



Mojave Desert Network Selected Large Springs

Tassi Spring Benthic Macroinvertebrate and Springsnail Data 2012 to 2016

Natural Resource Data Series NPS/MOJN/NRDS—2017/1119



ON THE COVER

Looking northeast toward Tassi Spring and Tassi Ranch
Photograph courtesy of the National Park Service

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Natural Resource Data Series NPS/MOJN/NRDS—2017/1119

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September 2017

U.S. Department of the Interior
National Park Service
Natural Resource Stewardship and Science
Fort Collins, Colorado

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Please cite this publication as:

Bailard, J. L., and G. J. M. Moret. 2017. Mojave Desert Network selected large springs: Tassi Spring benthic macroinvertebrate and springsnail data 2012 to 2016. Natural Resource Data Series NPS/MOJN/NRDS—2017/1119. National Park Service, Fort Collins, Colorado.

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Abstract

As part of the Selected Large Springs protocol, the Mojave Desert Network Inventory and Monitoring Program (MOJN I&M) collects samples of benthic macroinvertebrates and measures water quality and discharge in several springs. The purpose of these actions is twofold:

- to provide data regarding the overall health of the aquatic ecosystems in the springs, and
- to monitor the endemic species present in the springs.

These data will be useful in determining the effects of any observed changes in hydrology or vegetation and will provide baseline information that can be used to evaluate management actions at the springs.

In 2012, 2014, and 2016, MOJN I&M visited Tassi Spring in Grand Canyon-Parashant National Monument to test and implement the methods proposed for benthic macroinvertebrate sampling and springsnail counting. This report presents the results of these monitoring efforts.

Acknowledgments

The authors would like to acknowledge the assistance of Rachael Fletcher, Jennifer Fox, and Mary Levandowski in the field. The Utah State University National Aquatic Monitoring Center (NAMC) provided taxonomic analysis of the benthic macroinvertebrate samples.

Overview

The National Park Service (NPS) Mojave Desert Network Inventory and Monitoring Program (MOJN I&M) has developed a protocol to monitor the hydrology and ecology of large springs in the network parks. The MOJN I&M Selected Large Springs (SLS) protocol covers springs in Death Valley National Park (DEVA), Joshua Tree National Park (JOTR), Lake Mead National Recreation Area (LAKE), Mojave National Preserve (MOJA), and Grand Canyon-Parashant National Monument (PARA). The springs have been selected for their high discharges, stability, and biological importance, and were chosen in cooperation with park staff to address the resource management priorities of each park.

As part of the SLS protocol, MOJN I&M collects samples of benthic macroinvertebrates (BMI) and measures water quality and discharge in several springs. The purpose of these actions is twofold:

- to provide data regarding the overall health of the aquatic ecosystems in the springs, and
- to monitor the endemic species present in the springs.

These data will be useful in determining the effects of any observed changes in hydrology or vegetation and will provide baseline information that can be used to evaluate management actions at the springs.

On December 4, 2012, February 10, 2014, and March 24, 2016, MOJN I&M visited Tassi Spring in PARA to test and implement the methods proposed for BMI sampling and springsnail counting. This report presents the results of these monitoring efforts.

Tassi Spring

Tassi Spring is located on the historic Tassi Ranch and is one of two springs in PARA that are monitored for the SLS protocol (Figure 1). The ranch was constructed in the 1930s and is one of the most important cultural resources in the monument due to its “association with the historical development of the cattle ranching industry in the remote, arid country of the Arizona Strip (Provencher and Warner 2003).”

At the time of sampling, Tassi Spring had three outlets: one that was piped to a watering trough, one that was captured by a springbox behind the ranch house, and one that flowed into an irrigation canal on the hill slope behind the ranch buildings (Figures 2 and 3). Since the time of sampling, the watering trough outlet has been destroyed by a flood. All three of the outlets were diverted during the era that the ranch was in use from the early 1930s until 1981, and these diversions still control the spring today. Discharge at Tassi Spring was measured to be 25 gpm in 1980 (Truini 2013).

