6-2020

The Prairie Naturalist, Vol. 52, Issue 1, June 2020

The Prairie Naturalist

Established in 1969

The Prairie Naturalist

Vol. 52, Issue 1, June 2020

TABLE OF CONTENTS

Editor's Note

Greetings GPNSS members from your new Editor. Dr. Christopher Jacques passed on his role of Editor-in-Chief for *The Prairie Naturalist* to me in April this year, after serving for 11 years. I become just the fourth editor of this journal over its 52-year history, and I look forward to taking forward for another five or more years.

Some background about me: I was a research wildlife biologist for the U.S. Geological Survey at Northern Prairie Wildlife Research Center in North Dakota for 29 years, retiring two and a half years ago; I remain engaged as an emeritus scientist. My research and publications focused largely on waterbirds and their wetland habitat across the northern prairies but also extended to the Intermountain West and the Upper Peninsula of Michigan, and with a brief foray into prairie butterflies. My editorial experience includes serving as associate editor for *Wetlands*, *Condor*, and lead editor of the 14th and 15th Proceedings of the North American Crane Working Group. I was lead editor of *Cranes and Agriculture: A Global Guide for Sharing the Landscape* (2018; published by the International Crane Foundation, with many international authors) and co-editor of *Whooping Cranes: Biology and Conservation* (2019; Academic Press). I also helped with extensive writing, coordination, and editing of the *Crane Conservation Strategy* (2019, International Crane Foundation, with many international authors). Through these experiences and years of writing and reviewing manuscripts, I have learned a great deal about writing, editing, and the editorial process. I find it truly rewarding to help authors craft well-written and informative manuscripts, successfully address reviewers' concerns, and present their valuable findings to others in the field.

I have a long history with *The Prairie Naturalist* and its parent society, as both author (dating back to 1990) and member. I was active for many years as board member and president of the North Dakota Natural Science Society and as it transitioned to the Great Plains Natural Science Society (GPNSS). Hence, I have a deep affinity for this regional journal and its value to the region. Now retired from USGS, I can use my time, skills, and knowledge to contribute more directly to this journal. Among my goals are to improve the response rates and turnaround times for manuscripts, encourage more authors to consider *The Prairie Naturalist* as an outlet for their papers, and renew efforts to get the journal online and recognized in search engines such as the Web of Science.

One of the critical first steps is to improve the journal's online access and visibility. I and the GPNSS board are working with the University of Nebraska's Digital Commons to host all our past issues in their system. Paul Royster, head of Scholarly Communications with the University of Nebraska-Lincoln, is enthusiastic about hosting our journal and has already dowloaded articles from volumes 43–48 from the GPNSS website. GPNSS President Bill Jensen has shipped volumes 1–42 to him so they can be professionally scanned and added. Keep an eye on the DigitalCommons@University of Nebraska-Lincoln (https://digitalcommons.unl.edu/tpn) for our journal issues! The site is a wealth of information for the region and hosts other publications, such as Proceedings of the North American Prairie Conferences, Great Plains Research: A Journal of Natural and Social Sciences, and Proceedings of the North American Crane Workshop.

This being my first issue, I am still learning the publishing processes, particularly the final stages of getting papers into print. I am very grateful to the Associate Editors who are continuing in their role for the journal. Their expertise and network in their respective fields are invaluable for this journal that spans so many taxa and subjects. Dr. Melissa Wuellner has stepped aside as Associate Editor for fisheries to pursue other editorial work; I thank her for her years of assistance to the journal. I welcome Dr. Keith Koupal, Nebraska Game and Parks Commission, as her replacement for fisheries manuscripts, and Dr. Clint Otto, a research ecologist with the U.S. Geological Survey at Northern Prairie Wildlife Research Center, as new Associate Editor for herps and insects. Both graciously accepted my request for their help.

The staff at Minuteman Press in Sioux Falls, South Dakota, have been very helpful in this transition, responsive to my questions and communications. I greatly appreciate their timely and professional work in converting Word documents into well-formatted pages for page proofs publication, final clean-up, printing, and mailing to members.

This issue continues to provide valuable information on a diversity of taxa. Of interest to grassland managers is the study of small mammal communities in managed and restored grasslands at the Grand Forks Air Force Base in northeastern North Dakota. The study of larval freshwater drum in a Nebraska reservoir highlights the importance of key environmental factors influencing larval drum densities. Also in fisheries is the study that examines limitations of sampling gear for larval fish in the Minnesota River. Finally, a new record of wood frogs is documented in South Dakota. No book reviews were ready for this issue but will continue in future issues.

The Prairie Naturalist has long served a valuable role in promoting interest in and understanding of natural history of the Great Plains, encouraging conservation of natural

resources, and providing communication among individuals, institutions, and organizations of like interests. We publish full length articles, notes, and book reviews, but we also publish thesis and dissertation abstracts—an alternative way to get more awareness of students' studies. I also welcome other types of submissions, such as articles from regional workshops, commentaries, and articles or obituaries about important people in the region's natural history.

I welcome any comments and suggestions you have for articles, the editorial processes, and ideas to help improve and sustain *The Prairie Naturalist*. I look forward to learning much more about the natural history of the Great Plains through new manuscripts and articles.

—Jane E. Austin, Ph.D.
  Editor-in-Chief

Factors Associated with Larval Freshwater Drum Annual Peak Density in a Nebraska Irrigation Reservoir

BRETT T. MILLER[1], BRIAN C. PETERSON, KEITH D. KOUPAL, ᴀɴᴅ CASEY W. SCHOENEBECK[2]

Department of Biology, University of Nebraska at Kearney, Bruner Hall of Science,
2401 11th Street, Kearney, NE, 68849, USA (BTM, BCP, CWS)
Nebraska Game and Parks Commission, 1617 First Avenue, Kearney, NE, 68847, USA (KDK)
[1]Present Address: Kansas Department of Wildlife, Parks and Tourism,
1782 10th Road, Clay Center, KS, 67432, USA (BTM)
[2]Present Address: Minnesota Department of Natural Resources,
23070 North Lakeshore Drive, Glenwood, MN, 56334, USA (CWS)

ABSTRACT Freshwater drum (*Aplodinotus grunniens*) represent one of the most widely distributed fish species in North America. Identifying biotic and abiotic factors that influence larval freshwater drum densities can improve understanding of early life history. Our objective was to investigate correlations between annual peak density of larval freshwater drum and combinations of five variables (chlorophyll *a*, growing degree days [GDD], reservoir discharge, turbidity, and cladoceran density) from a long-term (2003–2017) monitoring program in a Nebraska irrigation reservoir. Twenty-eight a priori candidate models were assessed to determine the relative support of explanatory variables associated with annual peak density of larval freshwater drum using Akaike's information criterion. During the course of the study, larval freshwater drum annual peak densities ranged from <0.1 (±0.1 SE) to 4.5 (±0.8 SE) /m³ and variations were best explained by chlorophyll *a* (highest relative importance, 0.77). Chlorophyll *a* was positively associated with higher densities of larval freshwater drum. This study highlights the importance of chlorophyll *a* to larval freshwater drum annual peak densities and provides a greater understanding of freshwater drum early life history.

KEY WORDS chlorophyll *a*, early life history, freshwater drum, irrigation reservoir

Freshwater drum (*Aplodinotus grunniens*) are a member of the Sciaenidae family and inhabit the largest latitudinal range of any North American fish species (Barney 1926, Boschung and Mayden 2004). This species spawns when water temperatures are between 18º C and 25º C, are highly fecund, and can live for more than 10 years (Swedburg and Walburg 1970, Bur 1984, Pereira et al. 1995). Eggs are semi-buoyant, and after hatching the larvae drift at the surface for approximately two weeks while they absorb their yolk sac and develop orientated movement (Priegel 1967). Ichthyoplankton samples commonly report freshwater drum are the most abundant species in ichthyoplankton samples, and Wallus and Simon (2006) have summarized several studies that reflect the inter- and intra-annual variability in relative abundance demonstrated by this species. Freshwater drum dominate abundance in most egg and ichthyoplankton surveys; they can also exhibit high rates of mortality (Cada and Hergenrader 1980, Wallus 2006) and entrainment loss (Walburg et al. 1971).

While the role of freshwater drum in most aquatic systems is not well defined, they can represent a large proportion of the fish community biomass (Rypel 2007). Larval freshwater drum rely heavily on zooplankton as their primary food source, as other potential diet items are restricted by gape limitations (Swedburg and Walburg 1970, Schael et al. 1991, Sullivan et al. 2012). Previous studies have found adult freshwater drum to consume zebra mussels (*Dreissena polymorph*) in the Great Lakes (French and Love 1995, French and Bur 1996, Morrison et al. 1997) and Arkansas (Magoulick and Lewis 2002); however, the presence of these aquatic invasive species may also alter conditions for larval freshwater drum survival.

A variety of environmental factors have been associated with relative abundance, growth, survival, recruitment, year-class strength, and diet of freshwater drum in riverine systems (Braaten and Guy 2002, Wallus 2006, Jacquemin et al. 2014, Jacquemin et al. 2015); however, the factors that drive larval freshwater drum annual peak density in irrigation reservoirs are less understood. Because larval freshwater drum are normally absent as prey items of predatory fish in Nebraska reservoirs (Olson et al. 2007, Miller et al. 2019, Uphoff et al. 2019), we hypothesize that year-class strength may be driven by lower trophic or abiotic factors. For these reasons we explored how cladoceran density, turbidity, chlorophyll *a*, discharge, and growing degree days explain the variable recruitment of larval freshwater drum in Harlan County Reservoir, Nebraska. Cladoceran density was selected because larval freshwater drum in Harlan County

Reservoir positively selected for this prey taxa (Sullivan et al. 2012) and therefore the availability could be associated with larval survival. Water turbidity has been found to alter distribution (Matthews 1984) and negatively impact feeding ability in other species (Johnston and Wildish 1982, Zamor and Grossman 2007). Chlorophyll *a* concentrations have been associated with reservoir primary productivity and have been linked to relative abundance of higher trophic levels in irrigation reservoirs and Harlan County Reservoir specifically (Olds et al. 2014). The weak swimming ability of larval freshwater drum make them susceptible to entrainment loss in Midwest rivers (Walburg 1971) and reservoirs (Smith and Brown 2002, Fryda 2005) and may also impact relative abundance in Harlan County Reservoir. Growing degree days were also included because available temperature can influence fish growth rates and potentially subsequent survival (Neuheimer and Taggart 2007, Chezik et al. 2013, Uphoff et al. 2013). Understanding which factors impact larval freshwater drum densities in irrigation reservoirs is not well documented. Therefore, the objective of this study was to evaluate which factors influence annual peak density of larval freshwater drum within a Nebraska irrigation reservoir.

STUDY AREA

Harlan County Reservoir is an irrigation reservoir built in 1952 and is located on the Republican River drainage in south-central Nebraska. Harlan County Reservoir encompasses more than 5,362 ha, has 121 km of shoreline, and has mean and maximum depths of 4 m and 18 m (Uphoff et al. 2013). Daily inflows averaged 2.8 m^3/sec (SE = 1.0) from 2003–2017 (USBR 2018). During the study timeframe drought years were recorded that resulted in a net loss of inflow and nearly 50% loss of the conservation pool (Olds et al. 2011, Olds et al. 2014). Long-term monitoring and research at Harlan County Reservoir since 2003 has provided insight on changes in water quality (Olds et al. 2011), zooplankton (Olds et al. 2014), game fish species such as walleye (*Sander vitreus*; Uphoff et al. 2013– Miller et al. 2018*a*), white bass (*Morone chrysops*; Olson et al. 2007, Miller et al. 2018*a*), and larval fish including gizzard shad (*Dorosoma cepedianum*; Sullivan et al. 2011, Miller et al. 2018*b*) and freshwater drum (Sullivan et al. 2012).

METHODS

Since 2003, larval freshwater drum have been collected at dusk using bow-mounted ichthyoplankton push nets of two different diameters (1.0-m diameter with 1.80-mm mesh and 0.5-m diameter with 0.75-mm mesh) deployed simultaneously as one unit of sampling effort. A sample consisted of pushing the pair of nets for 5 min in a single direction at a speed of 4 km/hr (Sullivan et al. 2012). Each net was outfitted with a flowmeter (General Oceanics Inc.,

Miami, FL, USA) to estimate the volume of water sampled. Push-net sampling began in early June (2003–2004) or the last week of May (2005–2017) and was conducted once a week for eight consecutive weeks at standardized-GPS reservoir sites (Sullivan et al. 2012). Additional sites were added as the study progressed and ranged from eight sites in 2003 and 2004, to 24–48 sites during the remaining years (2005–2017). Collected larval fish were preserved in 70% ethyl alcohol and transported to the University of Nebraska at Kearney for identification, measurement (total length [TL]; mm), and enumeration.

Larval freshwater drum density at each site was determined by summing the number of freshwater drum <8-mm TL from the smaller-diameter net and the freshwater drum ≥8-mm TL from larger-diameter net and dividing by the respective volumes sampled. Freshwater drum <8-mm TL were counted from the smaller-diameter net and those ≥8 mm TL from the larger-diameter net to avoid double counting similar sized fish (Sullivan et al. 2012). Site-specific larval freshwater drum densities were averaged to determine a weekly mean. Annual peak larval density was therefore determined to be the week with the greatest density. Annual peak densities were used to be consistent with methodology in similar studies (Sullivan et al. 2011, Sullivan et al. 2012, Miller et al. 2018*b*) because an additive approach could introduce gear bias caused by catchability that may vary with freshwater drum length.

Zooplankton samples were collected concurrently with larval push-net samples at 15 standardized sites distributed across the reservoir using a Wisconsin plankton net (0.5-m diameter with 80-µm mesh) towed vertically from the bottom substrate to the surface (Peterson et al. 2005). Water depth (m) was recorded to calculate the water volume sampled. Samples were preserved in a sucrose-buffered 4% formalin solution to prevent osmotic distortion (Haney and Hall 1973) prior to being identified and quantified within the laboratory (Peterson et al. 2005). Cladoceran densities (number/L) were determined for each site and averaged for the sampling date across the reservoir (Sullivan et al. 2012).

Weekly water quality sampling was conducted to coincide with zooplankton and larval freshwater drum sampling at 15 standardized sites distributed across the reservoir, all of which were also sampling locations for larval freshwater drum. At each site, a Van Dorn bottle sampler collected water samples at 1 m and every subsequent 3 m at the sampling site (i.e., 1, 4, 7, and 10 m). All collected water samples from each site were pooled in a bucket and stirred to assumed homogeneity, at which time a subsample was processed. Turbidity (Formazin Attenuation Units, FAU) was measured using a Hach$^{(TM)}$ colorimeter and chlorophyll *a* (µg/L) was measured using a Turner Designs Aquafluor$^{(TM)}$ Handheld Fluorometer. Mean spring (April and May) values were used for the analysis of turbidity and chlorophyll *a* to coincide with initial larval freshwater drum development. Turbidity and chlorophyll *a* were restricted to 2004–2017 because data

were not collected in 2003. Discharge that coincided with annual peak density dates were obtained from 2003 through 2017 from the United States Bureau of Reclamation website (USBR 2015). Air temperature data were obtained for Republican City, Nebraska, for 2003–2017 from the National Oceanic and Atmospheric Administration's National Center for Environmental Information (NCEI 2018). Daily air temperature data were used to calculate growing degree-days (GDD) using the following:

$$GDD = \left[\frac{T_{max} + T_{min}}{2}\right] - T_{base}$$

where T_{max} is the maximum daily temperature, T_{min} is the minimum daily temperature, and T_{base} is the base temperature at which larval development and growth is thought to occur. In this case, T_{base} was set at 9° C, which is a species-specific value for freshwater drum (McInerny and Held 1995). Growing degree days were summed from 1 April through 31 May for each year between 2003 and 2017 in which the average air temperature was ≥9° C. Growing degree days were used instead of water temperatures because daily air temperatures were available, and air temperatures have been found to be strongly correlated to water temperatures (Shuter et al. 1983, Livingstone and Padisak 2007).

A set of 28 a priori candidate models were established to assess the relative support of explanatory variables using Akaike's information criterion (AIC; Akaike 1987). Due to small sample size relative to model parameters, second order Akaike's information criterion (AIC_c) was used to more conservatively rank competing models (Burnham and Anderson 2002). Models with the lowest difference between AIC_c values (Δ_i) and highest model weight (W_i) were chosen for model inference. Model averaging was used across all candidate models with associated parameter estimate and standard error by calculating,

$$\tilde{\beta} = \sum_{i=1}^{R} w_i \hat{\beta}_i$$

$$\widehat{var}\left(\tilde{\beta}\right) = \sum w_i \left[\widehat{var}\left(\tilde{\beta}\right) + \left(\beta_i - \tilde{\beta}\right)^2\right]$$

where $\tilde{\beta}$ is the parameter estimate, w_i is the perspective model weight, and β_i is the regression estimate for i (Burnham and Anderson 2002). Using the K-L method, AIC_c weights are summed for all models containing a predictor variable and models with zero weights are omitted to determine relative importance (Burnham and Anderson 2002). Variables with the largest total weight are considered to have the greatest relative importance for explaining the dependent variable (Burnham and Anderson 2004). Simple linear regression was performed between the predictor variable with the most

support and larval freshwater drum annual peak densities (α = 0.05; Fig. 1).

