a travel award program aimed at increasing student participation at our annual meetings. Oftentimes, I have found that student presentations are the highlight of the meetings, far outshining those given by fisheries professionals who are well into their careers. I can point to a number of instances where I hired someone or asked them to serve on a committee or review panel based on their impressive presentation at a professional society meeting. If our society considers student participation in our major meetings as a high priority and important for advancing their careers, why are federal and state agencies inhibiting attendance by their fisheries professionals at these very same meetings, deeming them nonessential?

Cutting travel of federal and state employees to scientific meetings is not a new issue confronting AFS and other professional societies. Silly as it seems in hindsight, I remember in the 1990s when only one regional employee of the National Marine Fisheries Service, the regional administrator, was authorized by the agency to attend the AFS Northeastern Division meeting. Many state agencies have been operating for years under the restriction on out-of-state travel for their employees, and for years both state and federal employees have taken personal time or leave without pay and covered their own travel costs, rather than risk falling further behind their fellow professionals on the learning curve by missing a key scientific meeting.

Recently, we have heard the term "budget sequester" bandied about on Capitol Hill. What many people in charge have lost sight of is the fact that the term "sequester" also means to put someone into isolation, exactly what is happening to state and federal fisheries professionals who have been denied authorization to attend scientific meetings. How can agencies expect to hire the best and the brightest scientists, and provide them with a work environment that is conducive to lifelong learning, if interaction with fellow professionals continues to be inhibited? The root of the issue is not the budget but perception. Although reduced budgets have been used as an excuse for restricting attendance at scientific meetings, it also reflects the mindsets of technocrats who have lost touch with the reasons why attendance is important to their employees. To put it bluntly, scientific meetings are not hobnobbing boondoggles.

In my charge to the AFS Electronic Services Advisory Board I asked the members to continue investigating the use of virtual attendance at AFS meetings. There are a number of issues to overcome, besides technological, including developing a means for the speaker and virtual audience to interact; establishing funding streams to support the technology by charging a fair fee for remote registration; and adjusting the venues of future meetings based on the possibility that in-person attendance may drop. If state and federal agencies continue to deem our scientific meetings as "nonessential" travel, perhaps they can contribute staff time and funding support to help the AFS develop the means for virtual attendance. This is just one of the many challenges that lie ahead. See you in Little Rock!

COLUMN
Fish Habitat Connections

What Exactly Is Fish Habitat and Why Must We Care?

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"Fish habitat" is a simple term. We can easily imagine a fish languishing under a log or in a kelp forest, and we can picture a school of forage fish zipping through the water column. We can also grasp that the preferred space for many species might change as the seasons change and the years pass by. But the rest of the story is not quite so simple, mostly because life is more complicated and knowledge is often limited. This month's "Fish Habitat Connections" seeks to demystify those details so we can appreciate the intricacies in the fish habitat world and become more emboldened to serve fish not just as a meal but as they deserve.

Let's begin with semantics. Each fish occupies its preferred niche in the ecosystem. The environmental conditions of that space define the fish's preference at each life stage—water temperature, depth, salinity, flow, bottom type, prey availability, annual cycles, and much more. It is important for us as professionals to place those variables in proper context so that individual fish can survive, fish stocks can flourish, fishery management can succeed, and society can benefit from our nation's waters.

That simplistic summary reflects our hopes, which are complicated by the reality that we know very little about our most basic habitat questions. With luck, we know where fish live throughout their life cycles. But oft times we have few insights into the shifting preferences of each life stage. Even that knowledge is elusive unless we have close observations from multidecadal stock assessments or the insights offered by a healthy fishery. Almost universally, we rarely understand the relationships between fish and their habitat. If a wetland is dredged, how will the local fish populations change over the short and long term? If a dam is breached, will the new hydrological regime support native species or invite invasive species? If an acre is protected or restored, how will the population respond? Will harvests increase?

These issues read like the final program at many an American Fisheries Society (AFS) conference. They have vexed us as a profession for decades. We must manage fisheries with the best available information, scant as it might be. And we must identify our primary needs so that gaps are addressed.

There is also the still-new concept of ecosystem-based approaches. Habitat must be an essential variable in stock assessments, but those analyses must be conducted with an ecosystem in mind. Those perspectives can be as important as data. Without that challenge, we won't even know we have a data gap.

Considering how complex this simple topic can be, and how it reflects human pressures from our coasts to the mountains, it is probably no surprise that we continue to lose habitat function at alarming rates. Along our oceans, marine and estuarine wetland loss was three times higher between 2004 and 2009 than in the previous 5 years (Stedman and Dahl 2008; Dahl 2011). Inland wetland loss is not as severe, but hundreds of rivers representing thousands of river miles are compromised by blockages that prevent fish movement upstream or downstream. The first-ever national fish habitat assessment found that 53% of our estuaries are at high or very high risk of habitat degradation (National Fish Habitat Board 2010). Given those numbers, it is unfortunate that those places provide vital nursery habitats for many of our favorite fish.

As fishery professionals from all disciplines, our assignment is to combine our skills to protect important habitats and restore those that are degraded. Our mission will be slightly less daunting if we and our partners can set a pace to match the steady pressure of human population growth and looming challenges such as climate change. AFS represents an incredible knowledge base. If anyone can analyze our habitat knowledge, fill our priority gaps, apply lessons learned, and improve habitats for the benefit of all, it is us.

Next month we will shift from the nuances of semantics to the harsh realities of the challenge before us. It is imperative that we engage now! Economic and ecological facts urge AFS, its units, each of us, and our home institutions to accept the challenge. We will explain the opportunities before us and how our collective skills are needed for success.

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Conservation Status of Freshwater Gastropods of Canada and the United States

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ABSTRACT: *This is the first American Fisheries Society conservation assessment of freshwater gastropods (snails) from Canada and the United States by the Gastropod Subcommittee (Endangered Species Committee). This review covers 703 species representing 16 families and 93 genera, of which 67 species are considered extinct, or possibly extinct, 278 are endangered, 102 are threatened, 73 are vulnerable, 157 are currently stable, and 26 species have uncertain taxonomic status. Of the entire fauna, 74% of gastropods are imperiled (vulnerable, threatened, endangered) or extinct, which exceeds imperilment levels in fishes (39%) and crayfishes (48%) but is similar to that of mussels (72%). Comparison of modern to background extinction rates reveals that gastropods have the highest modern extinction rate yet observed, 9,539 times greater than background rates. Gastropods are highly susceptible to habitat loss and degradation, particularly narrow endemics restricted to a single spring or short stream reaches. Compilation of this review was hampered by a paucity of current distributional information and taxonomic uncertainties. Although research on several fronts*

Estado de la conservación de los gasterópodos de Canadá y los Estados Unidos de Norteamérica

RESUMEN: *esta es la primera evaluación sobre el estado que guarda la conservación de los gasterópodos (caracoles) de Canadá y los EE.UU., realizada por el Subcomité para los Gasterópodos (Comité de Especies Amenazadas) de la Sociedad Americana de Pesquerías. Esta revisión comprende 703 especies, pertenecientes a 16 familias y 93 géneros, de las cuales 67 se consideran extintas o probablemente extintas; 278 están en peligro, 102 amenazadas, 73 vulnerables, 157 cuentan con poblaciones estables y 26 especies presentan un estado taxonómico incierto. De la totalidad de la fauna, 74% de los gasterópodos se encuentran en alguna categoría de vulnerabilidad (amenazados, en peligro o vulnerables) o extintos, lo cual excede al nivel de amenaza al que está sujeto el grupo de los peces (39%) y los langostinos (48%), pero es similar al de los mejillones (72%). Comparando las tasas de extinción actuales contra las tasas de extinción de fondo en el grupo de los gasterópodos, se tiene que en la actualidad son las más altas registradas: 9,539 veces la tasa de extinción de fondo. Los gasterópodos son altamente susceptibles a la degradación y pérdida de hábitat, en particular aquellas especies endémicas cuya distribución está restringida a un solo manantial o a arroyos pequeños. La compilación realizada para esta revisión se dificultó por la falta de información sobre la incertidumbre en la distribución y taxonomía del grupo. Si bien se necesita desarrollar investigación en distintos frentes como biología básica, fisiología, estrategias de conservación, historias de vida y ecología, se consideran como prioridades la sistemática, curación de colecciones museográficas y bases de datos acopladas con muestreos sistemáticos integrales (para establecer límites geográficos, identificación de amenazas).*

including basic biology, physiology, conservation strategies, life history, and ecology are needed, systematics and curation of museum collections and databases coupled with comprehensive status surveys (geographic limits, threat identification) are priorities.

INTRODUCTION

Freshwater gastropods (snails) are an important and diverse component of aquatic ecosystems worldwide. Gastropods have diversified into every conceivable aquatic habitat, including hypogean aquifers, springs, small streams, large rivers, ponds, lakes, and ephemeral to permanent wetlands. Most graze on periphytic or epiphytic algae and biofilms, though some are suspension or deposit feeders (Brown and Lydeard 2010). Unlike some of their terrestrial or marine counterparts, freshwater gastropods are not predatory (Burch 1989; Brown and Lydeard 2010). Gastropods dominate benthic stream communities in numbers (Hawkins and Furnish 1987; Johnson and Brown 1997) and often exceed 50% of the invertebrate biomass (Brown et al. 2008; Brown and Lydeard 2010). Gastropods are the principal grazers in many aquatic habitats (Huryn et al. 1995) and significantly influence algal primary productivity (e.g., Brown and Lydeard 2010), playing a pivotal role in aquatic food webs and nutrient cycling (Covich et al. 1999).

Gastropods were important dietary components of at least three extinct North American fishes, the Stumptooth Minnow *Stypodon signifier* (Miller et al. 1989), Harelip Sucker *Moxostoma lacerum* (Jenkins 1994), and Maryland Darter *Etheostoma sellare* (Neely et al. 2003). At least three rare fishes are gastropod molluscivores: the Copper Redhorse *Moxostoma hubbsi* (Jenkins and Burkhead 1994), Snail Darter *Percina tanasi* (Haag and Warren 2006), and Pygmy Sculpin *Cottus paulus* (Mettee et al. 1996). Other snail-eating fishes include diverse taxa from the Acipenseridae, Cyprinidae, Catostomidae, Ictaluridae, Centrarchidae, and Percidae (Boschung and Mayden 2004). Tetrapod molluscivores include the Stinkpot *Sternotherus odoratus* (Ford and Moll 2004) and map turtles *Graptemys* species (Cagle 1952; Vogt 1981), Snail Kite *Rostrhamus sociabilis* and Limpkin *Aramus guarauna* (Bourne 1993), and the Muskrat *Ondatra zibethicus* (Neves and Odum 1989).

Rough Hornsnail *Pleurocera foremani*, a federally endangered species from the lower Coosa River at Wetumpka, Elmore County, Alabama. A Coosa River endemic, its historical distribution was reduced by reservoir construction to isolated populations in lower Yellowleaf Creek and the Coosa River at Wetumpka. Photo Credit: Thomas Tarpley, ADCNR.

Native freshwater gastropods of Canada and the United States belong to three main clades: Neritimorpha, Caenogastropoda, and Heterobranchia (Bouchet and Rocroi 2005), representing numerous independent colonizations by marine or terrestrial ancestors (Strong et al. 2008). Most gastropods belong to the Caenogastropoda, which, along with the Neritimorpha, possess an operculum, respire with a gill, mature slowly, and are long-lived dioecious species with internal fertilization, and females generally attach eggs to firm substrates in late spring and early summer. Many species are narrow endemics associated with lotic habitats, often isolated in a single spring, river reach, or geographically restricted river basin. Neritimorpha differ from Caenogastropoda in gill, radula, and male penile morphology and are restricted to coastal river environments. In contrast, freshwater Heterobranchia (Valvatoidea, Pulmonata) are hermaphroditic, mature quickly, and generally have shorter generation times. Valvatoideans possess an external gill, an operculum, and lay small eggs much of the year (Burch 1989). Pulmonates lack both an operculum and gill, respiring with a modified mantle or "lung" (hence "pulmonate"), and lay large, gelatinous egg masses during warm months. Pulmonates are among the most ecologically tolerant snails and are widely distributed in lakes, ponds, rivers, bogs, and ephemeral bodies of water. Pulmonate endemism generally tends to be more pronounced in isolated lakes or springs in Canada and the northern United States (Brown and Lydeard 2010).

This is the first conservation assessment of freshwater gastropods published by the American Fisheries Society (AFS). Previous AFS conservation assessments have tracked freshwater fishes (Deacon et al. 1979; Williams et al. 1989; Jelks et al. 2008), marine fishes (Musick et al. 2000), and crayfishes (Taylor et al. 1996, 2007). Notably, the AFS freshwater mussel assessment by Williams et al. (1993) was a watershed contribution to mussel conservation. Its publication inspired scientific studies on the biology, conservation, and systematics of mussels. At this writing, second revision of mussel assessment is nearly complete (J. D. Williams, Florida Fish and Wildlife Conservation Commission, personal communication). Conservation assessments of mollusks demonstrate that they are among the most imperiled organisms on Earth (Lydeard et al. 2004; Bogan 2006; Lysne et al. 2008; Strong et al. 2008; Vaughn 2010; this assessment).

North America hosts the highest diversity of freshwater crayfishes and mussels in the world, and the gastropod fauna is among the richest (Neves et al. 1997; Bouchet and Rocroi 2005). High imperilment rates among freshwater groups have been repeatedly linked to habitat loss and destruction and introduction of nonindigenous species (Abell 2002; Heinz Center Report 2002; Taylor et al. 2007; Jelks et al. 2008; Lysne et al. 2008; Downing et al. 2010). Collectively, AFS assessments provide an important, contemporary snapshot of the state of the health of North American freshwater environments. These assessments indicate freshwater species have experienced dramatic declines. Estimated extinction rates of North American freshwater species are extraordinarily high (Abell et al. 2000; Master et al. 2000; Burkhead 2012b), nearing extinction rates observed in tropical rain forests, the greatest rate on the globe (Ricciardi and Rasmussen 1999).

PATTERNS OF IMPERILMENT

Each of the major freshwater gastropod clades evolved unique suites of anatomical features, life history traits, physiological tolerances, and ecological specialization. The rapid anthropogenic transformation of primarily riverine habitats exposed gastropods to degrees of change that simply exceed tolerances evolved over millions of years. For example, caenogastropods are slow maturing, often iteroparous, and geographically restricted, with narrow ecological tolerances; hence, many species are highly sensitive to habitat degradation. Rapid environmental changes have resulted in significant population reductions and a phenomenal number of extinctions. Sensitive species with small distributions are most susceptible to extinction (Pimm et al. 2006). The loss of a single spring can result in extinction of more than one endemic species. For example, repeated desiccation of Big Spring in Huntsville, Alabama, resulted in the demise of the Olive Marstonia *Marstonia olivacea* and the Whiteline Topminnow *Fundulus albolineatus* (Miller et al. 1989; Burkhead 2012b).

In systems with exceptionally high endemism such as the Tennessee and Mobile River basins, extensive conversion of flowing river mainstems into impoundments resulted in extraordinary species loss. The most renowned example represents the largest single modern extinction event in North America. From 1914 to 1964, 34 species and at least three genera were driven to extinction by a succession of impoundments on the Coosa River (Bogan et al. 1995; Neves et al. 1997; Lydeard et al. 2004; Ó Foighil et al. 2011). The surviving species persist as fragmented populations isolated by impoundments and are highly vulnerable to localized disturbances.

THREATS

Previous AFS assessments (Williams et al. 1993; Taylor et al. 2007; Jelks et al. 2008) and other reviews (Neves et al. 1997; Strayer and Dudgeon 2010; Downing et al. 2010) provide thorough summaries of threats to aquatic habitats and species. Causes of habitat degradation and gastropod species loss include dams, impounded reaches, tailrace modifications (temperature, dissolved oxygen [DO], discharge alterations), channelization, erosion, excessive sedimentation (of fines), groundwater withdrawal, and associated impacts on surface streams (flows, temperature, DO), multiple forms of pollution (salts, metals particularly Cu, Hg, Zn, untreated sewage, agricultural runoff), and invasive species.

The vast majority of extinct freshwater gastropods (92.5%) were narrow endemics, with highly restricted ranges, occurring in a single river, spring, or lake. Habitat destruction in medium to large rivers caused by damming and channelization contributed to most extinctions (45 species, 67% of total), followed by drainage or diversions of lakes (8 species, 12%), alteration of springs (4 species, 6%), and possibly effects of exotic fish introduction (2 species, 3%). Only five species with historical distributions spanning multiple water bodies are extinct. Loss of rare and localized, predominantly endemic species is the prevailing pattern of modern extinctions (Pimm et al. 1995; Burkhead 2012b).

There is a paucity of toxicological data for snails, but recently recognized threats to freshwater mussels include ammonia, endocrine disruptors, and herbicide surfactants (Grabarkiewicz and Davis 2008). However, formal toxicity testing with freshwater gastropods, particularly caenogastropods, lags behind studies for other freshwater organisms (Besser et al. 2009). Caenogastropods show increased sensitivity to copper, ammonia, and pentachlorophenol in comparison to ubiquitous heterobranchs (Besser et al. 2009). The near absence of basic information on the physiological and environmental tolerances for freshwater mollusks (e.g., respiratory adaptations to temperature and pH tolerances) limits our understanding of toxicity risks (Grabarkiewicz and Davis 2008). Toxicology research would provide data necessary for development of specific conservation and recovery criteria (Abell 2002).

ASSESSMENT GOALS

The current knowledge of freshwater gastropods lags behind that of North American freshwater fishes and mussels and crayfishes from Canada and the United States (e.g., Williams et al. 1993; Taylor et al. 2007; Jelks et al. 2008). Due to a paucity of recent survey data, it is only possible at this time to provide a current list of gastropods from Canada and the United States, with provisional lists of species by state and provincial boundaries. We hope that this assessment attracts students to study freshwater gastropods: there are many species yet to be described (Hershler and Liu 2012), and even basic biological information is lacking for most taxa. Considering strong evidence of decline and extinction, the need for surveys and biological

Smooth Mudalia *Leptoxis virgata* from the Hiwassee River near Ducktown, Polk County, Tennessee. This species remains confined to a few Tennessee River system tributaries in the vicinity of Chattanooga, Tennessee. Photo Credit: Thomas Tarpley, ADCNR.

Marsh Ramshorn *Planorbella trivolvis* from hatchery ponds at the Alabama Aquatic Biodiversity Center in Perry County, Alabama. This species is broadly distributed throughout Canada and the United States. Photo Credit: Thomas Tarpley, ADCNR.

studies is exigent. Therefore, the major goals of this first assessment are to

1. update Turgeon et al. (1998) by adding newly described taxa and taxonomic revisions;
2. compile lists of species by state and province;
3. assign a conservation status to each species;
4. compile essential references on distribution, biology, and conservation status;
5. provide a brief description of each family;
6. identify future research and management needs;
7. provide examples of conservation success stories; and
8. create a companion online site where additional information will be provided, including additional success stories and images of gastropod species.

METHODS AND DEFINITIONS

This review provides an updated comprehensive list of 703 native gastropods from Canada and the United States, divided among 16 families and 93 genera, following family classification of Bouchet and Rocroi (2005) with minor modifications (e.g., Albrecht et al. 2007; Strong and Köhler 2009; Wilke et al. 2001). This list was derived from Turgeon et al. (1998) and updated with subsequently described species and systematic revisions. Subspecies are not recognized. Species occurrences within provincial and state boundaries were generated using primary literature, including provincial and state checklists where available, as well as personal communications with professionals who are knowledgeable about certain groups or regions. Although outside continental North America, Hawaiian species are included as in previous AFS fish assessments (Deacon et al. 1979; Williams et al. 1989).

Status Definitions

The following listing criteria were adopted from previous AFS lists (Taylor et al. 2007; Jelks et al. 2008). Status categories were developed by the AFS Endangered Species Committee.

Endangered (E): A species that is in imminent danger of extinction.
Threatened (T): A species that is imminently likely to become endangered throughout all or a significant portion of its range.
Vulnerable (V): A species that is imminently likely to become threatened throughout all or a significant portion of its range; equivalent to "Special Concern" as designated by Deacon et al. (1979) and Williams et al. (1989).
Currently Stable (CS): Species populations not currently at risk.
Extinct (X): A taxon for which no living individual has been documented in nature for 50 or more years despite repeated efforts to do so.
Possibly Extinct (X_p): A taxon that is suspected to be extinct as indicated by more than 20 but less than 50 years since last observed in nature.
Unknown (U): A taxon in which the conservation or taxonomic status is unknown.

To facilitate direct comparisons with state natural heritage programs and Canadian conservation data centers, G-ranks, as developed by The Nature Conservancy and NatureServe (Master et al. 2009), were also included. This system ranks taxa on a scale from 1 to 5 based on estimated number of population occurrences, as follows:

G1 = critically imperiled (at very high risk of extinction or elimination due to extreme rarity, very steep declines, or other factors)
G2 = imperiled (at high risk of extinction or elimination due to very restricted range, very few populations or occurrences, steep declines, or other factors)
G3 = vulnerable (at moderate risk of extinction or elimination due to a restricted range, relatively few populations or occurrences, recent and widespread declines, or other factors)
G4 = apparently secure (uncommon but not rare; some cause for long-term concern due to declines or other factors)
G5 = secure (common; widespread and abundant)
GX = presumed extinct (not located despite intensive searches and virtually no likelihood of rediscovery)
GH = possibly extinct (known from historical occurrences but still some hope of rediscovery)
GU = Unable to assign rank due to taxonomic uncertainty or incomplete distributional information (Master et al. 2009)

Both the AFS and G-rank criteria are based on occurrence data and status evaluation is independent of geopolitical boundaries. However, this review does not utilize the same formal criteria required to list a species under the U.S. Endangered Species Act of 1973. A species may be rare because of a naturally restricted range but may not qualify for protection under the

Endangered Species Act if specific threats to its continued existence are not imminent. In Canada, the Committee on the Status of Endangered Wildlife in Canada began to consider mollusks for listing in 1995. The Species at Risk Act designated the Committee on the Status of Endangered Wildlife in Canada as the official assessor of conservation status in Canada. Canadian status assessment criteria were in use by November 2001 and are based on the revised International Union for Conservation of Nature (IUCN) Red List categories (IUCN 2001).

