Lissachatina fulica

Figure 1. Giant African snail (Yuri Yashin, achatina.ru, Bugwood.org)

Scientific Name
Lissachatina fulica (Bowdich, 1822)

Synonyms:
Achatina fulica Bowdich, 1822

Common Names
giant African snail, giant east African snail, giant African land snail, escargot Géant, caramujo, caracol africano gigante

Type of Pest
Mollusk

Taxonomic Position
Class: Gastropoda, **Order**: Stylommatophora, **Family**: Achatinidae

Reason for Inclusion
CAPS Target: AHP Prioritized Pest List for FY 2011-present

Figure 2. Eggs of *Lissachatina fulica* with size reference (David Robinson, USDA-APHIS-PPQ).

Pest Description
From Bequaert (1950):

Eggs: "The egg is broadly ellipsoidal, 5 to 5.5 mm by 4 to 4.5 mm [approx $^{3}/_{16}$ in] (Fig. 2)… white to [bright yellow], with a very thin and brittle calcareous shell."

Adults: "When full-grown, the shell of *Lissachatina fulica* consists of from 7 to 9 (very exceptionally 10) whorls, with a moderately swollen body-whorl and a sharply conical spire, which is distinctly narrowed but scarcely drawn out at the apex. The outline varies greatly, even in the same colony, from very slender to moderately obese, the broader specimens tending to be shorter for the same number of whorls. All whorls are decidedly convex, due to the broadly impressed sutures. The aperture is relatively short, even in the broadest specimens, being always shorter than the spire, often considerably so. The outer lip is usually sharp and thin, rarely somewhat thickened or even slightly expanded in very old specimens; it is very convex, evenly curved into a regular semi-ellipse, and inserted on the body-whorl at a sharp, open angle, the upper part of the body-whorl being scarcely or not flattened behind the lip. The columella is more or less concave, sometimes rather weakly so, in which case may be slightly or even much twisted; it tends to be more concave in the broader shells. It should be noted that in *L. fulica* all stages, from the nepionic shell on, have the umbilical slit completely closed and the columella truncate. In all specimens seen, which on general shape and sculpture were referable to *L. fulica*, both columella and parietal callus are white or bluish- white, without any trace of pink."

Figure 3. *Lissachatina fulica* shell (David Robinson, USDA-APHIS-PPQ).

The newly hatched, nepionic shell is 5-5.5 mm. long and about 4.5 mm wide [approx $^{3}/_{16}$ in], of 2½ nearly smooth whorls, without any granulation or decussation and with only the weakest traces of vertical wrinkles. The first half post-nepionic whorl is more distinctly wrinkled vertically, but not yet decussate; after this the vertical growth striae become gradually stronger and are now cut by spiral engraved lines, into elongate, vertical welts; at first weak, the decussation gains in strength on the fourth and fifth whorls, where it is visible to the naked eye, after which it decreases again, being superficial on the sixth and usually lacking on the seventh and succeeding whorls. In young shells, the periostracum of the early post-nepionic whorls shows in addition to the decussation a superficial, microscopic criss-cross texture, as if a finely woven cloth had been pressed onto the surface; but no trace of this remains on older shells. In the largest full-grown adult shells, of 7-9 whorls, the body whorl is nearly even, the growth-striae being very low or superficial, except below the suture where they form short, strong folds, very lightly crenulating the irregular sutural line. When the periostracum is fresh and intact, the terminal whorls appear not only smooth, but also glossy. When present in the adult, the darker markings appear almost at once on the first post-nepionic whorl as faint, vertical, straight, pale-brown streaks; in very young shells, these streaks stop at the periphery, forming there slightly deflected spots."

Shell coloration may be variable due to environmental conditions and diet (reviewed in USDA-APHIS, 2007). An identification guide for *L. fulica* can be found in USDA-APHIS (2007). A description of the shell and measurements can be found in Mitra et al. (2005).

Biology and Ecology

"This species is highly adaptable to a wide range of environments, modifying its life cycle to suit local conditions. It is one of the greatest threats to agriculture and the environment worldwide due to its reproductive capacity, destructiveness to plants, threat to human health, and large size. *Lissachatina fulica* prefers environments that are rich in calcium carbonate, such as limestone, marl, and developed areas with an abundance of cement or concrete" (USDA-APHIS, 2007). Optimal development of this species requires a calcium carbonate content of the substratum (calcium in the soil) of at least 3–4%. Lack of this results in slow growth, high mortality, cannibalism, and a cease to reproduction (Godan, 1983).

"Snails reach sexual maturity in less than one year. The average life span is 3-5 years, but individuals may reach the age of 9 years" (USDA-APHIS, 2007). *Lissachatina fulica* is a hermaphroditic snail but is usually required to mate to reproduce. This species uses neither a dart nor a spermatophore when mating (Chase, 2001). Egg laying can begin around six months of age with snails remaining fecund for approximately 400 days (Smith and Fowler, 2003). "Individuals produce from 10 to more than 400 eggs, 8-20 days after mating. Under optimum conditions, 300 to 1000 eggs in 3 to 4 batches may be laid each year" (USDA-APHIS, 2007). Hatching occurs after about 1-17 days. Hatchlings of *L. fulica* will remain beneath the soil surface for a few to several days while consuming their eggshells and organic matter within the soil (Raut and Barker, 2002).

After emergence from the soil, immature larvae begin to feed voraciously; snails ranging from 5-30 mm prefer living vegetation while smaller and older snails prefer decaying vegetation and detritus (Smith and Fowler, 2003). Growth rate of this species is correlated with feeding and excretion rate, which is tied to dietary calcium concentrations (reviewed in Dimitriadis, 2001).