RESULTS

Five variables were assessed to determine which factors were associated with annual peak density of larval freshwater drum. Between 2003 and 2017, annual peak densities of larval freshwater drum averaged 1.3 larvae/m³ (SE = 0.3, n = 15) and ranged from <0.1 to 4.5 larvae/m³, most often peaking around mid- to late June. Cladoceran density (during peak weeks) ranged from 2.7 to 30.5 organisms/L with a mean of 15.6 organisms/L (SE = 4.0) from 2003 through 2017. Turbidity (during peak weeks) ranged from 10.7 to 39.7 FAU with a mean of 23.1 FAU (SE = 6.0) from 2004 to 2017. Spring (April–May) chlorophyll a averaged 56.1 µg/L (SE = 8.3) between 2004 and 2017 and ranged from 6.7 to 70.4 µg/L. Discharge (during peak weeks) averaged 6.6 m³/sec (SE = 1.7) between 2003 and 2017 and ranged from 0.0 to 18.5 m³/sec. Growing degree days from April to May averaged 253 days (SE = 17.4) between 2003 and 2017 and ranged from 162 to 387 days.

The best supported model (W_i = 0.41) included chlorophyll a (Table 1; Fig. 1) and explained 36% of the annual variability in annual peak density of larval freshwater drum. Additionally, chlorophyll a was present in four of the top five models offering support for this variable. Other models evaluated were not supported by the data (i.e., high Δ_i and low W_i; Table 1). Relative variable importance weights suggested that chlorophyll a had the greatest relative importance on larval freshwater drum annual peak density (W_i = 0.77; Table 2) and chlorophyll a was significantly related to larval freshwater drum annual peak density (P = 0.02).

DISCUSSION

We found that chlorophyll a was the most supported variable of those we examined, associated with annual peak density of larval freshwater drum within Harlan County Reservoir. Chlorophyll a has also been linked to increased density of crappie (*Pomoxis* spp.) (McInerny and Cross 1999, Bunnell et al. 2006), largemouth bass (*Micropterus salmoides*), threadfin shad (*Dorosoma petenense*), and gizzard shad (Siler et al. 1986, Allen et al. 1999). While commonly used to index trophic state of lakes and reservoirs (Carlson 1977), chlorophyll a is primarily responsible for energy absorption during photosynthesis (Brönmark and Hansson 2005). During primary production, chlorophyll a has been found to be linked to zooplankton production (Pace 1986), which ultimately supports and enhances fish production (Oglesby et al. 1987, Downing et al. 1990). The availability of chlorophyll a may also be linked to other potential variables we included as predictors because it can be related to phytoplankton blooms (Boyer et al. 2009), which decrease
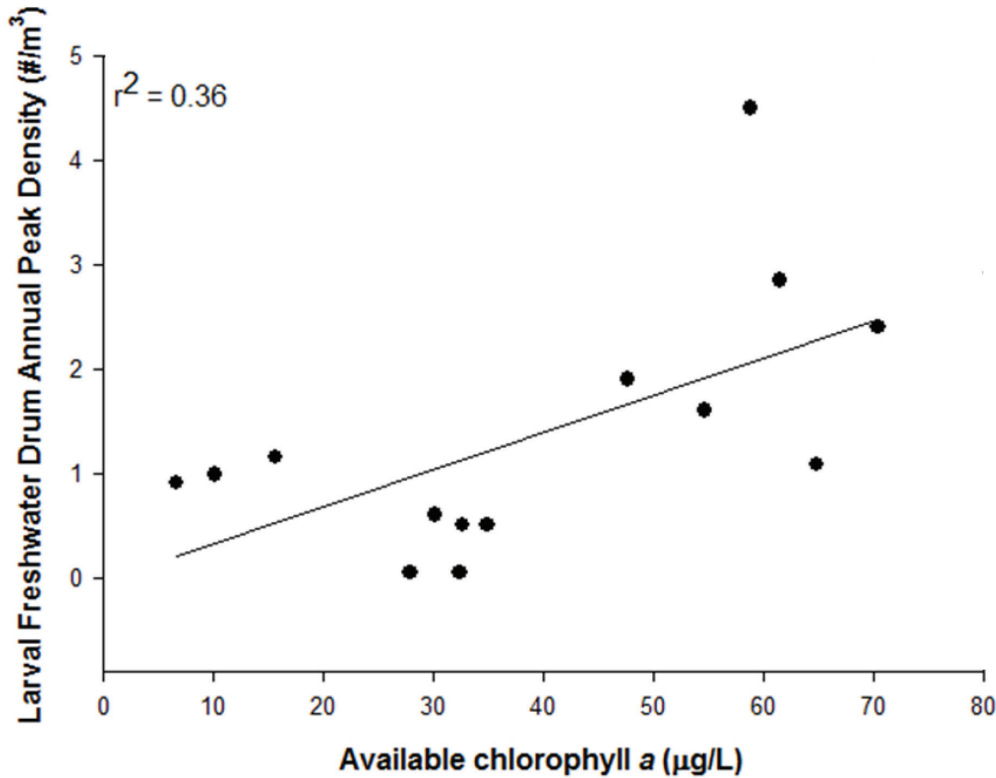
Figure 1. Larval freshwater drum annual peak densities (n/m^3) from Harlan County Reservoir, Nebraska, from 2004–2017 to compared chlorophyll a (µg/L) during the spring (April and May). Solid line indicates line of best fit from simple linear regression. The regression was significant ($P = 0.02$).

water clarity and manifest in higher productivity with warmer water temperatures (Elliot et al. 2006). Considering that discharge, turbidity, growing degree days, and cladoceran density all were included with chlorophyll a as weighted descriptors, it is likely that conditions conducive to nutrient rich waters offer a suite of survival advantages for hatching success and immediate larval survival.

Freshwater drum are not typically managed by biologists but understanding their ecological role in irrigation reservoirs is valuable. Historically, larval freshwater drum have been collected alongside larval gizzard shad in this reservoir (Sullivan et al. 2011, Sullivan et al. 2012, Miller et al. 2018*b*). Nutrient rich reservoirs can create size-selective feeding of particular taxa (Stenson 1976). A similar study found larval gizzard shad abundance was correlated to zooplankton density and reservoir elevation rather than chlorophyll a (Miller et al. 2018*b*). Larval densities of both species peak at similar times; however, they may have developed niche separation as larval gizzard shad primarily consumed copepod nauplii and cyclopoida taxa (Sullivan et al. 2011) and larval freshwater drum ate *Bosmina spp.* in this reservoir (Sullivan et al. 2012).

There is a need to investigate more factors associated with higher nutrient conditions to specifically identify the mechanisms driving year-class development of freshwater drum in irrigation reservoirs. As with all applications of AIC modelling, it needs to be recognized that this approach identifies which of the selected variables best describes the variability in relative abundance of larval freshwater drum. The use of AIC is common in the environmental field because it assists managers in identifying the relative importance of specific predictors for biological trends (Guthery 2008); however, it is limited by the biological interpretation of which variables can and should be included. Future studies could investigate spatial distributions of larval freshwater drum to determine if densities differ within reservoirs, especially considering that the availability of chlorophyll a has spatial patterns in this reservoir (Olds et al. 2011). Also, understanding what factors drive yearly densities for other species can allow for a wholistic management approach in assessing fish assemblages.

ACKNOWLEDGMENTS

Table 1. Coefficient of determination (r^2), number of parameters (K), AIC_c values, difference between successive AIC_c values (Δ_i), and model weights (W_i) describing support of 28 models to evaluate abiotic and biotic factors influencing annual peak density of larval freshwater drum in Harlan County Reservoir, Nebraska, during 2003–2017. Factors included chlorophyll *a* (CL), growing degree days (GDD), discharge (DI), turbidity (TB), and cladoceran density (CD).

Model	r^2	K	AIC_c	Δ_i	W_i
CL	0.36	3	5.43	0.00	0.41
CL + DI	0.41	4	8.10	2.66	0.11
DI	0.25	3	8.83	3.40	0.07
CL + TB	0.37	4	9.00	3.56	0.07
CL + GDD	0.44	4	9.10	3.66	0.07
CL + CD	0.36	4	9.25	3.81	0.06
TB	0.16	3	9.51	4.08	0.05
TB + DI	0.32	4	10.33	4.90	0.04
GDD + CL +DI	0.50	5	12.59	7.16	0.01
CD + DI	0.25	4	12.60	7.17	0.01
GDD + DI	0.39	4	12.65	7.21	0.01
CL + DI +TB	0.41	5	12.75	7.31	0.01
CL + DI + CD	0.41	5	12.75	7.32	0.01
CD	0.02	3	12.85	7.41	0.01
GDD	0.12	3	12.89	7.45	0.01
GDD + TB	0.33	4	13.02	7.58	0.01
CD + TB	0.18	4	13.10	7.67	0.01
GDD + CL + TB	0.47	5	13.57	8.13	0.01
GDD + CL + TB	0.37	5	13.66	8.23	0.01
CL + TB + CD	0.45	5	13.76	8.32	0.01
TB + DI +CD	0.32	5	14.98	9.55	0.00
GDD + TB + DI	0.45	5	14.99	9.56	0.00
GDD + CD	0.12	4	16.55	11.11	0.00
GDD + DI + CD	0.40	5	17.26	11.83	0.00
GDD + TB + CD	0.33	5	17.58	12.15	0.00
CL + TB + DI +CD	0.41	6	18.56	13.13	0.00
GDD + TB + DI + CD	0.46	6	20.81	15.38	0.00
GDD + TB + CL + DI + CD	0.53	7	25.88	20.44	0.00

Table 2. Final model averaged estimates, standard error, and relative variable importance for chlorophyll *a* (CL), growing degree days (GDD), discharge (DI), turbidity (TB), and cladoceran density (CD). AICc weights are summed for all models containing a predictor variable and models with zero weights are omitted to determine relative importance. The relative importance is the summation of the model weights for each variable.

i	Parameter estimate	SE	Relative importance
Chlorphyll *a* (CL)	0.03	0.05	0.77
Growing degree days (GDD)	0.00	0.01	0.14
Discharge (DI)	-0.02	0.01	0.28
Turbidity (TB)	0.01	0.02	0.21
Cladoceran density (CD)	0.00	0.01	0.11

LITERATURE CITED

Akaike, H. 1987. Factor analysis and AIC. Psychometrika 52:317–332.

Allen, M. S., J. C. Greene, F. J. Snow, M. J. Maceina, and D. R. DeVries. 1999. Recruitment of largemouth bass in Alabama reservoirs: relations to trophic state and larval shad occurrence. North American Journal of Fisheries Management 19:67–77.

Barney, R. L. 1926. The distribution of the fresh-water sheepshead, *Aplodinotus grunniens*, in respect to the glacial history of North America. Ecology 7:351–364.

Boschung, H. T., and R. L. Mayden. 2004. Fishes of Alabama. Smithsonian Books, Washington D.C., USA.

Boyer, J. N., C. R. Kelble, P. B. Ortner, and D. T. Rudnick. 2009. Phytoplankton bloom status: chlorophyll *a* biomass as an indicator of water quality condition in the southern estuaries of Florida, USA. Ecological Indicators 9:S56–S67.

Braaten, P. J., and C. S. Guy. 2002. Life history attributes of fishes along the latitudinal gradient of the Missouri River. Transactions of the American Fisheries Society 131:931–945.

Brönmark, C., and L. A. Hansson. 2005. The biology of lakes and ponds. Second edition. Oxford University Press, Oxford, UK.

Bunnell, D. B., R. S. Hale, M. J. Vanni, and R. A. Stein. 2006. Predicting crappie recruitment in Ohio reservoirs with spawning stock size, larval density, and chlorophyll concentrations. North American Journal of Fisheries Management 26:1–12.

Bur, M. T. 1984. Growth, reproduction, mortality, distribution, and biomass of fresh-water drum in Lake Erie. Journal of Great Lakes Research 10:48–58.

Burnham, K. P., and D. R. Anderson. 2002. Model selection and multimodel inference: a practical information theoretic approach. Second edition. Springer Science, New York, USA.

Burnham, K. P., and D. R. Anderson. 2004. Multimodel inference: understanding AIC and BIC in model selection. Sociological Methods and Research 33:261–304.

Cada, G. F., and G. L. Hergenrader. 1980. Natural mortality rates of freshwater drum larvae in the Missouri River. Transactions of the American Fisheries Society 109:479–483.

Carlson, R. E. 1977. A trophic state index for lakes. Limnology and Oceanography 22:361–369.

Chezik, K. A., N. P. Lester, and P. A. Venturelli. 2013. Fish growth and degree-days I: selecting a base temperature for a within-population study. Canadian Journal of Fisheries and Aquatic Sciences 71:47–55.

Downing, J. A., C. Plante, and S. Lalonde. 1990. Fish production correlated with primary productivity, not the morphoedaphic index. Canadian Journal of Fisheries and Aquatic Sciences 47:1929–1936.

Elliot, J. A., I. D. Jones, and S. J. Thackeray. 2006. Testing

the sensitivity of phytoplankton communities to changes in water temperature and nutrient load, in a temperate lake. Hydrobiologia 559:401–411.

French, J. R. P. III, and J. G. Love. 1995. Size limitation on zebra mussels consumed by freshwater drum may preclude the effectiveness of drum as a biological controller. Journal of Freshwater Ecology 10:379–383.

French, J. R. P. III, and M. T. Bur. 1996. The effect of zebra mussel consumption on growth of freshwater drum in Lake Erie. Journal of Freshwater Ecology 11:283–289.

Fryda, N. J. 2005. Adult abundance and larval production of crappie (*Pomoxis* spp.) and fish entrainment from a Nebraska irrigation reservoir. Thesis. The University of Nebraska at Kearney, Kearney, USA.

Guthery, F. S. 2008. A primer on natural resource science. Texas A&M University Press, College Station, USA.

Haney, J. F., and D. J. Hall. 1973. Sugar-coated *Daphnia*: A preservation technique for Cladocera. Limnology and Oceanography 18:331–333.

Jacquemin, S. J., M. Pyron, M. Allen, and L. Etchison. 2014. Wabash River freshwater drum *Aplodinotus grunniens* diet: effects of body size, sex, and river gradient. Journal of Fish and Wildlife Management 5:133–140.

Jacquemin, S. J., J. C. Doll, M. Pyron, M. Allen, and D. A. S. Owen. 2015. Effects of flow regime on growth rate in freshwater drum, *Aplodinotus grunniens*. Environmental Biology of Fishes 98:993–1003.

Johnston, D. D., and D. J. Wildish. 1982. Effect of suspended sediment on feeding by larval herring (*Clupea harengus harengus* L.). Bulletin of Environmental Contamination and Toxicology 29:261–267.

Livingstone, D. M., and J. Padisak. 2007. Large-scale coherence in the response of lake surface-water temperatures to synoptic-scale climate forcing during summer. Limnology and Oceanography 52:896–902.

Magoulick, D. D., and L. C. Lewis. 2002. Predation on exotic zebra mussels by native fishes: effects on predator and prey. Freshwater Biology 47:1908–1918.

Matthews, W. J. 1984. Influence of turbid inflows on vertical distribution of larval shad and freshwater drum. Transactions of the American Fisheries Society 113:192–198.

McInerny, M. C., and J. W. Held. 1995. First-year growth of seven co-occurring fish species of Navigation Pool 9 of the Mississippi River. Journal of Freshwater Ecology 10:33–41.

McInerny, M. C., and T. K. Cross. 1999. Effects of lake productivity, climate warming, and intraspecific density on growth and growth patterns of black crappie in southern Minnesota lakes. Journal of Freshwater Ecology 14:255–264.

Miller, B. T., C. W. Schoenebeck, and K. D. Koupal. 2018a. Gear- and season-specific catch rates of age-0 walleyes

and white bass: standard sampling recommendations for Great Plains reservoirs. North American Journal of Fisheries Management 38:903–910.

Miller, B. T., B. C. Peterson, K. D. Koupal, and C. W. Schoenebeck. 2018b. Long-term changes in biotic and abiotic factors influence larval gizzard shad (*Dorosoma cepedianum*) annual peak density. Journal of Freshwater Ecology 33:173–181.

Miller, B. T., C. W. Schoenebeck, and K. D. Koupal. 2019. Summer food habits and prey taxa and size electivity of age-0 white bass in a South-Central Nebraska irrigation reservoir. Journal of Freshwater Ecology 34:293–303.

Morrison, T. W., W. E. Lynch Jr., and K. Dabrowski. 1997. Predation on zebra mussels by freshwater drum and yellow perch in western Lake Erie. Journal of Great Lakes Research 23:177–189.

National Center for Environmental Information [NCEI]. 2018. Climate data online. Daily summaries station details. https://www.ncdc.noaa.gov/cdo-web/datatools/findstation. Accessed 17 July 2018.

Neuheimer, A. B., and C. T. Taggart. 2007. The growing degree-day and fish size-at-age: the overlooked metric. Canadian Journal of Fisheries and Aquatic Sciences 64:1571–1581.

Oglesby, R. T., J. H. Leach, and J. Forney. 1987. Potential *Stizostedion* yield as a function of chlorophyll concentration with special reference to Lake Erie. Canadian Journal of Fisheries and Aquatic Sciences 44 (S2):166–170.

Olds, B. P., B. C. Peterson, K. D. Koupal, K. M. Farnsworth-Hoback, C. W. Schoenebeck, and W. W. Hoback. 2011. Water quality parameters of a Nebraska reservoir differ between drought and normal conditions. Lake and Reservoir Management 27:229–234.

Olds, B. P., B. C. Peterson, K. D. Koupal, C. W. Schoenebeck, K. M. Farnsworth-Hoback, and W. W. Hoback. 2014. Zooplankton density increases in an irrigation reservoir during drought conditions. Transactions of the Nebraska Academy of Sciences 34:27–32.

Olson, N. W., C. S. Guy, and K. D. Koupal. 2007. Interactions among three top-level predators in a polymictic Great Plains reservoir. North American Journal of Fisheries Management 27:268–278.