The outlet that flows into the irrigation canal was chosen for monitoring because it provides habitat for the Grand Wash springsnail (*Pyrgulopsis bacchus*; Sada 2005), which is endemic to a small number of springs in the Grand Wash drainage. Tassi Spring is also a refugium for the regionally endemic relict leopard frog (*Rana onca*; Jaeger and Rivera 2017). Vegetation at the spring is actively managed to preserve the irrigation canal as a cultural resource.

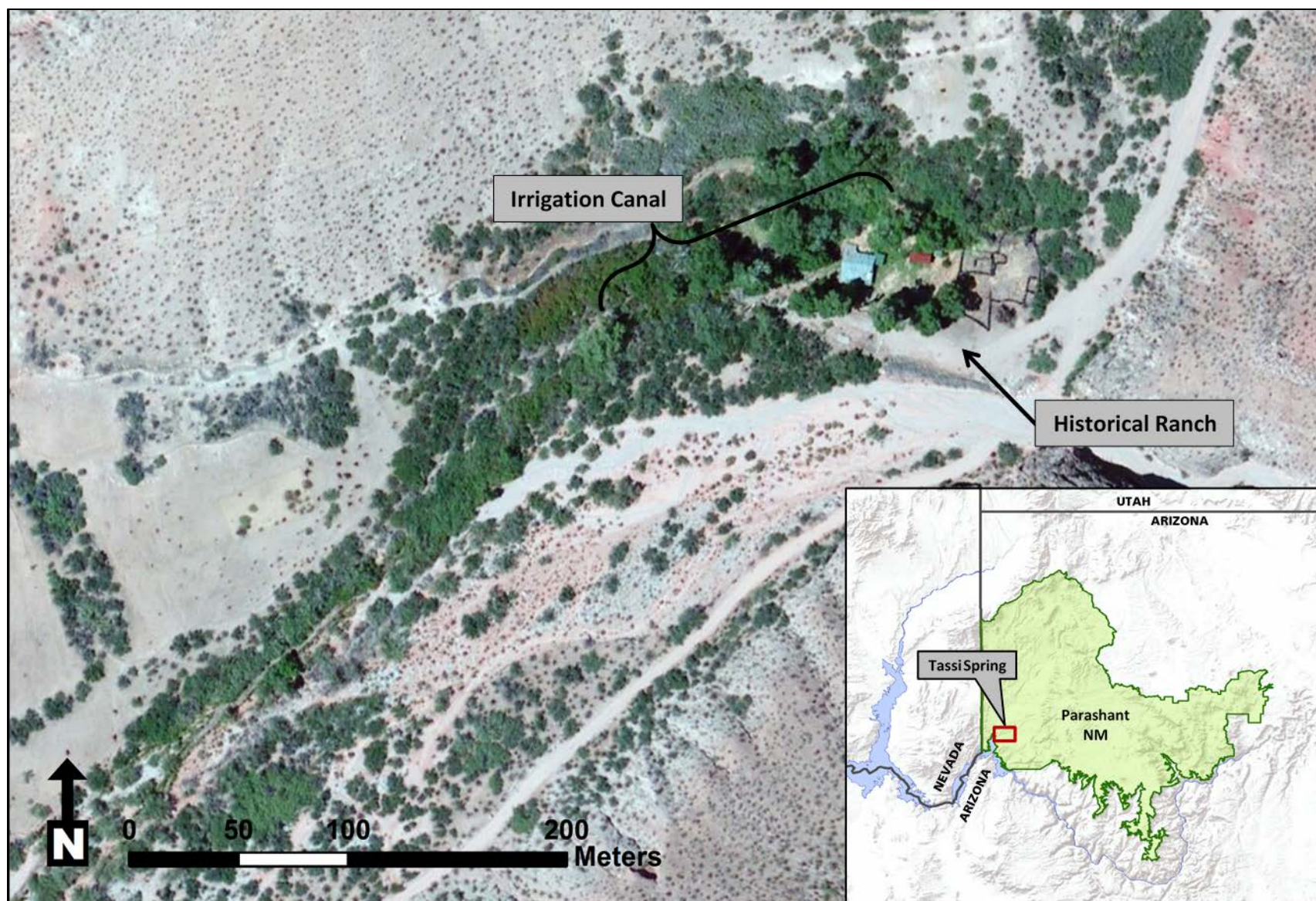


Figure 1. Aerial view of Tassi Spring. Inset: Location of Tassi Spring in PARA.