Because the approximate number of extinct gastropods is known, we can estimate modern to background extinction rates (M:BER) using the method described by Burkhead (2012b) but as corrected by Stuart Pimm (S. Pimm, Duke University, personal comunication; see corrigendum in Burkhead 2012a). The calculation of an M:BER ratio is similar to that of extinctions per million species years (Pimm et al. 1995, 2006), except that the mean species duration interval reported for gastropods—one extinction per 10 million years (Stanley 1985)—is used as the background extinction rate. To estimate M:BER, the sum of species-years—that is, the cumulative total of species described each year multiplied by the years observed from 1758 to the present (each year a species was described)—was determined to be 70,241 (see corrigendum examples in Burkhead 2012a). The extinction rate (or extinctions/species-years) is the number of extinct species (67) divided by the sum of species-years (70,241) = 0.0009539. Multiplying the latter product by the background extinction rate (10 million) = 9,539 M:BER. Hence, modern gastropod extinctions are estimated to be 9,539 times greater than the background extinctions.

At this time, the Mexican gastropod fauna lack comprehensive documentation and only seven hydrobiid species are currently listed as endangered (Secretary of the Environment and Natural Resources of Mexico 2010). Given the pervasiveness of stressors to aquatic habitats in Mexico (Alcocer et al. 2000; Contreras-Balderas et al. 2008; Alcocer and Bernal-Brooks 2010), high levels of aquatic endemism (Dinger et al. 2005), and the effects of human population growth on aquatic habitats, freshwater gastropods of Mexico likely have similar or greater extinction rates than those estimated for Canada and the United States. When it is possible to include Mexican species in the future conservation assessments of North American freshwater gastropods, modern to background extinction rates will certainly be higher.

Caveats

The systematics of most North American gastropod families are poorly understood. Even at higher levels, freshwater gastropod classification is still evolving, as illustrated, for example, by elevation of the pleurocerid subfamily Semisulcospirinae to family rank (Strong and Köhler 2009), the elevation of three hydrobiid subfamilies (Amnicolidae, Cochliopidae, and Lithoglyphidae) to family rank (Wilke et al. 2001), and the subsumation of Ancylidae within Planorbidae (Bouchet and Rocroi 2005; Albrecht et al. 2007). At the species level, systematics is similarly problematic for large portions of the freshwater gastropod fauna. In general, families with species that attain large adult size occurring in eastern North America (e.g., Viviparidae, Pleuroceridae) have historically received the most attention and typically have the most complex taxonomic histories. For example, over 800 nominal species of Pleuroceridae (Graf 2001) have been reduced to 162 species currently considered valid (Burch 1989; Turgeon et al. 1998; Appendix). Ecophenotypic variation along clines or intraspecific variation has led to widespread confusion about species circumscription and the names that should be applied to them (Minton et al. 2008). In contrast, families of small-sized species (e.g., Assimineidae, Cochliopidae, Hydrobiidae, Lithoglyphidae) that remained largely unknown for much of the 19th century now benefit from modern descriptions, including molecular data, detailed anatomical diagnoses, and museum vouchering of type material (e.g., Hershler et al. 2007a). However, knowledge of actual species diversity for even well-researched groups is still incomplete (Hershler and Liu 2012). Modern inventories (within the last 30 years) are lacking for most states and Canadian provinces or territories, leaving large gaps in knowledge of current species distributions. Targeted surveys in Alabama revealed isolated populations of several species previously considered extinct—for example, the Tulotoma *Tulotoma magnifica*, Teardrop Elimia *Elimia lachryma*, Wicker Ancylid *Rhodacmea filosa*, Oblong Rocksnail *Leptoxis compacta*—or critically imperiled species—for example, Cylindrical Lioplax *Lioplax cyclostomaformis* (Hershler et al. 1990; Ó Foighil et al. 2011; Whelan et al. 2012b; P. D. Johnson and J. T. Garner, unpublished data). Museum databases have not kept pace with the rapidly evolving taxonomic landscape and often reflect outdated information. These outdated records can perpetuate identification errors and often result in the extension of species distributions outside known ranges (i.e., false positives).

Olive Nerite *Neritina usnea* from the Blakeley River, Baldwin County, Alabama. This species is broadly distributed in creeks along the Gulf Coast and occasionally ventures into rivers. Photo Credit: Thomas Tarpley, ADCNR.

LIST OF TAXA (APPENDIX)

This compilation includes 703 species, of which 67 are presumed extinct (9.5%), 278 are endangered (39.5%), 102 are threatened (14.5%), 73 are vulnerable (10.4%), 157 are currently stable (22.3%), and another 26 (3.7%) are unknown (Figure 1). Considering that 74% of all species are imperiled or extinct, freshwater gastropods have the highest imperilment level of any taxonomic group evaluated by the AFS. The 74% imperilment rate for gastropods is higher than fishes (39%; Jelks et al. 2008), and crayfishes (48%; Taylor et al. 2007) and similar to the 72% imperilment rate for freshwater mussels (Williams et al. 1993). The complete taxon list is presented in the Appendix.

The Appendix is arranged alphabetically by family, genus, and species. Data for each species include scientific name, taxonomic authority, common name, AFS conservation status (Extinct, Endangered, Threatened, Vulnerable, and Currently Stable), NatureServe status (GX, G1, G2, G3, etc.), and legal status if applicable (online version only). Distribution data are presented in alphabetical order by the two-letter postal code for each state, Canadian province, or territory. In several instances, distributions include extralimital occurrences for native species introduced outside of their known historical ranges (e.g., Ampullariidae, Viviparidae, Lymnaeidae, Physidae). Approximately 30 species from 11 families not native to Canada or the United States (Turgeon et al. 1998) were excluded from this evaluation.

AQUATIC GASTROPOD FAMILIES

The following section is a brief synopsis of diagnostic characters, size range, life history traits, distribution patterns, and conservation summary for the 16 families recognized herein (Table 1). Families are organized alphabetically by clade (Caenogastropoda, Heterobranchia, Neritimorpha).

Caenogastropoda—Ampullarioidea
Ampullariidae—Applesnails

Represented in North America by a single native species, the Florida applesnail (*Pomacea paludosa*; Appendix; Plate 1) is native to southern Alabama, Georgia, and Florida, and introduced in North Carolina (Appendix). It is the largest native North American freshwater gastropod species, often exceeding 60 mm in adult shell length. All members of the family are capable of respiring with both gill and "lung," enabling them to tolerate low DO and prolonged periods of aerial exposure (Burch 1989). Males have a modified section of mantle that forms a penis. Females lay masses containing hundreds of eggs on emergent vegetation and other firm surfaces above the water line from spring to early fall; juveniles drop into the water after hatching (Sharfstein and Steinman 2001). Individuals apparently live several years (Estoy et al. 2002). The Florida Applesnail is the predominant prey in peninsular Florida of the Snail Kite, a federally protected bird (Beissinger 1990). The species is currently considered stable (Appendix).

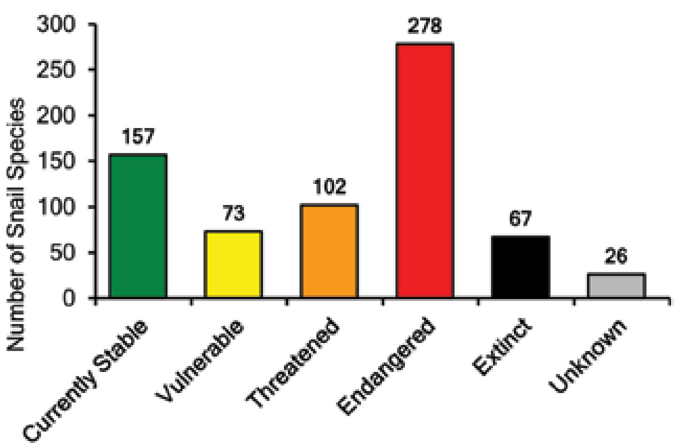

Figure 1. Summary of AFS conservation statuses for freshwater gastropods from Canada and the United States based on species status reviews in the Appendix.

Caenogastropoda—Rissooidea
Amnicolidae—Dusky Pebblesnails

With 18 North American species in four genera, these small gastropods (<5 mm adult shell length) are found in a wide variety of habitats. About 25% of species are restricted to subterranean streams (Appendix; Plate 2).The remaining four species occur predominantly in rivers and creeks in the eastern United States and southeastern Canada. Some appear to graze on algae and biofilm on hard substrates (Kesler 1981). Males have a highly modified penis on the side of the neck, which, as in other Rissooidea, provides the primary diagnostic character used in their identification (Hershler and Ponder 1998). Females typically attach eggs singly to vegetation or other firm surfaces in the spring and early summer (Davis 1961) and generally have a life span of less than 2 years (Servos et al. 1985). With 11 species currently classified as extinct, endangered, threatened, or vulnerable, the family has a 61% imperilment rate.

Caenogastropoda—Rissooidea Assimineidae—Badwater Pebblesnails

This largely marine family is represented in North America by two inland species (<5 mm adult shell length) specifically adapted to moderately saline springs in west Texas and California (Appendix; Plate 2). However, recent molecular work (Hershler and Liu 2008) suggests that there may be at least three undescribed California taxa. Males possess a distinctive penis like other Rissooidea (Hershler et al. 2007b). Life histories of these species are not well known, but a Japanese reed marsh species has a lifespan of 3–5 years (Kurata and Kikuchi 1999). Though some other pebblesnail families have species that occur in saline springs, assimineids are exclusive to these isolated habitats, typically occupying the spring margins and emergent vegetation (Sada 2001). The highly restricted ranges explain the 100% imperilment rate for the family.

Caenogastropoda—Rissooidea Cochliopidae—Tryonia Pebblesnails

Including 48 North American species in 14 genera, these small gastropods (<5 mm adult shell length) are found in many aquatic habitats, including caves, freshwater springs, saline

Table 1. Taxonomic distribution, percentage imperiled, and number of extinct Canadian and United States freshwater gastropods assessed herein. Classification follows Bouchet and Rocroi (2005). The category "Officially listed" lists the number of endangered, threatened, or candidate species formally designated by COSEWIC and the USFWS.

Family	Genera	Species	Percentage imperiled	Number extinct	Officially listed
Ampullariidae	1	1	0	0	0
Amnicolidae	4	18	61	1	0
Assimineidae	1	2	100	0	1
Cochliopidae	14	48	91	0	6
Hydrobiidae	16	185	92	4	14
Lithoglyphidae	11	73	64	4	2
Pleuroceridae	7	162	79	33	8
Pomatiopsidae	1	6	66	1	0
Semisulcospiridae	1	11	91	1	0
Viviparidae	4	21	24	0	3
Neritidae	1	5	60	0	0
Acroloxidae	1	1	100	0	0
Lymnaeidae	9	61	60	10	3
Physidae	5	47	55	1	3
Planorbidae	16	52	44	10	1
Valvatidae	1	10	50	1	0
Total	93	703		67	23

springs, and brackish waters (Appendix; Hershler 2001; Plate 2). Most are highly localized in streams or springs and, consequently, the family has a high imperilment rate (91%). A single widely distributed species that inhabits saline springs, the Saltmarsh Hydrobe *Spurwinkia salsa*, is also known from Canada. The life histories of most species are unknown, but males possess a distinctive penis on the side of the neck (Hershler 2001). Females of some species lay eggs singly on hard substrates (Taylor 1987), and at least one species is parthenogenic (Hershler et al. 2005). Although formal studies are lacking, it is likely that these species have a lifespan of less than 2 years, similar to other hydrobiids. Most are restricted to the southern and western United States, with a single Canadian species (Appendix).

Caenogastropoda—Rissooidea Hydrobiidae—Pyrg Pebblesnails

This is the most diverse North American gastropod family, with 185 species in 14 genera; the genus *Pyrgulopsis* alone contains 124 species (Appendix). Most are very small, <5 mm adult shell length. Typically found in springs, creeks, and small to medium rivers, many are restricted in range, with more than 151 species known from fewer than 10 localities (92% imperilment rate). They reach their highest diversity in the southwestern and southeastern United States, with only five species known from Canada (Appendix; Plate 2). Most species are dioecious, with males possessing a distinctive penis (Hershler and Ponder 1998). Females of several genera lay eggs singly on hard substrates, including the shells of other gastropods (Johnson and Garner, unpublished data). Few detailed life history studies have been completed, but the maximum age of at least one species is 2 years (Mladenka and Minshall 2001).

Caenogastropoda—Rissooidea Lithoglyphidae—River Pebblesnails

This diverse family includes 73 North American species in 11 genera. They inhabit rivers and creeks of the southeastern and western United States, with several species from the Midwest and three from Canada. Most species are small (adult shell length <5 mm) and endemic to a single river system (Appendix; Plate 1). Consequently, the family has a high rate of imperilment (64%). Males possess a distinctive penis (Hershler and Ponder 1998) and females usually lay eggs singly in the spring. However, the Flat Pebblesnail *Lepyrium showalteri* lays a "superclutch" to which multiple females contribute (Figure 2). Many species appear to be annual species, with most individuals dying soon after the reproductive season; for example, *Somatogyrus* spp. and *Lepyrium showalteri* (Johnson, unpublished data).

Caenogastropoda—Cerithioidea Pleuroceridae—Freshwater Periwinkles

Recent molecular studies of pleurocerids have revealed that the current classification requires substantial revision in order to reflect evolutionary history (e.g., Holznagel and Lydeard 2000; Minton and Lydeard 2003; Hayes et al. 2007; Dillon and Robinson 2009; Dillon 2011). Interim taxonomic rearrangements (e.g., Dillon 2011) are likely inadequate. Consequently, herein we retain the Turgeon et al. (1998) classification until a synthetic and comprehensive taxonomy of pleurocerids is constructed.

Pleurocerids are the second most diverse group of North American freshwater gastropods and one of the most imperiled (79%). With 162 species in seven genera, they occur east of the continental divide primarily in rivers and creeks, attaining their highest diversity in drainages of the southeastern United States. Only two wide-ranging species have distributions that extend into Canada (Appendix; Plate 1). Adult shell length ranges from 1 to 5 cm and shell morphology can be highly variable within and among species (Burch 1989; Whelan et al. 2012a). Males lack a penis (Strong 2005) and females attach egg capsules to firm substrates singly, in lines, or in well-defined concentric clutches (Whelan et al. 2012a, 2012b). Juveniles often reach maturity in one year and the maximum life span seems to be 2–6 years for most species (Brown et al. 2008; P. D. Johnson, unpublished data). In some rivers, pleurocerids can achieve extraordinary densities, exceeding 1,500/m^2 (Johnson and Brown 1997). Slow growth, prolonged maturation, and narrow ecological tolerances contribute to their exceptional vulnerability (Brown and Johnson 2004); pleurocerids account for over half of the 67 gastropod extinctions reported here (Appendix; Plate 3).

Plate 1. Apertural views of assorted North American freshwater gastropods. Top Row (L-R): *Acella haldemani*, USNM 569406, Fishtrap Lake, Wisconsin; *Lioplax pilsbryi*, USNM 709961, Chipola River, Florida; *Juga plicifera*, USNM 12135, Oregon; *Aplexa elongata*, ANSP 73703, Belle Isle, Michigan. Second Row (L-R): *Bulimnaea megasoma*, USNM 569420, Kashabowie Lake, Ontario; *Neritina usnea*, USNM 835884, Lake Seminole, Florida; *Lanx alta*, ANSP 345218, Trinity River, California; *Pomacea paludosa*, Swamps Pompano, Florida. Third Row (L-R): *Io fluvialis*, USNM 119349, Clinch River, Tennessee; *Pomatiopsis lapidaria*, ANSP 192844, White River, Arkansas; *Lymnaea stagnalis*, USNM 41020, Oneida Lake, New York: *Fluminicola virens*, USNM 883676, Willamette River, Oregon. Bottom Row (L-R) *Gyraulus deflectus*, USNM 336597, Stillwater River, Maine; *Campeloma crassulum*, USNM 106143, New Harmony, Indiana; *Physella hendersoni*, USNM 251132, Charleston, South Carolina; *Lithasia armigera*, USNM 121760, Cumberland River, Tennessee. Scale bars next to gastropods are 1, 5 or 10 mm in length (photos by Thomas Tarpley, ADCNR).

Plate 2. Apertural views of assorted North American freshwater gastropods. Top Row (L-R): *Stiobia nana*, USNM 854934, Coldwater Spring, Alabama; *Lyogyrus pupoides*, USNM 336437, Stillwater River, Maine; *Galba perpolita*, USNM, 473102, Agattu Island, Alaska; *Leptoxis dilatata*, USNM 1155170, Indian Creek, West Virginia. Second Row (L–R): *Pyrgophorus platyrachis*, USNM 874863, Sulphur Spring, Florida; *Valvata bicarinata*, USNM 76627, Philadelphia, Pennsylvania; *Tryonia clathrata*, USNM 791488, Pyramid Lake, Nevada; *Lepyrium showalteri*, USNM 672419, Cahaba River, Alabama. Third Row (L–R): *Assiminea pecos*, USNM 1155172, Bitter Lake National Wildlife Refuge, New Mexico; *Acroloxus coloradensis*, USNM 883768, Hudson Bay, Montana; *Rhodacmea filosa*, USNM 1155171, Choccolocco Creek, Alabama; *Antroselates spiralis*, USNM 854700, Valley Cave, Kentucky; Alabama. Bottom Row (L-R) *Pyrgulopsis coloradensis*, USNM 854641, Blue Point Spring, Nevada; *Amnicola limosus*, USNM 451730, Cambridge, Massachusetts; *Erinna newcombi*, ANSP 162210, Hanakapiai, Kauai, Hawaii; *Lithasia lima*, ANSP 124850, Elk River, Tennessee. Scale bars next to gastropods are 1 or 5 mm in length (photos by Thomas Tarpley, ADCNR).

Figure 2. A clutch of eggs deposited by the Flat Pebblesnail *Lepyrium showalteri*, a federally endangered gastropod endemic to the Cahaba River system in central Alabama. Multiple females contribute to this large "super clutch." Each small, orange-colored egg is surrounded by a large fluid-filled capsule. Females lay eggs from March through May, after which more than 85% senesce and die. Newly hatched juveniles must reach reproductive size within a few months, prior to cooler winter temperatures. Photo Credit: Randall Haddock, Cahaba River Society.

Caenogastropoda—Rissooidea Pomatiopsidae—Amphibious Walker

This family contains six North American species in the genus *Pomatiopsis* that range from the St. Lawrence River basin to Pacific drainages along the California and Oregon coast. Only a single widely distributed species, the Slender Walker *Pomatiopsis lapidaria*, is known from Canada (Appendix; Plate 1). They are generally found in seeps, along spring margins, in flowing water, and in lakes (Burch 1989). These small gastropods (usually ≤5 mm adult shell length) live at least 2 years (Dundee 1957) and have a curious loping mode of locomotion (hence "walkers"). They apparently feed on detritus deposited along channel margins (van der Schalie 1959). Males possess a distinctive penis; females deposit egg capsules attached to gravel or coarse sand (van der Schalie and Dundee 1956). Three Pacific taxa are narrow endemics (66% imperilment rate) and the single species from northern Alabama is considered extinct (Plate 3).

Caenogastropoda—Cerithioidea Semisulcospiridae—Pacific Slope Periwinkles

Previously a subfamily of Pleuroceridae (Strong and Köhler 2009), this family currently includes 11 species in the genus *Juga* restricted to Pacific drainages north of the Sacramento River to British Columbia (Strong and Frest 2007). Two species are currently known from British Columbia (Appendix; Plate 1). Semisulcospirids are generally large (up to 4 cm) and graze on periphyton in streams and rivers. In some streams, population densities can exceed 500 m^2, representing over 90% of the invertebrate grazing biomass (Hawkins and Furnish 1987). Females lay a large gelatinous clutch of eggs in the spring (Clarke 1976). All but one species are considered imperiled (91%) and one may be extinct (Appendix; Plates 1 and 4).

Caenogastropoda—Viviparoidea Viviparidae—Mystery Snails

Native to drainages east of the Continental Divide, these large species (>3 cm adult shell length) occur predominately in rivers, but several are associated with lentic habitats where they may be very abundant (Brown and Lydeard 2010). Of the 21 species in four genera native to North America, five species are imperiled (24%), including three federally protected narrow endemics native to Alabama (Appendix; Plate 1). Only three species are known from Canada, but one has questionable taxonomic status (Appendix). All species are ovoviviparous, with crawling juveniles released at ≈ 3 mm in shell length. Viviparids are detritivores or facultative suspension feeders (Richardson and Brown 1989). Population densities are dependent on the organic content of associated sediments (Brown et al. 1989). They live several years and densities of some species in large rivers can be very high (see Tulotoma Recovery, p. 261). Males possess a penis formed by a modified right cephalic tentacle (Burch 1989); however, some species are parthenogenetic, which complicates genetics and confounds species boundaries (S. C. Johnson 1992; Katoh and Foltz 1994; Crummett and Wayne 2009).

Neritimorpha—Neritoidea Neritidae—Nerites

Most members of this family are marine species, but five occur in fresh to brackish waters in estuaries and coastal southeastern rivers (two species) and Hawaii (three species). Two of the Hawaiian species are endemic to the islands (Appendix). They are of moderate size (≈2 cm shell length; Plate 1) and are typically found on vegetation or firm substrates where females attach eggs capsules (Brasher 1997). Males possess a penis adjacent to the right cephalic tentacle (Burch 1989). Veliger larvae emerge from the egg capsules at hatching and drift downstream before settling as crawling juveniles (Brasher 1997; Resh et al. 1992). Individuals migrate back upstream during their lifespan of two or more years (Brasher 1997). The Hawaiian species have a restricted range, giving the fresh to brackish members of the family a 60% imperilment rate (Appendix).

Heterobranchia—Pulmonata—Acroloxoidea Acroloxidae—Capshells

This family is represented in North America by one species, the Rocky Mountain Capshell *Acroloxus coloradensis*, which is restricted to isolated mountain lakes in Canada and the United States (Appendix). Although a Canadian status review suggests the possibility of more than one species (Lee and Ackerman 2001), relatively few populations of Rocky Mountain capshell are known (100% imperilment rate for this family). Capshells are small (<5 mm adult shell length), with limpet-like shells (Plate 2). They are hermaphroditic and lay yellowish clutches of two to three eggs on rocks, plant stems, or leaves during summer and likely have a lifespan up to 2 years (Harrold and Guralnick 2008).