Pace, M. L. 1986. An empirical analysis of zooplankton community size structure across lake trophic gradients. Limnology and Oceanography 31:45–55.

Pereira, D. L., C. Bingham, G. R. Spangler, D. J. Conner, and P. K. Cunningham. 1995. Construction of a 110-year biochronology from sagittae of freshwater drum (*Aplodinotus grunniens*). Recent developments in fish otolith research. University of South Carolina Press, Columbia, South Carolina, USA.

Peterson, B. C., N. J. Fryda, K. D. Koupal, and W. W. Hoback. 2005. *Daphnia lumholtzi*, an exotic zooplankton invading a Nebraska reservoir. The Prairie Naturalist 37:11–19.

Priegel, G. R. 1967. The freshwater drum, its life history,

ecology, and management. Wisconsin Department of Natural Resources Publication 236, Madison, Wisconsin, USA.

Rypel, A. L. 2007. Sexual dimorphism in growth of freshwater drum. Southeastern Naturalist 6:333–342.

Schael, D. M., L. G. Rudstam, and J. R. Post. 1991. Gape limitation and prey selection in larval yellow perch *Perca flavescens*, freshwater drum *Aplodinotus grunniens*, and black crappie *Pomoxis nigromaculatus*. Canadian Journal of Fisheries and Aquatic Sciences 48:1919–1925.

Shuter, B. J., D. A. Schlesinger, and A. P. Zimmerman. 1983. Empirical predictors of annual surface water temperature cycles in North American lakes. Canadian Journal of Fisheries and Aquatic Sciences 40:1838–1845.

Siler, J. R., W. J. Foris, and M. C. McInerny. 1986. Spatial heterogeneity in fish parameters within a reservoir. Pages 122–136 *in* G. E. Hall and M. J. Van Den Ayvle, editors. Reservoir Fisheries Management: Strategies for the 80's. American Fisheries Society, Bethesda, Maryland, USA.

Smith, K. A., and M. L. Brown. 2002. Seasonal composition and abundance of icthyoplankton entrained through Big Bend Dam, South Dakota. Journal of Freshwater Ecology 17:199–207.

Stenson, J. A. E. 1976. Significance of predator influence on composition of *Bosmina* spp. populations. Limnology and Oceanography 21:814–822.

Sullivan, C. L., C. W. Schoenebeck, K. D. Koupal, W. W. Hoback, and B. C. Peterson. 2011. Patterns of age-0 gizzard shad abundance and food habits in a Nebraska irrigation reservoir. The Prairie Naturalist 43:110–116.

Sullivan, C. L., K. D. Koupal, W. W. Hoback, B. C. Peterson, and C. W. Schoenebeck. 2012. Food habits and abundance of larval freshwater drum in a south central Nebraska irrigation reservoir. Journal of Freshwater Ecology 27:111–121.

Swedburg, D. V., and C. H. Walburg. 1970. Spawning and early life history of the freshwater drum in Lewis and Clark Lake, Missouri River. Transactions of the American Fisheries Society 99:560–570.

Uphoff, C. S., C. W. Schoenebeck, W. W. Hoback, K. D. Koupal, and K. L. Pope. 2013. Degree-day accumulation influences annual variability in growth of age-0 walleye. Fisheries Research 147:394–398.

Uphoff, C. S., C. W. Schoenebeck, K. D. Koupal, K. L. Pope, and W. W. Hoback. 2019. Age-0 walleye *Sander vitreus* display length-dependent diet shift to piscivory. Journal of Freshwater Ecology 34:27–36.

U.S. Bureau of Reclamation [USBR]. 2018. Current reservoir data for Harlan County Dam, NE. https://www.usbr.gov/gp-bin/arcweb_hcne.pl. Accessed 17 July 2018.

Walburg, C. H., G. L. Kaiser, and P. L. Hudson. 1971. Lewis and Clark Lake tailwater biota and some relations of the tailwater and reservoir fish populations. Pages 449–467 *in* G. E. Hall, editor. Reservoir Fisheries and Limnology. Special Publication Number 8, American Fisheries Society, Washington, D.C., USA.

Wallus, R. 2006. Family Sciaenidae: drums and croakers. Pages 277–302 *in* R. Wallus and T. P. Simon, editors. Reproductive biology and early life history of fishes in the Ohio River drainage Aphredoderidae through Cottidae, Moronidae, and Sciaenidae, volume 5. CRC Press, Boca Raton, Florida, USA.

Wallus, R., and T. P. Simon. 2006. Reproductive biology and early life history of fishes in the Ohio River drainage Aphredoderidae through Cottidae, Moronidae, and Sciaenidae, volume 5. CRC Press, Boca Raton, Florida, USA.

Zamor, R. M., and G. D. Grossman. 2007. Turbidity affects foraging success of drift-feeding rosyside dace. Transactions of the American Fisheries Society 136:167–176.

Small mammal communities in grasslands at the Grand Forks Air Force Base, North Dakota, USA

LYNDA R. LAFOND, ELIZABETH H. RAVE, AND KATHRYN A. YURKONIS

University of North Dakota, Starcher Hall Room 101,
10 Cornell Street Stop 9019, Grand Forks, ND 58202-9019 (LRL, KAY)
Bemidji State University, 1500 Birchmont Drive NE #27, Bemidji, MN 56601-2699, USA (EHR)

ABSTRACT Small mammals are important in grasslands but are often overlooked in management and reconstruction efforts. We sampled small mammals in three sites on the Grand Forks Air Force Base (GFAFB) located in central Grand Forks County, North Dakota, USA. The study sites varied in their management history and represented the three major types of grasslands (reconstructed prairie, old field, and hay field) within Grand Forks County. We captured 463 individuals of six species with Sherman live traps in summer (June, July, August) 2014 and 2015. We captured the most individuals and species (295 individuals of 5 species), including all shrews (*Sorex arcticus* and *Sorex spp.*) and an ermine (*Mustela erminea*; a new record for GFAFB), in an upland reconstructed prairie, and we captured the least in a lowland hay field (5 individuals of 1 species). Meadow voles (*Microtus pennsylvanicus*) were captured most frequently (96% of individuals) and were affected marginally by changes in vegetation height density over time. Our findings reinforce the notion that not all grasslands equally serve small mammals and that managers need to focus on landscape-scale heterogeneity to support diverse small mammal communities in grasslands.

KEY WORDS Grand Forks Air Force Base, grassland management, hay field, fire management, small mammal abundance, North Dakota, restored prairie, tallgrass prairie.

Small mammals play an important role in grasslands. Herbivorous small mammals provide seed dispersal and nutrient cycling services, insectivores regulate insect populations, and both support grassland carnivores (Sieg 1987, Churchfield et al. 1991, Willson and Traveset 2000). In the absence of fire and grazing, small mammals play a key role in grassland nutrient cycling (Howe and Lane 2004, Howe et al. 2006). These contributions to grassland function vary as small mammal populations fluctuate within and across years (Diffendorfer et al. 1999). One major factor that affects small mammal presence and diversity in grasslands is the temporal variation in vegetation structure and composition that results from grassland management (Getz 1985, Kaufman and Kaufman 1990, Burel et al. 2004, Matlack et al. 2008). As grasslands recover from haying, grazing, and fire, the somewhat predictable change in vegetation structure affects small mammal communities (Grant et al. 1982, Kaufman et al. 1990, Kaufman and Kaufman 2008). Likewise, as grasslands are left idle and are invaded by non-native or woody plant species, small mammal communities transition toward species more tolerant of greater cover and litter depth (Matlack et al. 2008). This gradation in grassland types and management regimes exists in many landscapes formerly dominated by tallgrass prairie and is particularly notable among the grasslands of Grand Forks County in northeastern North Dakota.

Historically, the northern most reaches of the tallgrass prairie region extended into the Red River Valley of eastern North Dakota and western Minnesota (Omernik and Griffith 2014). Bordered by aspen parkland on the east and drift plains on the west, this ecoregion is a transition zone known to host plant species of eastern and western origins (Ralston 1968). Small mammal communities of the region were likewise comprised of species whose distributions extended farther north and south (Iverson et al. 1967, Grant and Birney 1979). In eastern North Dakota, grassland small mammal communities include meadow vole (*Microtus pennsylvanicus*), deer mouse (*Peromyscus maniculatus*), meadow jumping mouse (*Zapus hudsonius*), short-tailed shrew (*Blarina brevicauda*), masked shrew (*Sorex cinereus*) or Hayden's shrew (*Sorex haydeni*), Arctic shrew (*Sorex arcticus*), western jumping mouse (*Zapus princeps*), and house mouse (*Mus musculus*) (Whitaker 1972, Kirkland and Schmidt 1996, Seabloom 2011). Grasslands close to landfills or housing many also contain Norway rat (*Rattus norvegicus*). Other species that may be captured but are considered rare to uncommon (< 1/ha) are the prairie vole (*Microtus ochrogaster*), pygmy shrew (*Sorex hoyi*), and northern grasshopper mouse (*Onychomys leucogaster*).

It is well established that grassland management affects small mammals. Populations of meadow voles, prairie voles, and short-tailed shrews often decline following fire and haying as a result of reduced plant litter and cover (Kaufman et al. 1989, Clark and Kaufman 1990, Kaufman et al. 1990, Neuhaus 2015). In contrast, *Peromyscus* spp. populations often increase following fire and haying (Sietman et al. 1994, Kaufman and Kaufman 2008, Neuhaus 2015). In the absence of disturbance, grasslands become more homogeneous

(Fuhlendorf et al. 2006, Gibson 2009) and are often invaded by woody plant species (Kulmatiski and Beard 2013). Woody growth often leads to a reduction in small mammal species that are more commonly affiliated with grasses (Zimmerman 1992, Ratajczak et al. 2012). Given these species-specific responses, it is not surprising that the highest landscape diversity for plants and small mammals occurs when multiple disturbances occur at different time scales (Zimmerman 1992, Fuhlendorf et al. 2006, Fuhlendorf et al. 2010), making heterogeneity important to consider as we manage grassland landscapes affected by grassland conversion and land use changes.

The meadow vole is of particular interest when considering comparative effects of grassland management on small mammals. Meadow voles are herbivores and serve as ecosystem engineers by selectively consuming vegetation (legumes and cool-season grasses) and affecting plant community composition (Howe et al. 2002, Howe et al. 2006). When their populations are high, their collective consumption of plant material has been thought to rival effects of prescribed fire (French et al. 1975). Meadow voles also serve as prey for many grassland predators such as barn owls (*Tyto alba*), in some cases accounting for 55–84 % of prey consumed (Colvin and McLean 1986). Given their potentially large populations, meadow voles can also limit other small mammal species through direct interactions and indirect effects on cover and food availability (Wolff 1989, Brady and Slade 2001).

Of all the common grassland small mammals, meadow voles are particularly responsive to changes in grassland structure. Their populations decrease in locations with reduced litter depth, and they are often less abundant in hay fields or grasslands the first growing season following fire (LoBue and Darnell 1959, Klatt and Getz 1987, Kaufman et al. 1990). Typically, meadow voles are more numerous in grasslands with greater cover (Birney et al. 1976, Getz 1985, Matlack et al. 2008). Because of their habitat responsiveness and relatively high abundance, meadow voles are particularly useful for assessing small mammal response to grassland management. Our objective was to compare the small mammal and meadow vole populations among three common types of grasslands in northeastern North Dakota and test the hypothesis that meadow voles are more numerous at sites with greater vegetation density.

STUDY AREA

Grand Forks Air Force Base (hereafter GFAFB) is a 2,336-ha United States Air Force military installation located approximately 24 km west of Grand Forks, North Dakota (Fig. 1). The GFAFB is located in the Red River Valley of eastern North Dakota within the boundaries of the historic glacial Lake Agassiz (Wali et al. 1973). The soils at GFAFB are composed mainly of Antler-Gilby-Svea, Bearden-Antler,
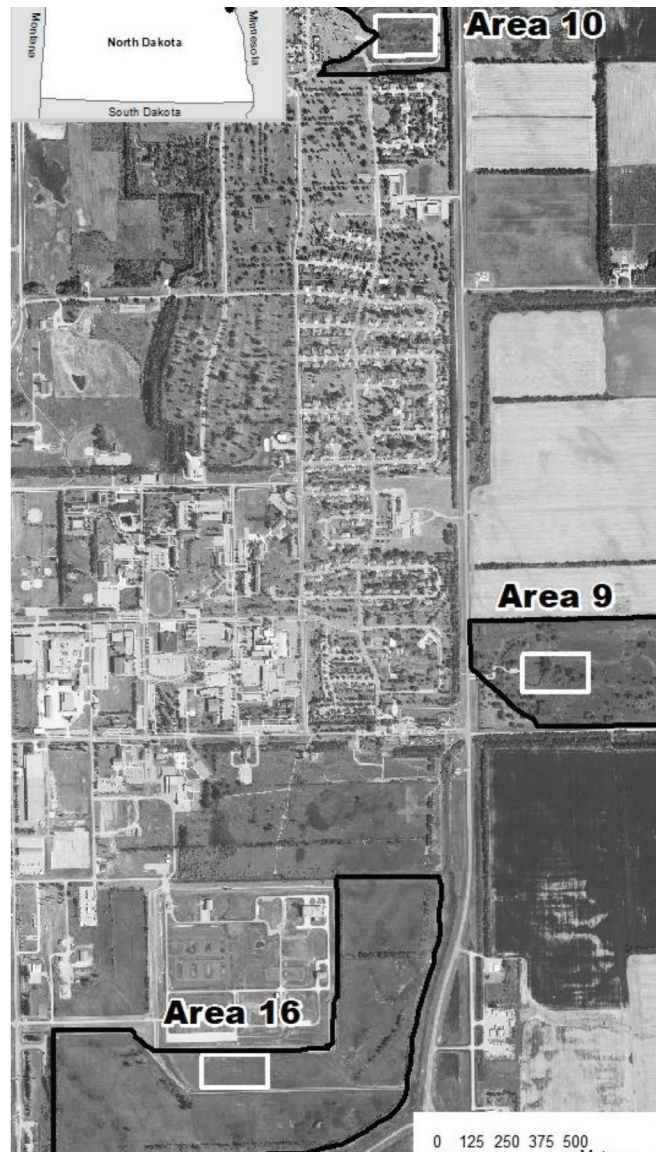
Figure 1. Aerial photo of the eastern half of the Grand Forks Air Force Base (North Dakota, USA; black dot, inset). We trapped small mammals with trap arrays (white outlines) in three large, non-restricted access grassland sites (black outlines) in summer 2014 and 2015.

Hecla, and Ojata series formed in glacial till (Wali et al. 1973). Like most soils within Grand Forks County, GFAFB's soils are poorly to moderately well drained and are moderately saline to very strongly saline. Prior to European settlement, tallgrass and mixed grass prairie were the dominant vegetation types (Hadley and Buccos 1967, Redmann 1972, Wali et al. 1973). Prior to the U.S. Department of Defense purchasing the land in 1955, most of the lands that make up GFAFB and the greater Grand Forks County had been extensively tilled and cultivated (Redmann 1972).

Currently, about half of the GFAFB land is not developed, the majority of which is some type of grassland. A 2008 vegetation survey noted that non-native species including leafy spurge (*Euphorbia esula*) and Canada thistle (*Cirsium arvense*) are prevalent throughout the base. Smooth brome (*Bromus inermis*), quackgrass (*Elymus repens*), and Kentucky bluegrass (*Poa pratensis*) were the most dominant grasses (GFAFB 2010). The GFAFB Integrated Natural Resources Management Plan regulates management of these grasslands (Rundquist et al. 2005), and it includes goals related to grassland reconstruction and management through prescribed fire, mowing, and woody vegetation removal. For this study, we sampled three distinct management areas that are the largest accessible sites of their respective types on the base.

The first grassland management area, listed as Area 10 or the Prairie View Nature Preserve in GFAFB documents (hereafter reconstructed prairie), is a 17-ha upland site reclaimed in 2000 after the demolition of a base housing complex. This site is bordered to the north by a dense shelterbelt (approximately 70 m wide). At the center of the site is a 4.86-ha reconstructed upland prairie (centroid: 47.970889, -97.367101) ringed by the old housing development roadways (Fig. 1). The site contains a mowed, gravel-base walking path and a small butterfly garden at the western entrance. After the soil surface was reclaimed, the site was plowed and seeded in spring 2000 with a mixture of 11 native grass cultivars (Millborn Seeds, Brookings, SD). The seed mix included 'Rosana' western wheatgrass (*Pascopyrum smithii*), 'Lodorm' green needlegrass (*Nassella viridula*), 'Revenue' slender wheatgrass (*Elymus trachycaulus*), 'Dacotah' switchgrass (*Panicum virgatum*), 'Pierre' sideoats grama (*Bouteloua curtipendula*), 'Bad River' blue grama (*Bouteloua gracilis*), 'Itasca' little bluestem (*Schizachyrium scoparium*), 'Bison' big bluestem (*Andropogon gerardii*), 'Tomahawk' Indiangrass (*Sorghastrum nutans*), 'Red River' prairie cordgrass (*Spartina pectinata*), and 'Mandan' Canada wildrye (*Elymus canadensis*). This site has been managed with spring prescribed fire as per the GFAFB Wildland Fire Management Plan on a four-year return interval (2004, 2008, and 2012), and the species pool was further augmented with a forb mixture broadcast-seeded and rolled after the spring 2004 burn (K. Rundquist, personal communication). Native forbs include oval-leaf milkweed (*Asclepias ovalifolia*), common milkweed (*Asclepias syriaca*), white prairie clover (*Dalea candida*), purple prairie clover (*Dalea purpurea*), black Samson (*Echinacea angustifolia*), Philadelphia fleabane (*Erigeron philadelphicus*), common gaillardia (*Gaillardia aristata*), wild licorice (*Glycyrrhiza lepidota*), Maximilian sunflower (*Helianthus maximiliani*), black medic (*Medicago lupulina*), wild bergamot (*Monarda fistulosa*), common evening primrose (*Oenothera biennis*), and blackeyed susan (*Rudbeckia hirta*) (GFAFB 2010).