Figure 2. Springsnail habitat in irrigation canal at Tassi Spring.



Figure 3. Historical Tassi Ranch downhill of the irrigation canal.

Methods

BMI Samples

BMI sampling occurred on February 10, 2014, and March 24, 2016, in the irrigation canal outlet of Tassi Spring. The samples were collected from 0 m to 70 m downstream of the spring source at eight 10 m intervals. At each interval, material was collected from a 10 cm x 10 cm quadrat using a 15-cm kicknet. The substrate within the quadrat was disturbed with a paintbrush for 30 seconds, allowing the current to carry material into the kicknet. The submerged vegetation in the quadrat was then jabbed three times with the kicknet. The material collected at the eight intervals was combined into a single composite reachwide sample during each visit. For more detailed information on the BMI collection methods used at the spring, refer to SOP 8: Water Sampling, Benthic Macroinvertebrate Sampling, and Springsnail Monitoring (Moret et al. 2016).

BMI samples collected at Tassi Spring were preserved in ethanol and shipped to the Utah State University National Aquatic Monitoring Center (NAMC) for processing. There, the samples were sorted, identified to the lowest resolute taxon, and enumerated. Identifications were made by taxonomists certified by the Society for Freshwater Science using the taxonomic keys recommended by the Southwest Association of Freshwater Invertebrate Taxonomists (Richards and Rogers 2011). In order to provide a more complete taxa list, the samples were visually searched for 10 minutes for additional big and/or rare taxa. The NAMC's full procedures can be reviewed on their website (<http://www.usu.edu/buglab/SampleProcessing/LaboratoryProcedures/>, accessed 26 July 2017). The specimens from the samples are archived by the NAMC under the terms of a long-term loan agreement with the NPS.

Springsnail Field Counts

While springsnails were included in the BMI sampling discussed above, MOJN I&M also monitored the density and spatial extent of the springsnail population in the irrigation canal outlet of Tassi Spring. Springsnail monitoring occurred on February 10, 2014, based on methods tested during a visit on December 4, 2012. The methods were modified from those outlined by Sada (2009) and were designed to monitor the extent and density of the springsnail (Hydrobiidae) population.

The extent of the springsnail population was determined by observing the presence or absence of springsnails at accessible locations. At each location, a sample of substrate was brushed into a sieve. The portion of the substrate retained in the sieve was then spread across a white plastic tray and visually inspected for springsnails. Monitoring points were selected along the springbrook between the spring source and a point just beyond the furthest downstream presence of springsnails.

In 2012, the farthest downstream monitoring point was at 66 m. A measuring tape was used to select monitoring points 10 m to 20 m apart, for a total of five monitoring points including the source. In 2014, the farthest downstream monitoring point was at 70 m. A measuring tape was used to select monitoring points 10 m apart, for a total of eight monitoring points including the source. These monitoring points coincided with the locations of BMI sample collection.

At each monitoring point, a quadrat was placed on the bottom of the springbrook. In 2012, a 5 cm x 5 cm quadrat was used, and in 2014, a 10 cm x 10 cm quadrat was used. Surface material and loose substrate were brushed from the quadrat into a sieve. The material retained from the sieve was transferred to a sorting tray, and approximately 1 cm of water was added. The number of springsnails in the tray was then counted. All springsnails were returned to the spring.

In 2016, quantitative springsnail field counts ceased, and monitoring of their spatial extent was integrated into the BMI sample collection. Material collected from each 10 cm x 10 cm quadrat was scanned visually for springsnails, and the presence or absence of springsnails was recorded at each location along the transect where BMI sampling occurred.

Results

Water Quality

Water quality parameters were measured at the source of the irrigation canal outlet during visits from 2012 to 2016. No BMI or springsnails were sampled on October 21, 2016, but a field crew working at the spring were able to measure water quality parameters on that day. The values of these measurements are provided in Table 1.