Plate 3. Apertural views of North American freshwater gastropods considered extinct. Top Row (L-R): *Stagnicola utahensis*, ANSP 187633, Lifton Bear Lake, Idaho; *Pomatiopsis hinkleyi*, ANSP 68449, Tennessee River, Alabama; *Lithasia jayana*, USNM 121760, Caney Fork, Tennessee; *Elimia impressa*, USNM 336364, Coosa River, Alabama. Second Row (L-R): *Amphigyra alabamensis*, ANSP 100980, Coosa River, Alabama; *Gyrotoma excisum*, ANSP 174777, Coosa River, Alabama; *Planorbella traski*, USNM 571751, Kern Lake, California; *Lithasia hubrichti*, USNM 636136, Big Black River, Mississippi. Third Row (L-R): *Athearnia crassa*; USNM 119636, Holston River, Tennessee; *Stagnicola pilsbryi*, ANSP 98545, Fish Springs National Wildlife Refuge, Utah; *Elimia clausa*, 177083, Coosa River, Alabama; *Somatogyrus crassilabris* USNM 271763, White River, Arkansas; Bottom Row (L-R): *Neoplanorbis carinatus*, ANSP 10112, Coosa River, Alabama; *Pyrgulopsis nevadensis*, USNM 31272, Pyramid Lake, Nevada; *Marstonia olivacea*, USNM 528038, Big Spring, Huntsville, Alabama; *Clappia umbilicata*, USNM 451821, Coosa River, Alabama. Scale bars next to gastropods are 1 or 5 mm in length (photos by Thomas Tarpley, ADCNR).

Cylinder Campeloma *Campeloma regulare* from the Alabama River near Claiborne, Monroe County, Alabama. This species is broadly distributed throughout the Mobile River Basin and is considered stable. Photo Credit: Thomas Tarpley, ADCNR.

Heterobranchia—Pulmonata—Lymnaeoidea Lymnaeidae—Elegant Pondsnails

With 61 North American species in nine genera, this family is most diverse in ponds and lakes of northern and western United States and Canada (Burch 1989). Nearly half of all North American species are found in Canada and two are endemic to Hawaii (Appendix; Plate 2). A recent phylogeny suggests a single well-supported clade for North American taxa (Correa et al. 2010). Twenty-six species (42%) have distributions restricted to two or fewer states/provinces, giving the family an overall 61% imperilment rate (Appendix). Most of these hermaphroditic species lay eggs in large gelatinous masses and juveniles grow quickly, often with multiple generations produced in a single year (Burch 1989). Species longevity may vary from several months to 3 years but is generally longer at northern latitudes (Burch 1989). Some lake species can reach substantial size, exceeding 30 mm in length; for example, the Mammoth Lymnaea *Bulimnaea megasoma*, (Plate 1).

Heterobranchia—Pulmonata—Planorboidea Physidae—Tadpole Pondsnails

This family has been the subject of several recent taxonomic revisions, not all of which agree (Taylor 2003; Dillon et al. 2007, 2011; Wethington and Lydeard 2007; Pip and Franck 2008; Wethington et al. 2009). Given this instability, the new species of Taylor (2003), Pip (2004), and Wethington et al. (2009) are herein recognized, but the classification in Turgeon et al. (1998) is retained.

These species are most commonly found in lentic environments, although some are restricted to rivers and springs. Forty-seven North American species in five genera are recognized (Appendix; Plate 1), most occurring in northern and western states, and 21 species in Canada (55% imperilment rate). Physids are hermaphroditic and generally lay large gelatinous egg masses during warmer months (Burch 1989; Dillon et al. 2011; Lepitzki 2013). Juveniles mature rapidly and multiple generations can be produced in a single year, but species from northern latitudes commonly live 2 years or more (DeWitt 1954; Pip and Stewart 1976).

Heterobranchia—Pulmonata—Planorboidea Planorbidae—Ramshorn Snails

Represented in North America by 52 species in 16 genera, most species have planispiral shells of variable size (5- to 25-mm shell width; Plates 3 and 4). Species in the subfamily Ancylinae have secondarily adopted a limpet-like shell shape and are now recognized as highly modified planorbids (Bouchet and Rocroi 2005; Walther et al. 2006, 2010), although European classifications have long recognized their planorbid affinities (e.g., Hubendick 1978). There are 25 species distributed across Canada (Appendix). Several genera are restricted to rivers, but many species utilize ponds, lakes, and bogs, including some low-DO environments (Burch 1989). Eggs from these hermaphroditic species are deposited singly or in large gelatinous clutches on firm substrates. Many species produce multiple generations in a year, and others may take a year to reach maturity (Burch 1989). Ten species (19%) are presumed to be extinct (Appendix; Plate 3), and several others have highly restricted distributions (44% imperilment rate).

Heterobranchia—Valvatoidea Valvatidae—Gilled Flatsnails

Valvatids are Holarctic, occurring in large lakes and rivers (Burch 1989). They are typically small (<8 mm shell width), operculate, and possess a unique gill that protrudes outside the mantle that allows them to tolerate low DO concentrations (Burch 1989). They are hermaphroditic with a penis positioned just beneath the right cephalic tentacle; some species have been reported to lay eggs between March and October (Lysne and Koetsier 2006). Of 10 North American species, seven have broad distributions, four are imperiled, and one is presumed extinct (50% imperilment rate; Appendix; Plate 2). The U.S. Fish and Wildlife Service (USFWS) recently delisted the only federally protected species in the family—the Desert Valvata *Valvata utahensis*—based upon new occurrence discoveries that expanded its known range.

SUMMARY AND CONCLUSIONS

This assessment determined that of 703 gastropod species, only 157 are currently stable. Of the remaining gastropods, 73 are vulnerable, 102 are threatened, 278 are endangered, 67 are extinct or possibly extinct, and the conservation or taxonomic status is ambiguous for 26 species (U or GU in the Appendix). The 74% imperilment rate of freshwater gastropods exceeds all other biota previously evaluated by AFS committees (Williams et al. 1993; Musick et al. 2000; Taylor et al. 2007; Jelks et al. 2008), but this rate may be marginally eclipsed by the pending AFS mussel assessment (J. D. Williams, Florida Fish and Wildlife Conservation Commission, personal communication). This assessment agrees with earlier models and summaries for North America (Ricciardi and Rassmussen 1999; Abell 2002). This pattern of decline reflects the degree of freshwater habitat

Plate 4. Apertural views of assorted North American freshwater gastropods. Top Row (L-R): *Planorbella trivolvis*, USNM 519355, Joliet, Illinois; *Viviparus subpurpureus*, ANSP 157362, Wabash River, Indiana; *Vorticifex effusa*, USNM 742157, Klamath River, Oregon; *Elimia boykiniana*, Flint River, Georgia. Second Row (L-R): *Juga acutifilosa*, USNM 425495, Klamath River, California; *Lithasia geniculata*, USNM, 129026, Cumberland River, Kentucky; *Pleurocera foremani*, ANSP 175693, Kelly Creek, Alabama; *Birgella subglobosus*, ANSP 57043, Iowa River, Iowa. Third Row (L-R): *Elimia hydei*, ANSP 122405, Black Warrior River, Alabama; *Lithasia duttoniana*, ANSP 334338, Duck River, Tennessee; *Viviparus georgianus*, ANSP 115729, Chicago River, Illinois; *Tulotoma magnifica*, USNM 176002, Coosa River, Alabama. Bottom Row (L-R): *Pleurocera alveare*, USNM 272182, Black River, Arkansas; *Campeloma decampi*, USNM 511325, Tennessee River, Alabama; *Lioplax sulculosa*, USNM 528050, Cedar River, Iowa; *Elimia floridensis*, ANSP 27526, Alexander Spring Creek, Florida. Scale bars next to gastropods are 1, 5 or 10 mm in length (photos by Thomas Tarpley, ADCNR).

Helmet Rocksnail *Lithasia duttoniana* from the Duck River near Columbia, Maury County, Tennessee, is endemic to the middle and lower Duck River; this species is usually found along channel margins. Photo Credit: Thomas Tarpley, ADCNR.

Smooth Hornsnail *Pleurocera prasinata* from its type locality, the Alabama River near Claiborne, Monroe County, Alabama. This species is currently stable and broadly distributed throughout the Mobile River basin. Photo Credit: Thomas Tarpley, ADCNR.

degradation and loss across the continent. In comparison to other sensitive ecosystems, including deserts, coastal marine environments, and forests, freshwater environments are the most threatened habitats in North America (Master et al. 2000; Heinz Center Report 2002; Burkhead 2012b). Only caves qualify as similarly imperiled ecosystems with moderate endemism but low diversity (Noss 2000).

Significant progress has been made in understanding ecological roles of freshwater invertebrates; however, our current knowledge of their distribution, systematics, biology, and ecology lags far behind our knowledge of freshwater fishes. The inherent human bias toward terrestrial systems is even evident in studies of freshwater fishes; for example, only about one third of North American freshwater fishes have been the focus of detailed life history studies (Etnier and Starnes 1994; Jenkins and Burkhead 1994; Boschung and Mayden 2004).

Distributional surveys in Canada are more comprehensive than comparable efforts for much of the United States (Figure 3; inset), but inventories in the United States are hampered by high diversity, lack of state or regional guides with keys, and unstable taxonomy for some groups. Although some states have completed recent reviews (Colorado, Connecticut, Florida, Indiana, New York, Missouri, Pennsylvania, Utah), state faunal guides are rare. The lack of surveys results from the relatively few biologists trained in the biology and systematics of freshwater gastropods and associated collection and preservation techniques.

The M:BER ratio of 9,539 is the highest modern to background extinction rate reported for any group of organisms on Earth (Pimm et al. 2006; Burkhead 2012b). Higher modern to background extinction rates (as extinctions per million species years) have been reported but these were based on future projections of models (Pereira et al. 2010a, 2010b; Barnosky et al. 2011). Considering the millions of years over which the fauna evolved and that nearly a tenth of known taxa from Canada and the United States have become extinct in only 112 years, the modern to background extinction ratio reported here seems intuitively low.

Mollusks have the highest numbers of documented extinctions among major taxonomic groups. The most extreme example may be that land snails endemic to tropical Pacific islands, which numbered in the thousands of species, have experienced even higher declines on a per island basis (Lydeard et al. 2004). Given the current rates of anthropogenic degradation of aquatic habitats (Vitousek et al. 1997; Ehrlich and Pringle 2008; Rockström et al. 2009) and the numbers of aquatic biota in jeopardy of future extinctions in North America (Williams et al. 1993; Taylor et al. 2007; Jelks et al. 2008; Burkhead 2012b; this study) and worldwide (IUCN 2012), it is self-evident that future rates of biodiversity loss will increase unless significant changes are made to the way humans use natural resources and modify landscapes.

Future priority conservation actions for freshwater gastropods include, but are not limited to (1) research on taxonomy, distribution, and basic biology; (2) modern surveys including detailed distributional and ecological requirements; (3) modernization mollusk collections including incorporating modern nomenclature, verification of identifications, and georeferencing of localities; (4) protection and restoration of relict habitats and freshwater gastropod assemblages; and (5) promoting freshwater species and ecosystem conservation and restoration to the general public.

EXAMPLES OF CONSERVATION SUCCESS

Though the overall conservation status of freshwater gastropods from Canada and the United States is disconcerting, we provide two examples of conservation successes that resulted from decreased threats and habitat restoration.

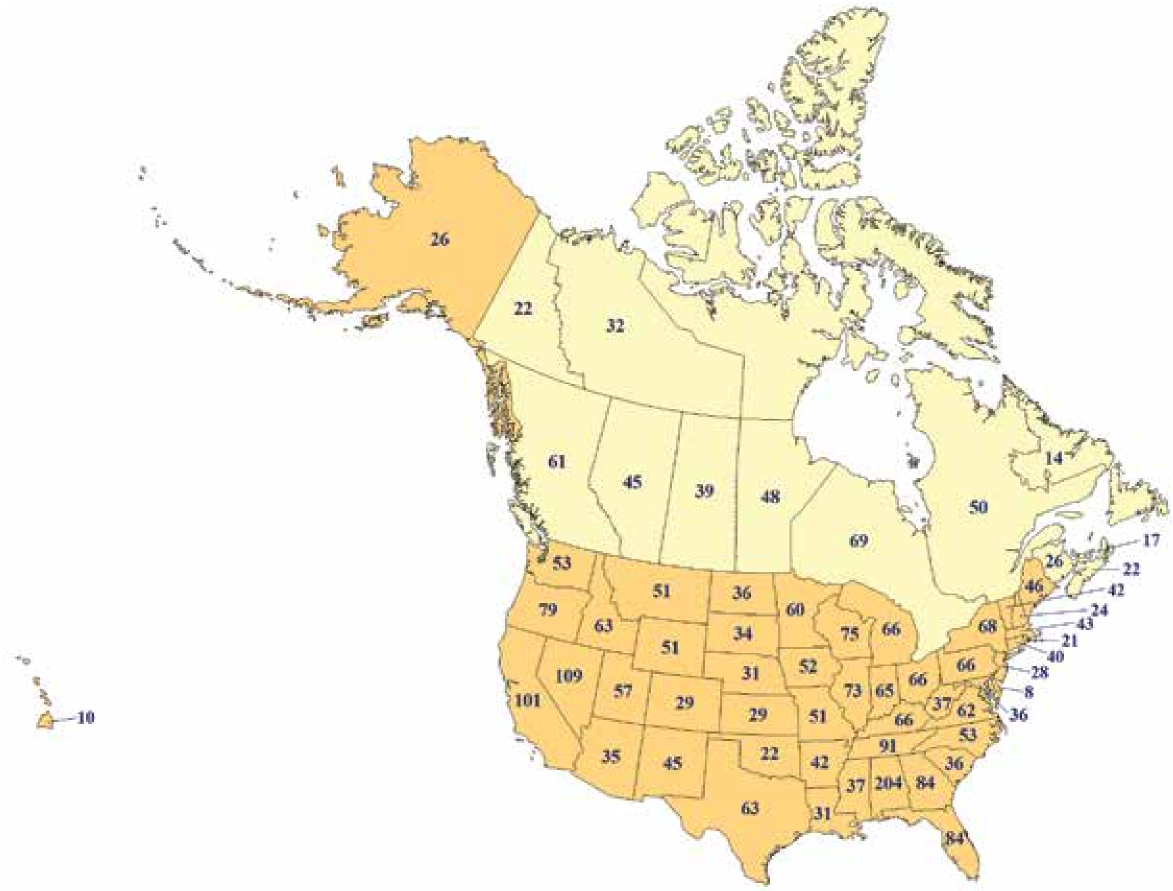

Figure 3. Map depicting approximate numbers of freshwater gastropod species by province and state for Canada and the United States.

Recovery of Tulotoma *Tulotoma magnifica* in Alabama

Once considered extinct, the Tulotoma was rediscovered in the lower Coosa River, Alabama, in 1988 (Hershler et al. 1990). This large viviparid was thought to have gone extinct because of hydroelectric dam construction and water quality problems throughout its 960-km historical distribution in the Coosa and Alabama rivers. The Tulotoma is a sedentary filter feeder, clustering on the undersides of large boulders, forming aggregations or "colonies" that can contain hundreds of individuals (Figure 4; USFWS 2000). Subsequent survey efforts confirmed five extant populations in the Coosa River basin and the snail was listed as endangered by the USFWS in 1991. Flow restoration in the Coosa River below Jordon Dam by the Alabama Power Company in the early 1990s dramatically improved water quality and increased downstream population levels of Tulotoma (USFWS 2010). Subsequent surveys by Auburn University located five additional populations in Coosa tributaries (DeVries 2005). In 2006, the Alabama Department of Conservation and Natural Resources (ADCNR) biologists found small numbers of Tulotoma in the Alabama River and surveys completed in 2010 located four new Alabama River populations that were more than 160 km distant from the Coosa River populations. The abundances observed in new populations are attributed to moderate water quality improvements in the Alabama River, which increased populations to detectable thresholds. Because all but one of the known populations have been stable or increasing for over a decade, along with the newly discovered populations in the Alabama River, the USFWS formally downlisted the species from endangered to threatened in June 2011. This represents the first successful down-listing of a freshwater mollusk under the Endangered Species Act. If Tulotoma populations continue to improve over the next decade, it may be possible to delist the species. An adult female Tulotoma is shown in Figure 5.

Habitat Recovery in the Cahaba River, Alabama

Located in central Alabama, the 304-km-long Cahaba River is the second largest tributary in the Alabama River system. The Cahaba River harbors one of the most species-rich faunas of mollusks and fishes in North America, although decades of poor land management and point and nonpoint source pollution have severely degraded the river (O'Neil and Shepard 2000). Cahaba River headwaters located in Birmingham receive more than 40 million gallons of discharge from 26 wastewater treatment plants daily (Shepard et al. 1994). With nutrient levels exceeding legal limits, the Environmental Protection Agency forced Birmingham to upgrade and construct new wastewater treatment facilities. Most of this work was completed by 2001. Although problems remain, water quality improved dramatically, and in 2004 the USFWS established the Cahaba River National Wildlife Refuge. Recent fish and mollusk inventories by the Geological Survey of Alabama, University of Alabama, and ADCNR documented 131 fish, 39 mussel, and 32 snail species extant in the system. The Cahaba River basin hosts 11 federally

Figure 4. A colony of the federally threatened Tulotoma attached to the underside of a small boulder from lower Choccolocco Creek, Talladega County, Alabama. Inset shows a large colony on the underside of a boulder from the lower Coosa River, Elmore County, Alabama. Photo Credit: Paul Johnson.

Figure 5. A female Tulotoma from Choccolocco Creek, Talladega County, Alabama. Photo Credit: Thomas Tarpley, ADCNR.

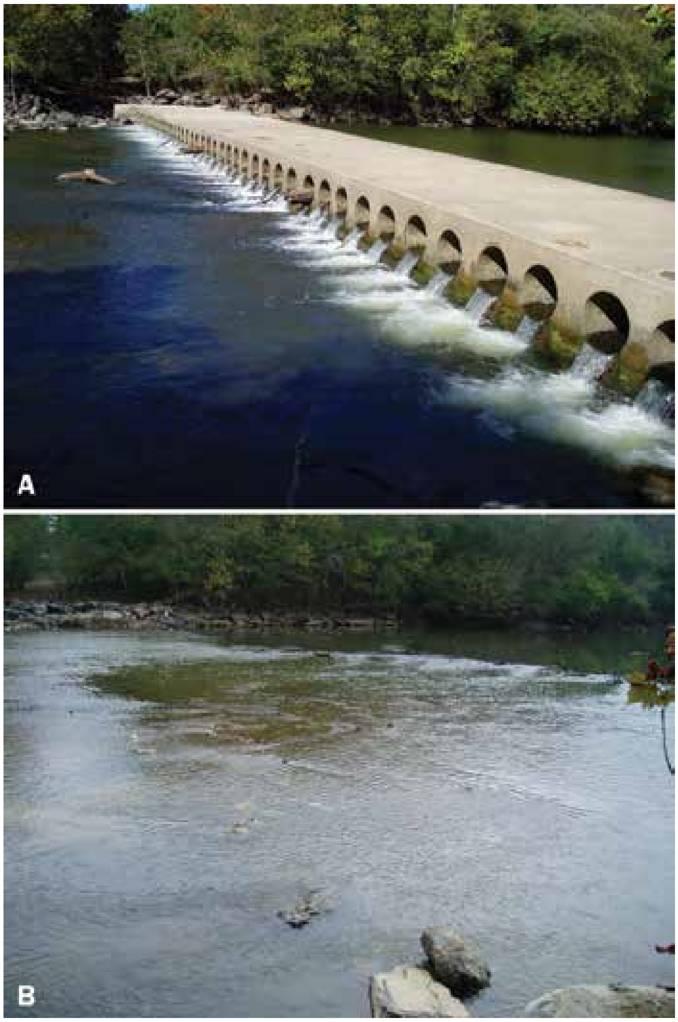

Figure 6. (A) Former Marvel Bridge located in the Cahaba River north of the Cahaba National Wildlife Refuge. The bridge (slab) was constructed by a mining company in the 1970s to move coal across the river and remained after the mine closed. (B) Efforts by the Nature Conservancy of Alabama culminated in its removal in late 2004, which improved habitat conditions over a kilometer of river and eliminated a barrier to fish passage. Photo Credit: Paul Freeman, the Nature Conservancy of Alabama.

listed species, including three snails. All listed fish and mollusks have shown range expansions and increasing numbers in recent years, presumably due to improving water quality.

The Nature Conservancy of Alabama recently led efforts to restore habitat by removing a large low-head concrete bridge (slab) just upstream of the new Cahaba River National Wildlife Refuge (Figure 6). Located in a section of river with exceptional fish and mollusk diversity, the 64-m-long × 7-m-wide × 2-m-high concrete bridge was an intermittent barrier to fish passage and disrupted flows above and below the structure (Figure 6). Pooled water behind the slab extended over 150 m upstream, and water passing through the 47 culverts scoured the channel bottom to bedrock downstream. With assistance from dozens of individuals representing various government and private conservation groups, mollusks were collected and removed in a large area above and below the concrete slab and translocated upstream. The slab was removed over a 3-day period in October 2004.

Slab removal initiated dramatic increases in snail densities, not only in the slab footprint and pool but downstream as well (Figure 7). Snail recovery was rapid and over the next few years, densities grew nearly exponentially. Importantly, densities of two federally listed snails increased more than 50-fold at the site. Subsequent monitoring of the fish community showed considerable expansion of the federally threatened Goldline Darter *Percina aurolineata* (B. Kuhadja, Tennessee Aquarium Conservation Institute, personal communication).

ADDITIONAL INFORMATION

The species database is available at the joint U.S. Geological Survey/AFS website (Johnson et al. 2013), along with extensive supplementary bibliographic information for North American freshwater gastropods and additional examples of recovery successes. The gastropod database and forthcoming AFS mussel conservation assessment will also be hosted by the Freshwater Mollusk Conservation Society (FMCS 2013), along with other general information about freshwater mollusks. Updated G-ranks, heritage conservation status, and global, national, and subnational distributions can be found at the NatureServe website (NatureServe 2013).