The second grassland management area, listed as Area 9 on GFAFB documents (hereafter old field), is an 18-ha field (centroid: 47.949367, -97.359069; Fig. 1) also located on a reclaimed housing complex. In this case, demolitions were completed in 2010 and plant species were allowed to naturally recolonize the site from the local propagule pool. Old gravel roadbeds and the mature trees sporadically distributed throughout the development remain on the site. Smooth brome and Kentucky bluegrass are the dominant grass species. Forbs include field pennycress (*Thlaspi arvense*), false mayweed (*Tripleurospermum maritimum*), bigbract verbena (*Verbena bracteata*), American vetch (*Vicia americana*), sleepy silene (*Silene antirrhina*), oxeye daisy (*Leucanthemum vulgare*), and lambsquarter (*Chenopodium album*) (GFAFB 2010). The remaining trees and low shrubs include Amur maple (*Acer ginnala*), paper birch (*Betula papyrifera*), redosier dogwood (*Cornus sericea*), ponderosa pine (*Pinus ponderosa*), prairie rose (*Rosa arkansana*), and Siberian elm (*Ulmus pumila*) (GFAFB 2010). Additionally, there are notable stands of Russian olive (*Elaeagnus angustifolia*) and willow (*Salix* sp.) saplings. As a result, the old field had the highest tree and shrub density of the three sites. This site is also included in the GFAFB Wildland Fire Management Plan and was most recently burned in spring 2013 (K. Rundquist, personal communication) prior to our study. Additionally, portions were mowed for invasive species in early August 2014 (sample year one; affected three trap points) at a height of 7–14 inches (17.8–35.5 cm).

The final grassland area sampled, listed as Area 16 on GFAFB documents (hereafter hay field), is a 67.5-ha (centroid: 47.936446, -97.374804) lowland wet prairie site managed as a hay field since base inception (Fig. 1). The site was most recently augmented in 2005 with a grass mixture including big bluestem, little bluestem, Indiangrass, switchgrass, sideoats grama, slender wheatgrass, Canada wildrye, green needlegrass, and western wheatgrass (GFAFB 2010). The dominant grasses identified during the 2008–09 vegetation survey were rough bentgrass (*Agrostis scabra*), creeping bentgrass (*Agrostis stolonifera*), foxtail barley (*Hordeum jubatum*), reed canarygrass (*Phalaris arundinacea*), prairie cordgrass (*Spartina pectinata*), prairie wedgescale (*Sphenopholis obtusata*), rough dropseed (*Sporobolus clandestinus*), prairie dropseed (*Sporobolus heterolepis*), and intermediate wheatgrass (*Thinopyrum intermedium*) (GFAFB 2010). Currently, the area is hayed once per year in late August or September.

METHODS

Small Mammal Sampling

We delineated a 260 × 160 m (4.16 ha) trapping area positioned 20 m from edge features in each of the three sites (Fig. 1). Within each trapping area, we established eight parallel traplines spaced 20 m apart, and each consisted of

25 trap points spaced 10 m apart (8 traplines × 25 traps/line = 200 traps/site). We placed a Sherman live trap (5.08 × 6.35 × 16.51 cm) containing a ball of 100% cotton and baited with a peanut butter-rolled oat mixture at each trap point (Sikes and Gannon 2011). Traps were set for five consecutive nights during each summer month (June, July, and August) of 2014 and 2015 (3 months × 2 years = 6 sample times). For each trapping night, we opened traps in the late afternoon and checked them beginning a half hour before sunrise the following morning (sprung traps were not recorded). We recorded the species, sex, relative age (adult or subadult), and breeding status (breeding or nonbreeding) of each individual captured. Additionally, we recorded visible external parasitism events by second and third instar bot fly larvae (*Cuterebra* spp.) in August of both years. Bot fly parasitism only occurred on individuals in the reconstructed prairie. We determined an individual's sex and breeding status through visual examination of the vulva, mammary glands, and testes. To identify animals recaptured within a consecutive five-night period, we temporarily marked individuals with permanent marker on their stomachs and released them at their trap point. We did not quantify recaptures from month to month. Over the course of two years we set 18,000 traps (3 sites × 8 traplines × 25 traps/line × 5 nights × 3 months × 2 years = 18,000 traps). All small mammals were captured and handled in accordance with live capture guidelines outlined by the American Society of Mammalogists (Sikes and Gannon 2011) and approved by the Institutional Animal Care and Use Committee at Bemidji State University (Reference Number: BSU2015-003).

For each of the six trapping periods, we determined the total number and the species richness of small mammals captured within each trapline exclusive of recaptured individuals (study average recapture rate = 11.99 ± 2.7% per five-night period). Because two regional mouse (deer mouse and white-footed mouse) and shrew (masked shrew and Hayden's shrew) species are indistinguishable in the field (Hazard 1982, Seabloom 2011), individuals within each genera were considered a single species for species richness summations. Additionally, we calculated the sex ratio (total males/total females for each site at each sample time), fecundity ratio (nonbreeding females/breeding females), and breeding ratio (breeding individuals/nonbreeding individuals) for meadow voles, the most frequently captured species (Carey and Wilson 2001). We excluded individuals of unknown sex (14 of 419 captured meadow voles) from this summarization.

Vegetation Sampling

We measured vegetation height density (visual obstruction reading; VOR) at three randomly selected points in each trapline during each trapping session using a 185-cm modified Robel pole (Robel et al. 1970). Given the close

proximity of trap points and the vegetation structure in each site, this subsampling reasonably captured within-site vegetation heterogeneity. We recorded vegetation density (50% visibility) to the nearest 5 cm in each cardinal direction at 1-m height and 4 m from the Robel pole on days when the winds were less than 32.2 km/h (Robel et al. 1970). We averaged the readings from the four cardinal directions for each point and then averaged the three readings per trapline to generate a single VOR value for each trapline.

Data Analysis

Because we captured so few additional individuals of other species, our grassland comparison analysis focused on meadow voles. To test for differences in meadow vole captures among sites and over time, we used a generalized linear mixed effects model (PROC GLIMMIX; SAS version 9.4, SAS Inc., Cary, NC, USA). We included site and sampling time as fixed effects and natural log transformed vegetation height density as a covariate. To account for repeated measures on each trapline, we included a random term (i.e., trapline nested in site). Because only five meadow voles were caught in the hay field over the entire two-year study, we excluded the hay field from the site comparison analysis. The meadow vole capture model was based on a negative binomial distribution with a log link function. We used Tukey-Kramer post hoc tests to test for pairwise site-sampling time differences. We tested for differences in meadow vole male-to-female, fecundity, and breeding-to-non-breeding ratios over time using analysis of variance (PROC GLM; SAS version 9.4, SAS Inc., Cary, NC, USA), with Tukey-Kramer post hoc tests to assess pairwise time differences.

RESULTS

We captured 463 individuals from six small mammal species over two years, 96% of which were meadow voles (*n* = 419; Table 1). We captured the majority of non-meadow vole individuals (31 of 103 individuals [30%] in the reconstructed prairie, 5 of 28 individuals [18%] in the old field) in August 2015. Most individuals (64%), including all shrews and the sole ermine (*Mustela erminea*), were captured in the reconstructed prairie. Although our trapping effort was similar across all sites, trapping yielded few captures (2.2% of the total) in the hay field (Table 1). Botflies affected one-third of meadow voles captured (11 of 33 individuals) in the reconstructed prairie in August 2014 and one of 72 individuals in August 2015. At times, the 2014 infestations were substantial; one female individual hosted 10 larvae and had evidence of their successful emergence.

Meadow vole captures were similar between the old field and reconstructed prairie at all times (Site $F_{1,14}$ = 1.27, *P* = 0.28). However, the effect of sampling time on meadow

Table 1. Composition of small mammals captured in three differently managed grasslands of the Grand Forks Air Force Base, North Dakota, USA, in summer 2014 and 2015. Numbers are the totals for each site across all sampling times.

Common Name (Scientific Name)	Reconstructed Prairie	Old field	Hay field
Meadow vole (*Microtus pennsylvanicus*)	258	156	5
Deer/white-footed mouse (*Peromyscus* spp.)	21	6	0
Meadow jumping mouse (*Zapus hudsonius*)	0	1	0
Arctic shrew (*Sorex arcticus*)	8	0	0
Masked/Hayden's shrew (*Sorex* spp.)	7	0	0
Ermine (*Mustela erminea*)	1	0	0
Total	295	163	5
Trapline richness ± SE	1.33 ± 0.11	0.96 ± 0.07	

vole captures differed among sites (Time $F_{5,63}$ = 2.35, P = 0.051; Site × Time $F_{5,63}$ = 3.91, P = 0.004; Fig. 2). Although meadow vole captures were consistent over time in the reconstructed prairie, in the old field we captured more meadow voles in July 2015 than in June and August of 2014 (Fig. 2). Meadow vole captures were marginally and variably affected by vegetation height density over time (VOR $F_{1,63}$ = 2.91, P = 0.09; VOR × Site $F_{1,63}$ = 0.73, P = 0.40; VOR × Time $F_{5,63}$ = 2.01, P = 0.09; Fig. 3). The strongest positive effects of VOR on meadow vole captures was in the second year in the reconstructed prairie when the site had the greatest variation in VOR (Fig. 3). During this period, tall sweet clover (*Melilotus officinalis*) naturally increased in abundance in two large patches between the first and second year. Although meadow vole fecundity ($F_{5,6}$ = 1.81, P = 0.25) and breeding ratios ($F_{5,6}$ = 0.38, P = 0.85) were consistent over time, male-to-female ratios fluctuated over time ($F_{5,6}$ = 7.32, P = 0.02). We captured more males relative to females in June 2014 compared to June 2015 (Fig. 4).

DISCUSSION

Our capture results indicated that the prairie reconstruction effort was effective at creating conditions that support the grassland small mammals of the region. Likewise, although the old field reclamation effort did not include a diverse plant mixture, the old field supported meadow voles at the same level as the reconstructed prairie. In contrast, we only captured 2% of the total individuals in the continuously managed hay field. Although this site was the largest and had been in grassland cover the longest of all the sites sampled, the haying management combined with the area being the most low-lying site appears to be keeping it from effectively serving as grassland small mammal habitat.

We captured half (7 of 13) of the grassland small mammal species previously recorded in Grand Forks county (Seabloom 2011). Although we would reasonably expect to find northern short-tailed shrews (*Blarina brevicauda*) and prairie voles (*Microtus ochrogaster*) (Iverson et al. 1967, Yahner 1983, George et al. 1986, Stalling 1990, Seabloom 2011), we did not capture any and have not recorded them in ongoing sampling efforts in nearby remnant grasslands (L. R. LaFond, unpublished data). The remaining five species previously recorded in the county, but not captured in our study, include three relatively rare species (pygmy shrew, western jumping mouse, and northern grasshopper mouse) and two introduced pest species (house mouse and Norway rat).

Our captures of the Arctic shrew and the ermine are notable for the region. The Arctic shrew is a Level III Species of Conservation Priority in North Dakota (Dyke et al. 2015) and had not been documented on GFAFB in over 20 years (GFAFB 1994). Little is known about the species regionally (Buckner 1966, Iverson et al. 1967, Kirkland
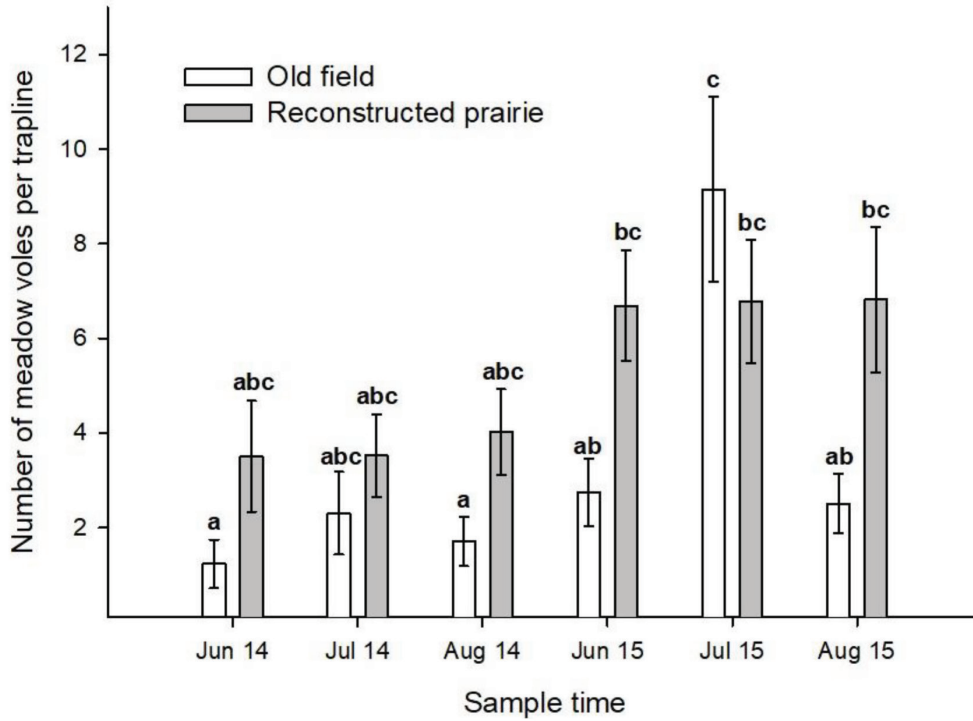
Figure 2. Number (LS Mean ± 1 SE) of meadow voles (*Microtus pennsylvanicus*) captured per trapline in an old field and a reconstructed prairie of Grand Forks Air Force Base, North Dakota, USA, in summer (June, July, and August) 2014 and 2015. Means with different letters are significantly different (Tukey's Post-Hoc test).
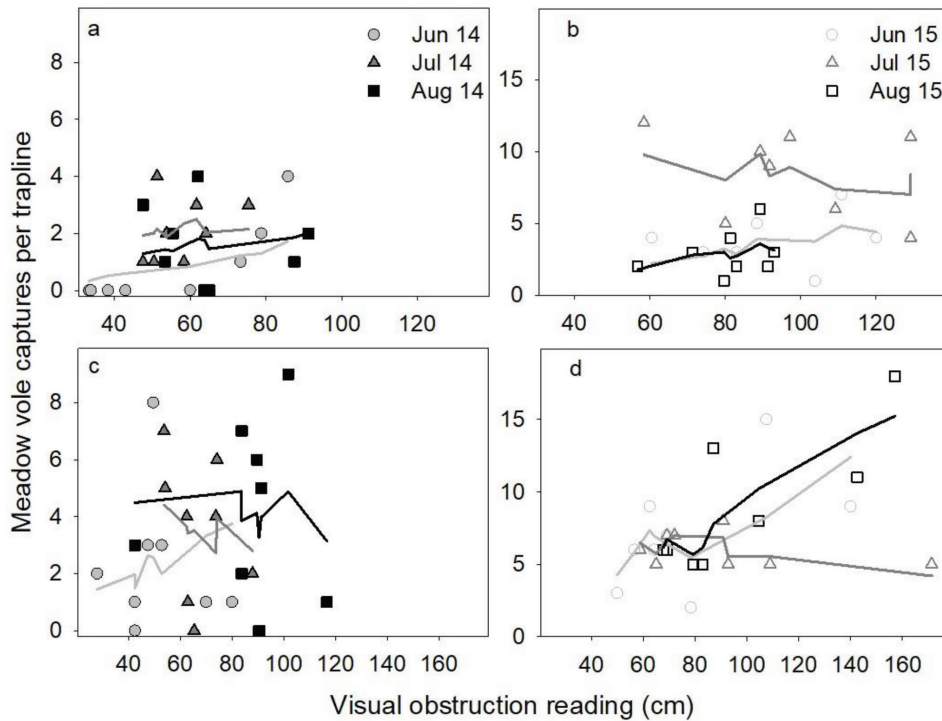

Figure 3. Effect of plant height density (visual obstruction reading; VOR) on meadow vole captures in an old field (a-b) and a reconstructed prairie (c-d) in 2014 (a, c) and 2015 (b, d) of the Grand Forks Air Force Base, North Dakota, USA. Lines are model predictions for VOR observations in each month.
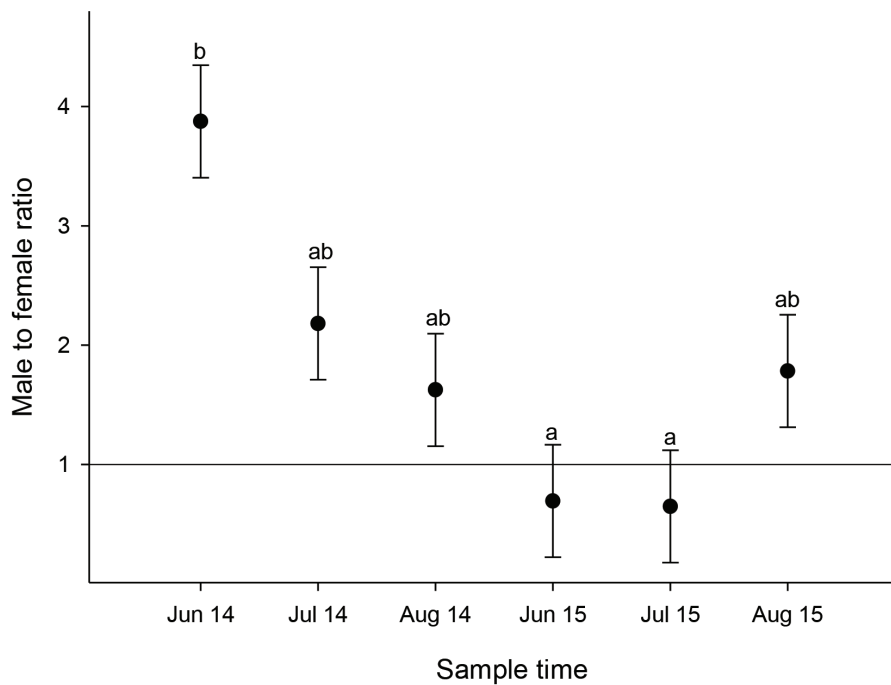
Figure 4. Ratio of male-to-female (LS Mean ± 1 SE) meadow voles (*Microtus pennsylvanicus*) in summer (June, July, and August) 2014 and 2015, summarized across an old field and reconstructed prairie grassland sites of the Grand Forks Air Force Base, North Dakota, USA. Means above the solid line indicate sex ratios where males exceed females. Means with different letters are significantly different (Tukey's Post-Hoc test).

and Schmidt 1996, Perry et al. 2004), and it is listed as uncommon and of unknown status by the North Dakota Game and Fish Department (Dyke et al. 2015). However, it is likely that Arctic shrews are widely distributed in the Grand Forks county grasslands, as we have captured several in ongoing studies in a nearby remnant grassland (L. R. LaFond, unpublished data). The sole ermine captured was not a target species and had not been previously observed on GFAFB (Rundquist et al. 2005). Although not listed as a Species of Conservation Priority, ermine are uncommon in North Dakota and are found in areas of high small mammal density (King 1983, Seabloom 2011).