Gradually, the smaller snails will disperse from their hatching site and establish home sites. Each day, the snails forage throughout the night and return to their home sites before dawn (Raut and Barker, 2002).

"This species is nocturnal, but may become active at twilight if the day is overcast and the soil is moist and warm. The snail is extremely sensitive to high rates of evaporation. Under moisture stress, it becomes inactive and begins estivating within 24 hours [estivation is when an animal becomes dormant during hot and dry weather conditions]. However, estivation can occur independent of moisture. Scientists believe that estivation may be cyclic. Snails may estivate as they cling to objects, aiding in their inadvertent spread to new areas on cargo, vehicles or machinery.

During unfavorable periods, the snail buries itself 10-15 cm (4-6 inches) deep in soft soil and may become inactive for up to a year, losing 60% of its weight. Physiological changes in blood and certain organs occur before and during the period of inactivity. This species can reproduce in areas that are too dry for other large snails (Hardouin et al., 1995) (Snail Draft Generic Action Plan, 1986; modified by IICA, 2002; Srivastava, 1985)" (USDA-APHIS, 2007).

Although *L. fulica* is considered a tropical species, it can remain active in temperatures ranging from 9-29°C (48.2-84.2°F). *Lissachatina fulica* can hibernate to survive temperatures of 2°C (35.6°F) and estivate in temperatures of 30°C (86°F) (Smith and Fowler, 2003). *Lissachatina fulica* can remain in this state for several months (Smith and Fowler, 2003). Estivation occurs in moist soil but can occur at sites above-ground (Raut and Barker, 2002).

Figure 4. Damage to Leaves of *Dieffenbachia* spp. Caused by *Lissachatina fulica* (Department of Plant Industry, Florida)

Damage

Lissachatina fulica prefers to feed on plants in the seedling or nursery stage. Damage to the seedling stage can be so severe that farmers change crop species grown. In more mature plants, the type of damage can vary depending on the plant species. Symptoms can range from defoliation to damage of stems, fruits or flowers (Fig. 4, 5, 6) (Raut and Barker, 2002). Due to their large size these snails can eat voraciously (Wilson, 2017).

Figure 5. Damage to Leaves of *Heliconia* spp. caused by *Lissachatina fulica* (Department of Plant Industry, Florida).

Visual signs of *L. fulica* can include chewing or rasping damage to plants, presence of eggs, juveniles and adults, empty snail shells, mucus and slime trails, large-ribbon like feces, and/or an increase in rat population densities in an area (USDA-APHIS, 2007).

Pest Importance

Lissachatina fulica is considered a significant pest of agricultural crops as well as a vector of a number of human, plant, and animal pathogens.

Efforts by the United States to eradicate *L. fulica* from Florida in the late 1960s into the 1970s cost an estimated $700,000. If the infestation had gone undetected, it is estimated that annual losses would have reached $11 million (in 1969 dollars) (USDA-APHIS, 1982). Raut and Barker (2002) also state that “opportunity losses associated with enforced changes in agricultural practice, such as limiting the crop species to be grown in a region to those resistant to *L. fulica*" may occur. In 2011, a new eradication program began in Florida after a new population of *L. fulic*a was discovered there (Fontanilla et al, 2014; Roda, 2016).

Figure 6. Damage to leaves of *Sanchezia nobilis* caused by *Lissachatina fulica* (Department of Plant Industry, Florida).

The species is believed to have originated in the coastal Kenya and Tanzania in East Africa (Raut and Barker, 2002), but was introduced to the Indian Ocean islands of Madagascar, Mauritius, the Comoros, Mayotte and Reunion prior to 1800 (Fontanilla et al, 2014). Based on genetic analysis, outside of East Africa and the neighboring Indian Ocean islands, the snails show little genetic diversity-an example of the founder effect (Fontanilla et al, 2014). *Lissachatina fulica* has spread rapidly over the last 200 years across the tropics and has caused problems such as: previously threatened production of bananas and coconuts in Samoa; displaced small agricultural producers in Brazil, causing an increase in food prices and importation of food; and caused severe vegetable crop damage in parts of India (Cowie, 2006; Fontanilla et al., 2014; reviewed in USDA-APHIS, 2007). It is a pest of *Gossypium* (cotton) seedlings in Mauritius, *Artocarpus* (bread fruit tree) in Saipan and various Pacific Islands, *Crotalaria* (East Indian hemp) in Indonesia, *Coffea* (coffee tree) in Tanzania, *Theobroma cacao* (cocoa tree) in Sri Lanka, *Hevea* (rubber tree) in Malaysia and Sri Lanka, *Crotalaria* (rattlebox) in Indonesia, Leguminosae in Malaysia, *Phalaenopsis* (Orchidaceae) in Philippines and Java, and *Asplenium nidus* (bird's nest fern) in Saipan (reviewed in Godan, 1983). In the tropics, this species is a known pest of vegetables including: cabbage, beans, peas, tobacco, and young plants of *Ipomoea* and *Hevea* (reviewed in Godan, 1983).

Other negative consequences of *L. fulica* establishment could include damage to native plants, alterations to nutrient cycling in the environment, adverse effects to native mollusks (through competition or control measures applied), and effects on human health (Raut and Barker, 2002). When populations are high, the calcium carbonate in the shells can neutralize acid soils, altering the soil properties and affecting the types of plants that can grow in the soil (reviewed in Cowie, 2006).