ACKNOWLEDGMENTS

We thank Jamie Smith, North Carolina Museum of Natural Sciences, for generating Figure 3. We are grateful to the numerous museum curators who provided access to the collections under their care during the preparation of this database. We also thank AFS *Fisheries* staff and especially Endangered Species Chair Howard Jelks for their assistance in publication. We thank Paul Callomon, Academy of Natural Sciences Philadelphia, for his technical assistance in photographing the micro planorbids. We thank Steve Ahlstedt for his assistance with the review process and his innumerable contributions to freshwater mollusk conservation over the years. Randall Haddock of the Cahaba River Society and Paul Freeman of the Nature Conservancy of Alabama contributed photographs. Thanks are extended to Buck Albert (Cherokee Nation Technology Solutions) and Howard Jelks (U.S. Geological Survey), Gainesville, Florida, for development of the website. We also gratefully acknowledge the contributions of two anonymous reviewers. Finally, a special thanks to Jim Williams, whose invaluable assistance facilitated completion of this assessment. This work was supported in part from various funding sources including the Alabama Department of Conservation and Natural Resources, the Smithsonian Institution, North Carolina Museum of Natural Sciences, and the U.S. Fish and Wildlife Service. Any use of trade, product, or firm names is for descriptive purposes only and does not imply endorsement by the U.S. Government.

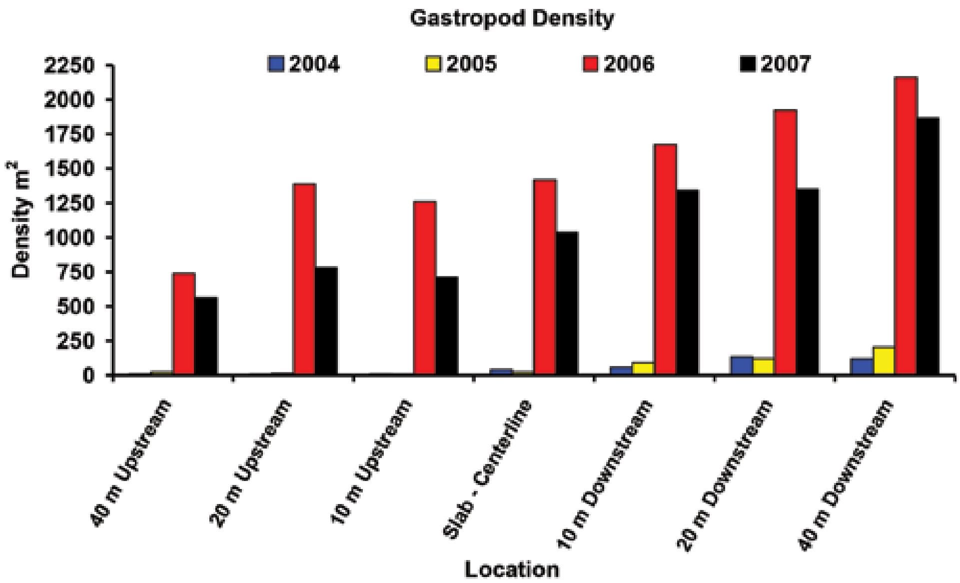

Figure 7. Gastropod densities in the Cahaba River above and below the Marvel Slab, from before slab removal in 2004 and after its removal (2005, 2006, and 2007); data courtesy of the the Nature Conservancy of Alabama. Bars indicate mean gastropod densities compiled from 10 Surber samples collected at each monitoring location.

Furrowed Lioplax *Lioplax sulculosa* from the Tennessee River near Florence, Lauderdale County, Alabama. This species is broadly distributed throughout the Mississippi River basin. Photo Credit: Thomas Tarpley, ADCNR.

Pleated Juga *Juga plicifera* from the Willamette River near Corvallis, Benton County, Oregon. The Pleated Juga is distributed in the Pacific Northwest from California to British Columbia, Canada. Photo Credit: Thomas Tarpley, ADCNR.

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From the Archives

The art of practical trout culture has, however, a very brief history. It is true that fish culture has been practiced, from time immemorial, by the southern Asiatics; that it was common among the Romans before the Christian era; that fish eggs were artificially impregnated and hatched by a monk in the middle ages. It is also true that a German army officer hatched salmon and trout about the middle of the eighteenth century, that experiments of a similar character were made in Great Britain and Norway and the United States, and that the French organized and kept in operation a large government fish-breeding establishment, till their late disastrous war with the Germans; but it was not--and I say it with pride--it was not till the persevering and far-seeing efforts of Stephen H. Ainsworth, and the wonderful genius of Seth Green, had been directed to the subject, that trout culture passed from the stage of experiment to that of a popular and practical branch of industry.

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APPENDIX. The 2012 AFS list of freshwater gastropods from Canada and the United States. Column headings are taxon (binomen) and species author(s), AFS common names [uncertain classification is denoted within brackets], AFS status and NatureServe G-ranks, and inferred distribution (alphabetic listing of states and provinces in which species are believed to occur); bold family names are followed by number of genera and species (or monotypic). Status abbreviations are provided in the text.

Taxon	AFS common name	AFS status	G-rank	Inferred distribution
Family Acroloxidae	**1 Genus, 1 species**			
Acroloxus coloradensis (Henderson, 1930)	Rocky Mountain Capshell	V	G3	CO, MT; Canada: AB, BC, ON, QC
Family Lymnaeidae	**9 Genera, 61 species**			
Acella haldemani (Binney, 1867)	Spindle Lymnaea	V	G3	IL, MI, MN, NY, OH, VT, WI; Canada: ON, QC
Bulimnaea megasoma (Say, 1824)	Mammoth Lymnaea	CS	G4G5	IA, MI, MN, NY, OH, VT, WI; Canada: MB, ON, QC
Erinna aulacospira (Ancey, 1899)	Hawaiian Bugle	Xp	GH	HI
Erinna newcombi Adams and Adams, 1855	Newcomb's Bugle	E	G1	HI
Fisherola nuttalli (Haldeman, 1841)	Shortface Lanx	T	G2	ID, MT, OR, UT, WA, WY; Canada: BC
Galba alberta Baker, 1919	Alberta Fossaria	E	G1Q	Canada: AB
Galba bulimoides (Lea, 1841)	Prairie Fossaria	CS	G5	AR, CA, CO, ID, KS, MN, MO, MT, NE, OR, SD, TX, UT, WA; Canada: AB, BC, MB, SK
Galba cockerelli Pilsbry and Ferriss, 1906	[uncertain classification]	V	G3G4Q	AZ, ID, NE, NM, SD, TX, WA; Canada: AB, BC
Galba cubensis (Pfeiffer, 1839)	Carib Fossaria	CS	G5	AL, CA, FL, GA, LA, MS, NC, NM, SC, TX
Galba cyclostoma (Walker, 1808)	Bugle Fossaria	Xp	GH	MI, NY
Galba dalli (Baker, 1907)	Dusky Fossaria	CS	G5	AZ, IL, IN, KS, MI, MN, MO, MT, ND, NE, NY, OH, PA, SD, TX, VA, WI, WV, WY; Canada: AB, BC, MB, ON, SK
Galba exigua (Lea, 1841)	Graceful Fossaria	CS	G5Q	AL, CT, IA, ID, IL, IN, KY, MA, ME, MI, MN, MO, NY, OH, OR, PA, TN, VA, WA, WI, WV; Canada: MB, ON, QC
Galba galbana (Say, 1825)	Boreal Fossaria	CS	G5	CT, ME, MI; Canada: , AB, BC, MB, NT, NU, ON, QC, SK
Galba humilis (Say, 1822)	Marsh Fossaria	CS	G5	KY, MD, ME, MO, NC, NJ, NY, OH, PA, SC, VA; Canada: ON, QC, PE
Galba modicella (Say, 1825)	Rock Fossaria	CS	G5	AK, AL, AZ, CA, CT, FL, IA, ID, IL, IN, LA, MA, MD, ME, MI, MN, MO, MS, MT, ND, NE, NH, NM, NV, NY, OH, OK, OR, PA, RI, SD, TN, TX, UT, VT, WA, WI, WV, WY; Canada: AB, BC, MB, NB, NS, NT, NU, ON, PE, QC, SK, YT
Galba obrussa (Say, 1825)	Golden Fossaria	CS	G5	AK, AL, AR, AZ, CA, CO, CT, DE, FL, GA, IA, ID, IL, IN, KS, KY, LA, MA, MD, ME, MI, MN, MO, MS, MT, NC, ND, NE, NH, NJ, NM, NV, NY, OH, OK, OR, PA, RI, SC, SD, TN, TX, UT, VA, VT, WA, WI, WV, WY; Canada: AB, MB, NF, NS, NT, SK
Galba parva (Lea, 1841)	Pygmy Fossaria	CS	G5	AZ, CO, CT, IA, ID, IL, IN, KS, KY, LA, MA, MD, ME, MI, MN, MO, MT, ND, NE, NM, NV, NY, OH, OK, PA, SD, TN, TX, UT, VA, WI, WY; Canada: AB, BC, MB, NT, NU, ON, QC, SK
Galba peninsulae (Walker, 1908)	[uncertain classification]	CS	G5Q	ME, MI, WI
Galba perplexa Baker and Henderson, 1929	[uncertain classification]	E	G1G2Q	CA, WA
Galba perpolita (Dall, 1905)	Glossy Fossaria	Xp	GH	AK
Galba rustica (Lea, 1841)	Rusty Fossaria	CS	G5Q	CO, CT, IL, IN, KS, MA, ME, MI, MO, NE, NM, NY, PA, UT, VT, WV; Canada: AB, MB, NS, NT, NU, ON, SK
Galba sonomaensis Hemphill, 1906	Sonoma Fossaria	T	G2Q	CA
Galba tazewelliana (Wolf, 1870)	Tazwell Fossaria	Xp	GH	IA, IL
Galba techella Haldeman, 1867	[uncertain classification]	V	G3G4Q	AR, AZ, CA, KS, LA, MO, NE, NM, NV, OK, TX, UT; Canada: AB, BC
Galba truncatula (Muller, 1774)	Attenuate Fossaria	CS	G5	AK; Canada: BC, YT
Galba vancouverensis Baker, 1939	[uncertain classification]	Xp	GHQ	WA; Canada: BC
Lanx alta (Tryon, 1865)	Highcap Lanx	T	G2	CA, OR
Lanx klamathensis Hannibal, 1912	Scale Lanx	E	G1	CA, OR
Lanx patelloides (Lea, 1856)	Kneecap Lanx	E	G1	CA
Lanx subrotunda (Tryon, 1865)	Rotund Lanx	T	G2	OR
Lanx sp	Banbury Springs Limpet	E	G1	ID
Lymnaea atkaensis Dall, 1884	Frigid Lymnaea	CS	G4G5	AK; Canada: BC, NT, YT
Lymnaea producta (Mighels, 1845)	[uncertain classification]	V	G3	HI
Lymnaea rubella Lea, 1841	Aloha Lymnea	Xp	GH	HI
Pseudosuccinea columella (Say, 1817)	Mimic Lymnaea	CS	G5	AL, AR, AZ, CA, CT, FL, GA, HI, IA, ID, IL, IN, KS, KY, LA, MA, MD, ME, MI, MN, MO, MS, NC, NH, NJ, NM, NY, OH, OK, OR, PA, RI, SC, TN, TX, VA, VT, WA, WI, WV, WY; Canada: AB, BC, MB, NB, NS, ON, QC
Stagnicola apicina (Lea, 1838)	Abbreviate Pondsnail	CS	G5	ID, MI, MN, MT, ND, OR, SD, WA, WI, WY; Canada: BC, ON
Stagnicola arctica (Lea, 1864)	Arctic Pondsnail	CS	G5	AK; Canada: AB, BC, LB, MB, NF, NT, NU, ON, QC, SK, YT
Stagnicola bonnevillensis (Call, 1884)	Fat-Whorled Pondsnail	E	G1	UT, WY
Stagnicola caperata (Say, 1829)	Wrinkled Marshsnail	CS	G5	AK, AL, CA, CO, IA, ID, IL, IN, MA, MD, ME, MN, MO, MT, ND, NE, NM, NV, NY, OH, OR, PA, SD, TX, UT, WA, WI, WV, WY; Canada: AB, BC, MB, ON, SK, YT

Taxon	AFS common name	AFS status	G-rank	Inferred distribution
Stagnicola catascopium (Say, 1817)	Woodland Pondsnail	CS	G5	CT, IA, IL, IN, MA, MD, ME, MI, MN, MT, ND, NH, NJ, NY, OH, OR, PA, RI, SD, VT, WA, WI, WY; Canada: AB, BC, MB, NB, NT, NS, ON, PE, QC, SK
Stagnicola contracta (Currier, 1881)	Deepwater Pondsnail	E	G1	MI
Stagnicola elodes (Say, 1821)	Marsh Pondsnail	CS	G5	AK, CA, CO, CT, IA ID, IL, IN, KY, KS, MA, ME, MI, MN, MO, MT, NE, ND, NH, NJ, NM, NY, OH, OR, PA, RI, SD, UT, VT, WA, WI, WY; Canada: AB, BC, LB, MB, NB, NF, NS, NT, NU, ON, PE, QC, SK, YT
Stagnicola elrodi (Baker and Henderson, 1933)	Flathead Pondsnail	E	G1Q	MT
Stagnicola elrodiana Baker, 1935	Longmouth Pondsnail	E	G1Q	MT
Stagnicola emarginata (Say, 1821)	St Lawrence Pondsnail	CS	G5	IA, ME, MI, MN, NH, NY, NT, OH, PA, VT, WI; Canada: NB, ON, QC
Stagnicola exilis (Lea, 1834)	Flat-Whorled Pondsnail	CS	G5	IA, IL, IN, KS, MI, MN, OH, WI; Canada: AB, MB, ON, QC, SK
Stagnicola gabbi (Tryon, 1865)	Striate Pondsnail	E	G1	CA
Stagnicola hinkleyi (Baker, 1906)	Rustic Pondsnail	T	G2	ID
Stagnicola idahoensis (Henderson, 1931)	Shortspire Pondsnail	E	G1	ID
Stagnicola kennicotti Baker, 1933	Western Arctic Pondsnail	T	G2	Canada: NT, NU
Stagnicola mighelsi (Binney, 1865)	Bigmouth Pondsnail	E	G1G2	ME
Stagnicola montanensis (Baker, 1913)	Mountain Marshsnail	V	G3	ID, MT, NV, UT, WY; Canada: AB, BC
Stagnicola neopalustris (Baker, 1911)	Piedmont Pondsnail	Xp	GH	VA
Stagnicola oronoensis (Baker, 1904)	Obese Pondsnail	T	G2G3	ME; Canada: ON
Stagnicola petoskeyensis (Walker, 1908)	Petosky Pondsnail	Xp	GH	MI
Stagnicola pilsbryi (Hemphill, 1890)	Fish Springs Marshsnail	X	GX	UT
Stagnicola traski (Tryon, 1863)	Widelip Pondsnail	V	G3	CA, ID, MT, OR, UT, WA, WY; Canada: AB, BC
Stagnicola utahensis (Call, 1884)	Thickshell Pondsnail	X	GX	UT
Stagnicola walkeriana Baker, 1926	Calabash Pondsnail	CS	G4	IL, IN, MI, MN, WI; Canada: ON
Stagnicola woodruffi (Baker, 1901)	Coldwater Pondsnail	T	G2G3	IL, IN, MI, MN, NY, WI; Canada: MB, ON
Family Physidae	**5 Genera, 47 species**			
Aplexa elongata (Say, 1821)	Lance Aplexa	CS	G5	AK, CO, CT, DC, IA, ID, IL, IN, MA, MD, ME, MI, MN, MT, ND, NE, NH, NV, NY, OH, OR, PA, SD, UT, VA, VT, WA, WI, WY; Canada: AB, BC, MB, NB, NS, NT, NU, ON, PE, QC, SK, YT
Archiphysa ashmuni Taylor, 2003	San Rafael Physa	E	G1	NM
Archiphysa sonomae Taylor, 2003	Sonoma Physa	E	G1	CA
Laurentiphysa chippuvarum Taylor, 2003	Chippewa Physa	E	G1	WI
Physa carolinae Wethington, Dillon, Wise, 2009	Carolina Physa	CS	G4	GA, NC, SC, VA
Physa jennessi Dall, 1919	Obtuse Physa	CS	G5	AK, ID, MN, MT, ND, WY; Canada: BC, MB, NT, NU, ON, QC, SK, YT
Physa megalochlamys Taylor, 1988	Cloaked Physa	V	G3	CO, ID, MT, OR, UT, WA, WY; Canada: AB, BC, SK
Physa natricina Taylor, 1988	Snake River Physa	E	G1	ID
Physa sibirica Westerlund, 1876	Frigid Physa	CS	G4G5	AK; Canada: NT, YT
Physa skinneri Taylor, 1954	Glass Physa	CS	G5	AK, CO, CT, IA, ID, IL, MA, MI, MN, MT, ND, NE, NV, NY, OH, PA, RI, SD, UT, WA, WI, WY; Canada: AB, BC, MB, NT, ON, QC, SK, YT
Physa vernalis Taylor and Jokinen, 1984	Vernal Physa	V	G3	CT, MA, MI, NY, OH, PA, RI; Canada: ON, NF
Physella ancillaria (Say, 1825)	Pumpkin Physa	CS	G5Q	CT, MA, ME, MI, MN, NH, NJ, NY,OH, PA, RI, VA, VT, WI, WY; Canada: NB, NF, QC
Physella bermudezi (Aguayo, 1935)	Lowdome Physa	CS	G4Q	FL
Physella bottimeri (Clench, 1924)	Comanche Physa	V	G3Q	NM, OK, TX
Physella boucardi (Cross and Fischer, 1881)	Desert Physa	CS	G5Q	CA, NV
Physella columbiana (Hemphill, 1890)	Rotund Physa	T	G2	MT, OR, WA, WY; Canada: BC
Physella conoidea (Fischer and Crosse, 1886)	Texas Physa	V	G3Q	TX
Physella cooperi (Tryon, 1865)	Olive Physa	V	G3	CA, ID, NV, OR, WA, WY
Physella costata (Newcomb, 1861)	Ornate Physa	E	G1	CA
Physella cubensis (Pfeiffer, 1839)	Carib Physa	CS	G5Q	AL, FL, GA
Physella globosa (Haldeman, 1841)	Globose Physa	V	G3Q	KY, OH, TN
Physella gyrina (Say, 1821)	Tadpole Physa	CS	G5	AK, AL, AR, AZ, CA, CO, CT, DE, FL, GA, IA, ID, IL, IN, KS, KY, LA, MA, MD, ME, MI, MN, MO, MS, MT, NC, ND, NE, NH, NJ, NM, NV, NY, OH, OK, OR, PA, RI, SC, SD, TN, TX, UT, VA, VT, WA, WI, WY; Canada: AB, BC, MB, NT, NU, ON, QC, SK, YT
Physella hemphilli Taylor, 2003	Idaho Physa	E	G1	ID
Physella hendersoni (Clench, 1925)	Bayou Physa	CS	G5Q	AL, FL, GA, MO, MS, NC, SC, TN, VA, WV
Physella heterostropha (Say, 1817)	Pewter Physa	CS	G5Q	AL, AR, CO, CT, FL, GA, IA, IL, IN, KS, KY, MA, MD, ME, MO, NC, NH, NJ, NY, OH, PA, RI, SC, TN, TX, VA, VT, WI, WV, WY; Canada: BC, NB, NF, NS, PE, QC

