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Herpetological Collections at Pittsburg State University: Assessing Collecting Patterns and Analysis of Spatial and Temporal Origin of Specimens.

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HERPETOLOGICAL COLLECTIONS AT PITTSBURG STATE UNIVERSITY: ASSESSING COLLECTING PATTERNS AND ANALYSIS OF SPATIAL AND TEMPORAL ORIGIN OF SPECIMENS.

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An Abstract of the Thesis by
Natalia Agostini Schneider

Natural History Collections are a rich source of biological data. Each specimen contains data for that species' presence for a specific location and time, providing researchers with essential biological information. Importantly, this information can be preserved and re-evaluated for hundreds of years. To maintain specimens through time, good curation protocols are essential. The Herpetology Collection (henceforth HC) at Pittsburg State University houses 1,631 specimens, representing 181 species and subspecie collected from 23 U.S. states, Mexico and Manitoba, Canada. The majority of specimens (78.6%) were collected from the four-state area (Kansas, Missouri, Arkansas and Oklahoma). Specimens collected exclusively in Kansas comprise 56.6% of the HC, and were collected in 37 counties, with major collecting effort on Crawford, Cherokee and Bourbon counties. Spatial analysis revealed many unique and unduplicated spatiotemporal records confirming the importance of the PSU herpetology collection as a local repository and source of herpetofaunal data. The temporal analysis showed continual collecting from 1961 to 1970 and from 1981 to 2002 during the months of March through June. Collecting peaks occurred by month in April, by year in 1964, and by decade during the 1960s. The curatorial work this project did on the long-neglected collections was crucial to reverse degradation, it demonstrated that specimens can be curated to 21st century standards with appropriate efforts. Out of 1,631 specimens, 147 were lost during past physical moves; out of 1,484 specimens left, 221 were rehydrated (14.8%); 757 required change of preservative fluids (51.0%); and 457 specimens (30.8%) did not require further curation other than new jars. Data limitations often were present due to somewhat incomplete descriptions of locality and habitat. Collecting biases in or nearby urban areas, along roads, and in areas of known higher biodiversity levels were identified for Kansas specimens.

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CHAPTER I

INTRODUCTION

Natural History Collections

Natural History Collections (NHCs henceforth) comprise items from all fields of biology and geology, including specimens of contemporary fauna and flora, genetic material, microscope slides, and paleontological material. NHCs also include documents such as field collection notebooks, which often are of historical value and interest. For example, the Smithsonian Institution, the Missouri Botanical Garden, and many other large research institutions routinely archive the collecting books of more productive researchers because of their historical value and marginalia, often which include important information when viewed historically.

Approximately 3 billion, and possibly 5 billion (Funk, 2017) biological specimens are preserved in natural history collections worldwide, which collectively provide a critical and irreplaceable window into global biodiversity (Shaffer *et al.*, 1998; Pyke and Ehrlich, 2009; Smith and Blagoderov, 2012). NHCs are a rich source of biological information and contain the spatiotemporal information provided by each specimen collected. Equally if not more importantly, they contain data related to biogeography, habitat variables, and provide the irreplaceable baseline data that underpins all of taxonomy and systematics (NatSCA, 2005; Drew, 2011; Lister *et al.*, 2011). According to Drew (2011), NHCs were also influential in the development of bioinformatics and online databases.

The value and importance of NHCs are increasing due to the decline in numbers and ranges of some species, resulting from habitat destruction, climate change, pollution, wildlife and plant trade, mortality on roadways, other anthropogenic disturbances, and emerging pathogens such as the chytrid that is causing chytridiomycosis in amphibians

and the snake fungal disease (Pough *et al.*, 2004; NatSCA, 2005; Lister *et al.*, 2011; Lavoie, 2013; Dirzo, 2014; Lujan and Page, 2015; McCallum, 2015).

History and Origins of Natural History Collections

Natural history collections have a long and rich history. They had originated by the 16th Century, when the upper socioeconomic classes in Europe sometimes had exotic specimens set aside in a designated room called a Cabinet of Curiosities. Owners typically invited members of higher social classes to walk through the Cabinet of Curiosities and discuss the origin and history behind each item (Appendix A) (Imperato, 1599; Spary, 2000; Impey and MacGregor, 2001; Simmons, 2015; Cribb, 2017; Friis, 2017). Nature enthusiasts, including clergy involved with "Natural Theology", explorers and researchers have been collecting specimens systematically for at least 500 years, which has necessitated creating and discovering new ways to preserve and exhibit specimens (MacGregor, 2007).

One of the earliest models for the modern museums dates back to the 3rd Century BCE (Before Current Era), which was established by Ptolemy I Soter in Alexandria and called the *Mouseion*. According to Simmons (2015), the *Mouseion* gathered scholars such as Archimedes and Erasistratus around a botanical garden, zoo, library, collections of specimens and other artifacts. The inspiration for displaying and presenting specimens came from a range of models that date back to the 1st Century, two of the most influential of whom were Pliny the Elder, author of *Historia Naturalis*, and Pedanius Dioscorides, author of *De Materia Medica* (MacGregor, 2007).

Cabinets of Curiosities were present in Italy by 1500 and flourished in Europe during the 16th Century (Impey and MacGregor, 2001; MacGregor, 2007). They were described by Francis Bacon as a sample of nature made private (MacGregor, 2007), as rooms were filled with specimens, ranging from plants and animal parts (including abnormal human parts) to illustrations and books that pictured the natural history of the most exotic locations and organisms (Impey and MacGregor, 2001; Simmons, 2015). Between 1556 and 1560 approximately 970 collections in Europe were distributed among noblemen, scholars and private citizens such as Albrecht V Duke of Bavaria, in Munich; Ulisse Aldrovandi, a professor of botany and natural history in Bologna; and Ferrante

Imperato, in Naples, engraved the first record of a Cabinet of Curiosity (Appendix A). In 1712 Sir Hans Sloane of England purchased the manor of Chelsea, which included the natural history collections of many important naturalists and explorers, such as those of Engelbert Kaempfer's from Japan, William Dampier's from Australia, and at least ten other notable collectors (MacGregor, 2007; Cribb 2017).

During the 16th and 17th centuries the shape of collections took a turn. The central concepts of diversity and curiosity were replaced by one that emphasized organization and classification of specimens. MacGragor (2007) cited Major (1674), who described the new organization of cabinets in the form of *naturalia* and *antiquaria*. *Naturalia* comprised mathematical instruments, applied arts, guns and armor, and biological specimens, whereas *antiquaria* comprised sculptures, antiques, coins and books. The then-new forms of organization became the foundation of most natural history museums as they exist today.

One of the first state-sponsored natural history museums was the Museum national d'Histoire naturelle, in Paris. Established in 1635 and made public some years later, it was the most complete and distinguished facility, housing collections from much of the world (Spary, 2000; MacGregor, 2007; MNHN, 2017). Another early museum, The Kunstkamera, was the first museum established in Russia in 1719 by Czar Peter the Great, which contained a variety of items, from biological specimens to art pieces (Impey and MacGregor, 2001; Pyke and Ehrlich, 2009). In the United Kingdom, the Natural History Museum in Kensington, London, was established in 1756. Initially and still known by many as the "British Museum" (now formally the Natural History Museum), it is considered that country's first institution dedicated exclusively to biological collections (Pyke and Ehrlich, 2009).

In South America, the first museum was established by the Dutch, Johan Mauritus of Nassau, Governor-General of Brazil (Scheurleer, 1985). Nassau established the Palace of Vrijburg, between 1639 and 1642, in the city known today as Recife (De Almeida *et al.*, 2011). Inspired by the European models, the Palace held a zoo and a botanical garden along with a collection of *naturalia* from South America including paintings portraiting Brazilian landscapes, native tribes, zoological and botanical species (Gaspar, 2009; Scheurleer, 1985). For the first-time paintings and academic papers were published from

Brazil, including *Historia Naturalis Brasiliae* by Georg Marcgrave (1648) and *De Medicina Brasiliensi* by Willem Piso (1648), sharing with Europe the biological, medical and cultural aspects of the New World (Gaspar, 2009; De Almeida *et al.*, 2011; UOL, 2016). Sadly, the majority of Nassau's collections were lost after he left Brazil in 1644 (MTH, 2006; Dobbin, 2009). During the 1800's, John VI of Portugal, Emperor of Brazil, established the Rio de Janeiro Botanical Garden (1808) and the National Museum of Brazil (1818) (Zaher and Young, 2003; SiBBr, 2016).