As reported in other small mammal studies in the Great Plains (Iverson et al. 1967, Grant and Birney 1979, Mihok et al. 1985, Sietman et al. 1994, Richardson 2010, Mulligan et al. 2013), meadow voles were the dominant species and *Peromyscus* spp. were less abundant. In most cases, *Peromyscus* spp. occur sporadically and in low numbers in tallgrass prairie (Moretti and Schramm 1972, Getz and Hofmann 1999), which may be because meadow voles dominate interspecies interactions with *Peromyscus* spp. (Reich 1981). Our *Sorex* spp. captures were also low, which may be an outcome associated with our trapping methods. Within our study, *Sorex* spp. captures were most likely incidental as they fed on invertebrates (e.g., millipedes,

Eurymerodesmus spp.) attracted to the bait (Patric 1970). Additionally, although Sherman live traps are presumed effective at capturing *Peromyscus* spp., some have questioned their effectiveness at capturing *Sorex* spp. (Williams and Braun 1983, Mengak and Guynn Jr 1987). That said, this outcome is consistent with previous studies that reported low *Sorex* spp. numbers in unmanaged, burned, mowed, and hayed grasslands (Tester and Marshall 1961, Kaufman and Kaufman 1989, Neuhaus 2015).

Overall, meadow vole numbers were consistent between years in the sites and fall within the range of values reported in previous studies. Mihok (1984) found meadow vole numbers ranging from 10 to 350 individuals per 3.24 ha and Neuhaus (2015) reported from 7 to 69 individuals in 0.25 ha plots. The only temporal exception in our study was the peak in meadow vole abundance in July 2015 in the old field, which likely reflected a population fluctuation that is common for the species. A 10-year study of meadow voles in a 3.24-ha old-field found abundance to increase by more than 100 individuals with a four-week period (Mihok 1984). Such within-season variation of voles has been well documented, yet there is no firm understanding of the cause of these fluctuations (Krebs et al. 1969, Mihok 1984, Mihok et al. 1985). The most recent hypotheses include a combination of patterns of life history and age of sexual maturity (Oli and

Dobson 2001).

We found that not all of the GFAFB grasslands serve small mammals equally. Small mammals were least abundant in the hay field, an outcome consistent with previous studies that reported low small mammal diversity in intensively hayed sites, presumably because of reduced cover and seed and insect food sources (LoBue and Darnell 1959, Getz 1985, Kaufman and Kaufman 2008). In contrast, the reconstructed prairie had the greatest meadow vole numbers and a well-developed small mammal community. This could be due to inputs from the shelterbelt and adjacent agricultural fields but, presumably, this effect would have also occurred in the hay field. In the old field and reconstructed prairie, captures marginally increased with vegetation density (VOR), an effect that was particularly notable in areas with higher sweet clover cover (August 2015). Most likely, all captures were affected additionally by litter depth and plant species composition, site characteristics that we did not measure in this study. These results reinforce the notion that not all grassland serve small mammals equally and that fostering heterogeneous management may be key to effectively maintaining small mammal populations across landscapes (Fuhlendorf et al. 2010).

ACKNOWLEDGMENTS

LITERATURE CITED

Birney, E. C., W. E. Grant, and D. D. Baird. 1976. Importance of vegetative cover to cycles of *Microtus* populations. Ecology 57:1043–1051.

Brady, M. J., and N. A. Slade. 2001. Diversity of a grassland rodent community at varying temporal scales: the role of ecologically dominant species. Journal of Mammalogy 82:974–983.

Buckner, C. H. 1966. Populations and ecological relationships of shrews in tamarack bogs of southeastern Manitoba. Journal of Mammalogy 47:181–194.

Burel, F., A. Butet, Y. R. Delettre, and N. Millàn de la Peña. 2004. Differential response of selected taxa to landscape context and agricultural intensification. Landscape and Urban Planning 67:195–204.

Carey, A. B., and S. M. Wilson. 2001. Induced spatial heterogeneity in forest canopies: responses of small mammals. Journal of Wildlife Management 65:1014–1027.

Churchfield, S., J. Hollier, and V. Brown. 1991. The effects of small mammal predators on grassland invertebrates, investigated by field exclosure experiment. Oikos 60:283–290.

Clark, B. K., and D. W. Kaufman. 1990. Short-term responses of small mammals to experimental fire in tallgrass prairie. Canadian Journal of Zoology 68:2450–2454.

Colvin, B. A., and E. B. McLean. 1986. Food habits and prey specificity of the common barn owl in Ohio. Ohio Journal of Science 86:76–80.

Diffendorfer, J. E., M. S. Gaines, and R. D. Holt. 1999. Patterns and impacts of movements at different scales in small mammals. Pages 63–88 *in* G. W. Barrett and J. D. Peles, editors. Landscape ecology of small mammals. Springer-Verlag, New York, New York, USA.

Dyke, S. R., S. K. Johnson, and P. T. Isakson. 2015. North Dakota state wildlife action plan. North Dakota Game and Fish Department, Bismarck, North Dakota, USA.

French, N. R., D. M. Stoddart, and B. Bobek. 1975. Patterns of demography in small mammal populations. Pages 73–102 *in* F. B. Golley, K. Petrusewicz, and L. Ryszkowski, editors. Small mammals: their productivity and population dynamics. Cambridge University Press, London, UK.

Fuhlendorf, S., W. Harrell, D. Engle, R. Hamilton, C. Davis, and D. Leslie Jr. 2006. Should heterogeneity be the basis for conservation? Grassland bird response to fire and grazing. Ecological Applications 16:1706–1716.

Fuhlendorf, S. D., D. E. Townsend II, R. D. Elmore, and D. M. Engle. 2010. Pyric-herbivory to promote rangeland heterogeneity: evidence from small mammal communities. Rangeland Ecology & Management 63:670–678.

George, S. B., J. R. Choate, and H. H. Genoways. 1986. *Blarina brevicauda*. Mammalian Species 261:1–9.

Getz, L., and J. Hofmann. 1999. Diversity and stability of small mammals in tallgrass prairie habitat in central Illinois, USA. Oikos 85:356–363.

Getz, L. L. 1985. Habitats. Pages 286–309 *in* R. H. Tamarin, editor. Biology of new world *Microtus*. The American Society of Mammalogists, Shippensburg, Pennsylvania, USA. Special Publication 8.

GFAFB. 1994. Grand Forks Air Force Base Biological Survey, Product #4. Prepared by the North Dakota Natural Heritage Inventory and North Dakota Parks and Recreation Department, Bismarck, North Dakota, USA.

GFAFB. 2010. Final 2009 biological survey Grand Forks Air Force Base, Grand Forks County, North Dakota. Prepared by Science Applications International Corporation, USA.

Gibson, D. 2009. Grasses and grassland ecology. Oxford University Press, New York, New York, USA.

Grant, W., and E. C. Birney. 1979. Small mammal community structure in North American grasslands. Journal of Mammalogy 60:23–36.

Grant, W. E., E. C. Birney, N. R. French, and D. M. Swift. 1982. Structure and productivity of grassland small mammal communities related to grazing-induced changes in vegetative cover. Journal of Mammalogy 63:248-260.

Hadley, E. B., and R. P. Buccos. 1967. Plant community composition and net primary production within a native eastern North Dakota prairie. American Midland Naturalist 77:116–127.

Hazard, E. B. 1982. The mammals of Minnesota. University of Minnesota Press, Minneapolis, USA.

Howe, H. F., J. S. Brown, and B. Zorn-Arnold. 2002. A rodent plague on prairie diversity. Ecology Letters 5:30–36.

Howe, H., and D. Lane. 2004. Vole-driven succession in experimental wet-prairie restorations. Ecological Applications 14:1295–1305.

Howe, H., B. Zorn-Arnold, A. Sullivan, and J. Brown. 2006. Massive and distinctive effects of meadow voles on grassland vegetation. Ecology 87:3007–3013.

Iverson, S. L., R. W. Seabloom, and J. M. Hnatiuk. 1967. Small-mammal distributions across the prairie-forest transition of Minnesota and North Dakota. American Midland Naturalist 78:188–197.

Kaufman, D., E. Finck, and G. Kaufman. 1990. Small mammals and grassland fires. Pages 46–80 *in* S. Collins and L. Wallace, editors. Fire in North American tallgrass prairie. University of Oklahoma Press, Norman, USA.

Kaufman, D., and G. Kaufman. 1989. Population Biology. Pages 233–271 *in* G. Kirkland, Jr. and J. Layne, editors. Advances in the study of *Peromyscus* (Rodentia). Texas University Press, Lubbock, Texas, USA.

Kaufman, D., and G. Kaufman. 1990. Influence of plant litter on patch use by foraging *Peromyscus maniculatus* and *Reithrodontomys megalotis*. American Midland Naturalist 124:195–198.

Kaufman, G., and D. Kaufman. 2008. Effects of haying on small mammals in mixed grass prairie of central Kansas. Transactions of the Kansas Academy of Science 111:275–282.

Kaufman, D., G. Kaufman, and E. Finck. 1989. Rodents and shrews in ungrazed tallgrass prairie manipulated by fire. Proceedings of the North American Prairie Conference 11:173–178.

King, C. 1983. *Mustela erminea*. Mammalian Species 195:1–6.

Kirkland Jr., G., and D. Schmidt. 1996. *Sorex arcticus*. Mammalian Species 524:1–5.

Klatt, B. J., and L. L. Getz. 1987. Vegetation characteristics of *Microtus ochrogaster* and *M. pennsylvanicus* habitats in east-central Illinois. Journal of Mammalogy 68:569–577.

Krebs, C. J., B. L. Keller, and R. H. Tamarin. 1969. *Microtus* population biology: demographic changes in fluctuating populations of *M. ochrogaster* and *M. pennsylvanicus* in southern Indiana. Ecology 50:587–607.

Kulmatiski, A., and K. H. Beard. 2013. Woody plant encroachment facilitated by increased precipitation intensity. Nature Climate Change 3:833–837.

LoBue, J., and R. M. Darnell. 1959. Effect of habitat disturbance on a small mammal population. Journal of Mammalogy 40:425–437.

Matlack, R. S., D. W. Kaufman, and G. A. Kaufman. 2008. Influence of woody vegetation on small mammals in tallgrass prairie. American Midland Naturalist 160:7–19.

Mengak, M. T., and D. C. Guynn Jr. 1987. Pitfalls and snap traps for sampling small mammals and herpetofauna. American Midland Naturalist 118:284–288.

Mihok, S. 1984. Life history profiles of boreal meadow voles (*Microtus pennsylvanicus*). Carnegie Museum of Natural History Special Publication 10:91–102.

Mihok, S., B. N. Turner, and S. L. Iverson. 1985. The characterization of vole population dynamics. Ecological Monographs 55:399–420.

Moretti, L.H. , and P. Schramm. 1972. A comparative survey of small mammal populations in various grassland habitats with emphasis on restored prairie. Proceedings of the Midwest Prairie Conference 3:79–84.

Mulligan, M. P., R. L. Schooley, and M. P. Ward. 2013. Effects of connectivity and regional dynamics on restoration of small mammal communities in midwestern grasslands. Restoration Ecology 21:678–685.

Neuhaus, R. 2015. A comparison of the effects of burning, haying and mowing on plants and small mammals in a tallgrass prairie reconstruction. Electronic Theses and Dissertation Paper 216. University of Northern Iowa.

Oli, M. K., and F. S. Dobson. 2001. Population cycles in small mammals: the α-hypothesis. Journal of Mammalogy 82:573–581.

Omernik, J. M., and G. E. Griffith. 2014. Ecoregions of the conterminous United States: evolution of a hierarchical spatial framework. Environmental Management 54:1249–1266.

Patric, E. F. 1970. Bait preference of small mammals. Journal of Mammalogy 51:179–182.

Perry, N. D., D. T. Stewart, E. M. Madden, and T. J. Maier. 2004. New records for the Arctic shrew, *Sorex arcticus* and the newly recognized Maritime shrew, *Sorex maritimensis*. The Canadian Field-Naturalist 118:400–404.

Ralston, R. D. 1968. The grasslands of the Red River valley. University of Saskatchewan, Saskatoon, Saskatchewan, Canada.

Ratajczak, Z., J. B. Nippert, and S. L. Collins. 2012. Woody encroachment decreases diversity across North American grasslands and savannas. Ecology 93:697–703.

Redmann, R. E. 1972. Plant communities and soils of an eastern North Dakota prairie. Bulletin of the Torrey Botanical Club 99:65–76.

Reich, L. 1981. *Microtus pennsylvanicus*. Mammalian Species 159:1–8.

Richardson, M. 2010. Effects of grassland succession on communities of small mammals in Illinois, USA. Biologia 65:344–348.

Robel, R. J., J. N. Briggs, A. D. Dayton, and L. C. Hulbert. 1970. Relationships between visual obstruction measurements and weight of grassland vegetation. Journal of Range Management 23:295–297.

Rundquist, K., H. Ourako, S. Braun, A. Kammer, C. Klaus, H. Nelson, L. Olson, K. Porteck, and R. Lopez. 2005. Integrated natural resources management plan Grand Forks Air Force Base, North Dakota. 2004–2008. 319 Civil Engineer Squadron, Grand Forks Air Force Base, North Dakota, USA.

Seabloom, R. 2011. Mammals of North Dakota. North Dakota Institute for Regional Studies, North Dakota State University, Fargo, North Dakota, USA.

Sieg, C. H. 1987. Small mammals: pests or vital components of the ecosystem. Great Plains Wildlife Damage Control Workshop Proceedings 97:88–92.

Sietman, B., W. Fothergill, and E. Finck. 1994. Effects of haying and old-field succession on small mammals in tallgrass prairie. The American Midland Naturalist 131:1–8.

Sikes, R., W. Gannon, and the Animal Care and Use Committee of the American Society of Mammalogists. 2011. Guidelines of the American Society of Mammalogists for the use of wild mammals in research. Journal of Mammalogy 92:235–253.

Stalling, D. T. 1990. *Microtus ochrogaster*. Mammalian Species 335:1–9.

Tester, J. R., and W. H. Marshall. 1961. A study of certain plant and animal interrelations on a native prairie in northwestern Minnesota. Occasional Papers No. 8. Minnesota Museum of Natural History, University of Minnesota, USA.

Wali, M. K., G. W. Dewald, and S. M. Jalal. 1973. Ecological aspects of some bluestem communities in the Red River Valley. Bulletin of the Torrey Botanical Club 100:339–348.

Whitaker, J. O. Jr. 1972. *Zapus hudsonius*. Mammalian Species 11:1–7.

Williams, D. F., and S. E. Braun. 1983. Comparison of pitfall and conventional traps for sampling small mammal populations. Journal of Wildlife Management 47:841–845.

Wilson, M. F., and A. Traveset. 2000. The ecology of seed dispersal. Pages 85–110 *in* R. Gallagher, editor. Seeds: the ecology of regeneration in plant communities. CAB International, Boston, Massachusetts, USA.

Wolff, J. 1989. Social Behavior. Pages 271–292 *in* G. Kirkland Jr., editor. Advances in the study of *Peromyscus* (Rodentia). Texas Tech University Press, Lubbock, Texas, USA.

Yahner, R. H. 1983. Small mammals in farmstead shelterbelts: habitat correlates of seasonal abundance and community structure. Journal of Wildlife Management 47:74–84.

Zimmerman, J. L. 1992. Density-independent factors affecting the avian diversity of the tallgrass prairie community. Wilson Bulletin 104:85–94.