Taxon	AFS common name	AFS status	G-rank	Inferred distribution
Physella hordacea (Lea, 1864)	Grain Physa	E	G1Q	OR, WA; Canada: BC
Physella humerosa (Gould, 1855)	Corkscrew Physa	V	G3Q	AZ, CA
Physella integra (Haldeman, 1841)	Ashy Physa	CS	G5Q	CO, IA, IL, IN, KY, MI, MN, ND, NY, OH, PA, SD, TN, TX, VT, WI, WV, WY; Canada: BC, MB, ON, QC
Physella johnsoni (Clench, 1926)	Banff Springs Physa	E	G1	Canada, AB
Physella lordi (Baird, 1863)	Twisted Physa	CS	G5Q	CA, ID, MI, MT, NV, OR, UT, WA, WI; Canada: BC
Physella magnalacustris (Walker, 1901)	Great Lakes Physa	T	G2Q	ME, MI, WI; Canada: ON
Physella mexicana (Philippi, 1841)	Polished Physa	CS	G4Q	AZ, ID, NM, OR, TX, UT
Physella microstriata (Chamberlain and Berry, 1930)	Fish Lake Physa	X	GX	UT
Physella osculans (Haldeman, 1841)	Cayuse Physa	V	G3Q	AZ, CA, NV
Physella parkeri (Currier, 1881)	Broadshoulder Physa	T	G2Q	ME, MI, WI; Canada: ON, QC
Physella pomilia Conrad, 1834	Claiborne Physa	CS	G5	AL, FL, GA, KS, KY, LA, MO, MS, NC, NE, SC, TN, VA, WV
Physella propinqua (Tryon, 1865)	Rocky Mountain Physa	CS	G5Q	CA, ID, MT, NV, OR, UT, WA, WY; Canada: BC
Physella spelunca (Turner and Clench, 1974)	Cave Physa	E	G1	WY
Physella squalida (Morelet, 1851)	Squalid Physa	CS	G5Q	TX
Physella traski (Lea, 1864)	Sculpted Physa	T	G2G3Q	CA, OR
Physella utahensis (Clench, 1925)	Utah Physa	T	G2Q	CO, UT, WY
Physella vinosa (Gould, 1847)	Banded Physa	CS	G5Q	MI, MN, MT, NY, WI; Canada: ON
Physella virgata (Gould, 1855)	Protean Physa	CS	G5Q	AR, AZ, CA, HI, IA, IL, KS, KY, LA, MN, MT, ND, NE, NM, NV, OK, SD, TX, UT, WI, WY
Physella virginea (Gould, 1847)	Sunset Physa	CS	G4Q	CA, ID, OR, WA; Canada: BC
Physella winnipegensis Pip, 2004	Lake Winnipeg Physa	E	G1	Canada, MB
Physella wrighti Te and Clarke, 1985	Hotwater Physa	E	G1	Canada, BC
Physella zionis (Pilsbry, 1926)	Wet-rock Physa	E	G1	UT
Family Planorbidae	**16 Genera, 52 species**			
Amphigyra alabamensis Pilsbry, 1906	Shoal Sprite	X	GX	AL
Biomphalaria havanensis (Pfeiffer, 1839)	Ghost Ramshorn	CS	G5	AZ, CA, FL, ID, LA, SC, TX
Ferrissia fragilis (Tryon, 1863)	Fragile Ancylid	CS	G5	AL, AR, AZ, CA, CO, CT, FL, GA, IA, ID, IL, IN, KS, KY, LA, MA, MD, ME, MI, MO, MS, MT, NC, NE, NM, NV, NY, OH, OK, OR, PA, SC, SD, TN, TX, VA, VT, WA, WI, WV, WY; Canada: AB, BC, ON, QC
Ferrissia rivularis (Say, 1817)	Creeping Ancylid	CS	G5	AL, AR, AZ, CA, CO, CT, DE, FL, GA, IA, ID, IL, IN, KS, KY, LA, MA, MD, ME, MI, MN, MO, MS, MT, NC, ND, NE, NH, NJ, NM, NV, NY, OH, OK, OR, PA, RI, SC, SD, TN, TX, UT, VA, VT, WA, WI, WV, WY; Canada: AB, BC, MB, NB, NF, NS, ON, PE, QC, SK
Gyraulus circumstriatus (Tryon, 1866)	Disc Gyro	CS	G5	AZ, CA, CO, CT, ID, IN, KS, MA, MI, MN, MT, ND, NE, NH, NM, NY, OH, OR, PA, SD, UT, VT, WA, WI, WV, WY; Canada: AB, BC, MB, NB, NS, NT, ON, PE, QC, SK, YT
Gyraulus crista (Linnaeus, 1758)	Star Gyro	CS	G5	AK, CA, ID, ME, MI, MN, MT, ND, NM, NY, OR, VT, WA, WI, WY; Canada: AB, BC, MB, NT, ON, QC, SK
Gyraulus deflectus (Say, 1824)	Flexed Gyro	CS	G5	AK, CT, IA, ID, IL, IN, KY, MA, MD, ME, MI, MN, MO, MT, NC, ND, NE, NH, NY, OH, PA, SC, SD, VA, WA, WI, WY; Canada: AB, BC, LB, MB, NB, NF, NS, NT, NU, ON, PE, QC, SK, YT
Gyraulus hornensis Baker, 1934	Tuba Gyro	CS	G4Q	ND, WI; Canada: ON, QC, NT, SK
Gyraulus parvus (Say, 1817)	Ash Gyro	CS	G5	AK, AL, AR, AZ, CA, CO, CT, DE, FL, GA, IA, ID, IL, IN, KS, KY, LA, MA, MD, ME, MI, MN, MO, MS, MT, NC, ND, NE, NH, NJ, NM, NV, NY, OH, OK, OR, PA, RI, SC, SD, TN, TX, UT, VA, VT, WA, WI, WV, WY; Canada: AB, BC, LB, MB, NB, NF, NT, NS, NU, ON, PE, QC, SK, YT
Gyraulus vermicularis (Gould, 1847)	Pacific Coast Gyro	CS	G4Q	CA, ID, OR, WA; Canada: BC, YT
Hebetancylus excentricus (Morelet, 1851)	Excentric Ancylid	CS	G5	AL, FL, GA, LA, MS, NC, OK, SC, TX, VA
Helisoma anceps (Menke, 1830)	Two-ridge Ramshorn	CS	G5	AK, AL, AR, AZ, CA, CO, CT, DE, FL, GA, IA, ID, IL, IN, KS, KY, LA, MA, MD, ME, MI, MN, MO, MS, MT, NC, ND, NE, NH, NJ, NM, NV, NY, OH, OK, OR, PA, RI, SC, SD, TN, TX, VA, VT, WA, WI, WV, WY; Canada: AB, BC, MB, NB, NT, NS, ON, PE, QC, SK, NU
Helisoma minus (Cooper, 1870)	[uncertain classification]	E	G1Q	CA
Helisoma newberryi (Lea, 1858)	Great Basin Ramshorn	E	G1Q	CA, ID, NV, OR, UT, WY
Laevapex fuscus (Adams, 1841)	Dusky Acylid	CS	G5	AL, AR, CT, DE, FL, GA, IA, IL, IN, KS, KY, LA, MA, MD, MN, MO, MS, NC, NJ, NY, OH, OK, PA, RI, SC, TN, TX, VA, VT, WI, WV; Canada: ON, QC
Menetus opercularis (Gould, 1847)	Button Sprite	CS	G5	AK, CA, ID, MT, OR, UT, WA; Canada: AB, BC
Micromenetus brogniartianus (Lea, 1842)	Disc Sprite	CS	G5Q	AL, FL, MO, OH, VA
Micromenetus dilatatus (Gould, 1841)	Bugle Sprite	CS	G5	AL, AR, CA, CT, FL, GA, IA, IL, IN, KY, LA, MA, MD, ME, MO, MS, NC, NH, NJ, NY, OH, OK, PA, RI, SC, TN, TX, VA WV; Canada: NS, ON
Micromenetus floridensis (Baker, 1945)	Penny Sprite	CS	G5	FL
Micromenetus sampsoni (Ancey, 1885)	Sampson Sprite	T	G2G3Q	AR, KY, MO, IL
Neoplanorbis carinatus Walker, 1908	Carinate Flat-top Snail	X	GX	AL

Taxon	AFS common name	AFS status	G-rank	Inferred distribution
Neoplanorbis smithi Walker, 1908	Angled Flat-top Snail	X	GX	AL
Neoplanorbis tantillus Pilsbry, 1906	Little Flat-top Snail	X	GX	AL
Neoplanorbis umbilicatus Walker, 1908	Umbilicate Flat-top Snail	X	GX	AL
Pecosorbis kansasensis (Berry, 1966)	New Mexico Ramshorn	V	G3	KS, NM
Planorbella ammon (Gould, 1855)	Jupiter Ramshorn	U	GU	CA, CO
Planorbella binneyi (Tryon, 1867)	Coarse Ramshorn	CS	G4G5Q	CA, OR, UT, WA; Canada: AB, BC
Planorbella campanulata (Say, 1821)	Bellmouth Ramshorn	CS	G5	CT, IA, IL, IN, MA, ME, MI, MN, ND, NY, OH, PA, VT, WI; Canada: MB, NB, NF, NS, ON, PE, QC, SK
Planorbella columbiensis (Baker, 1945)	Caribou Ramshorn	Xp	GH	Canada: BC
Planorbella corpulenta (Say, 1824)	Corpulent Ramshorn	T	G2	MN Canada, MB, ON
Planorbella duryi (Wetherby, 1879)	Seminole Ramshorn	CS	G5	CA, FL, HI, ID, NC, NM, WY
Planorbella magnifica (Pilsbry, 1903)	Magnificent Ramshorn	E	G1	NC
Planorbella multivolvis (Case, 1847)	Acorn Ramshorn	X	GX	MI
Planorbella occidentalis (Cooper, 1870)	Fine-lined Ramshorn	V	G3	CA, OR, WA; Canada: BC
Planorbella oregonensis (Tryon, 1865)	Lamb Ramshorn	E	G1	OR, UT
Planorbella pilsbryi (Baker, 1926)	File Ramshorn	CS	G4G5	MA, MI, MN, MT, ND, NY, OH, PA, WI; Canada: AB, MB, ON, NB, QC, SK
Planorbella scalaris (Jay, 1839)	Mesa Ramshorn	CS	G5	CO, FL, WY
Planorbella subcrenata (Carpenter, 1857)	Rough Ramshorn	CS	G5	AK, CA, CO, ID, MN, MO, MT, ND, NM, NV, OR, SD, UT, WA, WY; Canada: AB, BC, MB, NT, NU, ON, SK, YT
Planorbella tenuis (Dunker, 1850)	Mexican Ramshorn	CS	G5	AZ, CA, ID, NM, TX
Planorbella traski (Lea, 1856)	Keeled Ramshorn	X	GX	CA
Planorbella trivolvis (Say, 1817)	Marsh Ramshorn	CS	G5	AK, AL, AR, CA, CO, CT, DE, FL, GA, IA, ID, IL, IN, KS, KY, LA, MA, MD, ME, MI, MN, MO, MS, MT, NC, ND, NE, NH, NJ, NY, OH, PA, RI, SC, SD, TN, TX, UT, VA, VT, WI, WV, WY; Canada: MB, NB, NF, NS, NU, ON, PE, QC, SK
Planorbella truncata (Miles, 1861)	Druid Ramshorn	V	G3G4	IA, IL, MI, WI
Planorbula armigera (Say, 1821)	Thicklip Ramshorn	CS	G5	AL, AR, CT, FL, GA, IA, IL, IN, KY, LA, MA, MD, ME, MI, MN, MO, MS, MT, NC, ND, NE, NH, NJ, NY, OH, PA, RI, SC, SD, TN, VA, VT, WI; Canada: AB, BC, MB, NB, NT, NS, NU, ON, PE, QC, SK, YT
Planorbula campestris (Dawson, 1875)	Meadow Ramshorn	CS	G5	MT, ND, NM, SD, WY; Canada: AB, BC, MB, NT, ON, SK, YT
Promenetus exacuous (Say, 1821)	Sharp Sprite	CS	G5	AK, AR, AZ, CA, CO, CT, GA, IA, ID, IL, IN, KS, KY, MA, ME, MI, MN, MT, NC, ND, NE, NH, NM, NV, NY, OH, OK, OR, PA, SD, TN, TX, UT, VA, VT, WA, WI, WY; Canada: AB, BC, MB, NB, NT, NS, NU, ON, PE, QC, SK, YT
Promenetus umbilicatellus (Cockerell, 1887)	Umbilicate Sprite	CS	G4	AK, CA, CO, ID, IL, IN, KS, MN, MT, ND, NE, NM, NV, NY, OH, OK, OR, PA, SD, UT, WA, WI, WY; Canada: AB, BC, MB, ON, SK
Rhodacmea cahawbensis (Walker, 1917)	Cahaba Ancylid	E	G1	AL
Rhodacmea elatior (Anthony, 1855)	Domed Ancylid	E	G1	KY, TN
Rhodacmea filosa (Conrad, 1834)	Wicker Ancylid	E	G1	AL
Rhodacmea hinkleyi (Walker, 1908)	Knobby Ancylid	Xp	GHQ	AL, AR, IL, IN, KY, TN
Vorticifex effusa (Lea, 1856)	Artemesian Ramshorn	V	G3	CA, ID, WA, OR
Vorticifex solida (Dall, 1870)	[uncertain classification]	Xp	GHQ	CA, NV
Family Neritidae	**1 Genus, 5 species**			
Nertina cariosa (Wood, 1828)	Pip'wai	T	G1G3	HI
Neritina clenchi Russel, 1940	[uncertain classification]	CS	G5Q	FL
Nertina granosa Sowerby, 1825	Hihiwai	E	G1	HI
Neritina usnea (Roding, 1798)	Olive Nerite	CS	G5	AL, FL, MS, LA, TX
Neritina vespertina Sowerby, 1849	Hapawai	E	G1G2	HI
Family Viviparidae	**4 Genera, 21 species**			
Campeloma brevispirum Baker, 1928	[uncertain classification]	CS	G5Q	WI
Campeloma crassulum Rafinesque, 1819	Ponderous Campeloma	CS	G5	AR, IA, IL, IN, KY, KS, MN, MO, NC, OH, TN, WI
Campeloma decampi (Binney, 1865)	Slender Campeloma	E	G1	AL
Campeloma decisum (Say, 1817)	Pointed Campeloma	CS	G5	AL, AR, CT, GA, IA, IL, IN, KY, LA, MA, MD, ME, MI, MN, MS, NC, ND, NE, NH, NJ, NY, OH, OK, PA, RI, SC, TN, TX, VA, VT, WI, WV; Canada: MB, NB, NS, ON, QC
Campeloma floridense Call, 1886	Purple-throat Campeloma	CS	G5	FL
Campeloma geniculum (Conrad, 1834)	Ovate Campeloma	CS	G5	AL, FL, GA
Campeloma limum (Anthony, 1860)	File Campeloma	CS	G5	FL, GA, NC, SC
Campeloma milesi (Lea, 1863)	[uncertain classification]	CS	G5Q	WI; Canada: ON

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Campeloma parthenum Vail, 1979	Maiden Campeloma	CS	G5	AL, FL
Campeloma regulare (Lea, 1841)	Cylinder Campeloma	CS	G4	AL, GA, MS, TN
Campeloma rufum (Haldeman, 1841)	[uncertain classification]	CS	G5Q	CT, IA, IL, IN, KY, MA, ME, MI, MN, NY, OH, PA, VT, WI
Lioplax cyclostomaformis (Lea, 1841)	Cylindrical Lioplax	E	G1	AL, GA
Lioplax pilsbryi Walker, 1905	Choctaw Lioplax	CS	G5	AL, FL, GA
Lioplax subcarinata (Say, 1817)	Ridgid Lioplax	CS	G4G5	MD, NC, NJ, NY, PA, SC, VA, WV
Lioplax sulculosa (Menke, 1827)	Furrowed Lioplax	CS	G5	AL, AR, IA, IL, IN, KY, MN, MO, OH, TN, WI
Tulotoma magnifica (Conrad, 1834)	Tulotoma	T	G2	AL
Viviparus georgianus (Lea, 1834)	Banded Mysterysnail	CS	G5	AL, AR, CT, FL, GA, IA, IL, IN, KY, LA, MA, MD, MI, MN, MO, MS, NC, NJ, NY, OH, PA, SC, TN, VA, VT, WI; Canada: ON, QC
Viviparus goodrichi Archer, 1933	Globose Mysterysnail	V	G3G4	FL, GA
Viviparus intertextus (Say, 1829)	Rotund Mysterysnail	CS	G4	AL, AR, FL, GA, IA, IL, KY, LA, MN, MO, MS, NC, SC, TN, TX, WI
Viviparus limi Pilsbry, 1918	Ochlockonee Mysterysnail	V	G3G4	FL, GA
Viviparus subpurpureus (Say, 1829)	Olive Mysterysnail	CS	G5	AL, AR, IA, IL, IN, KY, LA, MO, MS, SC, TN, TX, WI
Family Ampullaridae	**1 Genus, 2 species**			
Pomacea paludosa (Say, 1829)	Florida Applesnail	CS	G5	AL, FL, GA, NC
Family Assiminidae	**1 Genus, 2 species**			
Assiminea infima Berry, 1947	Badwater Snail	E	G1	CA
Assiminea pecos Taylor, 1987	Pecos Assiminea	E	G1	NM, TX
Family Amnicolidae	**4 Genera, 18 species**			
Amnicola cora Hubricht, 1979	Foushee Cavesnail	E	G1	AR
Amnicola dalli (Pilsbry and Beecher, 1892)	Peninsula Amnicola	CS	G5	FL
Amnicola decisus Haldeman, 1845	[uncertain classification]	E	G1Q	ME, NY, PA
Amnicola limosus (Say, 1817)	Mud Amnicola	CS	G5	AL, AR, CO, CT, IA, IL, IN, KS, KY, LA, MA, MD, ME, MI, MN, MO, MS, MT, NC, ND, NE, NH, NJ, NM, NY, OH, OK, PA, RI, SC, SD, TN, UT, VA, VT, WI, WY; Canada: AB, MB, NB, NS, ON, PE, QC, SK, LB, NF
Amnicola rhombostoma Thompson, 1968	Squaremouth Amnicola	Xp	GH	FL
Amnicola stygius Hubricht, 1971	Stygian Amnicola	E	G1	MO
Colligyrus convexus Hershler, Frest, Liu, and Johannes, 2003	Canary Duskysnail	E	G1G2	CA
Colligyrus depressus Hershler, 1999	Harney Basin Duskysnail	E	G1	OR
Colligyrus greggi (Pilsbry, 1935)	Rocky Mountain Duskysnail	CS	G4	ID, MT, UT, WY; Canada: BC
Dasyscias franzi Thompson and Hershler, 1991	Shaggy Ghostsnail	E	G1	FL
Lyogyrus bakerianus (Pilsbry, 1917)	Baker's Springsnail	Xp	GH	NY
Lyogyrus browni (Carpenter, 1872)	Slender Duskysnail	T	G1G3Q	MA, RI
Lyogyrus granum (Say, 1822)	Squat Duskysnail	CS	G5	AL, CT, GA, MA, MD, MS, NC, NJ, NY, PA, SC, VA, VT; Canada: NB, NS
Lyogyrus latus Thompson and Hershler, 1991	Cobble Sprite	T	G2	GA
Lyogyrus pilsbryi (Walker, 1906)	Lake Duskysnail	CS	G4	IL, IN, OH, WI
Lyogyrus pupoideus (Gould, 1841)	Pupa Duskysnail	CS	G5	CT, MA, ME, NY, PA, VT
Lyogyrus retromargo (Thompson, 1968)	Indented Duskysnail	CS	G4	FL, GA, SC
Lyogyrus walkeri (Pilsbry, 1898)	Canadian Duskysnail	V	G3G4	MI, MN, NY, OH, PA, VT, WI; Canada: QC, MB, ON
Family Cochliopidae	**14 Genera, 48 species**			
Antrobia culveri Hubricht, 1971	Tumbling Creek Cavesnail	E	G1	MO
Antroselates spiralis Hubricht, 1963	Shaggy Cavesnail	V	G3	IN, KY
Aphaostracon asthenes Thompson, 1968	Blue Spring Hydrobe	E	G1	FL
Aphaostracon chalarogyrus Thompson, 1968	Freemouth Hydrobe	E	G1	FL
Aphaostracon hypohyalinum Thompson, 1968	Suwanee Hydrobe	T	G2	FL
Aphaostracon monas (Pilsbry, 1899)	Wekiwa Hydrobe	E	G1	FL
Aphaostracon pachynotum Thompson, 1968	Thick-shelled Hydrobe	V	G3	FL
Aphaostracon pycnus Thompson, 1968	Dense Hydrobe	E	G1	FL
Aphaostracon rhadinum Thompson, 1968	Slough Hydrobe	T	G2	FL

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Aphaostracon theiocrenetum Thompson, 1968	Clifton Spring Hydrobe	E	G1	FL
Aphaostracon xynoelictum Thompson, 1968	Fenney Spring Hydrobe	E	G1	FL
Balconorbis uvaldensis Hershler and Longley, 1986	Balcones Ghostsnail	E	G1G2	TX
Cochliopina riograndensis Pilsbry and Ferriss, 1906	Spiral Pebblesnail	T	G2G3	TX
Eremopyrgus eganensis Hershler, 1999	Steptoe Hydrobe	E	G1	NV
Ipnobius robustus (Hershler, 1989)	Robust Tryonia	E	G1G2	CA
Juturnia kosteri (Taylor, 1987)	Koster Springsnail	T	G2	NM
Juturnia tularosae Hershler, Liu, and Stockwell, 2002	Tularosa Springsnail	E	G1	NM
Littoridinops monroensis (Frauenfeld, 1863)	Cockscomb Hydrobe	CS	G5	AL, FL, GA, LA, MS, TX
Littoridinops palustris Thompson, 1968	Bantam Hydrobe	V	G3	AL, FL, MS
Littoridinops tenuipes (Couper, 1844)	Henscomb Hydrobe	CS	G5	CT, FL, GA, MA, MD, NC, NJ, NY, SC, VA
Pseudotryonia adamantina (Taylor, 1987)	Diamond Tryonia	E	G1	NM, TX
Pseudotryonia alamosae (Taylor, 1987)	Caliente Tryonia	E	G1	NM, NV
Pseudotryonia brevissima (Pilsbry, 1890)	Regal Hydrobe	E	G1	FL
Pseudotryonia grahamae Thompson, 2001	Salt Spring Hydrobe	E	G1	AL
Pyrgophorus platyrachis Thompson, 1968	Serrate Crownsnail	CS	G5	FL
Pyrgophorus spinosus (Call and Pilsbry, 1886)	Spiny Crownsnail	V	G3	TX
Spurwinkia salsa (Pilsbry, 1905)	Saltmarsh Hydrobe	CS	G4G5	CT, FL, MA, MD, ME, NH, NJ; Canada: NB
Stygopyrgus bartonensis Hershler and Longley, 1986	Barton Cavesnail	E	G1	TX
Tryonia aequicostata (Pilsbry, 1889)	Smooth-ribbed Hydrobe	V	G3	FL
Tryonia angulata Hershler and Sada, 1987	Sportingoods Tryonia	E	G1	NV
Tryonia brunei Taylor, 1987	Brune's Springsnail	E	G1	TX
Tryonia cheatumi (Pilsbry, 1935)	Phantom Tryonia	E	G1	TX
Tryonia circumstriata (Leonard and Ho, 1960)	Gonzales Springsnail	E	G1	TX
Tryonia clathrata Stimpson, 1865	Grated Tryonia	T	G2	NV
Tryonia diaboli (Pilsbry and Ferriss, 1906)	Devil Tryonia	E	G1	TX
Tryonia elata Hershler and Sada, 1987	Point of Rocks Tryonia	E	G1	NV
Tryonia ericae Hershler and Sada, 1987	Minute Tryonia	E	G1	NV
Tryonia gilae Taylor, 1987	Gila Tryonia	E	G1	AZ, NM
Tryonia imitator (Pilsbry, 1899)	Mimic Tryonia	T	G2G3	CA
Tryonia margae Hershler, 1989	Grapevine Springs Elongate Springsnail	E	G1	CA
Tryonia metcalfi Hershler, Liu, and Landye, 2011	Metcalf's Tryonia	E	G1	TX
Tryonia monitorae Hershler, 1999	Monitor Tryonia	E	G1	NV
Tryonia oasiensis Hershler, Liu, and Landye, 2011	Carolinae Tryonia	E	G1	TX
Tryonia porrecta (Mighels, 1845)	Desert Tryonia	V	G3	CA, NV, UT
Tryonia quitobaquitae Hershler, 1988	Quintobaquito Tryonia	E	G1	AZ, NM
Tryonia rowlandsi Hershler, 1989	Grapevine Springs Squat Tryonia	E	G1	CA
Tryonia salina Hershler, 1989	Cottonball Marsh Tryonia	E	G1	CA
Tryonia variegata Hershler and Sada, 1987	Amargosa Tryonia	T	G2	CA, NV
Family Hydrobiidae	**15 Genera, 185 species**			
Birgella subglobosus (Say, 1825)	Globe Siltsnail	CS	G4	AL, AR, IA, IL, IN, GA, KY, MI, MN, MO, MS, NY, OH, PA, TN, VT, WI, WV; Canada: MB, ON, QC
Cincinnatia integra (Say, 1821)	Midland Siltsnail	CS	G5	AL, AR, IL, IN, KS, KY, LA, ME, MD, MI, MN, MO, MS, ND, NE, NY, OH, OK, PA, SD, TN, TX, VA, VT, WI; Canada: MB, ON, SK
Floridobia alexander (Thompson, 2000)	Alexander Siltsnail	E	G1	FL
Floridobia floridana (Frauenfeld, 1863)	Hyacinth Siltsnail	CS	G5	GA, FL
Floridobia fraterna (Thompson, 1968)	Creek Siltsnail	T	G2	FL
Floridobia helicogyra (Thompson, 1968)	Crystal Siltsnail	E	G1	FL
Floridobia leptospira (Thompson, 2000)	Flatwood Siltsnail	E	G1G2	FL