Zoological specimens in museums initially were displayed not in an artistic manner, but rather scientifically in standard poses, mounted on simple wooden bases with a plain background, to facilitate comparison and classification. The first and most extravagant museum to display artistic taxidermies of zoological specimens in natural history cases was the Bullock's Museum in England, established in 1809 by William Bullock. The museum astonished visitors by its several rooms that displayed unique habitats, animal behavior and food habits, and high-quality taxidermies organized in a manner both scientifically and visually appealing (MacGregor, 2007). Thereafter, many museums and herbaria were established in western countries in the 18^{th} and 19^{th} centuries, promoting a new field in science known as Natural History.

Natural History, which among many carries a negative connation of only half-competent nature-enthusiasts, at the time referred to those who were considered experts in local flora and fauna (Winsor, 2009), before subdisciplines such as "botany" and "zoology" had solidified as areas of specialization. Natural history as a field of inquiry aroused the interest of many scholars and led many to reorganize and classify specimens preserved in collections. Sir Hans Sloane (1660 – 1753) had the largest collection in England, all meticulously catalogued (Cribb, 2017). The Swedish botanist Carl Linnaeus was among its prominent visitors, although the exact number of times he visited is unknown. Linnaeus criticized Sloane's method of classification, which used the polynomial system of nomenclature. Some years later Linnaeus published *Systema Naturae*, which consisted of binomial system of nomenclature. Moreover, between the 16^{th} and 17^{th} century, the Italian naturalist Ulisse Aldrovandi became an important influence after reorganizing mammal classifications, and several collectors such as Ole Worm, Ferdinando Cospi and Nehemiah Grew followed his system (Impey and

MacGregor, 1985). Aldrovandi regrouped mammals into solid-hoofed, cloven-hoofed and clawed, and included whales in the mammalian group. These curiosity collectors played an important role in systematics and taxonomy. Besides collecting for their own pleasure to enhance their collections, they also made use of scientific methods to classify and organize their collections, providing information for those who were studying the natural world.

Preservation of Collections and Specimens

The proper preservation of specimens is an important component of NHC maintenance. The age of specimens (from tens to hundreds of years), may compromise their quality, for reasons such as: inadequate storage; lack of past maintenance; poor or improper fluid for preservation; lack of basic curation (unprepared [backlogged] and unidentified specimens); misidentifications; and application of inadequate curation techniques (Snow, 2005; Simmons, 2014, 2015). Specimen quality is often compromised because of limitations in data, such as errors, incomplete information, and missing data (Pyke and Ehrlich, 2009; Newbold 2010). Proper curation can overcome many of these limitations, thereby increasing the scientific value and usefulness of NHCs.

Proper preservation also is necessary because each lost specimen contains irreplaceable biological and geographical data. For example, the gene pool and selective forces acting on a a fish species collected in the Missouri River in 1890 are not identical to those 127 years later in 2017. Apart from long-lived, geographically proximate individuals from which repeated sampling is possible (such as woody plants), we cannot re-collect an individual specimen. However, when maintained properly the value of a museum specimen continues indefinitely and sometimes even increases.

During the 18th Century, apart from organizational considerations, those involved with the care of collections faced the even bigger challenge of devising tecnhines to preserve specimens properly for the years to come. The loss of biological specimens, especially zoological specimens, was extremely common. Some of the challenges faced by collectors and curators were the putrification of specimens in the field, or of prepared specimens that were skinned improperly, or of taxidermied mounts damaged by insect pests (MacGregor, 2007).

Due to these kinds of problems, authors as early as 1700s published collecting procedures and best practices for preservation and pest control in an attempt to have better quality specimens (MacGregor, 2007; Simmons, 2015). During the mid-17th Century it was discovered that specimens could be preserved in alcohol, more specifically in spirits of wine or alcohol. At that time, rum and brandy were popular choices and said to be stronger preservatives (MacGregor, 2007; Simmons, 2014; Simmons, 2015). According to Simmons (2015), the first specimens preserved in alcohol were presented to the Royal Society of London, by William Croone in June of 1662. The specimens were two dog embryos preserved in spirits of wine. MacGregor (2007) and Simmons (2014) report that in 1664 Robert Boyle also presented a linnet bird and a snake preserved in wine. Simmons (2014) noted records of a human fetus preserved in alcohol from 1695 by T. Coxe and of insects preserved in 1670 by Jan Swammerdam.

With the discovery of spirits as an effective preservative, wet collections became increasingly common between the mid-17th and 18th centuries, which coincided with the development of standardized techniques for long-term preservation of specimens (Simmons, 2015).

Wet collections after the 17th Century increased rapidly worldwide. During the development of standardized techniques of fluid preservation, ethyl alcohol was commonly mixed with additives such as glycerine, first used in 1883; formaldehyde, in 1893; and isopropyl alcohol, in 1928 (Simmons, 2014). Until the 19th century many collectors also made use of pure glycerine, buffered 10% formalin, and 40 to 70% isopropyl alcohol to preserve specimens, among many other fluids. Before the discovery of formaldehyde, in 1858, specimens were fixed in ethyl alcohol. Formaldehyde fixation may deteriorate specimen DNA, increase specimen discoloration, cause swelling, demineralization of bones and tissue, among other issues, therefore many curators advocate for the reduction of formalin use (Simmons, 2014). The effects of preservative fluids and fixative agents over the long-term preservation of specimen has not been comprehensively studied (Simmons, 2014), however, one of the many 21st century best practices recommendations are the use of 70% ethanol for long-term preservation and fixation.

To improve the usefulness of biological collections, institutions needed to devise new ways to maintain preserved specimens. By the early 1990s (and in some places a decade earlier), many larger institutions began to enter data corresponding to each specimen into data bases. Initially, data bases were used to track specimens internally (Snow, 2005), but soon thereafter, or in some cases earlier (e.g., TROPICOS® at Missouri Botanical Garden), made the data freely available online.

Digitization is a broad term that includes the process of transcribing physical data from specimen labels, hand-written record books, field notebooks, documents, and ultimately the specimens themselves, into a digital database or archive online (Nelson *et al.*, 2012). Commencing on a large scale in the early 2000s, museums and herbaria began to digitally image type specimens. One important reason for focusing on type specimens initially was to reduce the wear and tear that accumulates inevitably when speicmens are loaned between institutions. Shortly thereafter, many began to digitally image non-type specimens as well (usually by project-driven priorities), and in many cases to retroactively georeferenced collections. Since then digitization has been promoted by many organizations (e.g. SPNHC, iDigBio) as it facilitates access to data worldwide reducing costs and time for researchers that would need to travel to several institutions to have access to collections data. It also increases the long-term conservation of specimens by avoiding sending specimens from one institution to the next. While digital images cannot replace a physical specimen, they can aid researchers during preliminary specimen examination, species verification, and search for specific characterists eliminating possible unnecessary travels.

The ability to actively curate and digitize, and the pace at which they occur, typically varies according to the size of the institution. Smaller institutions generally have limited (if any) funding or staff devoted to curation. However, if all biological data are to be placed online to help assemble the world's biodiversity "jigsaw puzzle", as described and recommended by NatSCA (2005), then all specimens ultimately must be curated to high standards. Snow (2005), MacDonald and Ashby (2011), and Schnalke (2011) emphasized the benefits that small and historical biological collections bring to the faculty, students, and the universities themselves. Smaller facilities, if actively curated, result in enhanced research possibilities, education, outreach, student training,

partnership with government agencies and other institutions, digitization and promoting the institution among the scientific community. Snow (2005) and Casas-Marce *et al.* (2012) added that small collections generally will have the best inventory of the local fauna and flora, and that smaller institutions often have important historical collections not duplicated in larger facilities.