Known Food Sources*

This species is polyphagous. The following food source list is from USDA-APHIS (2007). This host list is not all inclusive, as reported food sources in the literature tend to focus on economically important plant species.

Primary Food Sources	Common Name
Albizia lebbeck	Woman's tongue

Albizia spp.	Albizia
Alsophila spp.	Alsophila
Alstonia scholaris	Devil tree of India
Amaranthus blitum	Purple amaranth
Amaranthus gangeticus	Molten fire
Amaranthus tricolor	Chinese amaranth
Amaranthus viridis	Green amaranth
Amaranthus spp.	Amaranth
Annona muricata	Soursop
Arachis hypogaea	Peanut
Artocarpus altilis	Breadfruit
Artocarpus heterophyllus	Jackfruit
Artocarpus spp.	Breadfruit
Asplenium nidus	Bird's nest fern
Averrhoa bilimbi	Blimb
Basella alba	Ceylon spinach
Basella rubra	Malabar spinach
Blechum brownei	Browne's blechum
Boerhavia diffusa	Boerhavia
Bougainvillea speciabilis	Bougainvillea
Bougainvillea spp.	Bougainvillea
Brassica oleracea	Kohlrabi
Brassica oleracea var. acephala	Cabbage
Brassica oleracea var. botrytis	Cauliflower
Brassica oleracea var capitata	Cabbage
Brassica oleracea var. italica	Sprouting broccoli
Brassica spp.	Cabbage
Bryophyllum pinnatum	Air plant
Cajanus cajan	Pigeonpea
Calophyllum inophyllum	Indian laurel
Canavalia gladiata	Sword jackbean
Canna edulis	Arrowroot
Canna indica	Canna
Canna spp.	Canna
Carica papaya	Papaya
Cassia (=Chamaecrista) mimosoides	Chamaecrista
Cassia occidentalis	Septic weed
Centrosema pubescens	Flor de conchitas
Cestrum nocturnum	Night Queen
Cichorium endivia	Endive
Cichorium intybus	Chicory
Chrysanthemum spp.	Chrysanthemum
Cinnamomum tamala	Indian bay leaf
Citrullus vulgaris (=lanatus)	Watermelon
Citrus limon	Lemon
Citrus reticulata	Tangerine
Citrus sinensis	Sweet Orange
Citrus spp.	Citrus

Clitoria ternatea	Butterfly pea
Colubrina asiatica	Asian nakedwood
Corchorus capsularis	Jute
Corchorus spp.	Corchorus
Cosmos spp.	Cosmos
Crinum spp.	Crinum lily
Crotalaria anagyroides	Crotalaria
Crotalaria pallida var. obovata	Smooth rattlebox
Cucumis melo	Cantaloupe
Cucumis sativus	Cucumber
Cucumis spp.	Melon
Cucurbita maxima	Winter squash
Cucurbita pepo	Field pumpkin
Cucurbita spp.	Gourds
Cyathea lunulata	Tree fern
Daucus carota	Carrot
Dioscorea alata	Greater Yam
Dioscorea bulbifera	Air Yam
Dioscorea spp.	Yam
Dolichos spp.	Beans
Epipremnum aureum	Golden pothos
Erythrina lithosperma (=subumbrans)	Erythrina
Erythrina spp.	Erythrina
Falcataria moluccana	Peacocks plume
Ficus hispida	Fig
Fragaria x ananassa	Strawberry
Gazania rigens	Treasure-flower
Glycine max	Soybean
Glycine spp.	Beans
Gossypium herbaceum	Cotton
Gossypium spp.	Cotton
Grewia mariannensis	
Gynandropis speciosa	The queen's plume
Hevea brasiliensis	Rubber
Hibiscus esculentus	Okra
Hibiscus mutabilis	Dixie rosemallow Land-lily
Hibiscus rosa-sinensis	China rose
Hibiscus spp.	Hibiscus
Hemigraphis colorata	Broad leaf flame ivy
Impatiens balsamina	Balsam
Indigofera suffruticosa	Anil de pasto
Ipomoea alba	Tropical white morning-glory
Ipomoea pes-caprae	Beach morning glory
Pachystachys coccinea	Cardinal's guard
Lablab purpureus	Lablab Bean
Lactuca indica	Lettuce
Lactuca sativa	Lettuce
Lactuca spp.	Lettuce

Lagenaria leucantha	Haired gourd
Lageneria siceraria	Bottle gourd
Lagenaria vulgaris	Gourd
Lagenaria spp.	Gourds
Laportea crenulata	Tree nettle
Leucaena leucocephala	White leadtree
Catharanthus) *rosea*	Madagascar periwinkle
Luffa aegyptiaca	Dishcloth gourd
Luffa acutangula	Angled luffa
Luffa cylindrica	Smooth luffa
Luffa spp.	Vegetable sponge
Manihot esculenta	Cassava
Melanolepis multiglandulosa	Alom
Mimosa invisa (=diplotricha)	Giant false sensitive plant
Morinda citrifolia	Indian mulberry
Momordica charantia	Balsam apple
Momordica cochinchinensis	
Momordica spp.	Momordica
Musa acuminata x balbisiana	French plantain
Musa x paradisiaca	Plantain
Musa sapientum	Banana
Musa spp.	Bananas
Pachyrhizus erosus	Yam bean
Parkia spp.	Parkia
Passiflora foetida	Fetid passionflower
Passiflora spp.	Passion flower
Pauinia cupana	Guarana
Vigna radiata	Bean
Phaseolus vulgaris	Kidney bean
Physalis peruviana	Peruvian groundcherry
Piper nigrum	Pepper
Pipturus albidus	Waimea pipturus
Pipturus argenteus	Native mulberry
Portulaca grandiflora	Purslane
Portulaca oleracea	Little hogweed
Portulaca spp.	Nine-O'Clock
Prunus persica	Peach
Pueraria montana var. lobata	Kudzu
Raphanus sativus	Radish
Ricinus communis	Castor
Sanchezia nobilis	Sanchezia
Scaevola sirecea	Naupaka
Sesamum indicum	Sesame
Spilanthes acmella	Paracress
Spinacea oleracea	Garden spinach
Tabernaemontana divaricata	Pinwheel flower
Tagetes erecta	African marigold
Tagetes patula	Indian marigold