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Floridobia mica (Thompson, 1968)	Ichetucknee Siltsnail	E	G1	FL
Floridobia monroensis (Dall, 1885)	Enterprise Siltsnail	E	G1	FL
Floridobia parva (Thompson, 1968)	Pygmy Siltsnail	E	G1	FL
Floridobia petrifons (Thompson, 1968)	Rock Springs Siltsnail	E	G1	FL
Floridobia ponderosa (Thompson, 1968)	Ponderous Siltsnail	E	G1	FL
Floridobia porterae (Thompson, 2000)	Green Cove Springsnail	E	G1	FL
Floridobia vanhyningi (Vanatta, 1934)	Seminole Siltsnail	E	G1	FL
Floridobia wekiwae (Thompson, 1968)	Wekiva Siltsnail	E	G1	FL
Floridobia winkleyi (Pilsbry, 1912)	New England Siltsnail	V	G3	CT, MA, ME
Fontigens aldrichi (Call and Beecher, 1886)	Hoosier Springsnail	CS	G4	IL, MO
Fontigens antroecetes (Hubricht, 1940)	Missouri Cavesnail	T	G2	IL, MO
Fontigens bottimeri (Walker, 1925)	Potomac Springsnail	T	G2	MD, VA
Fontigens cryptica Hubricht, 1963	Hidden Springsnail	E	G1	IN
Fontigens morrisoni Hershler, Holsinger, and Hubricht, 1990	Morrison's Springsnail	E	G1	VA
Fontigens nickliniana (Lea, 1838)	Watercress Snail	CS	G5	AL, IL, IN, KY, MD, MI, NC, NY, OH, PA, TN, VA, WI, WV
Fontigens orolibas Hubricht, 1957	Blue Ridge Springsnail	V	G3	MD, PA, VA
Fontigens proserpina Hubricht, 1940	Proserpine Cavesnail	E	G1	MO
Fontigens tartarea Hubricht, 1963	Organ Cavesnail	T	G2	WV
Fontigens turritella Hubricht, 1976	Greenbrier Cavesnail	E	G1	WV
Hoyia sheldoni (Pilsbry, 1890)	Storm Ghostsnail	E	G1	WI
Marstonia agarhecta Thompson, 1969	Ocmulgee Marstonia	E	G1	GA
Marstonia angulobasis Thompson, 2005	Angled Marstonia	E	G1	AL, TN
Marstonia arga Thompson, 1977	Ghost Marstonia	CS	G5	AL, TN
Marstonia castor Thompson, 1977	Beaverpond Marstonia	E	G1	GA
Marstonia comalensis (Pilsbry and Ferriss, 1906)	Comal Marstonia	E	G1	TX
Marstonia gaddisorum Thompson, 2005	Emily's Marstonia	E	G1	GA
Marstonia halcyon Thompson, 1977	Halcyon Marstonia	T	G2	GA
Marstonia hershleri (Thompson, 1995)	Coosa Pyrg	E	G1	AL
Marstonia letsoni (Walker, 1901)	Gravel Pyrg	CS	G5	MI, NY, OH, PA; Canada: ON
Marstonia lustrica (Pilsbry, 1890)	Boreal Marstonia	CS	G5	IA, IL, IN, MA, ME, MI, MN, NY, OH, PA, VT, WI; Canada: MB, ON, NT, QC, NB
Marstonia ogmorhaphe Thompson, 1977	Royal Marstonia	E	G1	TN
Marstonia olivacea (Pilsbry, 1895)	Olive Marstonia	X	GX	AL
Marstonia ozarkensis (Hinkley, 1915)	Ozark Pyrg	E	G1	AR, MO
Marstonia pachyta Thompson, 1977	Armored Marstonia	E	G1	AL
Marstonia scalariformis (Wolf, 1870)	Moss Pyrg	V	G3	AL, IA, IL, MO
Notogillia sathon Thompson, 1969	Satyr Siltsnail	V	G3	GA
Notogillia wetherbyi (Dall, 1885)	Alligator Siltsnail	CS	G5	AL, FL, GA
Phreatodrobia coronae Hershler and Longley, 1987	Crowned Cavesnail	E	G1G2	TX
Phreatodrobia imitata Hershler and Longley, 1986	Mimic Cavesnail	E	G1	TX
Phreatodrobia micra (Pilsbry and Ferriss, 1906)	Flattened Cavesnail	T	G2	TX
Phreatodrobia nugax (Pilsbry and Ferriss, 1906)	Domed Cavesnail	V	G3G4	TX
Phreatodrobia plana Hershler and Longley, 1986	Disc Cavesnail	T	G2	TX
Phreatodrobia punctata Hershler and Longley, 1986	High-hat Cavesnail	T	G2	TX
Phreatodrobia rotunda Hershler and Longley, 1986	Beaked Cavesnail	E	G1G2	TX
Probythinella emarginata (Kuster, 1852)	Delta Hydrobe	CS	G5	AL, AR, IA, IL, IN, KS, KY, LA, ME, MI, MN, MO, MS, MT, NC, ND, NE, NY, OH, OK, PA, SD, TN, TX, WI; Canada: AB, MB, NT, NU, ON, QC, SK
Pyrgulopsis aardahli Hershler, 1989	Benton Valley Springsnail	E	G1	CA
Pyrgulopsis aloba Hershler, 1998	Duckwater Pyrg	E	G1	NV
Pyrgulopsis amargosae Hershler, 1989	Amargosa Springsnail	E	G1	CA

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Pyrgulopsis anatina Hershler, 1998	Southern Duckwater Pyrg	E	G1	NV
Pyrgulopsis anguina Hershler, 1998	Longitudinal Gland Pyrg	E	G1	NV, UT
Pyrgulopsis archimedis Berry, 1947	Archimedes Pyrg	E	G1	CA, OR
Pyrgulopsis arizonae (Taylor, 1987)	Apache Springsnail	E	G1	AZ
Pyrgulopsis augustae Hershler, 1998	Elongate Cane Spring Pyrg	E	G1	NV
Pyrgulopsis aurata Hershler, 1998	Pleasant Valley Pyrg	E	G1	NV
Pyrgulopsis avernalis (Pilsbry, 1935)	Moapa Pebblesnail	E	G1G2	NV
Pyrgulopsis bacchus Heshler, 1988	Grand Wash Springsnail	E	G1	AZ
Pyrgulopsis basiglans Hershler, 1998	Large Gland Carico Pyrg	E	G1	NV
Pyrgulopsis bedfordensis Hershler and Gustafson, 2001	Bedford Pyrg	E	G1	MT
Pyrgulopsis bernardina (Taylor, 1987)	San Bernardino Springsnail	E	G1	AZ
Pyrgulopsis bifurcata Hershler, 1998	Small Gland Carico Pyrg	E	G1	NV
Pyrgulopsis blainica Hershler, Liu, Gustafson, 2008	Blane Pyrg	E	G1	MT
Pyrgulopsis breviloba Hershler, 1998	Flat Pyrg	E	G1	NV
Pyrgulopsis bruesi Hershler and Sada, 2000	Fy Ranch Pyrg	E	G1	NV
Pyrgulopsis bruneauensis Hershler, 1990	Bruneau Hot Springsnail	E	G1	ID
Pyrgulopsis bryantwalkeri Hershler, 1994	Cortez Hills Pebblesnail	E	G1	NV
Pyrgulopsis californiensis (Gregg and Taylor, 1965)	Languna Mountain Springsnail	V	G3G4	CA
Pyrgulopsis carinata Hershler, 1998	Carinate Duckwater Pyrg	X	GX	NV
Pyrgulopsis carinifera (Pilsbry, 1935)	Moapa Valley Pyrg	E	G1	NV
Pyrgulopsis castaicensis Hershler and Liu, 2010	Middle Canyon Spring Pyrg	E	G1	CA
Pyrgulopsis chamberlini Hershler, 1998	Smooth Glenwood Pyrg	E	G1	UT
Pyrgulopsis chupaderae Taylor, 1987	Chupadera Springsnail	E	G1	NM
Pyrgulopsis cinerana Hershler, Frest, Liu, and Johannes, 2003	Ash Valley Pyrg	E	G1G2	CA
Pyrgulopsis coloradensis Hershler, 1998	Blue Point Pyrg	E	G1	NV
Pyrgulopsis conica Hershler, 1988	Kingman Springsnail	E	G1	AZ
Pyrgulopsis cruciglans Hershler, 1998	Transverse Gland Pyrg	E	G1	NV
Pyrgulopsis crystalis Hershler and Sada, 1987	Crystal Springsnail	E	G1	NV
Pyrgulopsis cybele Hershler and Liu, 2012	Nature Pyrg	E	G1	NV
Pyrgulopsis davisi (Taylor, 1987)	Limpia Creek Springsnail	E	G1	TX
Pyrgulopsis deaconi Hershler, 1998	Spring Mountains Pyrg	E	G1	NV
Pyrgulopsis deserta (Pilsbry, 1916)	Desert Springsnail	T	G2	AZ, UT
Pyrgulopsis diablensis Hershler, 1995	Diablo Range Pyrg	E	G1	CA
Pyrgulopsis dixiensis Hershler, 1998	Dixie Valley Pyrg	E	G1	NV
Pyrgulopsis eremica Hershler, 1995	Smoke Creek Pyrg	T	G2	CA
Pyrgulopsis erythropoma (Pilsbry, 1899)	Ash Meadows Pebblesnail	E	G1	NV
Pyrgulopsis fairbanksensis Hershler and Sada, 1987	Fairbanks Springsnail	E	G1	NV
Pyrgulopsis falciglans Hershler, Frest, Liu, and Johannes, 2003	Likely Pyrg	E	G1G2	CA
Pyrgulopsis fausta Hershler, 1998	Corn Creek Pyrg	E	G1	NV
Pyrgulopsis fresti Hershler and Liu, 2009	Owyhee Hot Springsnail	E	G1	OR
Pyrgulopsis fusca Hershler, 1998	Otter Creek Pyrg	E	G1	UT
Pyrgulopsis gibba Hershler, 1995	Surprise Valley Pyrg	V	G3	CA, NV
Pyrgulopsis gilae (Taylor, 1987)	Gila Springsnail	T	G2	NM
Pyrgulopsis giuliani Hershler and Pratt, 1990	Southern Sierra Nevada Springsnail	E	G1G2	CA
Pyrgulopsis glandulosa Hershler, 1988	Verde Rim Springsnail	E	G1	AZ
Pyrgulopsis gracilis Hershler, 1998	Emigrant Pyrg	E	G1	NV
Pyrgulopsis greggi Hershler, 1995	Kern River Springsnail	E	G1	CA
Pyrgulopsis hamlinensis Hershler, 1998	Hamlin Valley Pyrg	E	G1	UT

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Pyrgulopsis hovinghi Hershler, 1998	Upper Thousand Spring Pyrg	E	G1	NV
Pyrgulopsis hubbsi Hershler, 1998	Hubbs Pyrg	E	G1	NV
Pyrgulopsis humboldtensis Hershler, 1998	Humbolt Pyrg	E	G1	NV
Pyrgulopsis ignota Hershler, Liu, and Lang, 2010	Caroline Springs Pyrg	E	G1	TX
Pyrgulopsis imperialis Hershler, 1998	Kings River Pyrg	E	G1	NV
Pyrgulopsis inopinata Hershler, 1998	Carinate Glenwood Pyrg	E	G1	UT
Pyrgulopsis intermedia (Tryon, 1865)	Crooked Creek Springsnail	E	G1G2	OR
Pyrgulopsis isolata Hershler and Sada, 1987	Elongate-gland Springsnail	E	G1	NV
Pyrgulopsis kolobensis (Taylor, 1987)	Toquerville Springsnail	CS	G5	ID, NV, UT
Pyrgulopsis landyei Hershler, 1998	Landyes Pyrg	E	G1	NV
Pyrgulopsis lasseni Hershler, Frest, Liu, and Johannes, 2003	Willow Creek Pyrg	E	G1G2	CA
Pyrgulopsis lata Hershler, 1998	Butterfield Pyrg	E	G1	NV
Pyrgulopsis lentiglans Hershler, 1998	Critteden Pyrg	E	G1	NV
Pyrgulopsis leporina Hershler, 1998	Elko Pyrg	E	G1	NV
Pyrgulopsis limaria Hershler, 1998	Squat Mud Meadows Pyrg	E	G1	NV
Pyrgulopsis lockensis Hershler, 1998	Lockes Pyrg	E	G1	NV
Pyrgulopsis longae Hershler, 1995	Long Valley Pyrg	E	G1	CA
Pyrgulopsis longiglans Hershler, 1998	Western Lahontan Pyrg	T	G2G3	NV
Pyrgulopsis longinqua (Gould, 1855)	Salton Sea Springsnail	E	G1	CA
Pyrgulopsis marcida Hershler, 1998	Hardy Pyrg	T	G2	NV
Pyrgulopsis merriami (Pilsbry and Beecher, 1892)	Pahrangagat Pebblesnail	E	G1	NV
Pyrgulopsis metcalfi (Taylor, 1987)	Naegele Springsnail	E	G1	NM, TX
Pyrgulopsis micrococcus (Pilsbry, 1893)	Oasis Valley Springsnail	V	G3	CA, NV
Pyrgulopsis militaris Hershler, 1998	Northern Soldier Meadow Pyrg	E	G1	NV
Pyrgulopsis millenaria Hershler, 1998	Twentyone Mile Pyrg	E	G1	NV
Pyrgulopsis milleri Hershler and Liu, 2010	Pierpoint Spring Pyrg	E	G1	CA
Pyrgulopsis montana Hershler, 1998	Camp Valley Pyrg	E	G1	NV
Pyrgulopsis montezumensis Hershler, 1988	Montezuma Well Spring-snail	E	G1	AZ
Pyrgulopsis morrisoni Hershler, 1988	Page Springsnail	E	G1	AZ
Pyrgulopsis nanus Hershler and Sada, 1987	Distal-gland Springsnail	E	G1	NV
Pyrgulopsis neomexicana (Pilsbry, 1916)	Socorro Springsnail	E	G1	NM
Pyrgulopsis neritella Hershler, 1998	Neritiform Steptoe Ranch Pyrg	E	G1	NV
Pyrgulopsis nevadensis (Stearns, 1883)	Corded Pyrg	X	GX	NV
Pyrgulopsis nonaria Hershler, 1998	Ninemile Pyrg	E	G1	UT
Pyrgulopsis notidicola Hershler, 1998	Elongate Mud Meadows Pyrg	E	G1	NV
Pyrgulopsis orbiculata Hershler, 1998	Sub-globose Steptoe Ranch Pyrg	E	G1	NV
Pyrgulopsis owensensis Hershler, 1989	Owens Valley Springsnail	E	G1G2	CA, NV
Pyrgulopsis owyheensis Hershler and Liu, 2009	Owyhee Upland Pyrg	E	G1G2	OR
Pyrgulopsis papillata Hershler, 1998	Big Warm Spring Pyrg	E	G1	NV
Pyrgulopsis pecosensis (Taylor, 1987)	Pecos Springsnail	E	G1	NM
Pyrgulopsis peculiaris Hershler, 1998	Bifid Duct Pyrg	T	G2	NV, UT
Pyrgulopsis pellita Hershler, 1998	Antelope Valley Pyrg	E	G1	NV
Pyrgulopsis perturbata Hershler, 1989	Fish Slough Springsnail	E	G1G2	CA
Pyrgulopsis pictilis Hershler, 1998	Ovate Cain Spring Pyrg	E	G1	NV
Pyrgulopsis pilsbryana (Bailey and Bailey, 1952)	Bear Lake Springsnail	T	G2	ID, UT, WY
Pyrgulopsis pisteri Hershler and Sada, 1987	Median-gland Springsnail	E	G1	NV
Pyrgulopsis planulata Hershler, 1998	Flat-topped Steptoe Pyrg	E	G1	NV

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Pyrgulopsis plicata Hershler, 1998	Black Canyon Pyrg	E	G1	UT
Pyrgulopsis robusta (Walker, 1908)	Jackson Lake Springsnail	CS	G5	ID, OR, WA, WY
Pyrgulopsis roswellensis (Taylor, 1987)	Roswell Springsnail	E	G1	NM
Pyrgulopsis ruinosa Hershler, 1998	Fish Lake Pyrg	X	GX	NV
Pyrgulopsis rupinicola Hershler, Frest, Liu, and Johannes, 2003	Sucker Spring Pyrg	E	G1G2	CA
Pyrgulopsis sadai Hershler, 1998	Sada's Pyrg	E	G1	NV
Pyrgulopsis sathos Hershler, 1998	White River Valley Pyrg	T	G2	NV
Pyrgulopsis saxatilis Hershler, 1998	Sub-globose Snake Pyrg	E	G1	UT
Pyrgulopsis serrata Hershler, 1998	Northern Steptoe Pyrg	V	G3	NV
Pyrgulopsis simplex Hershler, 1988	Fossil Springsnail	E	G1G2	AZ
Pyrgulopsis sola Hershler, 1988	Brown Springsnail	E	G1	AZ
Pyrgulopsis stearnsiana (Pilsbry, 1899)	Yaqui Springsnail	T	G2	CA
Pyrgulopsis sterilis Hershler, 1998	Sterile Basin Pyrg	E	G1	NV
Pyrgulopsis sublata Hershler, 1998	Lake Valley Pyrg	E	G1	NV
Pyrgulopsis sulcata Hershler, 1998	Southern Steptoe Pyrg	E	G1	NV
Pyrgulopsis taylori Hershler, 1995	San Luis Obispo Pyrg	E	G1	CA
Pyrgulopsis texana (Pilsbry, 1935)	Phantom Cavesnail	E	G1	TX
Pyrgulopsis thermalis (Taylor, 1987)	New Mexico Hot Spring-snail	E	G1	NM
Pyrgulopsis thompsoni Hershler, 1988	Huachuca Springsnail	T	G2	AZ
Pyrgulopsis transversa Hershler, 1998	Southern Bonneville Pyrg	T	G2	UT
Pyrgulopsis trivialis (Taylor, 1987)	Black River Springsnail	E	G1	AZ, NM
Pyrgulopsis turbatrix Hershler, 1998	Southeast Nevada Pyrg	T	G2	NV
Pyrgulopsis umbilicata Hershler, 1998	Southern Soldier Meadow Pyrg	E	G1	NV
Pyrgulopsis variegata Hershler, 1998	Northwest Bonneville Pyrg	T	G2	NV, UT
Pyrgulopsis varneri Heshler, Liu, and Sada, 2007	Varner's Pyrg	E	G1	NV
Pyrgulopsis ventricosa Hershler, 1995	Clear Lake Pyrg	E	G1	CA
Pyrgulopsis villacampae Hershler, 1998	Duckwater Warm Springs Pyrg	E	G1	NV
Pyrgulopsis vinyardi Hershler, 1998	Vinyards Pyrg	E	G1	NV
Pyrgulopsis wongi Hershler, 1989	Wong's Pyrg	T	G2G3	CA, NV
Rhapinema dacryon Thompson, 1969	Teardrop Snail	CS	G5	AL, FL, GA
Spilochlamys conica Thompson, 1968	Conical Siltsnail	V	G3G4	FL, GA
Spilochlamys gravis Thompson, 1968	Armored Siltsnail	V	G3G4	FL
Spilochlamys turgida Thompson, 1969	Pumpkin Siltsnail	T	G2	GA
Stiobia nana Thompson, 1978	Sculpin Snail	E	G1	AL
Texapyrgus longleyi Thompson and Hershler, 1991	Striated Hydrobe	E	G1	TX
Lithoglyphidae	**11 Genera, 72 species**			
Antrorbis breweri Hershler and Thompson, 1990	Conical Siltsnail	E	G1	AL
Clappia cahabensis Clench, 1965	Armored Siltsnail	E	G1	AL
Clappia umbilicata (Walker, 1904)	Pumpkin Siltsnail	X	GX	AL
Fluminicola ahjumawi Hershler, Liu, Frest and Johannes, 2007	Sculpin Snail	V	G3	OR
Fluminicola anserinus Hershler, Liu, Frest and Johannes, 2007	Striated Hydrobe	E	G1	OR
Fluminicola caballensis Hershler, Liu, Frest and Johannes, 2007	Horse Creek pebblesnail	E	G1	OR
Fluminicola coloradoensis Morrison, 1940	Green River pebblesnail	T	G2G3	ID, UT, WY
Fluminicola dalli (Call, 1884)	Pyramid Lake pebblesnail	E	G1	NV
Fluminicola erosus Hershler, Liu, Frest and Johannes, 2007	Smokey Charley pebblesnail	E	G1	OR
Fluminicola favillaceus Hershler, Liu, Frest and Johannes, 2007	Ash Valley pebblesnail	E	G1	OR