Collection-Based Research Opportunities

Natural History Collections are commonly undervalued by the general public (Suarez and Tsutsui, 2004), but some scientists also fail to recognize their potential. Even some biologists believe that collections are of little value beyond their use as a tool for teaching classes. That perspective certainly was not uncommon in many smaller, primarily undergraduate institutions in the United States before the new millenium (N. Snow, pers. comm. 2015), but with the advent of initiatives such as the National Science Foundation's iDigBio, perceptions of the value of herbaria and museums have become more favorable. Besides playing important and crucial roles in the education of organismal biologists and land use managers, collections underlie many aspects of research, outreach and even public health, including:

- The treatment and spread of diseases, which can only be understood by the distribution and abundance of their vectors (Suarez and Tsutsui, 2004; IWGSC, 2009) and via genetic diversity baselines prior to the introduction of pathogens (Burrell *et al.,* 2014; Drew, 2011);
- Environmental contamination (e.g., mercury poisoning) (Suarez and Tsutsui, 2004; IWGSC, 2009).
- Extraction of genetic material from dried or wet collections for ecological, systematics and evolutionary research (Payne and Sorenson, 2003; Schander and Halanych, 2003; Zimmermann *et al.,* 2008; Casas-Marce *et al.,* 2009; Drew, 2011; Burrell *et al.,* 2014; Yong, 2016).
- The study of medicinal plants using herbarium specimens and in teaching of medical botany (Hedberg, 1993; Senchina, 2006; Eisenman *et al.,* 2012; Culley, 2013); and preservation of wild sources of genetic material to augment crop production (IWGSC, 2009);

- The origin and spread of agricultural pests, diseases, and weeds, including their rate of spread (Suarez and Tsutsui, 2004; Winker, 2004; NatSCA, 2005; IWGSC, 2009);
- Forensic science usage of collections to compare to samples collected at crime scenes (NatSCA, 2005; Zimmermann *et al.*, 2008);
- Archageological uses by ethnologists and decorative art curators to compare parts of artifacts (Burrell *et al.*, 2014; NatSCA, 2005).
- To guide scientific illustrations in popular field guides (NatSCA, 2005).

Shaffer *et al*. (1998), Pyke and Ehrlich (2009), Newbold (2010), Drew (2011), Lister *et al.* (2011), Casas-Marce *et al.* (2012), and Lavoie (2013) provided examples of collection-based research despite the limitations inherent with some collections. For example, data can be generated for studies involving: species richness and abundance; the production of current distribution maps (including community composition on a finer scale); historical species' ranges; changes in species' ranges; evolutionary changes (e.g., changes in flowering cycles or mating seasons); species distribution modeling; and species and/or areas in need of conservation.

Within the molecular field, newer technologies have emerged that have further enhanced the value of specimens, such as DNA barcoding (e.g., Peterson *et al.*, 2014). Older technologies have been improved (e.g., PCR, high-throughput DNA sequencing) (Burrell *et al.*, 2014; Zimmermann *et al.*, 2008), and an increasing number of DNA extraction protocols published, resulting in the development and use of an immense DNA library by a growing number of scientists (Payne and Sorenson, 2003; Schander and Halanych, 2003; Hebert *et al.*, 2004).

Most recently, new research possibilities have emerged through the digitization of biological collections for "STEM" areas (science, technology, engineering, and mathematics). During the process of digitization, it not uncommon for professionals from several areas to be working on the same project, since knowledge of informatics, photography and georeferencing is required. By data basing and digitizing collections, institutions make available a larger percentage of its collections. The additional data online facilitates the asking of new research questions, but equally important brings the

institution itself to the world by providing its data to the scientific community via the internet (Snow, 2005; Smith and Blagoderov, 2012).

Limitations of Collections Data

Newbold (2010) and Pyke and Ehrlich (2009) highlighted the importance of understanding the limitations of specimen-related data. Newbold (2010) discussed in particular spatial bias, which generally reflects concentrated sampling in places with easy access, such as roads, rivers, coastlines, cities, a collector's house or property; and in areas of greater interest such as those with rich biodiversity (protected areas, preserves, and biodiversity hotspots). Another important component of spatial bias is that collections reflect merely the presence of species, but do not register its presence in areas that were sampled but where the species was not observed; in other words, a false absence.

Bias in environmental coverage, or of a specified area such as protected area, can have spatial and temporal components. For example, thorough sampling should include all habitats within an area for their species, and should include extended collecting across all elevations and seasons. Thorough sampling often is unachievable given practical or cost-related factors. Temporal bias is often reflected in collecting peaks during particular years or seasons. The peaks may reflect the highest seasonal activity, presence, or visibility of a taxonomic group, as well as the collector's interests and available time. As an example, in temperate areas herpetological specimens are mainly collected from early Spring to late Summer, a period of time that most species are active due to warmer temperatures and breeding activity. Another temporal bias in nearly all university collections is the time of year (Fall, Spring, Summer) that college courses are taught (e.g., herpetology, plant taxonomy, ichthyology, etc.).

Another form of bias is taxonomic, which often reflects groups that are more charismatic (e.g., ferns, orchids, bromeliads, snakes), easiest to detect and/or capture (e.g., vertebrates, insects and plants), and which leads to an underrepresentation of many taxonomic groups in collections. To cite one example, probably less than one percent of the many hundreds of herbaria worldwide have strong, or approximately equal, representation across vascular plants, lichens, fungi, bryophytes and algae, even though

all typically are curated by herbaria. Taxonomic biases also can reflect groups that are difficult to physically handle and process, as for example in thistles and cacti, each with sharp prickles or spines, or palms, a single specimen of which may take up to two hours to properly collect and press, given its many (often large) component parts. Amongst zoological examples are large vertebrates such as ungulates, cartilaginous fish, or large reptiles (i.e. pythons, alligators), leading to small number of such specimens, or the intentional and biased collecting of juvenilles due to their smaller size. This is particularly true in tropical areas where at least three or four duplicate specimens are collected.

Pyke and Ehrlich (2009) additionally discussed phenotypic bias, which relates to the appearance of specimens, in which collectors sometimes are drawn to collecting individuals of particular size, age, sex, and common or abnormal appearance. Such bias is well represented in herbaria in at least three ways: 1) where showy plants (e.g., *Ipomopsis aggregata*, *Phlox divaricata*) are collected in abundance, whereas smaller, less easily seen plants are often overlooked (e.g., *Floerkea proserpenidoides*); 2) where specimens in fruit are typically bypassed in favor of those in flower (even though some species require fruits to be identified with confidence); and 3) the gross under-representation in most herbaria of submerged or emergent aquatic species. In herpetological collections, males and younger specimens are collected more often, likely due to the fact that males tend to move more frequently than females in search of new territory during the breeding season, and juveniles are easier to capture, preserve and require lest storage space.

Boakes *et al.* (2010) analyzed spatial and temporal bias in species occurrence data from museum data, literature, distributional data, ornithological atlases and website reports from citizen scientists, and concluded that museum data are the most comprehensive historical record of biodiversity, even though museum data also have spatial and temporal biases. Boakes *et al.* (2010) also discussed the critical need for understanding limitations and biases in museum data.

Decreases in Collecting

Collecting rates vary through time. For example, a dramatic increase of research in tropical biology in the Americas followed the founding of the Organization for Tropical Studies in 1963 in many areas south of the United States, where it continues unabated in many countries. The increases were aided by long-term commitments and institutional presences of institutions such as The Field Museum (Chicago), Duke University, the New York Botanical Garden, the Missouri Botanical Garden, and other programs. Likewise, North-South collaborations between museums and herbaria in Europe and Asia have increased collecting rates in some parts of Africa, southern and eastern Asia, and Malesia (e.g., Friis and Balslev, 2017).

However, in North America the rates of collecting have decreased substantially at many museums and herbaria, including those with large collections. Prather *et al.* (2004) analyzed data from 71 herbaria in the United States to evaluate whether collecting rates, as measured by decades, are decreasing. They found that the temporal pattern of collecting decreased substantially between 1980 and 2000, compared (in particular) to the 1950s through 1970s, although an earlier peak had also occurred in the 1930s (Prather *et al.*, 2004).