Table 1. Water quality measured at the source of Tassi Spring during visits in 2012, 2014, and 2016.

Parameter	04 Dec 2012	10 Feb 2014	24 Mar 2016	21 Oct 2016
Temperature (°C)	24.7	24.3	24.42	25.01
pH	7.80	7.43	8.98 *	7.55
Specific Conductivity (µS/cm)	–	528	455	472
Dissolved Oxygen (mg/L)	–	6.82	7.75	6.13
Dissolved Oxygen (%)	–	–	92.5	74.3

Asterisk (*) indicates that the reading was fluctuating and did not stabilize. Measurement likely inaccurate.

Dash (–) indicates that the parameter was not measured.

BMI Samples

The results of the BMI sample processing and taxonomic analysis conducted by the NAMC are shown in Table 2 for 2014 and Table 3 for 2016. Selected population metrics reported by the NAMC are shown in Table 4 for both visits.

Table 2. BMI abundance per square meter in Tassi Spring on February 10, 2014.

Phylum	Class	Order	Family	SubFamily	Genus	Species	Abundance
Annelida	Clitellata	–	–	–	–	–	67
Arthropoda	Insecta	Coleoptera	Elmidae	–	<i>Heterelmis</i>	–	867
Arthropoda	Insecta	Coleoptera	Elmidae	–	<i>Microcylloepus</i>	–	3733
Arthropoda	Insecta	Diptera	Ceratopogonidae	–	–	–	67
Arthropoda	Insecta	Diptera	Chironomidae	Chironominae	<i>Paratendipes</i>	–	67
Arthropoda	Insecta	Diptera	Chironomidae	Chironominae	<i>Polypedilum</i>	–	533
Arthropoda	Insecta	Diptera	Chironomidae	Chironominae	<i>Rheotanytarsus</i>	–	2267
Arthropoda	Insecta	Diptera	Chironomidae	Chironominae	<i>Xestochironomus</i>	–	67
Arthropoda	Insecta	Diptera	Chironomidae	Orthoclaadiinae	<i>Paraphaenocladus</i>	–	67
Arthropoda	Insecta	Diptera	Chironomidae	Orthoclaadiinae	<i>Thienemanniella</i>	–	133
Arthropoda	Insecta	Diptera	Chironomidae	Tanypodinae	<i>Alotanypus</i>	–	933
Arthropoda	Insecta	Diptera	Chironomidae	Tanypodinae	<i>Pentaneura</i>	–	133
Arthropoda	Insecta	Diptera	Dixidae	–	<i>Dixella</i>	–	67
Arthropoda	Insecta	Diptera	Dolichopodidae	–	–	–	67
Arthropoda	Insecta	Diptera	Simuliidae	Simuliinae	<i>Simulium</i>	–	133
Arthropoda	Insecta	Diptera	Tabanidae	–	–	–	67
Arthropoda	Insecta	Ephemeroptera	Baetidae	–	<i>Fallceon</i>	–	333
Arthropoda	Insecta	Ephemeroptera	Leptohyphidae	–	<i>Tricorythodes</i>	–	733
Arthropoda	Insecta	Odonata	Coenagrionidae	–	<i>Argia</i>	–	1479
Arthropoda	Insecta	Trichoptera	Helicopsychidae	–	<i>Helicopsyche</i>	–	1267
Arthropoda	Insecta	Trichoptera	Odontoceridae	–	<i>Marilia</i>	<i>flexuosa</i>	1279
Arthropoda	Malacostraca	Amphipoda	Hyalellidae	–	<i>Hyalella</i>	–	10825
Mollusca	Gastropoda	Neotaenioglossa	Hydrobiidae	–	–	–	17933
Platyhelminthes	Turbellaria	–	–	–	–	–	200

Table 3. BMI abundance per square meter in Tassi Spring on March 3, 2016.