Tagetes spp.	Marigolds
Tectaria spp.	Halberd fern
Tectona grandis	Teakwood
Tephrosia candida	White hoarypea
Tephrosia vogelii	Vogel's tephrosia
Theobroma cacao	Cacao
Thespesia populnea	Portia tree
Trichosanthes dioica	Pointed gourd
Triticum aestivum	Wheat
Vigna radiata	Mung bean
Vigna sinensis unguiculata	Blackeyed pea
Vitis vinifera	Grape
Vigna marina	Notched cowpea
Xanthosoma maffafa	Golden delicious

Secondary Food Sources	**Common Name**
Abelmoschus esculentus	Okra
Allangana lamarcana	Ballabhi-anga
Allium cepa	Onion
Alocasia indica	Arum
Alocasia macrorrhizos	Giant taro
Aloe indica	Aloe
Amorphophallus campanulatus (=paeoniifolius)	Elephant-Foot Yam
Antigonum lepiotus	Antigonon
Alocasia spp.	Elephant ear
Arctium lappa	Greater burrdock
Areca catechu	Betel nut palm
Averrhoa carambola	Carambola
Bauhinia accuminata	White dwarf orchard tree
Beta vulgaris	Common beet
Bikkia mariannensis	
Boehmeria nivea	Chinese grass
Brassica campestris	Field mustard
Broussonetia papyrifera	Paper mulberry
Callicarpa cana	
Camellia sinensis	Tea
Capparis cordifolia	Maiapilo
Capsicum annuum	Cayenne pepper
Capsicum baccatum	Locoto
Capsicum spp.	Chili peppers
Senna sophera	Kasuandi
Catharanthus roseus	Periwinkle
Cerbera manghas	Madagascar ordeal bean
Cereus hildmannianus	Hedge cactus
Cereus spp.	Cactus

Cleome gyandra	Spiderwisp
Clerodendron inerme	Glory bower
Coccinia cordifolia	Coccinea
Cocos nucifera	Coconut
Coffea arabica	Arabian coffee
Coffea canephora	Robusta coffee
Coffea spp.	Coffee
Colocasia antiguorum	Arum
Colocasia esculenta	Dasheen or taro
Dalbergia sissoo	Indian rosewood
Dhalia spp.	Dhalias
Dieffenbachia seguine	Dumbcane
Dracaena spp.	Dracaena
Edgaria darjeelingensis	Squash
Elaeis guineensis	Oil palm
Epipremnum pinnatum	Pothos
Eranthemum spp.	Eranthemum
Eucalyptus deglupta	Indonesian gum
Eucalyptus spp.	Australian gum
Eugenia spp.	Star apple
Euphorbia pulcherrima	Poinsettia
Euphorbia trigona	Sandmat
Ficus tinctoria	Fig
Galinsoga parviflora	Gallant-soldier
Gardenia augusta	Gardenia
Gliricidia sepium	Madre de Cacao
Commelina benghalensis	Tropical day flower
Gomphrena globosa	Globe amaranth
Helianthus annuus	Sunflower
Hernandea ovigera	
Ipomoea batatas	Sweet potato
Jasminum sambac	Jasmine
Kakanchoe pinnatum	Kalanchoe
Lycopersicon esculentum	Tomato
Mentha repens	Hortela
Monstera deliciosa	Split leaf philodendron
Montanoa hibiscifolia	Tree daisy
Moringa oleifera	Horseradish tree
Morus alba	White mulberry
Muntingia calabura	Strawberry tree
Nerium spp.	Oleander
Nicotiana spp.	Tobacco
Ochrosia mariannensis	Lipstick tree
Ochrosia oppositifolia	Bwa sousouri
Operculina turpethum	St. Thomas lidpod
Opuntia spp.	Cholla cactus
Pandanus spp.	Screwpine
Pemphis acidula	Small-leafed mangrove

Phalaenopsis spp.	Moth orchids
Pisum sativum	Garden pea
Pisum spp.	Pea
Pluchea indica	Pluchea
Pisidium guaja	Guava
Psychotria mariana	
Tradescentia spathacea	Moses in the cradle
Rosa spp.	Roses
Ruta graveolens	Common rue
Saccharum officinarum	Sugarcane
Salvia spp.	Sage
Sanseveria trifasciata	Snake Plant
Sechium edule	Chayote
Semibarbula orientalis	Moss
Sinapis arvensis	Charlock mustard
Solanum melongena	Eggplant
Solanum tuberosum	Potato
Swietenia mahogoni	Mahogany
Symphytum officinale	Common comfrey
Synedrella nodiflora	Synadrelia
Thea sinensis	Tea
Tradascantia spathacea	Moses in the boat or oysterplant
Trema orientale	Oriental trema
Trichosanthes anguina	Snake gourd
Tridax argentea	Tridex
Vanda spp.	Vanda orchid
Vanilla spp.	Vanilla
Vernonia scandens	Vernonia
Xanthosoma brasiliense	Cakakib
Zea mays	Corn
Zinnia linearis	Zinnia

Many mollusks find food through smell. *Lissachatina fulica* has been found to orient towards the smell of sliced cucumber or carrot (reviewed in Speiser, 2001).