For the herpetofauna of Kansas, Taggart *et al.* (2006) evaluated the temporal and spatial collecting patterns by gathering data from 36 North American institutions that housed specimens collected in Kansas. These authors concluded that the overall coverage of collecting in Kansas has provided the state with a better understanding of its herpetofaunal distribution than similar sized geographic regions in most parts of the world (Taggart *et al.*, 2006). This was also true for the temporal coverage, which was continuous from 1857 to 2005, with three main collecting peaks from the early 1920's to mid-1930's, early 1960's to beginning of 1980's, and the last peak occurring in 2004.

Curation and Research within Small Collections

Despite the immense value of museum specimens and their increased visibility in the current digital age, comparatively little research is occurring in smaller collections, including those transitioning towards 21st Century curatorial standards. In general, the better the facility has been curated historically (often over many decades or in some cases

over a century or more), the more research that comes out of the facility. Few smaller collections have been rigorously curated over long periods, given that successive curators' interests in, and relative commitment to, active or even intensivelevels of curation, vary significantly. Likewise, scant research has focused on tabulating the condition of most collections and assessing their limitations. Such analyses relate to the historical (e.g., by decade) and geographical origin of specimens (Prather *et al.*, 2004; Snow *et al.*, 2014), including temporal peaks in collecting, a comprehensive accounting (list) of species present in the collection, general geographical coverage of specimens (e.g., by State, county, etc.), presence of endangered or extinct species, curation techniques employed, and how that collections originated (e.g., by researchers, students, faculty).

Given a general lack of knowledge about historical patterns of collections in smaller facilities, this study assessed the temporal and spatial patterns of specimens in the herpetological collection at Pittsburg State University in Pittsburg, Kansas. To achieve these goals, the entire herpetological collection (snakes, lizards, salamanders, frogs, turtles) was curated at modern standards, including data basing of all specimens. The study summarizes the many limitations of the collections and their data, the poor curatorial quality of most specimens at the outset of the study, and presents updated data that is based on newly curated collections. The project opens new possibilities of research on specimens within a collection that had rarely if ever been the source of primary biodiversity data.

CHAPTER II

MATERIALS AND METHODS

History of the Herpetological Collection at Pittsburg State University

The herpetology collection (HC henceforth) at Pittsburg State University was established in February, 1967 by Dr. James Triplett (Emeritus Professor of Biology) when he was an undergraduate student. While working with new specimens collected on a field trip in 1964, he learned that the department had several specimens of amphibians and reptiles but no formal collection. The current HC contains all specimens collected by Dr. Triplett, which have been used primarily for teaching purposes, and all others collected by various individuals during the last four decades.

When this study commenced in 2015, the HC housed 622 specimens in its catalogued (written summary) collection, with several hundred specimens awaiting curation and cataloguing. The catalogued collection, which dates back to the late 1930s, includes specimens from 17 US states (Arizona, Arkansas, California, Colorado, Indiana, Kansas, Mississippi, Missouri, Nevada, New Mexico, North Carolina, Oklahoma, Oregon, South Carolina, Texas, Utah and Wisconsin) and the Canadian Province of Manitoba.

The collections have been housed in four different rooms in three buildings during the last 40 years, including Carnie Hall, until 1980 when the building was razed; Heckert-Wells, until 2013; and their current location in Hartman Hall Room 216. Due to these moves, which were made without the supervision of a collections manager, the collection became badly disorganized. Some specimens were lost due to broken jars or misplacement. Previous curation, when done at all, was a casual affair by students taking advanced topic classes, but with no clear best practices information to follow, or with

appropriate workspace. Moreover, the collective time for students and faculty members to curate minimally, let alone fully, was never sufficient. Specimens periodically were removed from the general collections for teaching purposes, whereas most were stored in the basement of Heckert Wells (location of the Department of Biology), along with the ichthyology collection.

Assessment of Wet Collections

For many years wet collections were used for teaching purposes only. When the collections were moved to the new location in Hartman Hall (Fig. 1), the contents of all jars were screened for their catalogue numbers, which had been recorded carefully by hand in a large scribe (ruled) notebook over many years, henceforth called the "catalogue" (Appendix B). Each jar was compared to the respective catalogue entry and notes were taken according to various curatorial issues that would need to be addressed. Such concerns included missing jars, dehydrated specimens, inadequate preservative in jars, mold on some specimens, the need for new preservatives, specimens destroyed or beyond normal conservation abilities.

Figure 1. Initial storage of the Herpetology Collection on the new collection rooms at Hartman Hall, Pittsburg State University. Pictures taken before assessments began in 2015 showing several different types and sizes of jars. At the time Herpetology specimens were still mixed with the Ichthyology and teaching collections.

Screening and Curation of the Herpetology Collection

After initial assessments, the condition of all reptile and amphibian specimens were meticulously screened in April 2015. The screening process included comparing every detail of the information in (or on) the jars and the specimens therein, to the data entered on the catalog. It involved all of the following: checking for missing jars; trying to locate missing specimens; assessing the biological contents (species kinds and numbers); assessing each jar for the condition of its specimen tags, preservative fluids and quality of its lid; and updating the catalogue to reflect missing information. Collectively, the screening indicated extreme measures were needed to update the curatorial quality of the HC.

To upgrade the quality of specimens, curation included changing out most jars, obtaining in many cases new specimen tags, changing the preservative fluid, refilling jars, verifying specimen identifications, and rehydrating specimens. These activities constituted a significant part of this MS project.

Curation protocols borrowed from the bibliography from Collins *et al.* (2014), Simmons (2014), Simmons (2015), the National Park Service Museum Handbook (NPS, 2000; NPS, 2005b), National Park Service Conserve O Gram (NPS, 2005a), and were adapted to the materials available at the lab. I also reviewed guidelines and protocols shared by curators, collection managers and researchers at The Natural History Collections list server (NHCOLL-L) hosted by Yale University, which is sponsored by the Society for the Preservation of Natural History Collections (SPNHC) and Natural History Collections Alliance (NSC). The author attended the SPNCH meetings in Florida in the Summer of 2015 at the Florida Museum of Natural History (University of Florida, Gainesville) and in Colorado in the Summer of 2017 at the Denver Museum of Natural History, and visited the Missouri Botanical Garden in St. Louis – Missouri and The Field Museum in Chicago – Illinois to meet and interact with colleagues actively curating herpetological specimens, and to hear presentations about curation in general.

Collection Jars. All food and canning jars with metal lids were replaced with O.Berk™ glass jars with polypropylene (PP) lids with liner in standardized sizes of 4oz (≈118mL), 8oz (≈236mL), 16oz (≈473mL), 32oz (≈946mL) and 1 gal (≈3.7L). The latter provide superior sealing efficiency with seal longevity greater than 20 years, whereas jars

with metal lids last less than 10 years (if new and used with liner or Teflon tape to avoid oxygen movement – which was not done in the HC) (Simmons, 2014). The glass jars with wire bail and rubber gaskets lids were replaced if the gasket was deteriorated or if the jar had to be opened. According to Simmons (2014) after the gasket is exposed to fluids, its sealing longevity might last less than 5 years, therefore regular inspections are required.

Specimens in plastic food containers made of polyethylene terephthalate (PET), polycarbonate (PC) and high-density polyethylene (HDPE) also were replaced with O.Berk™ glass jars. Five-gallon (≈19L) buckets made of HDPE that contained specimens were retained or replaced as needed, since stainless steel tanks were not available; Simmons (2014) indicates that sealing properties of HDPE containers may last more than 15 years.

Specimen Tags. Several different papers and pens had been used for specimen tags over the years, which had resulted in many faded and torn tags (Fig. 2A). There was no standard size for the tags, or standard thread to attach the tags to the specimens, resulting in several loose tags or thread tangled around the specimens (Fig. 2B). New tags were created using Resistall Paper 36-pound off-white linen ledger 100% rag paper, which maintains its structural stability when exposed to fluids. Sakura Pigma Micron pens were used to write data on tags. These pens use the Pigment Ink Process, which is acid free with neutral pH, providing markings that are resistant to fluids and UV light.