Phylum	Class	Order	Family	SubFamily	Genus	Species	Abundance
Arthropoda	Arachnida	Trombidiformes	–	–	–	–	100
Arthropoda	Insecta	Coleoptera	Elmidae	–	<i>Heterelmis</i>	–	400
Arthropoda	Insecta	Coleoptera	Elmidae	–	<i>Microcylloepus</i>	<i>pusillus</i>	1513
Arthropoda	Insecta	Diptera	Ceratopogonidae	Dasyheleinae	<i>Dasyhelea</i>	–	100
Arthropoda	Insecta	Diptera	Ceratopogonidae	Forcipomyiinae	–	–	200
Arthropoda	Insecta	Diptera	Chironomidae	Chironominae	<i>Micropsectra</i>	–	100
Arthropoda	Insecta	Diptera	Chironomidae	Chironominae	<i>Paratendipes</i>	–	500
Arthropoda	Insecta	Diptera	Chironomidae	Chironominae	<i>Polypedium</i>	–	900
Arthropoda	Insecta	Diptera	Chironomidae	Chironominae	<i>Rheotanytarsus</i>	–	800
Arthropoda	Insecta	Diptera	Chironomidae	Orthoclaadiinae	<i>Paraphaenocladus</i>	–	100
Arthropoda	Insecta	Diptera	Chironomidae	Tanypodinae	<i>Alotanypus</i>	–	2000
Arthropoda	Insecta	Diptera	Chironomidae	Tanypodinae	<i>Zavrelimyia</i>	–	100
Arthropoda	Insecta	Diptera	Culicidae	–	–	–	100
Arthropoda	Insecta	Diptera	Simuliidae	Simuliinae	<i>Simulium</i>	–	100
Arthropoda	Insecta	Ephemeroptera	Leptohyphidae	–	–	–	200
Arthropoda	Insecta	Odonata	Calopterygidae	–	<i>Hetaerina</i>	–	100
Arthropoda	Insecta	Odonata	Coenagrionidae	–	<i>Argia</i>	–	1100
Arthropoda	Insecta	Trichoptera	Helicopsychidae	–	<i>Helicopsyche</i>	–	500
Arthropoda	Insecta	Trichoptera	Odontoceridae	–	<i>Marilia</i>	<i>flexuosa</i>	300
Arthropoda	Malacostraca	Amphipoda	Hyalellidae	–	<i>Hyalella</i>	–	5275
Mollusca	Gastropoda	Neotaenioglossa	Hydrobiidae	–	–	–	57300

Table 4. Selected population metrics for 2014 and 2016 BMI samples from Tassi Spring.

Population Metric	10 Feb 2014	3 Mar 2016
# of Taxa*	15	12
Abundance	43317	71788
Dominant Family	Hydrobiidae	Hydrobiidae
% Dominant Family	41.4%	79.8%
# of EPT Taxa*	4	2
EPT Abundance	3612	1000
# of Shredder Taxa*	1	1
Shredder Abundance	1812	1200
# of Scraper Taxa*	1	1
Scraper Abundance	19200	57800
# of Collector-Filterer Taxa*	1	1
Collector-Filterer Abundance	2400	1000
# of Collector-Gatherer Taxa*	5	3
Collector-Gather Abundance	16292	6275
# of Predator Taxa*	4	3
Predator Abundance	2746	3600
# of Long-Lived Taxa*	4	4

* Values for richness-based metrics are standardized to operational taxonomic units and a fixed count of 300. These standardized metrics are indicated with an asterisk. Abundance-based metrics are presented as the estimated number of individuals per square meter. EPT stands for the insect orders Ephemeroptera (mayflies), Plecoptera (stoneflies), and Trichoptera (caddisflies).

In 2014, springsnails (Hydrobiidae) were the most dominant family. There were 17,933 springsnails per m², comprising 41.4% of the sample. Freshwater amphipods (*Hyalella sp.*) were also common, with 10,825 specimens per m². The most abundant insect family were the non-biting midges of Chironomidae, with 4,067 specimens per m². Riffle beetles (*Microcylloepus sp.*) were similarly abundant at 3,733 specimens per m². Other taxa with densities greater than 1,000 specimens per m² included pond damselflies (*Argia sp.*), and caddisflies (*Marilia flexuosa* and *Helicopsyche sp.*).