Evaluation of Four Larval Fish Sampling Methods in a Large Midwestern River

NATHAN J. LEDERMAN[1], ANTHONY R. SINDT, DOUGLAS J. DIETERMAN, JOHN D. KRENZ, AND SHANNON J. FISHER[2]

Minnesota State University, Mankato, Department of Biological Sciences,
Mankato, MN 56001, USA (NJL, JDK)
Minnesota Department of Natural Resources, 20596 Highway 7, Hutchinson, MN 55350, USA (ARS)
Minnesota Department of Natural Resources, 1801 S. Oak Street, Lake City, MN 55041, USA (DJD)
Water Resources Center, Minnesota State University, Mankato, Trafton Science Center S.,
Mankato, MN 56001, USA (NJL, SJF)
[1]Present Address: Illinois Department of Natural Resources,
13608 Fox Road, Yorkville, IL 60560, USA (NJL)
[2]Present Address: Minnesota Department of Natural Resources,
500 Lafayette Road, St. Paul, MN 55155, USA (SJF)

ABSTRACT Understanding limitations of larval fish capture gears is critical for developing appropriate sampling protocols and interpreting catch data. We evaluated genera richness, genera diversity, assemblage similarities, abundance indices (i.e., density or catch per unit effort [CPUE]), and sample size requirements between a surface slednet and glow-stick light traps used in 2014 and 2015 and a benthic slednet and light-emitting diode light (LED) traps used in 2015 in the Minnesota River. The surface slednet captured the greatest number of larval fish genera (15) while the LED light trap captured the fewest (1). Similarities of assemblages sampled was highest between surface and benthic slednets (58%) and lowest between the benthic slednet and LED light trap (0%). All evaluated gears had low and variable catch rates; the highest variability was observed for the LED light trap (CV = 800), and the lowest variability was observed for surface slednets (CV = 173). Slednets required less effort to detect a 25% change in total larval fish abundance compared to light traps. Low CPUEs or densities were possibly the result of suspended sediment loads (85.3 ± 8.5 Nephelometric Turbidity Units) that blocked light trap entrance slots and clogged net pores. Further, not targeting habitats critical to adult spawning and larval rearing (e.g., log jams or shallower or inside bends of meanders) may have influenced CPUEs and densities. We recommend modifications to evaluated sampling gears (e.g., nets with larger mesh sizes) or the evaluation of additional larval fish sampling methods (e.g., larval seines or pumps) coupled with a stratified random sampling protocol that incorporates complex habitats for sampling larval fish within the main channel of the Minnesota River or other river systems with similar high turbidity levels.

KEY WORDS larval fish relative abundance, larval fish densities, large rivers, sampling gear comparisons, standardized sampling

Assessing larval fish presence and abundance can help inform stocking decisions, index species restoration success, and identify environmental factors that regulate fish community dynamics (Avery 1996, Nemeth 2005, Kelso et al. 2012, Pulg et al. 2013). Interpretations of population dynamics and community structure of larval fishes vary depending on the habitat sampled and timing of sampling (Kelso et al. 2012) because of differential efficacy among fish species and habitats (Bonar et al. 2009). Thus, a variety of larval fish sampling methods have been used for collecting fish larvae (Kelso et al. 2012).

Riverine larval fish have typically been sampled with passive gears and active gears. The most commonly used passive gear for larval fish are light traps (Naus and Adams 2016) that attract and entrap positively phototaxic species

(Kelso et al. 2012). Phototaxic responses vary among species (Mueller and Neuhauss 2010) and by individuals of the same species (Bulkowski and Meade 1983). Phototaxic larvae may be unable to visually detect light sources due to water clarity or unable to reach the light trap due to current velocity (Marchetti et al. 2004, Lindquist and Shaw 2005). Active larval fish sampling gears have included electrofishers (King and Crook 2002), centrifugal pumps (Gale and Mohr 1978), and a variety of seines (e.g., beach, purse, small mesh; Kelso et al. 2012). Most contemporary large-river sampling protocols use an actively towed 500–1,000-µm mesh ichthyoplankton net that is pushed or pulled through the water (e.g., Nannini et al. 2012, Cheshire et al. 2015, Mapes et al. 2015). Ichthyoplankton nets capture larval fish by filtering known volumes of water at specified depths within

the water column (Kelso et al. 2012). Like passive gears, active gears also demonstrate bias depending on deployment times and depths (Bosley et al. 1986), larval gear avoidance (Gartz et al. 1999), and habitat accessibility (Hayes et al. 2012). In flowing waters, ichthyoplankton nets can also be held stationary allowing them to passively fish (e.g., Killgore and Baker 1996). Due to known gear limitations, studies assessing larval fish assemblages often use multiple gear types (Niles and Hartman 2007, Pritt et al. 2015). However, deployment of multiple gears may not always be feasible due to needs for broad geographic sampling within time and fiscal restraints (Bonar et al. 2009).

Riverine fisheries managers often establish main channel monitoring protocols to reliably track trends in larval fish abundance and species richness (Pritt et al. 2015), identify larval fish responses to changing conditions (e.g., climate change and invasive species establishment; Mapes et al. 2015), and verify reproduction for fishes of management interest (Braaten et al. 2008, Harvey et al. 2002). In the Minnesota River, Minnesota, USA, a goal of resource managers is to develop a sampling protocol that monitors trends in larval fish abundance and richness and reproductive activity for native species of concern such as lake sturgeon (*Acipenser fulvescens*), paddlefish (*Polyodon spathula*), and shovelnose sturgeon (*Scaphirhynchus platorynchus*), as well as for invasive species such as bighead carp (*Hypophthalmichthys nobilis*) and silver carp (*H. molitrix*; Minnesota Department of Natural Resources 2013).

Magnan (1991) suggested that larval fish monitoring protocols be based on efficiency, effectiveness, effort requirements, and time of year that will best meet management objectives. Such information is limited for the Minnesota River. Nickel (2014) provided initial information on larval fish sampling on the Minnesota River with a 1-year study using surface slednets and glow-stick light traps. Given temporal (1 year) and gear limitations (single light intensity for light traps and surface sampling with the slednet) of the Nickel (2014) study, annual variations in larval fish assemblages and gear effectiveness could not be sufficiently addressed. Our goal was to build upon the Nickel (2014) investigation by sampling similar time periods and locations but including gear modifications: adding slednet sampling at a different water depth and light trap sampling with increased light intensity using light-emitting diodes (LEDs). The objectives of the study were to 1) compare larval fish assemblages collected with slednets (benthic and surface) and light traps (glow-stick and LED); 2) quantify larval fish densities or relative abundance among gears; and 3) compare precision estimates and sample size estimates among the four gears. The collective results from these three objectives were then used to make recommendations for sampling larval fishes in the Minnesota River.

STUDY AREA

The Minnesota River originates at Big Stone Lake along the Minnesota-South Dakota border as a sixth-order river (Strahler 1957) and flows 530 km to its confluence with the Mississippi River near St. Paul, Minnesota (Musser et al. 2009). The Minnesota River is generally characterized as low gradient, productive, and turbid, with an annual hydrological regime driven by spring snowmelt and rainfall (Waters 1977). Approximately 79% of pre-settlement grasslands within the Minnesota River watershed have been converted to row-crop agriculture. Landscape conversion has reduced the time water spends on the landscape, increasing erosion potential (Thoma et al. 2005) and creating more intense hydrographs (Nelson 2015) that amplify sediment movement within the Minnesota River (Johnson et al. 2009).

Two reaches near the towns of Savage (river kilometer [RKM] 24–26) and Franklin (RKM 298–300), Minnesota, were sampled in 2014 and two reaches near Henderson (RKM 105–107) and New Ulm (RKM 234–236), Minnesota, were sampled in 2015 (Fig. 1). Reaches were selected to reflect habitat diversity present within the Minnesota River. Study reach channel widths varied from 55 to 140 m (mean [$\overline{X}$] ± 1 standard error [SE] = 70.3 ± 3.3). Median annual discharges from 2005 to 2015 recorded at United States Geological Survey gauging stations near study reaches at RKMs 64 and 313 were 98 and 40 m³/s, respectively (Minnesota Pollution Control Agency 2016). Mean turbidity (± SE) during the same time frame (2005–2015) at the same gauging stations was 202.0 ± 43.1 and 55.5 ± 3.5 Nephelometric Turbidity Units (NTUs), respectively (Minnesota Pollution Control Agency 2016).

METHODS

Larval fishes were sampled approximately every other week from 15 May to 15 August 2014 at the Franklin and Savage study reaches (5 surveys at each reach) and from 23 April to 15 August 2015 at the Henderson and New Ulm study reaches (7 surveys at each reach). Within each study reach, 10 transects were established at 200-m intervals. Each transect was initiated on the left bank and extended at a 30–40° upstream angle to encompass all habitats across the river where gears could be deployed (>0.75 m in water depth). During 2014, one glow-stick light trap sample and one surface slednet sample were collected at each transect during each survey. A benthic slednet and a LED-light-source light trap were added as additional gears in 2015. We randomly selected the light source for each light trap so half of the transects (*n* = 5) during each survey were equipped with a glow-stick and half (*n* = 5) were equipped with a LED. The net method used at each transect was also randomly selected in 2015 so that half of the transects during each survey were sampled with the surface slednet (*n* = 5) and
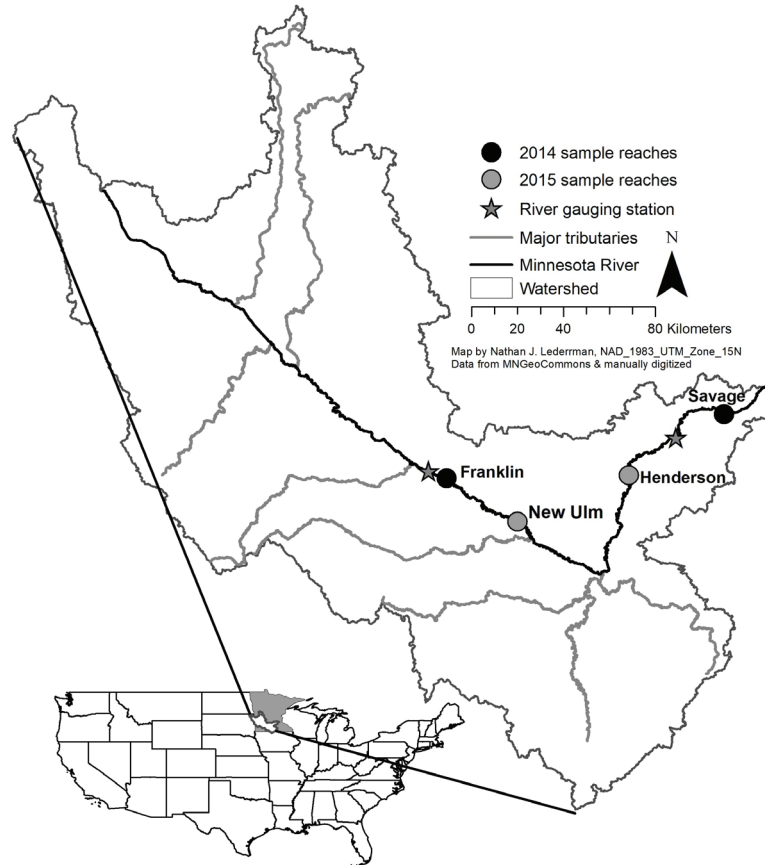
Figure 1. Study reaches where larval fish sampling gears were evaluated in the Minnesota River, Minnesota, USA, during 2014 and 2015. Included are U.S. Geological Survey river gauging stations (stars) near Morton and Jordan, Minnesota, USA.

half with the benthic slednet (*n* = 5).

Glow-stick light traps and LED light traps were the same quatrefoil design used by Nickel (2014) and included 2-mm slot openings and a light source suspended inside the trap from an eyebolt on the top plate (Fig. 2a). Glow-sticks (16 × 2 cm, FlashingBlinkyLights, California, USA) produced mean light intensity of 0.2 ± 0.1 lux directly outside of the light trap. Battery powered LEDs (two green LED lamps in a 12 × 4-cm polycarbonate resin body, KryptoLume, Utah, USA) increased light intensity to 1.2 ± 0.1 lux (379% increase) directly outside of the light trap compared to the glow-stick light source. Light traps were suspended directly beneath the water surface with a buoy tethered to a 9.1-kg cement block with 6 m of rope. Light traps were placed about 2 m from the bank in water deep enough to allow the light trap to be completely submerged but not deeper than the tether (i.e., 0.75–6.0 m deep). We set light traps between 0830 and 1200 and retrieved them 24 hr later.
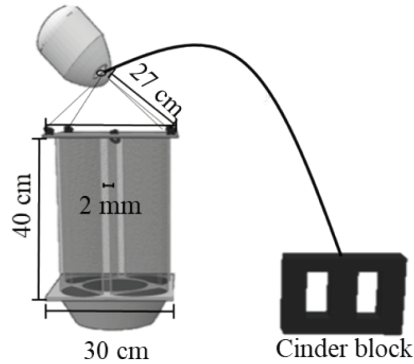
The surface slednet was designed by Nickel (2014) and used a 500-µm mesh driftnet with a polyvinyl chloride pipe frame (Fig. 2b). Surface slednets were towed upstream along the sample transect parallel to the side of the boat at ground speeds ~1.6 km/h for about 5 min. In 2015, we altered

the surface slednet to be able to sample near the benthic surface by attaching three detachable 13.5-kg sounding weights (Hoskin Scientific Ltd., British Columbia, Canada) that sank the sled to the river bed (Fig. 2b). We anchored the boat where the thalweg intersected the sample transect, lowered the weighted surface slednet to the river bottom, and allowed the slednet to soak for 5 min before lifting the net. A mechanical flow meter (General Oceanics, Miami, Florida, USA) suspended in the mouth of the net was used to estimate volume of water filtered for both surface and benthic samples. We conducted all slednet samples between 0830 and 1300.

All larval fish samples taken from all gears were immediately fixed in a 10% buffered formalin solution. After 48 hr, each sample was filtered through a 53-µm sieve (Newark Wire Cloth Company, New Jersey, USA), and contents were preserved in 90% ethanol. Larval fish were sorted from sample contents, identified to genera under a dissecting microscope (Olympus, Massachusetts, USA) using larval fish keys by Auer (1982), Kay et al. (1994), and Simon and Wallus (2005), and counted.

Genera richness, diversity, and community assemblage similarities were calculated and compared among larval fish

a) light trap

27 cm

40 cm

2 mm

30 cm

Cinder block

b) slednet

140 cm

100 cm

30 cm

45 cm

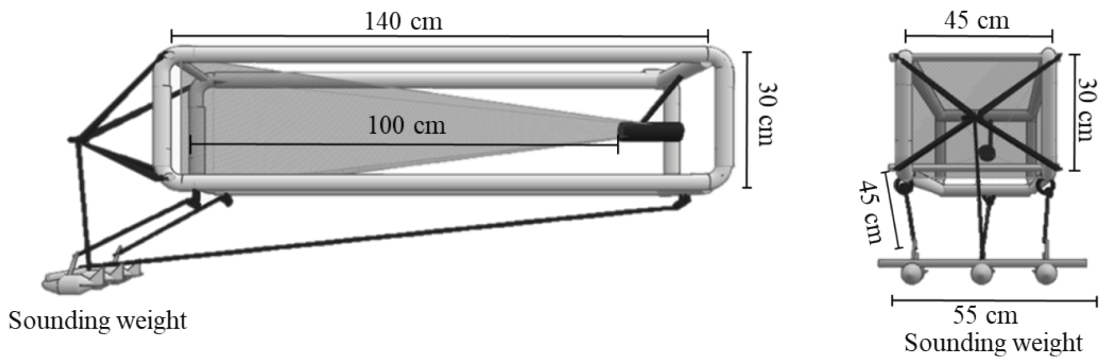30 cm

45 cm

55 cm

Sounding weight

Sounding weight

Figure 2. Schematic of the light trap (a) and slednet (b) used for sampling larval fish in the Minnesota River, Minnesota, USA, during 2014 and 2015. Sounding weights were attached to the surface net to adapt it to sampling at the benthic-water interface (i.e., benthic slednet).

gears. A Shannon-Weiner diversity index was used to index diversity of the larval fish assemblage captured within each gear type and was calculated using the "vegan" package (Oksanen et al. 2019) using the following equation:

$$H' = -\Sigma\, P_i * \log_e P_i,$$

where P_i is the proportion of species in a sample (Kwak and Peterson 2007). Values of H' closer to zero indicate a less-diverse fish assemblage and values further away from zero indicates a more-diverse fish assemblage. Schoener's percentage overlap index was calculated to quantify similarities in assemblages among each pairwise gear comparison (PSI; Schoener 1970). Schoener's percentage overlap index was calculated using the "spaa" package (Zhang 2016) using the following equation:

$$P_{jk} = \Sigma\, minimum\ (P_{ki}, P_{ji},),$$

where P_{ki} and P_{ji} are the count of species I in assemblage j and k, and minimum indicates the smallest of the two counts and is used in the summation (Kwak and Peterson 2007).

The total number of larval fish was used to calculate density (larvae/100 m^3 of water) for each slednet sample and total catch per unit effort (CPUE; larvae/trap night) for each light trap. Coefficient of variation (CV) was used as a measure of precision for larval fish density and CPUE for each gear type. A lower CV indicated higher precision (e.g., less variation) among samples (Zar 1996).

Sample size required to detect a 25% change in mean larval fish density or relative abundance was estimated for each gear with the power analysis formula described by Snedecor and Cochran (1989):

$$n = 2\left(z_\alpha + z_\beta\right)^2 (s^2/d^2)$$

where n is estimated sampling effort, z_α is the z-distribution deviate for the probability of a Type I error at a given level of significance, z_β is the z-distribution deviate for the probability of a Type II error at a given level of statistical power, s is the standard deviation of the abundance estimate (i.e., larvae per 100 m^3 of water for nets or larvae per trap for light traps) and d is the specified effect size. We used an alpha of 0.10, a beta of 0.20, and a power (1-" β") of 0.80 similar to

Dembkowski et al. (2012). All analyses were performed with Program R version 3.3.3 (R Development Core Team 2014).