*Terrestrial mollusks do not show host specificity and can feed on multiple crops as well as other materials, like decaying organic matter.

Pathogen or Associated Organisms Vectored

Human and Animal Pathogens

Lissachatina fulica vectors the nematode *Angiostrongylus cantonensis* (rat lungworm) which can cause eosinophilic meningoencephalitis (meningitis), as well as *A. costaricensis*, which causes abdominal angiostrongylosis (Thiengo et al., 2010). Of the two nematodes, *A. cantonensis* is the more concerning, especially with recent unexplained cases, and will be described in detail here.

Angiostrongylus cantonensis

Angiostrongylus cantonensis is a metastrongyloid nematode found widely in the Asia-Pacific region of which terrestrial and aquatic mollusks are the intermediate hosts in its parasitic lifecycle (Barratt et al., 2016). In Thailand, *L. fulica* is considered the most important intermediate host of *A. cantonensis*, and it is the country with the highest prevalence of human illness from the worm, with 47% of the cases reported worldwide (Barratt et al., 2016; Godan, 1983). However, *L. fulica* may not be the best host or biggest culprit there, after all. Vitta et al. (2016) found that only 3 of 275 *L. fulica* snails (1.1%) they tested were infected with the roundworm there, with a range of 5-15 larvae per snail, whereas *Cryptozona siamensis* snails had a 10.6% infection rate (45 out of 425 snails), with a range of 1-4858 larvae per snail. Hopefully this is an indication that *L. fulica* is not the preferred or ideal host for the worm, and that without the presence of *C. siamensis* in the U.S. the worm will not be able to reach the levels here that it has obtained in Thailand.

In the United States, *L. fulica* are infected with the nematode larvae on the main island of Hawaiʻi and on Oʻahu, but so are other terrestrial slugs, semi-slugs and other mollusk species, especially *Parmarion martensi*, which are carrying heavy loads of the parasite, maintaining its endemicity in Hawaiʻi (Barratt et al, 2016). Likewise, at least 10 other species of intermediate hosts have been found infected throughout Florida and in Louisiana as well, with five of those species newly recorded hosts in 2017, and with prevalence rates ranging from 1.2%-100% of those tested (Barratt et al., 2016; Stockdale Walden et al., 2017). For example, in Louisiana, *A. cantonensis* was found parasitizing 9% of the island apple snails, *Pomacea maculata*, an exotic species from South America, which is serving as a reservoir of the nematode in that region, and is used as a food source by people in New Orleans (Teem et al., 2013).

In any case, the spread and increasing prevalence of the nematode within these other intermediate hosts has impact on the worm loads in *L. fulica* as well. In 2010, on the main island of Hawaiʻi, *A. cantonensis* was detected in 5 out of 6 (83.3%) of the *L. fulica* snails tested, but that number grew slightly to 7 out of 8 (87.5%) on Hawaiʻi in 2013, and 5 out of 9 (55.6%) tested positive on Oʻahu. It was also found in only 4 out of 140 (2.9%) snails in Miami, Florida in 2013, but that number grew to 18 out of 50 (36%) in 2015 (Barratt et al., 2016; Iwanowicz et al., 2015; Teem et al., 2003). It is apparent that *L. fulica* is not the only host for the worm, nor the best host perhaps, but the trend of increasing levels of the nematode infecting intermediate hosts, including *L. fulica*, is unmistakable and alarming.

Rats (*Rattus rattus* and *R. norvegicus)* are the favored definitive/end hosts in the lifecycle of *A. cantonensis*, but at least 17 species of rodent may be a host and able to pass the larvae in their feces, which is then eaten by the mollusk (Barratt et al., 2016). Screening of *R. rattus* for *A. cantonensis* infection during the last 70 years in the Pacific has revealed infection rates ranging from 3% (Taiwan) to 20%–30% (Australia, Fiji, Japan) to 100% (Thailand) (Wang et al., 2008). On Hawaiʻi, several definitive hosts have been found to be infected with *A. cantonensis* larvae, including *R. rattus* (26 rats out of 26 tested were positive)*, R. norvegicus* (1 out of 1 tested) and *R. exulans* (10 out of 10) (Barratt et al., 2016). On the mainland in 2015 two *R.*

norvegicus rats (2 out of 36 tested) were infected in New Orleans, Louisiana, a *Sigmodon hispidus* (1 out of 30) (Hispid cotton rat) was positive for the nematode in McCurtain County in Oklahoma, and in 2017, 39 out of 171 (22.8%) *R. rattus* were positive from four counties in Florida (Barratt et al., 2016; Stockdale Walden et al., 2017).

Unfortunately, humans are also infected as an accidental dead-end host upon ingesting infected mollusks, paratenic hosts (monitor lizards, frogs, shrimp, fish and crabs), and fresh produce contaminated with mollusk slime containing nematode larvae (Barratt et al., 2016). *Angiostrongylus cantonensis* can infect drinking water that *L. fulica* have come in contact with as well (Godan, 1983), but that seems to be a minor route of spread. For examples, McBride et al. (2017) found that 29% of the 55 patients in southern Thailand who got meningitis from the nematode from 2008 to 2014 did report having eaten mollusks, but a woman in France who ate only vegetables and fish from her local supermarket contracted the worm, possibly from slime on the vegetables (Nguyen et al., 2017).