In 2016, springsnails (Hydrobiidae) were again the most dominant family. There were 57,300 springsnails per m², comprising 79.8% of the sample. *Hyalella sp.* was also common, with 5,275 specimens per m². The most abundant insect family were the non-biting midges of Chironomidae, with 4,500 specimens per m². Other common insects included the riffle beetle *Microcylloepus pusillus*, at 1,513 specimens per m², and the pond damselfly *Argia sp.*, at 1,100 specimens per m². Mayflies (order Ephemeroptera) and caddisflies (order Trichoptera) were also present in the sample. Additionally, several adult relict leopard frogs (*Rana onca*) were observed in the irrigation canal during the visit in 2016 (Figure 4).



Figure 4. Relict leopard frog (*Rana onca*) at Tassi Spring in 2009. Photo courtesy of J. Fox.

Springsnail Field Counts

In addition to collecting BMI samples, MOJN I&M also counted springsnails in the field. Tables 5 and 6 show the observed springsnail abundances from the methods test visit in 2012 and the pilot study visit in 2014. Table 7 shows the observed springsnail presence from the visit in 2016.

Table 5. Springsnail abundance observed in Tassi Spring on December 4, 2012.

Location on Transect 0 m = Source	Abundance in 5 x 5 cm (0.0025 m ²) Quadrat
0 m	17 alive, 0 dead
10 m	19 alive, 10 dead
20 m	11 alive, 2 dead
40 m	3 alive, 1 dead
66 m	2 alive, 3 dead

Table 6. Springsnail abundance observed in Tassi Spring on February 10, 2014.

Location on Transect 0 m = Source	Substrate	Comment	Abundance in 10 x 10 cm (0.01 m²) Quadrat
0 m	Fines, small gravel covered with dead cottonwood leaves	Full sun exposure	3
10 m	Gravel, fines	Shaded, 70% cover, 24.7 °C	3
20 m	–	Full sun exposure, 30% cover	7
30 m	Sand, fines	Partial shade from willow, 24.4 °C	23
40 m	–	Full sun exposure, juvenile cattails 18 in tall, 50 % cover, 24.8 °C	58
50 m	Small gravel, fines	Very little sun exposure, cattails, 24.7 °C	8
60 m	Small gravel, fines	Some sun exposure, 50% cover, 24.2 °C	8
70 m	–	Some sun exposure, lots of vegetation, 24.1 °C	0

Table 7. Springsnail presence observed in Tassi Spring on March 24, 2016.

Location on Transect 0 m = Source	Substrate	Comment	Springsnails Present?
0 m	Gravel, fines	Part shade, glide	Yes
10 m	Gravel	Part shade, glide	Yes
20 m	Gravel	Part shade, riffle	Yes
30 m	Gravel	Part shade, riffle	Yes
40 m	Gravel	Full shade, glide	Yes
50 m	Roots, gravel	Full shade, glide	Yes
60 m	Sand, fines	Full shade, glide	Yes
70 m	Sand, plants	Full shade, riffle	Yes

Springsnails were present at the spring source and throughout the irrigation canal during all three years of monitoring. In 2012, two living individuals were found at a distance of 66 m from the source, which was the farthest distance measured during that visit. In 2014, springsnails were found at a distance of 60 m from the source, but not at 70 m. In 2016, springsnails were found at both 60 m and 70 m from the source.

The highest population density during the 2012 visit was 7,600 specimens per m² at a distance of 10 m from the source. The average density was 4,160 specimens per m². The highest population density during the 2014 visit was 5,800 specimens per m² at a distance of 40 m from the source. The average density was 1,571 specimens per m² where springsnails were present.

These densities are considerably lower than the nearly 18,000 specimens per m² reported by the lab for the BMI sample collected in 2014. It is possible that juvenile springsnails were too small to discern with the naked eye, and that a large number of specimens went uncounted in the field.

Additionally, springsnails may have been buried within the substrate and overlooked in the tray. Hand counting of springsnails will be eliminated in future monitoring efforts as this method has proven to be inaccurate during field testing. Future monitoring will rely on laboratory counts to estimate springsnail population density.

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NPS 666/140234, September 2017

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