RESULTS

A total of 99 overnight glow-stick light trap samples and 100 surface slednet samples that filtered 22,515 m³ of water ($\bar{X} \pm SE = 225 \pm 5$ m³/sample) were collected in 2014. In 2015, 64 overnight glow-stick light trap samples, 64 LED light trap samples, 70 surface slednet samples that filtered 19,564 m³ of water (279 ± 8 m³/sample), and 65 benthic slednet samples that filtered 8,655 m³ of water (133 ± 15 m³/sample) were collected.

A total of 213 larval fish representing 18 genera were captured collectively among all gears, reaches, and years. The LED light trap detected the fewest genera (1) and the surface slednet detected the most genera (15; Tables 1, 2). The surface slednet had the highest diversity index (H') during both years of sampling (1.8 in 2014, 2.1 in 2015; Table 1) and the LED light trap had the lowest (0 in 2015; Table 2). Schoener's PSI was highest between the 2015 surface slednet and 2015 benthic slednet (58%) and lowest between the 2015 benthic slednet and 2015 LED light trap (0%; Table 3).

Samples where zero larval fish were captured were common among all four gears, reducing CPUE or density estimates for each of those gears. The LED light trap had the highest percentage of samples where zero larvae were caught and the surface slednet had the lowest (Tables 1 and 2). Density and relative abundance tended to be higher during July and August compared to April, May, and June (Fig. 3). The highest mean CPUE (0.2 ± 0.1 larvae/trap-night) was observed in the 2014 glow-stick light traps, and the highest observed mean larval density was in the 2015 benthic slednet (0.5 ± 0.1 larvae/100 m³; Table 1). Coefficients of variation of mean CPUE or density estimates were high among all gears, but higher CVs were observed among light trap methods compared to slednet methods (Tables 1, 2). Effort required to detect a 25% change in mean CPUE or density was highest for light traps (>100 samples) compared to slednets (<64 samples; Tables 1, 2).

DISCUSSION

The light trap and slednet represent some of the more commonly used large river larval fish sampling gears (Niles and Hartman 2007, Kelso et al. 2012, Mapes et al. 2015, Pritt et al. 2015). Few studies have compared these gears to each other or have tested modifications of those to increase catches of larval fishes in riverine systems. Despite efforts to increase larval catches, gears evaluated here may still be limiting abilities to draw inferences about larval fish production. Only 40% of the known genera in the Minnesota River were captured across all gears used in this study. Low catches of larval fish were also found among all gears leading to highly skewed data, creating challenges in identifying gears to monitor abundance, and quantifying specific sampling periods for larval fish within the Minnesota River. Despite the restrictiveness of results, we were still able to make recommendations for future larval sampling efforts.

Previous research has shown the ability to capture genera varies widely among larval fish gears. In the Kanawha River, West Virginia, USA, Rider and Margraf (1997) used a modified Isaacs-Kidd ichthyoplankton net (n = 912) and sampled 21 of 40 genera (53%) during the larval stage that Messinger and Chambers (2001) noted as being present. Niles and Hartman (2007) sampled 12 of the Kanawha River genera (30%) using benthic sleds (n = 720), larval activity traps (n = 360), and light traps (n = 360). Passive light traps captured 45% more genera compared to the benthic slednet (Niles and Hartman 2007). Gale and Mohr (1978) captured more species with an active ichthyoplankton net compared to a passive net. In flow-through backwaters of the upper Missouri River, Fisher (1999) captured 18 genera of larval fish, including 15 genera in glow-stick light traps and 13 genera in surface trawls. In our study, the glow-stick light trap and LED light trap captured fewer genera than expected based off the fish community present, but the benthic slednet and surface slednet performed at a level comparable to other studies.

Abundance indices among capture gears also vary markedly from prior research. Holland-Bartels et al. (1995) reported capturing nearly 1,000 times more larvae/100 m³ in the main channel of the Mississippi River than we did in the Minnesota River with similar surface trawls. Similarly, mean benthic slednet and light trap catch rates from our study were more than 50 times lower than catch rates of similar gears reported by Niles and Hartman (2007) from the main channel of the Kanawha River. However, our results were more similar to those of Nickel (2014; e.g., 0.1 v. 3.2 larval fish/trap night and 0.4 v. 1.0 larval fish/100 m³) than other research. Results indicate that abundance may vary among systems and gear selection should be system specific.

Various larval fish capture gears have been recommended depending on study system characteristics (e.g., fish population, spatial/temporal sampling, habitat). Niles and Hartman (2007) and Neal et al. (2012) recommend the use of light traps to sample larval fish in rivers. Those studies sampled shallow, slower water velocity areas within the river. Increases in discharge may reduce the effectiveness of light traps (Lindquist and Shaw 2005). Systems or sample areas that are deeper and have higher steady flow (i.e., main channel, channel boarders, side channels) may be more conducive to net gears (e.g., benthic slednet or surface slednet) that exploit the limited swimming capabilities of larvae instead of those requiring active swimming to the gear (glow-stick light trap or LED light trap).

Recommended capture gear should also take other system-specific factors, such as sediment levels, into consideration.

Table 1. Genera richness, Shannon-Weiner Diversity Index (H'), percentage of samples capturing no larvae, mean CPUE (no./trap night ±1 standard error), coefficient of variation of mean CPUE (CV), and the number of samples (*n*) required to detect a 25% change in larval fish abundance for light trap methods from the Minnesota River, Minnesota, USA, during 2014–2015.

Year	Gear	Genera richness	H'	Zero catch (%)	CPUE (no./trap night)	CV	*n*
2014	Glow-stick	6	1.2	89	0.17 (0.09)	525	109
2015	Glow-stick	0	NA	100	0.00 (0.00)	NA	NA
2016	LED	1	0.0	98	0.02 (0.02)	800	NA

Table 2. Genera richness, Shannon-Weiner Diversity Index (H'), percentage of samples capturing no larvae, mean density (no./100m³ of water ±1 standard error), coefficient of variation of mean densities (CV), and the number of samples (*n*) required to detect a 25% change in larval fish abundance for slednet methods from the Minnesota River, Minnesota, USA, during 2014–2015.

Year	Gear	Genera richness	H'	Zero catch (%)	CPUE (no./100m³)	CV	*n*
2014	Surface	10	1.8	61	0.33 (0.06)	171	15
2015	Surface	14	2.1	56	0.37 (0.08)	174	21
2015	Benthic	8	1.7	74	0.50 (0.14)	226	63

Table 3. Percentage similarity index (PSI; %; Schoener 1970) of larval fish assemblages between each pairwise comparison of evaluated gear by year. Comparisons to 2015 glow-stick light traps were omitted as no larval fish were captured with that gear during that year.

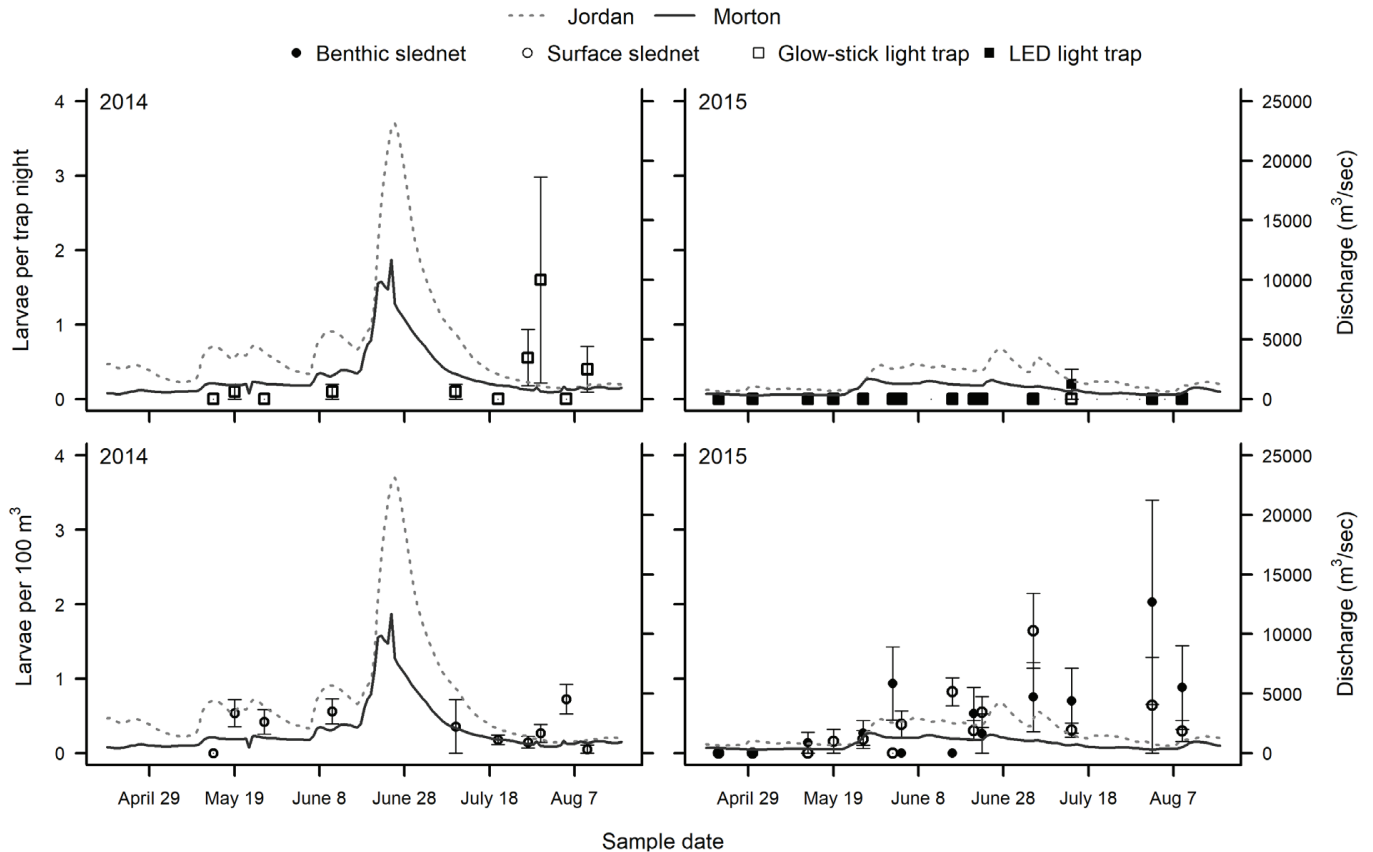|	2014 Glowstick light trap	2014 Surface slednet	2015 Benthic slednet	2015 LED light trap
2014 Surface slednet	21			
2015 Benthic slednet	36	58		
2015 LED light trap	11	8	0	
2015 Surface slednet	16	52	58	3

Figure 3. Larval fish abundance indices from the Minnesota River, Minnesota, USA, during 2014 (left panels) and 2015 (right panels) from light traps (top panels; larvae per net night) and slednets (bottom panels; larvae per 100 m³ of water). Bars indicate ±1 standard error. Discharge measures of the Minnesota River taken by the U.S. Geological Survey near Jordan, Minnesota (grey dotted line), and Morton, Minnesota (black line), are overlaid on each plot.

Deployed light traps slow water velocities, which appeared to cause sediment to accumulate in the entrance slots. This sedimentation potentially prevented larval fish from entering the trap, lowering CPUEs. Suspended sediment may have also clogged net pores, which could reduce filtration rates and increase net avoidance by larval fishes (Isermann et al. 2002), ultimately reducing abundance estimates. Reducing sample or soak time for any of these gears may result in lower catch rates among samples, but these shorter times may allow for more locations within specific time periods to be sampled, potentially increasing precision.

Another consideration to improve catch rates of larval fishes would be to test other sampling gears not included in this study that are less likely to be impacted by sedimentation. For example, larger mesh ichthyoplankton nets (i.e., 1,000-μm or larger) could increase filtration capabilities and filter water for longer time intervals (i.e., >5minutes) providing more representative density estimates and reducing the number of zero catches and variability. Durable pumps can intake water and have the ability to limit the effects of suspended sediment on filtering efficiency compared to

towed nets (Mohlenberg 1987). Another possible sampling gear would be a larval seine with a greater mesh size that reduces the impacts of sediment and could potentially detect more individuals (Post et al. 1995). An understanding of how additional capture gears function within the Minnesota River and the sample sizes required should be determined prior to including them in a monitoring program.

Location and timing of sampling may have also impacted observed catch rates and genera detected across gears evaluated in our study. Sample transects of this study were placed every 200 m, no matter the habitat present. Complex habitats (e.g., log jams) and shallower, slower water velocity areas (e.g., inside bends of meanders) within the main channel serve as critical adult spawning habitat and larval fish refuge (Slipke et al. 2005) but were not targeted in this study. Incorporating complex habitats using a stratified random sampling protocol may provide a more representative picture of the larval fish assemblage in the main channel. In addition, bi-weekly sampling may have missed important pulses of drifting larval fishes. Spawning periods of many fishes can be as short as a few days (Neal et al. 2012), and

pulses of drifting larvae may last only three days (Tan et al. 2010). Increasing sampling frequency (e.g., weekly or continuous) and number of samples collected may improve catch rates and potentially reduce variability in catches.

MANAGEMENT IMPLICATIONS

Larval fish sampling protocols should be based on the goals of sampling. If the goal is to determine genera richness, we recommend a multiple gear approach that includes the combination of the slednet and light trap. If only a single gear is to be used, slednets had lower variability in abundance, detected a greater number of species, produced higher diversity indices, and required fewer samples to assess larval fish assemblages compared to light trap methods and is recommended. However, because of low and variable catch rates of evaluated gears, monitoring abundance may be difficult with any of these gears and underscores the challenges associated with sampling larval fish in large river systems. Thus, continued evaluation of additional larval fish capture gears, such as larger mesh nets, pumps, or larval seines, in a high-frequency, habitat-stratified sampling design that encompasses complex habitats (e.g., log jams, inside bend of meanders) in a river system should be considered.

ACKNOWLEDGMENTS

LITERATURE CITED

Auer, N. A. 1982. Identification of larval fishes of the Great Lakes basin with emphasis on the Lake Michigan drainage. Great Lakes Fishery Commission Special Publication 82-3. Ann Arbor, Michigan, USA.

Avery, E. L. 1996. Evaluations of sediment traps and artificial gravel riffles constructed to improve reproduction of trout in three Wisconsin streams. North American Journal of Fisheries Management 16:282–293.

Bonar, S. A., S. Contreras-Balderas, and A. C. Iles. 2009. An introduction to standardized sampling. Pages 1–12 *in* S.A. Bonar, W.A. Hubert, and D.W. Willis, editors. Standard Methods for Sampling North American Freshwater Fishes. American Fisheries Society, Bethesda, Maryland, USA.

Bosley, T. R., C. H. Pennington, M. E. Potter, and S. S. Knight. 1986. Evaluation of larval fish sampling gears for use on large rivers. Technical Report E-86-9, U.S. Army Engineer Waterways Experiment Station, Vicksburg, Mississippi, USA.

Braaten, P. J., D. B. Fuller, L. D. Holte, R. D. Lott, W. Viste, T. F. Brandt, and R. G. Legare. 2008. Drift dynamics of larval pallid sturgeon and shovelnose sturgeon in a natural side channel of the upper Missouri River, Montana. North American Journal of Fisheries Management 28:808–826.

Bulkowski, L., and J. W. Meade. 1983. Changes in phototaxis during early development of walleye. Transactions of the American Fisheries Society 112:445–447.

Cheshire, K. M., Q. Ye, B. M. Gillanders, and A. King. 2015. Annual variation in larval fish assemblages in a heavily regulated river during differing hydrological conditions. River Research and Applications 32:1207–1219.

Dembkowski, D. J., D. W. Willis, and M. R. Wuellner. 2012. Comparison of four types of sampling gears for estimating age-0 yellow perch density. Journal of Freshwater Ecology 27:587–598.

Fisher, S. J. 1999. Seasonal investigation of native fishes and their habitats in Missouri River and Yellowstone River backwaters. Dissertation. South Dakota State University, Brookings, USA.

Gale, W. F., and H. W. Mohr. 1978. Larval fish drift in a large river with a comparison of sampling methods. Transactions of the American Fisheries Society 107:46–55.

Gartz, R. G., L. W. Miller, R. W. Fujimara, and P. E. Smith. 1999. Measurement of larval striped bass (*Morone saxatilis*) net avoidance using evasion radius estimation to improve estimates of abundance and mortality. Journal of Plankton Research 21:561–580.

Harvey, B. C., J. L. White, and R. J. Nakamoto. 2002. Habitat relationships and larval drift of native and nonindigenous fishes in neighboring tributaries of coastal California River. Transactions of the American Fisheries Society 131:159–170.

Hayes, D. B., C. P. Ferreri, and W. W. Taylor. 2012. Active fish capture methods. Pages 267–304 *in* A. V. Zale, D. L. Parrish, and T. M. Sutton, editors. Fisheries Techniques. Third edition. American Fisheries Society, Bethesda, Maryland, USA.

Holland-Bartels, L. E., M. R. Dewey, and S. J. Zigler. 1995. Ichthyoplankton abundance and variance in a large river system concerns for long-term monitoring. Regulated Rivers: Research and Management 20:1–13.

Isermann, D. A., P. A. Hanchin, and D. W. Willis. 2002. Comparison of two mesh sizes for collecting larval yellow perch in surface trawls. North American Journal of Fisheries Management 22:585–589.