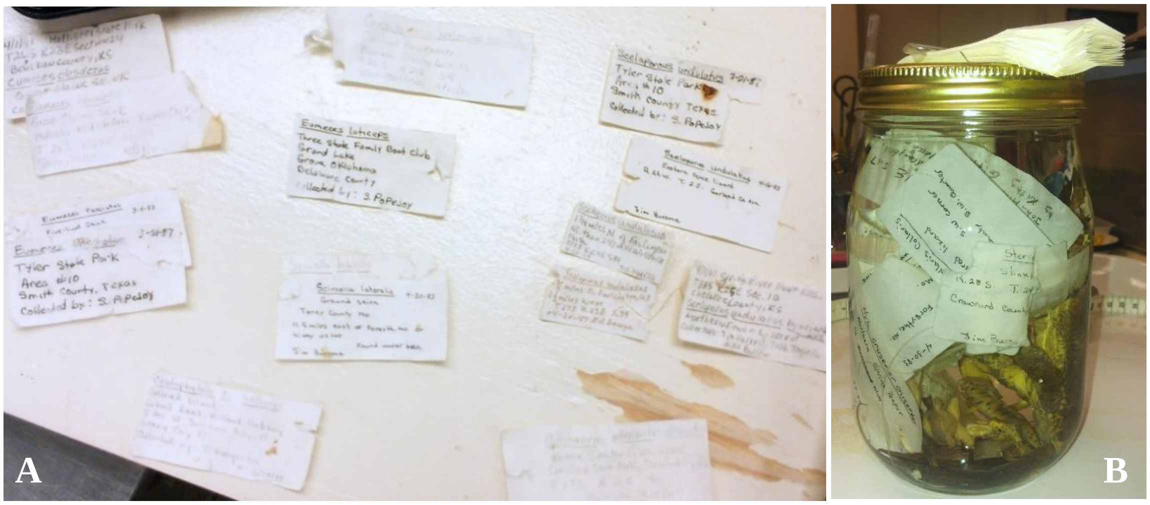

Figure 2. Degraded old specimen tags due to unsuitable paper and ink use. A – Old specimen tags used different sizes, types of paper, pens and pencils resulting in faded data, torn tags and ultimately data loss; **B –** Specimen jar showing numerous loose tags; tags piled up on the top of the jar belonged to specimens that had their tags removed and had not been put back on the same jar. It is also important to note that the ratio of paper, specimens and preservative solution was not ideal.

All torn or faded tags were removed from specimens, archived and replaced by new tags. Following the protocol by M. Revelez at Angelo State University in Texas (pers. comm., June 2, 2015), tags and strings were removed from specimens to allow the tags to air dry on a paper towel, after which the dry tags were archived in the Pioneer Archival Photo Album with archival quality paper (Fig. 3). Individual pockets were assigned to each catalogued jar, within each pocket a label containing the HC information and the jar's catalogued entry number was printed. All specimen tags belonging to the same jar were kept in the same pocket.

As the old tags were removed for archiving they were replaced with new Resistall tags measuring 7.2 x 2.5 cm. For stringing specimen tags, white 100% corespun polyester thread was used for small specimens, whereas white 100% cotton crochet thread was used for larger specimens. Following the protocol of Simmons (2015), tags were tied to each individual specimen on the left hind leg above the knee joint. If the left leg was missing then the tag was tied to the right hind leg, an arm, or around specimen's waist. For small specimens the tags were tied to specimen's waist or kept untied inside a small vial with the individual specimen. On limbless specimens (i.e. snakes) the tags were tied around the neck.

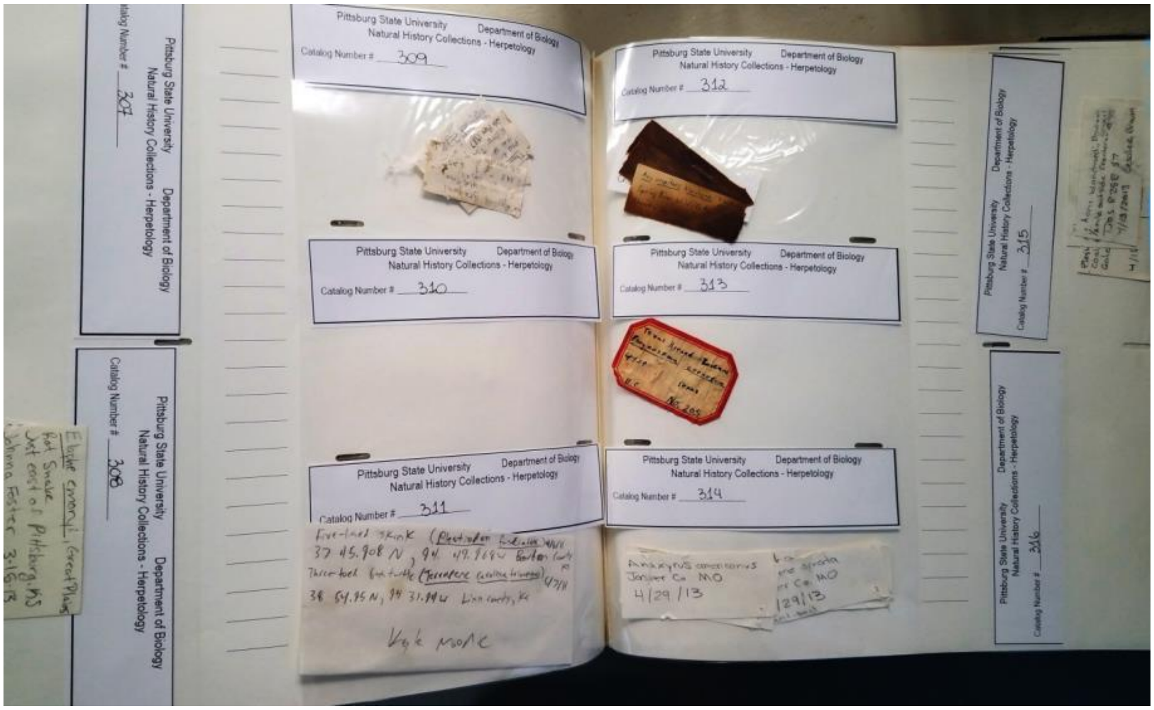

Figure 3. Archiving of old specimen tags. As old tags were replaced by new ones, they were archived in archival quality album containing individual pockets. Each pocket contains a label indicating the name of the institution, department, collection and catalogue number. All tags from the same catalogue/jar number were archived together.

Preservative Fluid. The majority of specimens in the HC had been preserved in 70% ethanol (ETOH). Some were preserved in 40% isopropanol, and a few in 10% formalin. In a few cases drops of glycerin had been added to the 70% ETOH solution putatively to help maintain a specimen's flexibility and color. However, Simmons (2014) reported that no studies corroborate that idea, and in fact that glycerin is a hygroscopic liquid and thus absorbs moisture from the air. Therefore, improperly sealed jars with glycerin absorbed water, diluting the 70% ETOH solution and facilitating the proliferation of bacteria and mold as the fluids slowly evaporate. Glycerin was also found to be the only preservative solution for a few larvae specimens in the HC.

Ethanol at concentrations of 70% acts as a biocide and as a preservative; higher or lower concentrations are not recommended, given that higher concentrations dehydrate the specimens and lower concentrations are not as effective as a biocide (Simmons, 2014).

Fluid changes can have a negative impact on the specimens, as the fluid acts as a "microhabitat" because the specimen may exchange body fluids and lipids (Simmons,

2015). As such, every time fluids are changed the specimen loses more of its contents into the new fluid.

Preservative solutions were changed in the following situations: 1) fluid color was too dark due to discoloration of specimens or rust was on the lid; 2) excessive residues in the bottom of the jar (usually from the tag or rusted lid); 3) specimens preserved in 10% formalin, 40% isopropanol or other unknown fluid; and 4) fluid level was less than 50% of the jar by volume.

The process of changing fluids, particularly when fluid was not 70% ethanol, followed the three staged concentration steps of 20% phases of ethanol, following recommendations of Simmons (2014, 2015). The staged concentration steps are important to prevent shrinkage and swelling that can change sizes in specimen due to osmotic pressure variation among different preservative solution (Simmons, 2014). Specimens thus were removed, rinsed in running water, staged for 24 hours each in concentration steps of 10%, 30% and 50% ETOH, and ultimately preserved in 70% ETOH. The ethanol was mixed with distilled water (Simmons, 2014) to avoid damage to specimens caused by the presence of chlorine, oxidation products, or calcium chlorine precipitates. If the previous fluid was not 70% ETOH, then specimens were washed in running water for 2 to 24 hours (according to specimen size) before the staged concentration steps. During this process specimens were monitored closely due to increased chances of bacterial and mold contamination.