These infections have resulted in morbidity, permanent neurological damage, and sometimes mortality in humans (Barratt et al., 2016). It was identified as a human pathogen in 1945 and has since become the leading cause of eosinophilic meningitis, with at least 2,877 human cases documented worldwide, with many more probably unreported (Barratt et al, 2016). In addition to eosinophilic meningitis, angiostrongyliasis infections can result in coma, convulsion, epilepsy, amentia, hypomnesia and death, however, patients more often present with headache, neck stiffness, paraesthesia, muscle weakness, Brudzinski's sign, fever, nausea and vomiting, among many other less common symptoms (Barratt et al., 2016).

In addition to humans, at least 10 other species in Australia, both domesticated and wild, have also been shown to become accidental hosts, including dogs and horses (Barratt et al., 2016). In the U.S., many species of captive animals have been infected including apes and monkeys, a lemur, a horse, and a falcon, but wildlife are also contracting the disease in the southeast and becoming reservoirs, including nine-banded armadillos, opossums, and a wood rat, such that the nematode is now considered endemic in southern states (Barratt et al., 2016; Dalton et al., 2017).

The spread of angiostrongyliasis has been correlated with the spread of *Lissachatina fulica,* or at least with the spread of the nematode into new regions, sometimes leading to delayed diagnosis and treatment due to a lack of familiarity with the disease by healthcare providers in those regions (Barratt et al, 2016; USDA-APHIS, 2007). For example, a 37-year-old woman in Israel acquired *A. cantonensis*, most likely from vegetables, and since the disease is largely unknown in Europe, a diagnosis and treatment with steroids was not started for a few days after admission to the hospital, during which time she rapidly deteriorated (Fellner et al., 2016). In the U.S., Mr. McCumber contracted the worm in Hawaii, and was originally misdiagnosed with the flu, delaying treatment (Scutti, April 11, 2017). Unfortunately, he sustained permanent neurological damage from being in a coma for three months (Scutti, April

11, 2017). A geriatric patient was misdiagnosed with an embolic stroke, and sadly, remained in a stupor after delayed treatment (Tseng, et al., 2016). Other cases are springing up, including an infant in Tennessee, an 11-year-old in New Orleans, and three toddlers in Houston, Texas, but no clear tie to *L. fulica* in particular has been established in those cases (Flerlage et al., 2017; Foster et al., 2016; Hammond et al., 2017).

Recent cases in Australia and the U.S. have spurred concern in those countries (Barratt et al., 2016), and in general, growing concern has led to increased effort, as 35% of all articles written and published about the snail are from the 2010s, even though there were 5 decades prior to that since the first article was written in 1966 (Pavanelli et al., 2017). In the U.S. there has been alarm over cases in the Hawaiian Islands. There were only 24 suspected cases reported between 2001 and 2005, but a new record of 11 cases in 2016 occurred, and 17 confirmed cases in 2017 (as of September), including the first case on Oʻahu since 2010 (Gutierrez, September 28, 2017; KITV4 August 30, 2017).

A joint task force was established in May of 2016 in Hawaii with members from nine institutions, and "(d)ue to growing concerns, lawmakers set aside $1 million over two years. Most of the money will be spent on public education. The rest of the funds will be used for controlling rats, slugs, and snails, as well as a statewide study of the pests" to be conducted via a collaboration of several agencies (Governor's Office News Release, August 2, 2017; Gutierrez, September 28, 2107). These efforts and expenditure are justifiable and warranted, as Jarvi et al. (2017) found that the number of people in Hawaii exposed to it is higher than previously thought, at around 30% (56 out of 186) of those tested by ELISA dot-blot, based on the presence of antibodies in blood samples. Fortunately, 36 of the 56 people with antibodies to the worm self-reported that they were never infected with it, so they apparently have some immunity to it without ever knowingly becoming ill, which is ideal. Unfortunately, 120 people of the larger study of 435 people were diagnosed previously by a clinician as having contracted the worm (15) or self-reported a previous infection (105) (Jarvi et al., 2017).

<u>*Other diseases*</u>

Achatinids can also carry other diseases that affect humans and animals (Raut and Barker, 2002), including the bacterium *Aeromonas hydrophila* (Cowie, 2006). *Lissachatina fulica* has been experimentally infected with *Anafilaroides rostratus*, a parasite of the bronchii of cats (Malek, 1980). Valente et al. (2017) mention *Angiostrongylus vasorum* in *L. fulica*, which infects the pulmonary artery and right ventricle of wild and domestic canids, and they also found a natural infection of *L. fulica* with the feline lungworm *Aelurostrongylus abstruses* in Argentina.

Plant Pathogens

Lissachatina fulica has been implicated in transmitting *Phytophthora palmivora* (in black pepper, betel pepper, coconut, papaya), *Phytophthora colocasiae* (in taro), and *Phytophthora parasitica* (in eggplant and tangerine); however, the relative importance of *L. fulica* as a transmission agent has not been well established (Raut and Barker, 2002). Plant pathogens are spread through the feces (reviewed in Cowie, 2006).