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Flumincola fremonti Hershler, Liu, Frest and Johannes, 2007	Fremont pebblesnail	E	G1	OR
Fluminicola fuscus (Haldeman, 1847)	Ashy pebblesnail	T	G2	ID, MT, OR, WA, WY; Canada: BC
Fluminicola gustafsoni Hershler and Liu, 2012	Salmon River pebblesnail	V	G3	ID, WA
Fluminicola insolitus Hershler, 1999	Strange pebblesnail	E	G1	OR
Fluminicola lunsfordensis Hershler, Liu, Frest and Johannes, 2007	Lunsford Pebblesnail	E	G1	CA
Fluminicola minutissimus Pilsbry, 1907	Pixie Pebblesnail	Xp	GH	ID
Fluminicola modoci Hannibal, 1912	Modoc Pebblesnail	E	G1	CA, OR
Fluminicola multifarius Hershler, Liu, Frest and Johannes, 2007	Shasta Pebblesnail	T	G2	OR
Fluminicola neritoides Hershler, Liu, Frest and Johannes, 2007	Willow Creek Pebblesnail	E	G1	OR
Fluminicola nuttallianus (Lea, 1838)	Dusky Pebblesnail	Xp	GH	OR
Fluminicola potemicus Hershler, Liu, Frest and Johannes, 2007	Potem Creek Pebblesnail	E	G1	OR
Fluminicola scopulinus Hershler, Liu, Frest and Johannes, 2007	Castle Creek Pebblesnail	E	G1	OR
Fluminicola seminalis (Hinds, 1842)	Nugget Pebblesnail	T	G2	CA
Fluminicola turbiniformis (Tryon, 1865)	Turban Pebblesnail	V	G3	CA, NV, OR
Flumincola umbilicatus Hershler, Liu, Frest and Johannes, 2007	Goose Valley Pebblesnail	E	G1	OR
Fluminicola virens (Lea, 1838)	Olympia Pebblesnail	T	G2	OR, WA
Fluminicola virginius Hershler, 1999	Virginia Mountains Pebblesnail	E	G1	NV
Fluminicola warnerensis Hershler, Liu, Frest and Johannes, 2007	Topaz Pebblesnail	T	G2	OR
Gillia altilis (Lea, 1841)	Buffalo Pebblesnail	CS	G5	MD, NC, NJ, NY, PA, SC, VA, VT, WV; Canada: ON
Holsingeria unthanksensis Hershler, 1989	Thankless Ghostsnail	T	G2	VA
Lepyrium showalteri (Lea, 1861)	Flat Pebblesnail	E	G1	AL
Phreatoceras taylori (Hershler and Longley, 1986)	Nymph Trumpet	E	G1G2	TX
Phreatodrobia conica Hershler and Longley, 1986	Hueco Cavesnail	E	G1	TX
Pristinicola hemphilli (Pilsbry, 1890)	Pristine Pyrg	V	G3	CA, ID, OR, WA
Somatogyrus alcoviensis Krieger, 1972	Reverse Pebblesnail	E	G1	GA
Somatogyrus amnicoloides Walker, 1915	Ouachita Pebblesnail	U	GU	AR
Somatogyrus aureus Tryon, 1865	Golden Pebblesnail	U	GU	AL, TN
Somatogyrus biangulatus Walker, 1906	Angular Pebblesnail	U	GU	AL
Somatogyrus constrictus Walker, 1904	Knotty Pebblesnail	U	GU	AL
Somatogyrus coosaensis Walker, 1904	Coosa Pebblesnail	U	GU	AL
Somatogyrus crassilabris Walker, 1915	Thick-lip Pebblesnail	Xp	GH	AR
Somatogyrus crassus Walker, 1904	Stocky Pebblesnail	U	GU	AL
Somatogyrus currierianus (Lea, 1863)	Tennessee Pebblesnail	U	GU	AL
Somatogyrus decipiens Walker, 1909	Hidden Pebblesnail	U	GU	AL
Somatogyrus depressus (Tryon, 1862)	Sandbar Pebblesnail	T	G2	IA, IL, MO, WI
Somatogyrus excavatus Walker, 1906	Ovate Pebblesnail	U	GU	AL
Somatogyrus georgianus Walker, 1904	Cherokee Pebblesnail	U	GU	AL, GA, TN
Somatogyrus hendersoni Walker, 1909	Fluted Pebblesnail	U	GU	AL
Somatogyrus hinkleyi Walker, 1904	Granite Pebblesnail	U	GU	AL
Somatogyrus humerosus Walker, 1906	Atlas Pebblesnail	U	GU	AL
Somatogyrus integra (Say, 1829)	Ohio Pebblesnail	V	G3	IL, IN, KY, OH, PA
Somatogyrus nanus Walker, 1904	Dwarf Pebblesnail	U	GU	AL
Somatogyrus obtusus Walker, 1904	Moon Pebblesnail	U	GU	AL
Somatogyrus parvulus Tryon, 1865	Sparrow Pebblesnail	E	G1G2Q	TN
Somatogyrus pennsylvanicus Walker, 1904	Shale Pebblesnail	V	G3	PA, VA, WV
Somatogyrus pilsbryanus Walker, 1904	Tallapoosa Pebblesnail	U	GU	AL
Somatogyrus pumilus (Conrad, 1834)	Compact Pebblesnail	U	GU	AL

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Somatogyrus pygmaeus Walker, 1909	Pygmy Pebblesnail	U	GU	AL
Somatogyrus quadratus Walker, 1906	Quadrate Pebblesnail	U	GU	AL
Somatogyrus rheophilus Thompson, 1984	Flint Pebblesnail	E	G1	GA
Somatogyrus rosewateri Gordon, 1986	Elk Pebblesnail	E	G1	MO
Somatogyrus sargenti Pilsbry, 1895	Mud Pebblesnail	U	GU	AL
Somatogyrus strengi Pilsbry and Walker, 1906	Rolling Pebblesnail	U	GU	AL
Somatogyrus substriatus Walker, 1906	Choctaw Pebblesnail	U	GU	AL, MS
Somatogyrus tenax Thompson, 1969	Savannah Pebblesnail	T	G2G3	GA
Somatogyrus tennesseensis Walker, 1906	Opaque Pebblesnail	U	GU	AL, MS, TN
Somatogyrus trothis Doherty, 1878	[uncertain classification]	U	GU	KY
Somatogyrus tryoni Pilsbry and Baker, 1927	Coldwater Pebblesnail	T	G2G3	IL, MN, WI
Somatogyrus virginicus Walker, 1904	Panhandle Pebblesnail	T	G2G3	NC, SC, VA
Somatogyrus walkerianus Aldrich, 1905	Gulf Coast Pebblesnail	T	G2G3	AL, FL
Somatogyrus wheeleri Walker, 1915	Channelled Pebblesnail	Xp	GH	AR
Taylorconcha insperata Hershler, Liu, Frest, Johannes, and Clark, 2006	Unexpected Pebblesnail	E	G1	ID, OR
Taylorconcha serpenticola Hershler, Frest, Johannes, Bowler, and Thompson, 1994	Bliss Rapids Snail	E	G1	ID
Family Pleuroceridae	**7 Genera, 162 species**			
Athearnia anthonyi (Redfield, 1854)	Anthony's Riversnail	E	G1	AL, GA, TN
Athearnia crassa (Haldeman, 1841)	Boulder Snail	X	GX	TN
Elimia acuta (Lea, 1831)	Acute Elimia	T	G2	AL, TN
Elimia alabamensis (Lea, 1861)	Mud Elimia	T	G2	AL
Elimia albanyensis (Lea, 1864)	Black-crest Elimia	V	G3	AL, FL, GA
Elimia ampla (Anthony, 1854)	Ample Elimia	E	G1	AL
Elimia annae Mihalcik and Thompson, 2002	Rainbow Elimia	V	G3	AL
Elimia annettae (Goodrich, 1941)	Lilyshoals Elimia	T	G2	AL
Elimia arachnoidea (Anthony, 1854)	Spider Elimia	T	G2	TN, VA
Elimia aterina (Lea, 1863)	Coal Elimia	T	G2	TN, VA
Elimia athearni (Clench and Turner, 1956)	Knobby Elimia	V	G3Q	FL
Elimia bellacrenata (Haldeman, 1841)	Princess Elimia	E	G1Q	AL
Elimia bellula (Lea, 1861)	Walnut Elimia	E	G1	AL
Elimia boykiniana (Lea, 1840)	Flaxen Elimia	T	G2	AL, GA
Elimia brevis (Reeve, 1860)	Short Spire Elimia	X	GX	AL
Elimia broccata Thompson, 2000	Brooch Elimia	E	G1	AL
Elimia buffyae Mihalcik and Thompson, 2002	Iris Elimia	CS	G4	AL, FL
Elimia bullula (Lea, 1861)	Yellowleaf Elimia	E	G1G2Q	AL
Elimia caelatura (Reeve, 1860)	Savannah Elimia	V	G3	GA
Elimia cahawbensis (Lea, 1841)	Cahaba Elimia	CS	G4	AL
Elimia capillaris (Lea, 1861)	Spindle Elimia	X	GX	AL, GA
Elimia carinifera (Lamarck, 1822)	Sharp-crest Elimia	CS	G5	AL, GA, TN
Elimia carinocostata (Lea, 1854)	Fluted Elimia	CS	G4Q	AL, GA
Elimia catenaria (Say, 1822)	Gravel Elimia	CS	G4	GA, NC, SC, VA
Elimia catenoides (Lea, 1842)	Lirate Elimia	U	GU	AL, GA
Elimia chiltonensis (Goodrich, 1941)	Prune Elimia	E	G1G2	AL
Elimia christyi (Lea, 1862)	Knotty Elimia	T	G2	NC, TN
Elimia clara (Anthony, 1854)	Riffle Elimia	V	G3	AL
Elimia clausa (Lea, 1861)	Closed Elimia	X	GX	AL
Elimia clavaeformis (Lea, 1841)	Club Elimia	CS	G4	NC, TN, VA
Elimia clenchi (Goodrich, 1924)	Slackwater Elimia	V	G3	AL, FL
Elimia cochliaris (Lea, 1868)	Cockle Elimia	E	G1	AL
Elimia comalensis (Pilsbry, 1890)	Balcones Elimia	T	G2	TX
Elimia comma (Conrad, 1834)	Hispid Elimia	T	G2	AL
Elimia costifera (Reeve, 1861)	Corded Elimia	V	G2G4	IL, KY

Taxon	AFS common name	AFS status	G-rank	Inferred distribution
Elimia crenatella (Lea, 1860)	Lacey Elimia	E	G1	AL
Elimia curreyana (Lea, 1841)	Amber Elimia	V	G3	KY, TN
Elimia cylindracea (Conrad, 1834)	Cylinder Elimia	T	G2	AL, MS
Elimia darwini Mihalcik and Thompson, 2002	Pup Elimia	E	G1	GA
Elimia dickinsoni (Clench and Turner, 1956)	Stately Elimia	V	G3	AL, FL
Elimia dislocata (Reeve, 1861)	Lapped Elimia	CS	G4Q	NC, SC, VA
Elimia dooleyensis (Lea, 1862)	Graphite Elimia	CS	G5	AL, FL, GA
Elimia ebenum (Lea, 1841)	Ebony Elimia	CS	G5	KY, TN
Elimia edgariana (Lea, 1841)	Cumberland Elimia	V	G3	KY, TN
Elimia exusta Mihalcik and Thompson, 2002	Fire Elimia	T	G2	AL
Elimia fascinans (Lea, 1861)	Banded Elimia	V	G3	AL
Elimia flava (Lea, 1862)	Yellow Elimia	CS	G4	AL
Elimia floridensis (Reeve, 1860)	Rasp Elimia	CS	G5	FL, GA
Elimia fusiformis (Lea, 1861)	Fusiform Elimia	X	GX	AL
Elimia gibbera (Goodrich, 1922)	Shouldered Elimia	X	GX	AL
Elimia glarea Mihalcik and Thompson, 2002	Gravel Elimia	V	G3	AL
Elimia godwini Thompson, 2000	Rusty Elimia	T	G2	AL
Elimia hartmaniana (Lea, 1861)	High-spired Elimia	X	GX	AL
Elimia haysiana (Lea, 1843)	Silt Elimia	V	G3	AL
Elimia hydeii (Conrad, 1834)	Gladiator Elimia	T	G2	AL
Elimia impressa (Lea, 1841)	Constricted Elimia	X	GX	AL
Elimia inclinans (Lea, 1862)	Slanted Elimia	E	G1G2Q	GA
Elimia interveniens (Lea, 1862)	Slowwater Elimia	T	G2	AL, TN
Elimia induta (Lea, 1862)	Gem Elimia	T	G2	GA
Elimia jonesi (Goodrich, 1936)	Hearty Elimia	X	GX	AL
Elimia lachryma (Reeve, 1861)	Teardrop Elimia	E	G1	AL
Elimia laeta (Jay, 1839)	Ribbed Elimia	X	GX	AL
Elimia laqueata (Say, 1829)	Panel Elimia	CS	G5	AL, KY, TN
Elimia lecontiana (Lea, 1841)	Rippled Elimia	V	G3	AL, GA
Elimia livescens (Menke, 1830)	Liver Elimia	CS	G5	IA, IL, IN, KY, MI, NY, OH, PA, VT, WI; Canada: ON, QC
Elimia macglameriana (Goodrich, 1936)	Wrinkled Elimia	X	GX	AL, GA
Elimia melanoides (Conrad, 1834)	Black Mudalia	T	G2	AL
Elimia mihalcikae Thompson, 2000	Latticed Elimia	E	G1	AL
Elimia modesta (Lea, 1845)	Coldwater Elimia	CS	G5	AL, GA
Elimia mutabilis (Lea, 1862)	Oak Elimia	T	G2Q	GA
Elimia nassula (Conrad, 1834)	Round-ribed Elimia	E	G1	AL
Elimia olivula (Conrad, 1834)	Caper Elimia	E	G1	AL
Elimia ornata (Lea, 1868)	Ornate Elimia	E	G1	GA
Elimia paupercula (Lea, 1862)	Sooty Elimia	CS	G4Q	AL
Elimia perstriata (Lea, 1852)	Engraved Elimia	E	G1	AL
Elimia pilsbryi (Goodrich, 1927)	Rough-lined Elimia	X	GX	AL
Elimia plicatastriata (Wetherby, 1876)	Carved Elimia	T	G2G3	KY, TN
Elimia porrecta (Lea, 1863)	Nymph Elimia	T	G2	TN
Elimia potosiensis (Lea, 1841)	Pyramid Elimia	CS	G5	AR, KS, MO, OK
Elimia proxima (Say, 1825)	Sprite Elimia	CS	G5	GA, NC, SC, VA, WV
Elimia pupaeformis (Lea, 1864)	Pupa Elimia	X	GX	AL
Elimia pupoidea (Anthony, 1854)	Bot Elimia	X	GX	AL
Elimia pybasii (Lea, 1862)	Spring Elimia	T	G2Q	AL
Elimia pygmaea (Smith, 1936)	Pygmy Elimia	X	GX	AL
Elimia semicarinata (Say, 1829)	Fine-ridged Elimia	CS	G5	IN, KY, OH
Elimia showalterii (Lea, 1860)	Compact Elimia	E	G1	AL
Elimia simplex (Say, 1825)	Smooth Elimia	CS	G5	NC, TN, VA, WV
Elimia striatula (Lea, 1842)	File Elimia	T	G2	GA, TN
Elimia strigosa (Lea, 1841)	Brook Elimia	T	G2	TN

Taxon	AFS common name	AFS status	G-rank	Inferred distribution
Elimia symmetrica (Haldeman, 1841)	Symmetrical Elimia	CS	G4Q	NC, VA
Elimia taitiana (Lea, 1841)	Dented Elimia	V	G3Q	AL, FL
Elimia teres (Lea, 1841)	Elegant Elimia	E	G1	TN
Elimia teretria Thompson, 2000	Auger Elimia	E	G1	AL
Elimia timida (Goodrich, 1942)	Timid Elimia	E	G1	GA
Elimia troostiana (Lea, 1838)	Mossy Elimia	E	G1	TN
Elimia ucheensis (Lea, 1862)	Creek Elimia	V	G3	AL
Elimia vanhyningiana (Goodrich, 1921)	Goblin Elimia	CS	G5	FL
Elimia vanuxemiana (Lea, 1843)	Cobble Elimia	E	G1Q	AL
Elimia varians (Lea, 1861)	Puzzle Elimia	T	G2Q	AL
Elimia variata (Lea, 1861)	Squat Elimia	T	G2Q	AL
Elimia viennaensis (Lea, 1862)	Slough Elimia	CS	G4	AL, GA
Elimia virginica (Say, 1817)	Piedmont Elimia	CS	G5	CT, MA, MD, NC, NJ, NY, PA, VA, WV
Gyrotoma excisa (Lea, 1843)	Excised Slitshell	X	GX	AL
Gyrotoma lewisii (Lea, 1869)	Striate Slitshell	X	GX	AL
Gyrotoma pagoda (Lea, 1845)	Pagoda Slitshell	X	GX	AL
Gyrotoma pumila (Lea, 1860)	Ribbed Slitshell	X	GX	AL
Gyrotoma pyramidata (Shuttleworth, 1845)	Pyramid Slitshell	X	GX	AL
Gyrotoma walkeri (Smith, 1924)	Round Slitshell	X	GX	AL
Io fluvialis (Say, 1825)	Spiny Riversnail	T	G2	AL, GA, TN, VA
Leptoxis ampla (Anthony, 1855)	Round Rocksnail	T	G2	AL
Leptoxis arkansensis (Hinkley, 1915)	Arkansas Mudalia	E	G1	AR, MO
Leptoxis carinata (Bruquiere, 1792)	Crested Mudalia	CS	G5	MD, NC, NJ, NY, PA, VA, WV
Leptoxis clipeata (Smith, 1922)	Agate Rocksnail	X	GX	AL
Leptoxis compacta (Anthony, 1854)	Oblong Rocksnail	E	G1	AL
Leptoxis dilatata (Conrad, 1835)	Seep Mudalia	V	G3	NC, PA, VA, WV
Leptoxis foremani (Lea, 1843)	Interrupted Rocksnail	E	G1	AL, GA
Leptoxis formosa (Lea, 1860)	Maiden Rocksnail	X	GX	AL, GA
Leptoxis ligata (Anthony, 1860)	Rotund Rocksnail	X	GX	AL
Leptoxis lirata (Smith, 1922)	Lirate Rocksnail	X	GX	AL
Leptoxis minor (Hinkley, 1912)	Knob Mudalia	X	GX	AL
Leptoxis occultata (Smith, 1922)	Bigmouth Rocksnail	X	GX	AL
Leptoxis picta (Conrad, 1834)	Spotted Rocksnail	E	G1	AL
Leptoxis plicata (Conrad, 1834)	Plicate Rocksnail	E	G1	AL
Leptoxis praerosa (Say, 1821)	Onyx Rocksnail	CS	G5	AL, GA, IL, IN, KY, OH, TN, VA
Leptoxis showalterii (Lea, 1860)	Coosa Rocksnail	X	GX	AL
Leptoxis taeniata (Conrad, 1834)	Painted Rocksnail	E	G1	AL
Leptoxis torrefacta (Goodrich, 1922)	Squat Rocksnail	X	GX	AL
Leptoxis trilineata (Say, 1829)	Broad Mudalia	X	GX	IN, KY, OH
Leptoxis umbilicata (Wetherby, 1876)	Umbilicate Rocksnail	E	G1Q	TN
Leptoxis virgata (Lea, 1841)	Smooth Mudalia	T	G2	AL, NC, TN, VA
Leptoxis vittata (Lea, 1860)	Stripped Rocksnail	X	GX	AL
Lithasia armigera (Say, 1821)	Armored Rocksnail	V	G3G4	AL, IL, IN, KY, OH, TN, WV
Lithasia curta (Lea, 1868)	Knobby Rocksnail	E	G1	AL, KY, TN
Lithasia duttoniana (Lea, 1841)	Helmet Rocksnail	T	G2	TN
Lithasia geniculata Haldeman, 1840	Ornate Rocksnail	V	G3	AL, IL, KY, TN
Lithasia hubrichti Clench, 1956	Big Black Rocksnail	X	GX	MS
Lithasia jayana (Lea, 1841)	Rugose Rocksnail	X	GX	TN
Lithasia lima (Conrad, 1834)	Warty Rocksnail	T	G2	AL, MS, TN
Lithasia obovata (Say, 1829)	Shawnee Rocksnail	CS	G4	IL, IN, KY, OH, PA, TN
Lithasia salebrosa (Conrad, 1834)	Muddy Rocksnail	V	G3	AL, KY, TN
Lithasia spicula Minton, Savarese, and Campbell, 2005	Harpeth Rocksnail	E	G1	TN
Lithasia verrucosa (Rafinesque, 1820)	Varicose Rocksnail	CS	G4	AL, AR, IN, IL, KY, NC, OH, PA, TN, WV