Nitrile gloves were used to handle specimens, as well as during all procedures involving fluids, due to the presence of formalin and other unknown preservatives. Formalin easily penetrates the skin and the mucus membranes, requiring the use of nitrile gloves, goggles and respirator with filters for formalin gas.

Topping off Fluids. The ideal volume of fluids is 90% of the total volume of the jar. Allowing 10% of the jar to remain empty reduces evaporation and provides better sealing properties (Simmons, 2014). Jars were refilled with 70% ETOH when fluid levels were between than 50% and 90% of the jar's total volume.

Rehydration. Dehydration typically is a result of problematic enclosure and lack of monitoring. Several cases of dehydrated specimens were found in the HC, and nearly all were properly rehydrated. Specimens that were completely dry (skin was crispy) were

not rehydrated, whereas specimens with some moisture content were rehydrated. The initial rehydration process used different protocols (Chmiel, 2014; Schneider, 2002; Simmons, 1999; Simmons, 2002; Singer, 2014; Smith, 2012), and different techniques were mixed to fit the availability of products and equipment of the laboratory, resulting in two rehydration techniques used in this project.

The first technique developed consisted of six steps using a surfactant to induce the absorption of water, which was used on drier specimens. The first step is to remove the specimen from the jar (if flexible enough to do so), add it to a beaker, immerse it in a 3% surfactant solution (Fisherbrand Sparkleen detergent or Decon 90), and warm it to 50°C using a hotplate. Specimens were kept in 3% solution for several days until additional morphological improvements were no longer observed. The second step immerses the specimen in a 5% surfactant solution (Fisherbrand Sparkleen detergent or Decon 90) and warms it to 50°C. (The second step can be omitted if the specimen was well rehydrated after first step). Overnight solution should be changed daily. Third, specimens were re-fixed overnight in buffered 10% formalin. (The third step was applied only to specimens with mold or bacterial growth previously to rehydration, or were severely dehydrated and required longer periods on surfactant.) Fourth, the specimen undergoes the staged concentration steps in ethanol as described in the Preservative Fluid section. The fifth step is to preserve specimens in 70% ethanol. The sixth and final step of the first technique is required for floating specimens, whereby air bubbles can be removed by gently applying pressure to the body of the specimen while it is submerged in ethanol (a step that applies to any rehydration process). This first rehydration technique is long, taking usually six days or more, and requires all the steps to be followed as described. Close daily monitoring is required to avoid mold or bacterial growth.

The second technique was used with specimens that were more humid and flexible, and requires only four steps that usually take five days. The first step is to submerge the specimen in warm distilled water overnight. If needed it is possible to allow a specimen to remain for 48h or 72h in the warm distilled water, although it should be changed every day to avoid growth of bacteria or mold. The second step was applied only to specimens that had not been properly fixed, or that had mold or bacterial growth; it included re-fixing specimen in buffered 10% formalin overnight. Third, the specimen

undergoes the staged concentration steps in ethanol. Finally, the fourth step is to preserve specimen in 70% ethanol. This technique is preferred since no surfactant is used.

After the acquisition of a new surfactant, Decon 90, a few other specimens were rehydrated, following the first technique described above. Decon 90 is the most common surfactant used in rehydrations, therefore it was acquired in order to compare its results to Sparkleen, which was available in the laboratory.

Mold and Bacterial Removal. Microorganisms can appear in jars with defective seals, in which the preservative solutions evaporated and levels dropped below 50% (Simmons, 2014). Specimens affected by bacterial or fungal growth were removed from the jar, rinsed with running water, and with the aid of a cotton swab or very soft brush, mold and bacteria were carefully removed under running water. The jar was washed with antibacterial liquid dish soap and carefully inspected before reusing it. Specimens were immediately immersed in 10% ethanol, following the staged concentration steps up to 70% ETOH. Contaminated fluid was discarded.

Storage Environment. The HC is stored within a small room (2.8 x 2.2 m) inside the Herpetology and Ichthyology Laboratory, at Hartman Hall Room 216. The temperature in the HC room and on the laboratory space is maintained at 21°C (ca. 70°F). Ideally, the temperature inside the HC room should be cooler (18°C) than the laboratory space, and humidity levels should be kept at around 50%. Fluctuations in temperature and humidity decrease sealing properties of jars, increase fire risk due to low flash point (16.6°C) of ethanol, increase evaporation of fluids, reduce binding of tags and ink to the paper, and cause the expansion of skeletal material losing teeth and smaller bones (Simmons 2014). However, only one air conditioner and heating system is available for both the HC room and laboratory space, which works independently from the building's air conditioner and heating systems. There is no humidity control or monitoring system at the laboratory. The collection room has its own light system, allowing the collection to be stored in the dark to avoid deterioration of specimens caused by chemical processes of deterioration, evaporation of fluids and ultraviolet radiation (Simmons, 2015).

Jars are stored in cardboard trays and organized numerically following the catalogue numbering system (Fig. 3A). Trays are stored on wooden shelves at a maximum height of 1.6 m from the ground. Cardboard trays are used to easily remove

jars from the shelves, since the wooden shelves are not movable. The trays also offer extra security to the jars since the shelves do not have a lip. Wet collections ideally should be stored in fireproof cabinets or on metal shelves with restraining bars due to the flammable characteristic of ethanol. Due to fire hazard of the ethanol, fire extinguishers are available in the lab.

The HC is organized numerically by jar, not taxonomically, as it is common in large collections. Jars were organized in a numerical ascending order, starting with number 1, following the catalogue numbering system to the most recent addition. Each catalogued jar was assigned a tag with a number; i.e. CAT 217, where CAT stands for 'catalogue' and 217 is the catalogue entry number (Fig. 4B).

Figure 4. Organization of jars within the collection room at Hartman Hall. Jars are organized numerically, inside cardboard trays, following the catalogue numbering system. Each cardboard tray has a tag indicating which jars are held within it (Figure 4A), as well as each jar has a catalogue number on top of the lid and inside the jar (Figure 4B).

Handwritten Catalogue Databasing and Initial Digitization of Specimen Data

The data for specimens originate from the hand-written catalogue (in a bound, ledger format) (Fig. 5A) and students' field notebooks. Hand-written index was created in 1983 to facilitate locating specific species within jars (Fig. 5B); however, there were no records of total number of specimens, which species were preserved, or how many specimens of each species were preserved. The records had not been updated since 1983. Without that data it was not possible to answer basic questions such as:

- Does PSU have a specimen of *Gastrophryne olivacea*?
- What is the most abundant species in the collection?
- What locations were sampled most frequently and which areas need more sampling effort?
- Is the collection representative of local biodiversity?
- Are Threatened and Endangered species present in the collection?

These kinds of question can be answered with confidence only after completely curating, cataloguing and commencing the digitization of the collection.

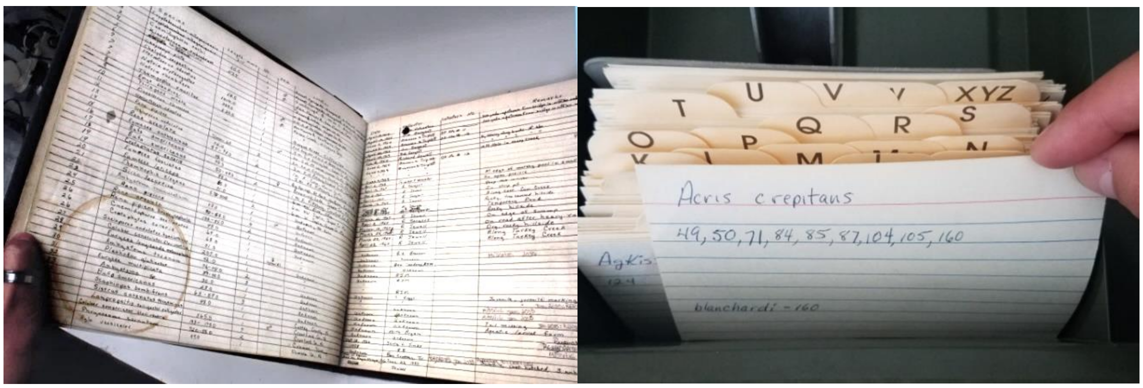

Figure 5. Original data entry and cataloguing system for the Herpetology Collection. A - Hand-written catalogue created in 1967 containing data for over 600 catalogued specimens. This image can be seen in greater detail on Appendix B. **B –** Index card system created in 1983 to help locating specific species in the collection. Following the species name are catalogued jar numbers wherein individuals of that species were preserved (e.g. *Acris crepitans* could be found on jars #49, #50, #71, etc.). Unlike the catalogue, the index card system has not been updated.