Known Distribution

Africa: Annobón (Equatorial Guinea), Comoros, Ethiopia, Ghana, Ivory Coast, Kenya, Madagascar, Mayotte Island (French territory), Morocco, Mozambique, Nigeria, Somalia, Seychelles, South Africa, Tanzania (including Zanzibar), and Togo; **Asia:** Andaman Islands, Bangladesh, Bhutan, Bonin Islands, Brunei, Burma, Cambodia, China* (including Hong Kong)*, Taiwan*, India, Indonesia, Japan, Laos, Malaysia, Maldives, Nepal, Nicobar Islands, Philippines, Singapore, Sri Lanka, Thailand*, and Vietnam; **Caribbean:** Anguilla, Antigua (Antigua and Barbuda), Aruba, Barbados, Cuba (since 2014), Dominica, Dominican Republic, Guadeloupe, Lesser Antilles (2017), Marie-Galante, Martinique, Puerto Rico, Saint Lucia, Saint Maarten, and Trinidad (Trinidad and Tobago); **Europe:** Spain. **North America:** Florida, Hawaii; **Oceania:** American Samoa, Christmas Island (Australia), Cook Islands, Federated States of Micronesia, French Polynesia (including Society Islands), Guam, Kiribati, Marshall Islands, Mauritius, New Caledonia, Palau, Papua New Guinea (including Bismarck Archipelago), Northern Mariana Islands, Réunion, Samoa, Solomon Islands, Tokelau (New Zealand), Tonga, Vanuatu, Wake Island, and Wallis and Futana Islands; **South America:** Argentina, Bolivia, Brazil, Colombia, Ecuador, Peru, and Venezuela.

(Robinson, 2002; Prasad et al., 2004; Venette and Larson, 2004; Mitra et al., 2005; Thiengo et al., 2007; NAPPO, 2008; Wang et al., 2008; Vazquez Perera and Sanchez Noda, 2014; Epelboin, L. et al., 2016; Fellner et al., 2016; Foster et al., 2016; Spanish News Today, 2016; Waugh et al., 2016; Dard et al., 2017; Dominican Today, 2017; Flerlage et al., 2017; Hammond, et al., 2017; Nguyen et al., 2017; Vázquez et al., 2017; Robinson, 2018, personal communication).

*Locations at which approximately 75% total of cases of angiostrongyliasis occur.

Pathway

Lissachatina fulica has been introduced both purposefully and accidentally into many parts of the world. Intentional movement for medicinal purposes, beauty products, food (escargot), and research purposes has occurred (reviewed in Cowie, 2006). In addition, *L. fulica* has been found to be smuggled for use in religious practices or to be sold as pets (Than, 2013).

Prior to 1997, live [achatinid] snails were seized by PPQ in Arizona, California, Florida and Ohio. During a blitz conducted by Safeguarding, Intervention, and Trade Compliance (SITC) in 2004, PPQ Officers seized and destroyed 6,719 achatinids in six states and 64 cities (USDA-APHIS, 2007).

Giant African snail has previously been intercepted in baggage, cargo, soil (with or without plants), flowers, seeds, and *Zoysia* sp. (peat) (Godan, 1983), among other things. The vast majority of intercepted snails have been intended for consumption, but unfortunately there have been nine documented attempts since 1984 to bring the snail from Hawaii to the mainland (usually into California) for propagation, and five interceptions from countries in Asia and the U.K. for propagation purposes as well have occurred (AQAS, 2017).

Potential Distribution within the United States

Lissachatina fulica has been found and subsequently eradicated several times in the United States. On the mainland, this pest was found in the gardens of San Pedro, California in the late 1940s (Abbott, 1949), in Arizona in 1958 (Mead, 1959a) and in Florida in the late 1960s (USDA-APHIS, 2007). This infestation in 1966 demonstrated how easily the snail can get established, because only 3 juveniles brought from Hawaii to Florida resulted in seven years of eradication efforts and a budget of $700,000 (in 1969 USD) (Roda et al., 2016). In 2004, it was discovered that *L. fulica* was being imported by the pet trade and educational institutions; over 6,700 snails were confiscated from nine states and Puerto Rico to prevent establishment (USDA-APHIS, 2007).

Due to illegal introductions in 2011, *L. fulica* from the West Indies were detected in Miami-Dade County in Florida. APHIS, in partnership with the Florida Department of Agriculture and Consumer Services, has been conducting a regulatory program to eradicate the species, resulting in the elimination of 128,000 snails within the first two years, and that number rose steadily to 168,355 by March 2, 2018 (Fontanilla et al, 2014; FDACS, 2018a). Treatment efforts have focused on debris removal, hand collection, and pesticide application (Roda et al., 2016). Public education and enlistment for help has proven very successful as well. Over 95% of the cases identified so far have been a result of calls from the public to the snail hotline (FDACS, 2018b). A success story in the making, there has been a significant decline in the snail populations, and 15 of the 32 “core” areas in Florida were decommissioned in 2016 and 2017, since it was 36 months or more since the last live snail was found in those areas, and with an additional 8 more planned for decommissioning in 2018 (FDACS, 2018b, 2018a). Unfortunately, success comes at a price. Since 2011, over $14.1 million federal dollars and almost $3.3 million state funds have been needed to combat the snails this time (Feiber, 2018, pers. comm.). For more information on the efforts in Florida, see the APHIS program page: https://www.aphis.usda.gov/aphis/resources/pests-diseases/hungry-pests/the-threat/giant-african-snail/giant-african-snail.
Outside of the mainland, this species was found in Hawaii in 1936 and was subsequently eradicated through intensive control measures. However, *L. fulica* reestablished and is currently present in Hawaii. (Godan, 1983; Iwanowicz et al., 2015; Roda et al., 206; Stockdale Walden et al., 2017). As mentioned previously, a joint task force was established in May of 2016 in Hawaii with members from nine institutions, and “(d)ue to growing concerns, lawmakers set aside $1 million over two years. Most of the money will be spent on public education. The rest of the funds will be used for controlling rats, slugs, and snails, as well as a statewide study of the pests” to be conducted via a collaboration of several agencies (Governor’s Office

News Release, August 2, 2017; Gutierrez, September 28, 2107).