Johnson, H. O., S. C. Gupta, A. V. Vecchia, and F. Zvomuya. 2009. Assessment of water quality trends in the Minnesota River using non-parametric and parametric methods. Journal of Environmental Quality 38:1010–1030.

Kay, L. K., R. Wallus, and B. L. Yeager. 1994. Reproductive and early life history of fishes in the Ohio River drainage. Volume 2: Catostomidae. CRC Press, Boca Raton, Florida, USA.

Kelso, W. E., M. D. Kaller, and D. A. Rutherford. 2012. Collecting, processing, and identification of fish eggs and larvae and zooplankton. Pages 363-451 *in* A. V. Zale, D. L. Parrish, and T. M. Sutton, editors. Fisheries Techniques. Third edition. American Fisheries Society, Bethesda, Maryland, USA.

Killgore K. J., and J. A. Baker. 1996. Patterns of larval fish abundance in a bottomland hardwood wetland. Wetlands 16:288–295.

King, A. J., and D. A. Crook. 2002. Evaluation of a sweep net electrofishing method for the collection of small fish and shrimp in lotic freshwater environments. Hydrobiologia 472:223–233.

Kwak, T. J., and J. T. Peterson. 2007. Community indices, parameters and comparisons. Pages 677–763 *in* C. S. Guy and M. L. Brown, editors. Analysis and Interpretations of Freshwater Fisheries Data. American Fisheries Society, Bethesda, Maryland, USA.

Lindquist, D. C., and R. F. Shaw. 2005. Effects of current speed and turbidity on stationary light-trap catches of larval and juvenile fishes. Fishery Bulletin 103:438–444.

Magnan, P. 1991. Unrecognized behavior and sampling limitations can bias field data. Environmental Biology of Fishes 31:403–406.

Mapes, R. L., M. R. DuFour, J. J. Pritt, and C. M. Mayer. 2015. Larval fish assemblage recovery: a reflection of environmental change in a large degraded river. Restoration Ecology 23:85–93.

Marchetti, M. P., E. Esteban, M. Limm, and R. Kurth. 2004. Evaluating aspects of larval light trap bias and specific in the Northern Sacramento River System: Do size and color matter? Pages 269–279 *in* F. Feyrer, L. R. Brown, R. L. Brown, and J. J. Orsi, editors. Early Life History of Fishes in the San Francisco Estuary and Watershed. American Fisheries Society, Symposium 39, Bethesda Maryland, USA.

Messinger, T., and D. B. Chambers. 2001. Fish communities and their relation to environmental factors in the Kanawha River Basin, West Virginia, Virginia, and North Carolina, 1997–1998. Water-Resources Investigations Report 01-4048, Charleston, West Virginia, USA.

Minnesota Department of Natural Resources. 2013. Minnesota River Fisheries Management Plan 2013–2017. Minnesota Department of Natural Resources, St. Paul, Minnesota, USA.

Minnesota Pollution Control Agency. 2016. Environmental Data Access. <https://cf.pca.state.mn.us/water/watershedweb/wdip/search_more.cfm>. Accessed 14 May 2017.

Mohlenberg, F. 1987. A submersible net-pump for quantitative zooplankton sampling; comparison with conventional net sampling. Ophelia 27:101–110.

Mueller, K. P., and S. C. F. Neuhauss. 2010. Behavioral neurobiology: How larval fish orient towards the light. Current Biology 20:159–161.

Musser, K., S. Kudelka, and R. Moore. 2009. Minnesota River Basin Trends. Minnesota State University, Mankato, USA.

Nannini, M. A., J. Goodrich, J. M. Dettmers, D. A. Soluk, and D. H. Wahl. 2012. Larval and early juvenile fish dynamics in main channel and backwater lake habitats of the Illinois River ecosystem. Ecology of Freshwater Fish 21:499–509.

Naus, C. J., and S. R. Adams. 2016. Fish nursery habitat function of the main channel, floodplain tributaries and oxbow lakes of a medium-sized river. Ecology of Freshwater Fish 27:4–18.

Neal, J. W., C. M. Adelsberger, and S. E. Lochmann. 2012. A comparison of larval fish sampling methods for tropical streams. Marine and Coastal Fisheries: Dynamics, Management and Ecosystem Science 4:23–29.

Nelson, B. D. 2015. Hydrologic and temperature regime influence on growth and recruitment of fishes in an upper Midwest riverine ecosystem. Thesis. Minnesota State University, Mankato, Mankato, USA.

Nemeth, R. S. 2005. Population characteristics of a recovering U.S. Virgin Islands red hind spawning aggregation following protection. Marine Ecology Progress Series 286:81–97.

Nickel, A. 2014. Investigation of the connectivity relationship with abiotic condition and community dynamics in Minnesota River backwater lakes. Thesis. Minnesota State University, Mankato, Mankato, USA.

Niles, J. M., and K. J. Hartman. 2007. Comparison of three larval fish gears to sample shallow water sites on a navigable river. North American Journal of Fisheries Management 27:1126–1138.

Oksanen, J., F. Guillaue Blanchet, M. Friendly, R. Kindt, P. Legendre, D. McGlinn, P. R. Minchin, R. B. O'Hara, G. L. Simpson, P. Solymos, M. Henry, H. Stevens, E. Szoecs, and H. Wagner. 2019. Vegan: Community Ecology Package. R package version 2.5-5.

Post, J. R., L. G. Rudstam, and D. M. Schael. 1995. Temporal and spatial distribution of pelagic Age-0 fish in Lake Mendota, Wisconsin. Transactions of the American Fisheries Society 124:84–93.

Pritt, J. J., E. F. Roseman, J. E. Ross, and R. L. DeBruyne. 2015. Using larval fish community structure to guide long-term monitoring of fish spawning activity. North American Journal of Fisheries Management 35:241–252.

Pulg, U., B. T. Barlaup, K. Sternecker, L. Trepl, and G. Unfer. 2013. Restoration of spawning habitats of brown trout (*Salmo trutta*) in a regulated chalk stream. River Restoration and Applications 29:172–182.

R Development Core Team. 2014. Version 3.3.3. R: a language and environment for statistical computing. R Foundation for Statistical Computing, Vienna, Austria.

Rider, S. J., and F. J. Margraf. 1997. Dynamics of ichthyoplankton in the Kanawha River, West Virginia. Journal of Freshwater Ecology 12:239–251.

Schoener, T. W. 1970. Nonsynchronous spatial overlap of lizards in patch habitats. Ecology 51:408–418.

Simon, T. P., and R. Wallus. 2005. Reproductive biology and early life history of fishes in the Ohio River Drainage. Volume 4: Percidae—Perch, Pikeperch, and Darters. CRC Press, Boca Raton, Florida, USA.

Slipke, J. W., S. M. Sammons, and M. J. Maceina. 2005. Importance of the connectivity of backwater areas for fish production in Demopolis Reservoir, Alabama. Journal of Freshwater Ecology 20:479–485.

Snedecor, G. W., and W. G. Cochran. 1989. Statistical methods. Iowa State University Press, Ames, USA.

Strahler, A. N. 1957. Quantitative analysis of watershed geomorphology. Transactions of the American Geophysical Union 38:913–920.

Tan, X., X. Li, S. Lek, Y. Li, C. Wang, J. Li, and J. Luo. 2010. Annual dynamics of the abundance of fish larvae and its relationship with hydrological variation in the Pearl River. Environmental Biology of Fishes 88:217–225.

Thoma, D. P., S. C. Gupta, J. S. Strock, and J. F. Moncrief. 2005. Tillage and nutrient source impacts on water quality and corn grain yield from a flat landscape. Journal of Environmental Quality 34:1102–1111.

Waters, T. F. 1977. The streams and rivers of Minnesota. University of Minnesota Press, Minneapolis, USA.

Zar, J. H. 1996. Biostatistical Analysis. Third edition. Prentice Hall. Upper Saddle River, New Jersey, USA.

Zhang J. 2016. spaa: SPecies Association Analysis. R package version 0.2.2.

NOTES

WOOD FROGS (*RANA SYLVATICA*) IN SOUTHWESTERN ROBERTS COUNTY AND WESTERN GRANT COUNTY, SOUTH DAKOTA — Historically, only six records of Wood Frogs (*Rana sylvatica*) existed from South Dakota, all represented by museum specimens. A single specimen was collected from Hartford Beach on Big Stone Lake, Roberts County in 1922 (National Museum of Natural History, Smithsonian Institution [USNM] 65452) and five Wood Frogs were collected near the outlet of Blue Dog Lake, Day County in 1929 (USNM 312618–312622; Fig. 1). After the 1920s, no other Wood Frogs were reported in South Dakota for the next six decades, and the species was presumed extirpated. Over (1923, 1943) reported the species was common in the wooded coulees located along the escarpment of the Prairie Coteau in western Roberts County, South Dakota. Later, Fishbeck and Underhill (1960) reported that Wood Frogs no longer occurred in these areas and surmised that droughts and over-harvesting of timber in the coulees had led to the species' extirpation. Unfortunately, no author was specific about the locations or names of the coulees. However in 1997, Wood Frog calls were recorded at two sites in northeastern Roberts County (Fisher 1998, Naugle et al. 2005), the first record of the species in South Dakota since 1929. Since 1997, numerous individuals have continued to document Wood Frogs in northeastern Roberts County (Backlund 2005; James Ford Bell Museum of Natural History, University of Minnesota [JFBM] 14426; Biodiversity Collections, University of Texas at Austin [TNHC] 108910–108914, 108916–108918).

It was not until 2016 that Wood Frogs were detected outside of northeastern Roberts County (Fig. 1). On 3 May 2016, a single juvenile Wood Frog was collected along Owens Creek, Ortley Game Production Area, southwestern Roberts County, South Dakota (45.34936°N, 97.20640°W; WGS 84; TNHC 108915). On 18 April 2019, co-author Dennis R. Skadsen (DRS) was notified that several male Wood Frogs were calling at 2330 h CST at Ortley Game Production Area. DRS visited the site on 23 April 2019 and recorded Wood Frogs calling from 2030 to 2130 h CST. Wood Frogs were in a large, rich fen wetland (45.34695°N, 97.21637°W; WGS 84) ca. 1.2 km west of 450th Ave, which runs parallel to the east boundary of Ortley Game Production Area. Prior to this, DRS had visited this site on several occasions from late March to April during 2017 and 2018 to listen for calling males during their breeding season, but no Wood Frogs were heard or observed. This population of Wood Frogs is ca. 48 km southwest of the Wood Frog records in northeastern Roberts County and 10 km east of historic specimens collected at Blue Dog Lake, Day County. Until these recent records, there had been no other reports of Wood Frogs from the Prairie Coteau or southwestern Roberts County.

Additional records of Wood Frogs from the Prairie Coteau were collected in 2019. On 9 August 2019, a Wood Frog was collected near Pearson Slough in southwestern Roberts County (45.30346°N, 97.16389°W; WGS 84; TNHC 114350). On 26 April 2019, Wood Frogs were heard calling from a wetland near the headwaters of the Big Sioux River in northwestern Grant County, South Dakota (45.28489°N, 97.16194°W; WGS 84), and on 2 May 2019, Wood Frogs were recorded calling near Meyer-Janssen Waterfowl Production Area, Grant County (45.17600°N, 96.92900°W; WGS 84; HerpMapper.org [HM] 283333 and TNHC 114348). These two records represent a range expansion farther south in South Dakota and are the first records of Wood Frogs in Grant County, South Dakota (Ballinger et al. 2000).

It remains unclear why Wood Frogs have only been recently detected in southwestern Roberts County and Grant County. Given the proximity to historic records from Blue Dog Lake, small numbers of Wood Frogs might have been present in southwestern Roberts County since the 1920s with an increased abundance during the last few years. Drainages and tributaries in this region, such as Owens Creek, may have facilitated movement of Wood Frogs throughout the area. Despite numerous amphibian and reptile surveys at wetland and coulee habitats across Roberts County, no Wood Frog populations have been detected between these two broadly separated regions. Further, given the proximity to recent records, surveys for Wood Frogs should target suitable habitat in Codington and Day counties in an attempt to better understand the distribution of this species in South Dakota.

—*Dennis R. Skadsen, Northeast Glacial Lakes Watershed Project, Day County Conservation District, Webster, South Dakota 57274, USA; Drew R. Davis, School of Earth, Environmental, and*
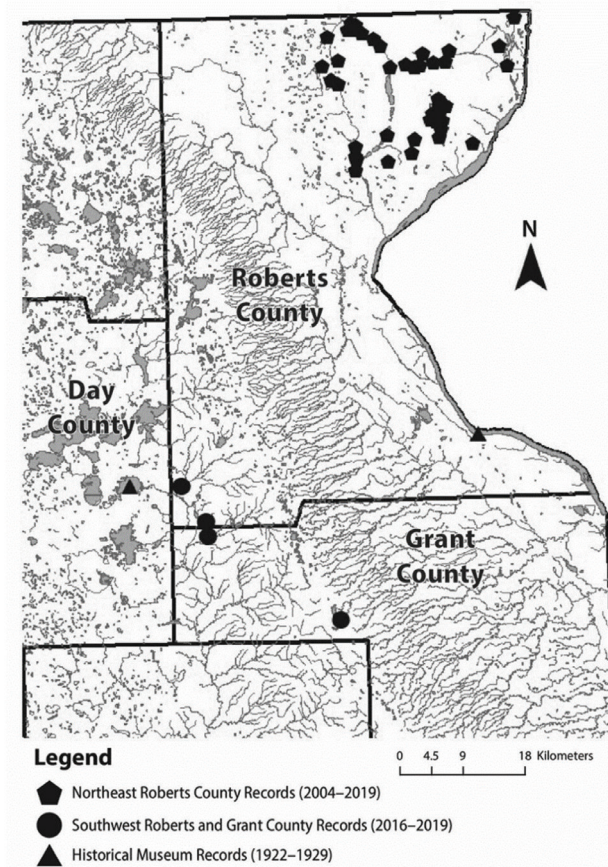
Figure 1. Current and historic Wood Frog (*Rana sylvatica*) reports and voucher records in northeastern South Dakota. The majority of recent records are restricted to northeastern Roberts County; however, recent specimens have been detected in southwestern Roberts County and western Grant County (2016–2019).

Marine Sciences, University of Texas Rio Grande Valley, South Padre Island, Texas 78597, USA; Corresponding author (dennis.skadsen@sd.nacdnet.net).

LITERATURE CITED

Backlund, D. 2005. South Dakota statewide herpetology survey 2004. Final report to South Dakota, Game, Fish, and Parks. 68 pp.

Ballinger, R. E., J. W. Meeker, and M. Thies. 2000. A checklist and distribution maps of the amphibians and reptiles of South Dakota. Transactions of the Nebraska Academy of Sciences 26:29–46.

Fischer, T. D. 1998. Anura of eastern South Dakota: their distribution and characteristics of their wetland habitats, 1997–1998. Thesis. South Dakota State University, Brookings, USA.

Fishbeck, D. W., and J. C. Underhill. 1960. Amphibians of eastern South Dakota. Herpetologica 16:131–136.

Naugle, D. E., T. D. Fischer, K. F. Higgins, and D. C. Backlund. 2005. Distribution of South Dakota anurans. Pages 283–291 *in* M. J. Lannoo, editor. Amphibian Declines : the Conservation Status of United States Species. University of California Press, Berkeley, California, USA.

Over, W. H. 1923. Amphibians and reptiles of South Dakota. South Dakota Geological and Natural History Survey Series 23, Bulletin 12:1–34, 18 plates.

Over, W. H. 1943. Amphibians and reptiles of South Dakota. University of South Dakota Natural History Studies 6:1–31.

The Great Plains Natural Science Society

The Great Plains Natural Science Society, formerly the North Dakota Natural Science Society, was founded in 1967 and seeks to promote interest in and understanding of natural history in the Great Plains, to encourage the conservation of natural resources, and to provide communication among individuals, institutions, and organizations of like interests. The GPNSS actively promotes the study of natural history of the Great Plains region, including geology, plants, birds, mammals, fish, insects, and other forms of life. Together with local, state, and national conservation organizations, the GPNSS fosters natural resource conservation and preservation of outstanding natural areas. The GPNSS publishes *The Prairie Naturalist*, a widely read, peer-reviewed journal which deals with the natural history and environment of the Great Plains region.

The GPNSS is currently composed of a diverse membership, many of whom are professional scientist. Society members have strong interests in sustainable management of Great Plains natural resources and their habitats. Natural history and ecology of the Great Plains is the primary focus and interest of the GPNSS, thus, the primary interests and goals of the Society seeks to promote increased scientific knowledge of the interactions of all Great Plains organisms with their natural environments, enhance professional stewardship of Great Plains natural resources and their habitats, and encourage use of applied research for informing Great Plains natural resource policy decisions.

First published in 1969 by the University of North Dakota, *The Prairie Naturalist* has been published by Minuteman Press since 2013 and fills an important role as the avenue of communication of research on the North American grasslands and their biota. Research topics include articles investigating Great Plains community and landscape ecologies, species-specific population dynamics, mammalogy, ornithology, invertebrate zoology, herpetology, ichthyology, botany, animal behavior, infectious diseases, biostatistics, and records of occurrence. This journal offers timely technical information for researchers, educators, students, and the interested public. Published twice a year, *The Prairie Naturalist* reaches subscribers throughout the United States and Canada, as well as libraries in Europe and Asia. A portion of each volume is devoted to shorter and less comprehensive communications (notes) and book reviews. Manuscripts containing original material not submitted elsewhere are considered for publication; all are reviewed by specialists in relevant fields.

Past issues of The Prairie Naturalist are available through
the DigitalCommons@University of Nebraska-Lincoln at https://digitalcommons.unl.edu/tpn.

Cover Photograph by
Lynda Lafond