After the screening and curation of the HC, the changes made to each jar were recorded in the catalogue as follows: missing yyyy (year that it was noticed the missing jar); missing specimen(s) yyyy; dehydrated; rehydrated mm/yyyy (month and year specimen was rehydrated, along with the description of rehydration process and final preservative solution); preparation changed mm/yyyy (along with final preservative fluid); refilled mm/yyyy (along with fluid used to refill jar).

Once the data on jars and specimens were cross-referenced with the handwritten records, the entire catalogue was databased on Excel. As new specimens were curated, the data retrieved from specimens tags were digitized into an Excel database, along with the existing records. The field names (columns) in the database on the table (Appendix

D) allows data sorting within any category: Catalogue Jar Number, Species (ID on tag), Species Name (updated nomenclature), Taxonomy Changes, Common Name, Total Specimens, Length (mm), Sex (♀♂), State, County, City, Location, TRS coordinate, Latitute, Longitude, GPS coordinate, Date, Year, Collector, Preparation, Preparation Changes, Notes, Remarks, Entry Date, Cataloguing Staff. The original handwritten catalogue had fewer categories, which were all retained in the digital version (Appendix B). A current database is critical for finding specimens, especially in this collection where the arrangement of specimens in jars is somewhat random. For example, specimen jars had been scattered around the Biology Building, so as older jars were added to the catalogue it somewhat disrupted the system as it was created initially. The initial organization followed a timeline, from the oldest to the newest specimen collected. Specimens of different species were added to the same jar if they were collected in the same location, or by the same collectors in the same field trip. This grouping system was done to save space and jars, which are both limited. Once the digital catalogue was established, however, all new specimens were curated and catalogued.

Field Notebook Databasing

Field notebooks are a valuable source of biological and ecological information, containing details about field trips, collection sites, weather, collectors, location, disposition of specimens and field photographs. They serve as complementary material for the specimens in the collection and thus often are preserved for more active collectors. Several students taking Herpetology classes donated their field notebooks to the HC.

Field notebooks were digitized in a similar manner to the HC catalogue. All notebooks from the same year were digitized within a single tab, the categories of which include: Species Name, Common Name, Total Individuals Collected, Total Adults Collected, Total Juvenilles Collected, Total Individuals ID in the Field (released), State, County, City, Location, GPS Coordinates, TRS Coordinates, Date, Year, Primary Collector, Other Collectors, Collecting Site Notes, Curator Notes. A reference column also indicates which specimens are preserved in the collection, along with their catalogue numbers (Specimens Catalogued, Total Number of Specimens Catalogued, Jar Number),

and which specimens have field photographs (Specimen Photo on Field Notebook) (Appendix E).

After databasing, all specimens in the collection were cross-referenced with the digital field notebooks to improve the data on the catalogue. If specimens collected by students were preserved in the collection, information regarding the catalogue number was added to the digital field notebook, along with how many specimens were preserved, and the collector(s) names. Field notebooks were archived chronologically in ULINE® archival-quality filing boxes.

Curation and Cataloguing Process of Uncatalogued Herpetological Specimens

Since 1974, the most recent entry in the catalogue, the HC continued to receive new specimen. However, without a curator or collections manager handling specimens, hundreds of specimens were added and stored haphazardly. As time passed many became teaching tools, resulting in disorganization and loss of data. The vast majority of uncatalogued specimens were collected during field trips and were prepared by students taking Herpetology; therefore, many deficiencies were present, including misidentifications and poor fixation and/or preservation techniques.

The first curatorial step was to sort uncatalogued jars and their specimens by year, location and collector. After each jar was assessed the specimens with data were added to the HC, and all specimens without data were transferred to the herpetology teaching collection. As needed fluids were changed, specimens were transferred to new collections jars and tags were redone, following the curatorial steps described above. The data available on tags from specimens collected by students during field trips were compared to the data in students' field notebooks, field trip class material and collecting permits to ensure higher quality of data. In some cases, it was necessary to identify specimens or to confirm identifications. Identification of reptiles and amphibians used dichotomous keys and field guides (Altig and McDiarmid, 2015; Blair, 1957; Collins, 1993; Collins *et al*., 2010; Conant and Collins, 1998; Powell *et al.*, 2012, Powell *et al.*, 2016; Stebbins, 2003). Specimens were measured in millimeters using digital calipers (Pittsburgh 150mm and Neiko 300mm), a ruler (150 mm) and measuring tape (1500mm), following the protocol

by Conant and Collins (1998) for taking measurements of reptiles and amphibians. Snout-vent length (SVL) was measured in snakes, lizards and salamanders; head-body length (HBL) was measured in frogs and toads; and length of shell was measured in turtles. Once specimen curation was completed, data were entered in the digital catalogue (as described on Handwritten Catalogue Databasing and Initial Digitization of Specimen Data section), and the jar was assigned a catalogue number.

Herpetology Teaching Collection

For many years the HC was used mainly as teaching resource, resulting in many cases of significant disorganization of specimens and data loss. After consultation with faculty members, the establishment of a dedicated but separate teaching collection was deemed as being crucial to the safekeeping of the scientific collection.

Specimens without data were moved to the new teaching collection. All specimens were identified and grouped by species in new jars. Besides O.Berk™ glass jars, 64oz (≈1.9L) and 1 gal (≈3.7L), polyethylene terephthalate (PET) containers also were used for this collection due to the wider mouth of containers, low price, and practicality for transporting specimens to classes. Old tags and strings were removed, fluids were changed to 70% ETOH and dehydrated specimens were rehydrated.

The freeze-dried specimens were sealed in Marvelseal 360© and stored in -20°C for three weeks to eliminate dermestid beetles, larvae or eggs, and any other pests, specimens were stored in ULINE® archival cardboard boxes lined with ethafoam. Specimen jars and boxes were organized taxonomically, and are temporarily stored on the same room as the research herpetology collection.

The Teaching Collection was databased in Microsoft Excel 2010 containing the Catalogue Number, Scientific and Common Names, Total Number of Specimens per Jar, Life Stage, Sex, Original Fluid Preparation, Fluid Changes, and Notes. The same design used for the herpetology catalogue was adopted for the Teaching Collection. A simplified version of the catalogue was printed to facilitate locating specific jars.

A phylogenetic list of taxa (species and subspecies) was created following the phylogeny of the Tree of Life Web Project (Laurin, 2011) and Collins *et al.* (2010). Jars were numbered following the phylogenetic list and are grouped by taxonomic Order.

Data analysis

Data were analyzed with the primary goal of understanding the historical aspects of the collection, including: representation by taxonomic groups (families, genera, species); geographic distributions (by country, state/province, and county/parish); and temporal information (monthly, annual, and by decade). Scientific names were updated, along with the common names, according to the Committee on Standard English and Scientific Names (Crother, 2012, 2017). Temporal and geographical analyses were done using Microsoft Excel 2010.

Spatiotemporal maps of collecting records were created for specimens collected in Kansas using QGIS 2.18, Google Maps, Google Earth and Earth Point. These maps show location for each specimen collected in KS, county delimitation, road system, river system and urban areas. Collecting radius was measured on Google Earth associated to Earth Point; however, a confidence radius was not determined for each specimen because geo-referencing was not the main purpose of this study, and a relatively steep learning curve exists for the intelligent and thorough use of georeferencing software programs. Even so, such maps may aid on finding areas for potential preservation and to identify areas in need of additional surveying, especially when merged with larger datasets. These can be applied to aspects of conservation biology, such as identifying hotspots and to studing changes in range through space and time. Given that many specimens were collected at the same location, points often overlap on a given map. Temporal distribution is also represented on the maps, with each collecting point color-coded to a specific decade, and with different-sized symbol to facilitate the visualization of overlapping decades. Different symbols were used on maps cointaining more than one species.