According to USDA-APHIS (2007) the range of this species is limited by temperature, moisture, and availability of calcium. *Lissachatina fulica* could potentially be found in environments that have a minimum temperature of 1°C (34°F), subtropical rainfall, and available calcium and soil pH of 7.0 – 8.0.

If *L. fulica* were to become established, it would most likely survive in the lower southeastern portion of the United States. Based on host availability and climate, states at increased risk include Alabama, Arkansas, Florida, Georgia, Louisiana, Mississippi, North Carolina, South Carolina, and Texas. Portions of California and Arizona also have increased risk. Smith and Fowler (2003) came up with a similar potential distribution for *L. fulica* in the continental United States. Populations were estimated to extend up to 38° latitude, including most of the southern states, up to Maryland in the east, through Texas to California on the west, and north to Washington and the Pacific Northwest.

Survey

CAPS-Approved Method*:

Visual. Specifics on survey information for L. fulica can be found in the New Pest Response Guidelines for Giant African Snails (USDA-APHIS, 2007). See the Introduction to the mollusk manual for specific information on visual surveys.

Survey Site Selection

New introductions of mollusks will likely be related to commerce and human- assisted movement. The habitat and land-use type of each survey site may be variable, ranging from agricultural land, to residential or industrial features.
When planning the survey route for a particular site, examine the following microhabitats:

a. Near heavily vegetated areas, especially gardens and fields where plants have been damaged by feeding;
b. Under rocks, asphalt or cement pieces that are in loose contact with the ground surface;
c. Under discarded wooden boards and planks, fallen trees, logs, and branches;
d. In damp leaf litter (not wet or soggy), compost piles, and rubbish heaps;
e. Under flower pots, planters, rubber mats, tires, and other items in contact with the soil;
f. Standing rock walls, cement pilings, broken concrete, and grave markers;
g. In gardens and fields where plants have been damaged by feeding snails and slugs; and
h. At the base of the plants, under leaves, or in the “heart” of compact plants, such as lettuce or cabbage.

This species can occur in agricultural areas, coastal and wetland areas, disturbed areas, natural and planted forests, riparian zones, scrublands and shrub-lands, and urban areas (reviewed in Cowie, 2006). It thrives in forest edge, modified forest, and plantation habitats (Raut and Barker, 2002).

Trap Placement
Trapping cannot be used alone but can be used to supplement visual surveying. Trapping for mollusks is not species-specific and will attract non-target species, including non-mollusks. Platform or baiting traps can be used to supplement visual inspection. Trap placement can occur in the same areas that visual surveys occur.

Time of year to survey
"Conduct detection surveys on an ongoing basis, with repeated visits at the beginning, during, and–or just after the rainy season. Keep in mind that *Lissachatina fulica* remains active at a range of 9–29°C (48–84°F). *Lissachatina fulica* begins hibernating at 2°C (35°F), and begins estivation at 30°C (86°F). Plan surveys for early morning and overcast days. Achatinids are active on warm nights, early mornings, and overcast and rainy days" (USDA- APHIS, 2007).

Note: While most cases of human infections result from consumption of raw or partially cooked snail meat, government inspectors, officers and field surveyors are at-risk due to the handling of live snail, samples, and potential exposure to mucus secretions. ***Wear gloves when handling mollusks and wash hands thoroughly after any mollusk survey or inspection activities.***

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Key Diagnostics/Identification

CAPS-Approved Method*:
Morphological. Identification should be verified by David Robinson at the USDA APHIS National Malacology Laboratory in Philadelphia, PA.

A key identification feature in *L. fulica* is the columella which ends abruptly; this feature is constant throughout the lifespan of the snail (USDA-APHIS, 2004).

A key to terrestrial mollusks (including *Lissachatina fulica*) is found here: http://idtools.org/id/mollusc/index.php

*For the most up-to-date methods for survey and identification, see Approved Methods on the CAPS Resource and Collaboration Site, at http://caps.ceris.purdue.edu/.

Easily Confused Species

This pest may be confused with *Euglandina rosea* or *Orthalicus* spp. both of which are present in the United States. This pest may also be confused with pests not currently present in the United States including *Achatina achatina*, *Archachatina marginata* and *Limicolaria aurora*, the latter two species being established on the French island of Martinique (Mead and Palcy, 1992).

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This datasheet was developed by USDA-APHIS-PPQ-CPHST staff. Cite this document as:

Molet, T. 2011. CPHST Pest Datasheet for *Lissachatina fulica*. Updated 2018 by L. Morales and D. Mackesy. USDA-APHIS-PPQ-CPHST.

Reviewed by: David Robinson, USDA-APHIS National Malacologist, March 2018.

Revisions

April 2014

1) Revised **Biology and Ecology** section
2) Revised **Pest Importance** section
3) Revised **Pathogen or Associated Organisms Vectored** section
4) Added **Pathway** section
5) Revised **Potential Distribution within the United States** section
6) Revised **Survey** section

August 2014

1) Revised **Known Distribution** section
2) Revised **Key Diagnostics/Identification** section

October 2016

1) NAPPFAST map removed
2) **Pathogen and Associated Organisms Vectored** section revised

April 2018

1) Comprehensive literature review and revision of entire datasheet