Taxon	AFS common name	AFS status	G-rank	Inferred distribution
Pleurocera acuta Rafinesque, 1831	Sharp Hornsnail	CS	G5	AR, IA, IL, IN, KS, KY, LA, MI, MN, MO, MS, NE, NY, OH, PA, TN, VT, WI, WV; Canada: ON, QC
Pleurocera alveare (Conrad, 1834)	Rugged Hornsnail	CS	G4	AL, AR, IL, IN, KY, MO, TN
Pleurocera annulifera (Conrad, 1834)	Ringed Hornsnail	V	G3	AL
Pleurocera brumbyi (Lea, 1852)	Spiral Hornsnail	T	G2	AL
Pleurocera canaliculata (Say, 1821)	Silty Hornsnail	CS	G5	AL, AR, IL, IN, KY, LA, MS, OH, PA, TN, VA, WV
Pleurocera corpulenta Anthony, 1854	Corpulent Hornsnail	E	G1	AL, TN
Pleurocera curta (Haldeman, 1841)	Shortspire Hornsnail	T	G2	AL, KY, TN
Pleurocera foremani (Lea, 1843)	Rough Hornsnail	E	G1	AL
Pleurocera gradata (Anthony, 1854)	Bottle Hornsnail	V	G3	TN, VA
Pleurocera nobilis (Lea, 1845)	Noble Hornsnail	T	G2Q	AL, TN
Pleurocera parva (Lea, 1862)	Dainty Hornsnail	V	G3	NC, TN
Pleurocera postelli (Lea, 1862)	Broken Hornsnail	T	G2	AL
Pleurocera prasinata (Conrad, 1834)	Smooth Hornsnail	CS	G4	AL
Pleurocera pyrenella (Conrad, 1834)	Skirted Hornsnail	T	G2	AL, GA
Pleurocera showalteri (Lea, 1862)	Upland Hornsnail	T	G2Q	AL, GA
Pleurocera striatum (Lea, 1863)	Striate Hornsnail	T	G2Q	AL, GA
Pleurocera trochiformis (Conrad, 1834)	Sulcate Hornsnail	T	G2Q	AL, GA, TN
Pleurocera uncialis (Reeve, 1861)	Pagoda Hornsnail	CS	G4	NC, TN, VA
Pleurocera vestita (Conrad, 1834)	Brook Hornsnail	V	G3	AL, GA
Pleurocera walkeri Goodrich, 1928	Telescope Hornsnail	V	G3	AL, GA, KY, TN
Family Semisulcospiridae	**1 Genus, 11 species**			
Juga acutifilosa (Stearns, 1890)	Topaz Juga	T	G2	CA, OR
Juga bulbosa (Gould, 1847)	Bulb Juga	E	G1	OR
Juga chacei (Henderson, 1935)	Chace Juga	E	G1	CA, OR
Juga hemphilli (Henderson, 1935)	Barrren Juga	T	G2	OR, WA; Canada: BC
Juga interioris (Goodrich, 1944)	Smooth Juga	E	G1	NV
Juga laurae (Goodrich, 1944)	Oasis Juga	E	G1	CA, NV
Juga newberryi (Lea, 1860)	Banded Juga	E	G1	OR
Juga nigrina (Lea, 1856)	Black Juga	V	G3	CA, NV, OR
Juga occata (Hinds, 1844)	Scalloped Juga	E	G1	CA
Juga plicifera (Lea, 1838)	Pleated Juga	V	G3	CA, OR, WA; Canada: BC
Juga silicula (Gould, 1847)	Glassy Juga	CS	G4	WA; Canada: BC
Family Pomatiopsidae	**1 Genus, 6 species**			
Pomatiopsis binneyi Tryon, 1863	Robust Walker	E	G1	CA, OR
Pomatiopsis californica Pilsbry, 1899	Pacific Walker	E	G1	CA, OR
Pomatiopsis chacei Pilsbry, 1937	Marsh Walker	E	G1	CA, OR
Pomatiopsis cincinnatiensis (Lea, 1840)	Brown Walker	CS	G4	IA, IL, IN, KY, MI, OH, TN, VA
Pomatiopsis hinkleyi Pilsbry, 1896	Tennessee River Walker	X	GXQ	AL, TN
Pomatiopsis lapidaria (Say, 1817)	Slender Walker	CS	G5	AL, AR, CT, DE, GA, FL, IA, IL, IN, KS, KY, LA, MA, MD, MI, MN, MO, MS, NC, NJ, NM, NY, OH, OK, PA, SC, SD, TN, TX, VA, WI, WV; Canada: ON, QC
Family Valvatidae	**1 Genus, 10 species**			
Valvata bicarinata Lea, 1841	Two-ridge Valvata	CS	G5	AL, AR, GA, IA, IL, IN, KY, MI, NC, NJ, NY, PA, TN, VA, WI
Valvata humeralis Say, 1829	Glossy Valvata	CS	G5Q	AZ, CA, CO, ID, MT, NV, OR, UT, WA, WY; Canada: BC
Valvata lewisi Currier, 1868	Fringed Valvata	CS	G5	AK, IA, IN, ME, MI, MN, MT, NY, VT, WA, WI; Canada: AB, BC, LB, MB, NB, NF, NS, NT, ON, PE, QC, SK, YT
Valvata mergella Westerlund, 1883	Rams-horn Valvata	T	G2	AK, WA; Canada: BC
Valvata perdepressa Walker, 1906	Purplecap Valvata	V	G3	IL, IN, MI, NY, OH, PA, WI; Canada: ON
Valvata sincera Say, 1824	Mossy Valvata	CS	G5	AK, CO, CT, IA, ID, IL, IN, MA, ME, MI, MN, MT, NC, ND, NH, NY, PA, SD, VT, WI, WY; Canada: AB, BC, LB, MB, NT, NU, ON, QC, SK, YT
Valvata tricarinata (Say, 1817)	Threeridge Valvata	CS	G5	AR, CT, IA, ID, IL, IN, KS, KY, MA, MD, ME, MI, MN, MT, ND, NE, NH, NJ, NY, OH, PA, RI, SD, VA, VT, WA, WI, WY; Canada: AB, BC, MB, NB, NT, ON, QC, SK
Valvata utahensis Call, 1884	Desert Valvata	E	G1	ID, UT
Valvata virens Tryon, 1863	Emerald Valvata	Xp	GH	CA
Valvata winnebagoensis Baker, 1928	Flanged Valvata	T	G2	MI, MN, WI; Canada: ON

Effects of Anthropogenic Chemicals on Chemosensation and Behavior in Fish: Organismal, Ecological, and Regulatory Implications

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During the 2012 American Fisheries Society annual meeting in St. Paul, Minnesota, a half-day session examined the effects of contaminants on chemosensation and behavior in fish. Talks were presented by Canadian and American scientists representing business, government, and academia, and they included diverse topics ranging from basic olfactory neurophysiology to regulatory implications of contaminant-induced behavioral effects. Session participants engaged in a stimulating discussion session following a full slate of talks, resulting in ideas about future research directions and policy implications.

Of the 10 presentations during the session, two focused on organic contaminants and the others focused primarily on effects of metals. The two presentations on organic contaminants included a case study involving organic contaminants released to the North Saskatchewan River from a wastewater treatment plant in Edmonton, Alberta, and a study of the effects of one class of polychlorinated biphenyls on neuromuscular pathways in fish, which could result in alteration of behaviors due to impaired motor function.

The remaining eight presentations covered a wide range of topics about the effects of metals on biochemical, cellular, behavioral, and toxicological responses and ecological and regulatory implications of chemosensory and behavioral responses. Although most work focusing on the effects of metals on fish olfaction and behavior takes place under controlled conditions using typical model species in the laboratory, some effort is being directed toward understanding contaminant effects on nonstandard fish species (e.g., White Sturgeon [*Acipenser transmontanus*]) and on wild fish populations (e.g., Yellow Perch [*Perca flavescens*] from metal-contaminated lakes in the industrial region of Sudbury, Ontario). Additionally, chemosensory and behavior studies are being conducted on other ecosystem components, including invertebrates. Although they are generally not considered to be recreationally or commercially important species, chemosensory effects on invertebrates can potentially lead to a trophic cascade of ecological effects caused by their morphological and/or behavioral responses.

Take-home messages from the symposium include the following:

1. In recent years, the breadth of testing of chemosensory responses to chemicals has increased considerably (i.e., more species and a finer scale at cellular and molecular/gene levels), thus increasing the understanding of organismal and potential ecological and regulatory implications of chemosensory responses.

2. A variety of organic chemicals and metals can cause chemosensory and behavioral impairment in fish and at least some aquatic invertebrates.

3. A variety of biochemical/physiological pathways can lead to those impairments and are not the same among all chemicals, and not even among the same classes of chemicals (e.g., at relatively low concentrations, copper and nickel target different olfactory sensory neurons).

4. A distinction should be made between "detection" and "perception," whereby the former refers to a fundamental physiological response to a chemosensory cue (for which there is some current understanding) and the latter refers to the potential subsequent interpretation of the cue in an ecological context by an organism (for which there is much less current understanding).

5. Although gene expression can be a useful forensic tool in many areas of toxicology, behavioral and neurophysiological assays are probably more appropriate for evaluating olfactory effects in wild fish than are gene expression assays.

6. The U.S. Environmental Protection Agency's (USEPA) ambient water quality criteria for copper appear to be protective against olfactory impairment in most fish studies conducted to date; however, levels of protection are improved if biotic ligand model (BLM)-based criteria are used instead of hardness-based criteria.

7. The USEPA's BLM-based and hardness-based copper criteria are generally protective against olfactory and behavior impairment in fish because the traditional growth and reproduction endpoints for invertebrates (which tend to "drive" metals criteria) are even more sensitive than fish olfaction;

therefore, for an olfaction or behavior endpoint in a fish species to be used to adjust water quality criteria downward, the olfactory or behavioral impairment would have to be judged "biologically important" and would have to occur in an "important species" at a concentration lower than the sensitive endpoints for invertebrates.

8. Studies that report contaminant-induced chemosensory and behavioral effects (especially for metals) should report relevant exposure–water chemistry, so the results can be interpreted in the proper context and can be used in olfactory- and behavior-parameterized BLMs.

9. Authors should report whether concentrations of chemicals that impair olfaction or behavior responses exceeded or did not exceed ambient water quality criteria concentrations that would be calculated for the water chemistry to which the organisms were exposed in the laboratory or in the field.

10. Caution should be used when extrapolating results of laboratory olfaction and behavior studies to the field, giving special consideration to the water to which the organisms are adapted.

11. Research is needed to determine whether metals that are taken up from the environment by way of the olfactory system and then accumulate in the brain can cause behavioral impairment.

12. More work is needed to understand the effects of metals on marine chemosensory systems and the role of competing cations in either ameliorating or exacerbating those effects.

13. Although there is some evidence that contaminant-impaired chemosensory systems can recover relatively rapidly (e.g., minutes to hours), the implications of that recovery to ecological fitness or for use in risk assessment are not known at this time.

14. More realistic experimental systems, including multiple species and multiple contaminants (possibly tested outside the laboratory), could help improve the ecological interpretation of the results regarding chemosensory and behavioral impairment.

15. Any models that are developed either to predict chemosensory or behavioral effects or to establish environmental criteria based on those effects should be tested and validated empirically.

ACKNOWLEDGMENTS

Preparation of this article was supported by funding from the Copper Development Association (to J.S.M.) and from the Natural Sciences and Engineering Research Council (to G.G.P.).

New and Ongoing Society Initiatives to Craft a Lasting Partnership between AFS and All Things Aquaculture

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The American Fisheries Society—representing all of the fisheries disciplines—is the common denominator for all those interested in aquaculture, fisheries, and related fields ... [this is] our most powerful role and greatest responsibility—to help create and shape the shoal of aquaculture stakeholders. (Trushenski et al. 2012, p. 396)

Or, to put it more succinctly, get up to speed, get engaged, and get going. The text above is quoted from an article appearing in *Fisheries* last year that challenged the American Fisheries Society (AFS) and its members to recognize our collective role in supporting the sustainable growth of commercial aquaculture. Since then, other items have appeared in *Fisheries*, highlighting the society's renewed interest in and commitment to all things aquaculture. Public or private, food fish or sport fish, imperiled stocks or commonplace ornamentals, the AFS is taking up the charge laid before it and assuming an active role in the effective production and prudent use of cultured fishes. As an AFS member, fisheries professional, fish culturist, aquarium hobbyist, voracious consumer of seafood, natural resource advocate, and enthusiastic (albeit mostly inept) angler, I am proud to see the society acting to ensure that fish culture and cultured fishes are effectively integrated into sound, science-based stewardship of aquatic resources.

The "AFS and Aquaculture" article (Trushenski et al. 2012) served to crystallize the intent of the AFS leadership to craft the society's contemporary position regarding commercial aquaculture. The society's positions are commonly articulated in the form of policy statements, and there is an existing policy addressing commercial aquaculture. The policy states, "The American Fisheries Society supports the continued development of commercial aquaculture as an important source of food, potential fisheries enhancement, and business opportunity" (AFS 1990). This much remains the same; if our collective position has changed at all, it has been to strengthen our interest in the sustainable growth of commercial aquaculture. Beyond this statement of support, however, the current policy on commercial aquaculture is anything but current: originally dating back to the 1980s, the policy does not reflect the aquaculture industry, demand for fishery products, or our society as they exist today. Recognizing the need to address aquaculture—as it is today and how the society views it—President John Boreman tasked the AFS Resource Policy Committee with revising the commercial aquaculture policy. Partnering with the National Aquaculture Association, the U.S. Aquaculture Society, and World Aquaculture Society, the Resource Policy Committee has assembled a diverse team to revise and reinvigorate the AFS policy on commercial aquaculture. Jim Bowker, Gary Fornshell, Jeff Hill, Jonathan Leiman, Randy MacMillan, Diane Windham, and Jesse Trushenski have outlined a revised policy and hope to bring a draft before the society later this year. The global community has come to rely on aquaculture: roughly 50% of all seafood now comes from farms. A sustainable seafood supply that meets demand for fish and shellfish now and in the future means aquaculture. Our revised policy on commercial aquaculture will reflect these realities and the society's principles of science-based, ethically and professionally sound advocacy.

Regarding public aquaculture and the use of cultured fish in fisheries enhancement and restoration, the Hatcheries and Management of Aquatic Resources (HaMAR) initiative (Trushenski and Bowker 2013) continues to progress, and the associated symposium is slated to be one of the largest at the upcoming AFS annual meeting in Little Rock, Arkansas. Beyond the efforts of the HaMAR committee, hatchery operation and the use of cultured fish are once again becoming integral components of AFS meeting programs (Trushenski 2013). Those attending the Western Division meeting heard the importance of even greater integration among the fisheries disciplines in Stuart Leon's (former Division Chief, Fish and Wildlife Service, Fisheries and Aquatic Resource Conservation) plenary presentation on the past, present, and future of fisheries stewardship. Hatcheries and hatchery-origin fish will continue to be a central part of fisheries conservation and contribute to the completion of management objectives. Greater representation of fish culture and allied disciplines within the society and at our meetings will undoubtedly facilitate greater "cross-pollination" and success in the field. Fish culturists returning to our society's ranks is a welcome sight, personally and professionally.

The AFS is getting up to speed, getting engaged, and getting going in aquaculture. Look for updates on the AFS's aquaculture-related projects and other advancements in the rearing and use of cultured fish and shellfish in a future concept issue of *Fisheries*.

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JOURNAL HIGHLIGHTS

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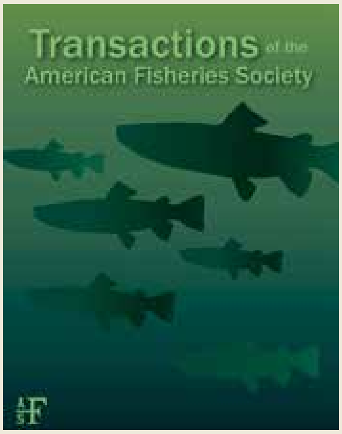
Volume 142, Number 3, May 2013



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CALENDAR
Fisheries Events

To submit upcoming events for inclusion on the AFS web site calendar, send event name, dates, city, state/province, web address, and contact information to sgilbertfox@fisheries.org.

(If space is available, events will also be printed in Fisheries magazine.)

More events listed at www.fisheries.org

DATE	EVENT	LOCATION	WEBSITE
June 24–28, 2013	9th Indo-Pacific Fish Conference	Okinawa, Japan	fish-isj.jp/9ipfc
June 25–27, 2013	2013 International Conference on Engineering & Ecohydrology for Fish Passage	Corvallis, OR	fishpassage.umass.edu Contact: Dr. Guillermo R. Giannico at giannico@oregonstate.edu
July 14–20, 2013	2nd International Conference on Fish Telemetry	Grahamstown, South Africa	oceantrackingnetwork.org
July 15–19, 2013	The World Conference on Stock Assessment Methods for Sustainable Fisheries	Boston, MA	ices.dk/iceswork/symposia/wcsam.asp
July 21–25, 2013	7th International Symposium on Sturgeon	Nanaimo, Canada	iss7.viu.ca
August 9–12, 2013	Aquaculture Europe 13	Trondheim, Norway	easonline.org/images/stories/Meetings/AE2013/AE2013_Brochure_final.pdf
August 19–23, 2013	Aquatic Science at the Interface	Hamilton, New Zealand	aquascience.org.nz
August 26–27, 2013	Trout Unlimited's 2013 Utah Single Fly Event - To protect Utah's rivers and fight the spread of aquatic invasive species.	Green River, Dutch John, UT	tu.org/events/2013UTSF
September 23–25, 2013	2nd Annual World Congress of Mariculture and Fisheries-2013 (WCMF-2013)	Hangzhou, China	bitconferences.com/wcmf2013/default.asp
September 23–26, 2013	OCEANS '13 MTS/IEEE - The Largest Ocean Conference in U.S. History	San Diego, CA	oceans13mtsieeesandiego.org.
September 28–October 4, 2013	2013 World Seafood Conference	Newfoundland and Labrador, Canada	wsc2013.com
October 7–11, 2013	AFS 40th Annual Meeting of the Alaska Chapter of AFS	Fairbanks, AK	afs-alaska.org/annual-meetings/2011-2
October 21–27, 2013	3rd International Marine Protected Areas Congress	Marseille, France	impac3.org
August 3–7, 2014	International Congress on the Biology of Fish	Edinburgh, United Kingdom	icbf2014.sls.hw.ac.uk

NEW AFS MEMBERS

Daniel Aboagye
Felix Ayson
Harry M Barnes MD
Bill Beckett
Maximillian Bertetti
Emily Campbell
Mikaela Campbell
Michael Ciaramella
Bud Downs
Shelley Edmundson
Joanna Field
Tyler Grabowski
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Liran Haller
David Hamilton
Jessica Helsley
Jeannie Heltzel
Kjetil Henderson
Nick Huber
Zach Klein
Seunghyung Lee
Sarah Moffitt
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Russell Nolan
Douglas Peck
Brian Perleberg
Shane Ramee
Charles Reeves
Bernard Sainte-Marie
Jacob Sengele
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Ronald Taylor
Rebecca Vito
Kristin Wright
Michel Ybarrondo
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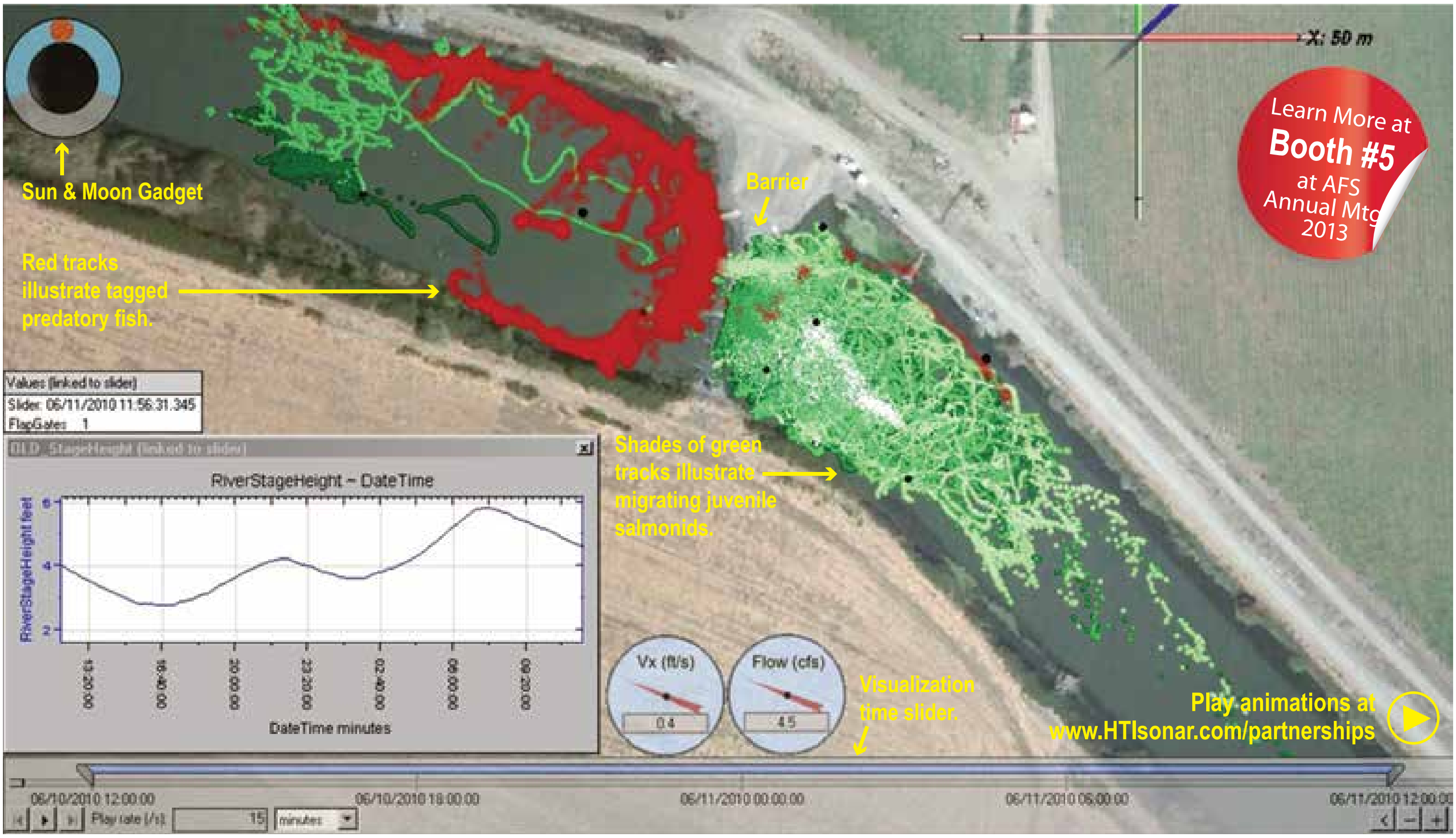

Study Data Courtesy: California Dept. of Water Resources Visualization Software: Eonfusion Acoustic Telemetry System: HTI

Example of a juvenile salmonid migrating to the ocean each spring. Predation is a significant challenge for juvenile salmonid survival. Smolt image courtesy fishbio.com.

Example of a salmon predator, the largemouth bass (sp. *Micropterus salmoides*) is found in North American river environments. Several countries report adverse ecological impact after introduction. Reference Fishbase.org & image courtesy fishbio.com.

The Importance of Partnerships in Fisheries Research

Collaboration is a vital part of fisheries research. Effective partnerships have specialized expertise and sensitivities to address complex challenges. With a more diversified perspective, we can better understand and see challenges and opportunities. We can deliver new knowledge with greater agility in developing new technologies that may even lead to the next generation of breakthroughs. We also build relationships benefitting the sciences as a whole.

A good example of the importance of partnerships for fisheries research is Eonfusion and HTI. Both support non-exclusive partnerships sharing the best of insights and stimulate new questions along the way. Eonfusion is a 4D analysis software application especially designed for time-varying challenges. HTI is a leading designer, manufacturer, and user of acoustic telemetry systems for monitoring fish survival, passage, and behavior (often over time). Together, they can do what may seem impossible, such as revealing high-resolution predator/prey behavior within unique environmental variables. Eonfusion uses time as a fully-fledged geospatial axis which can include a plethora of environmental data sources.

Within a scene, Eonfusion and HTI are able to animate HTI's acoustic tag track data concurrently with key variables, e.g., tidal data, velocity, flow, sun and moon gadget, as well as barrier/gate operations. The result is data-rich visualizations created within a geo-referenced study area, as shown in the salmon smolts/largemouth bass tracks illustrated above (courtesy of the California Department of Water Resources).

Together they create a concert of data unlike anything seen before in fisheries science. To see the animation of predator/prey example, visit www.HTIsonar.com/partnerships. To learn more about Eonfusion's fisheries ecology applications, visit www.eonfusion.com.

> **"I'm a big advocate of data visualization and the HTI & Eonfusion folks work very well together. The ability to visualize time series fish tracks along with environmental variables & hydrodynamics has improved our ability to understand complicated smolt & predator behavior."**
>
> - Jacob McQuirk, Supervising Engineer, California Dept. of Water Resources