CHAPTER III

RESULTS AND DISCUSSION

Curation of the Herpetology Collection

Initial assessments of the hand-written catalogue suggested 623 specimens of reptiles and amphibians in the HC. The lack of curation through time associated with faulty enclosures and inappropriate storage resulted in dried specimens (Fig. 6A), rusted metal lids (Fig. 6B), growth of bacteria and mold (Fig. 6C), and loss of specimens. When the project began, 141 specimens were missing. A total of 140 required rehydration, 126 required a change of fluids, and the jars of 89 specimens needed their fluids topped off. At the outset, only 26.3% of the collections appeared to be well curated.

At completion of the HC curation (Fig. 7A), 1,008 specimens added to the collection, totaling 1,631 catalogued specimens and 1,484 physical specimens preserved as of March, 2017 (Table 1). Approximately 330 specimen jars were replaced by O.Berk™ jars (Fig. 7B). The old jars and metal lids were recycled. More than half of the specimens needed their fluids changed, mostly due to rust on lids that contaminated the preservative solutions and allowed for evaporation of ethanol, or because specimens were not transferred from 10% formalin to ethanol after having been collected. A relatively small percentage of specimens were preserved appropriately at the outset (30.8 %). Approximately 1,000 liters of 70% ethanol were used for staging specimens, changes of fluid, rehydration and topping off; roughly another 300 liters of 70% ethanol were used as final preservative fluid.

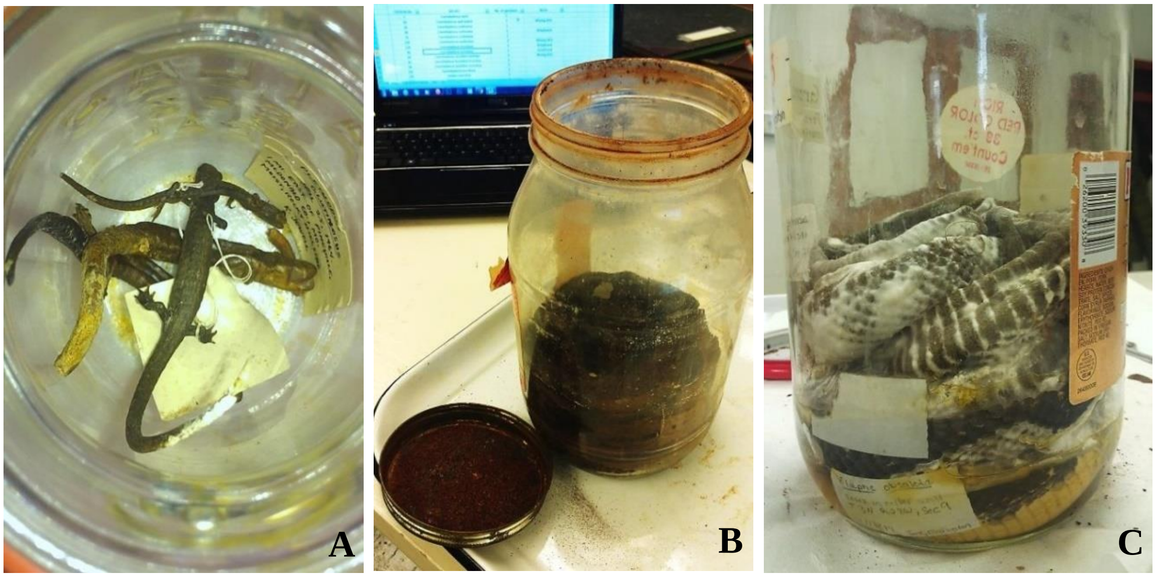

Figure 6. Results of decades without adequate specimen curation and monitoring. A – Severely dehydrated Small-mouthed Salamanders (*Ambystoma texanum)* showing whitish mold damage on the tails; **B –** Faulty enclosure showing rust on the lid and on the jar, which resulted in the evaporation of preservative fluids and partial dehydration of a Western Ratsnake (*Pantherophis obsoletus)*; **C –** Snakes covered by mold as a result of fluid evaporation on a faulty jar.

Table 1. Assessment of the Herpetology Collection at Pittsburg State University after conclusion of curation on March, 2017

HC Assessment Situation	Number of Jars	Number of Specimens	%
Entire jar missing	26	47	2.8*
Specimens missing within a jar	24	100	6.4*
Total specimens missing	-	147	9.0*
Total rehydrated specimens	57	221	14.8**
Change of fluids	154	757	51.0**
Topping off	56	151	10.2**
Well preserved specimens	102	457	30.8**
Total physical specimens preserved	372	1484	91.0*
Total catalogued specimens	398	1631	

* Percentage calculated over number of catalogued specimens. ** Percentage calculated over number of physical specimens preserved.

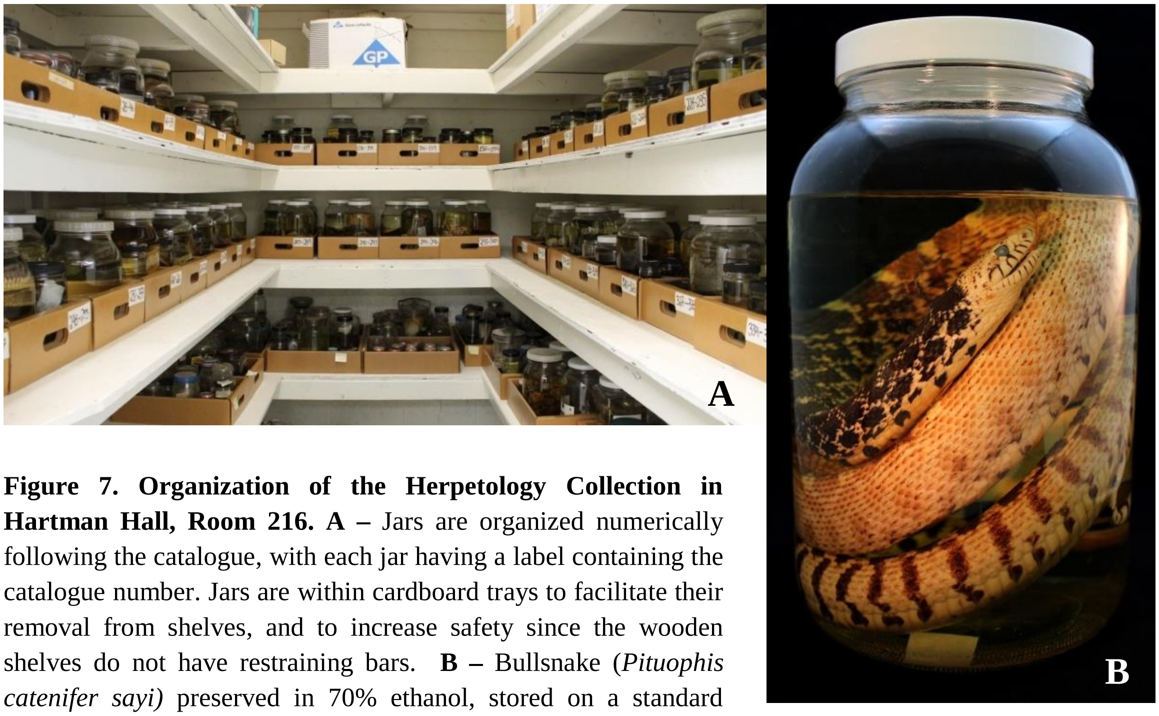

Figure 7. Organization of the Herpetology Collection in Hartman Hall, Room 216. A – Jars are organized numerically following the catalogue, with each jar having a label containing the catalogue number. Jars are within cardboard trays to facilitate their removal from shelves, and to increase safety since the wooden shelves do not have restraining bars. **B –** Bullsnake (*Pituophis catenifer sayi)* preserved in 70% ethanol, stored on a standard 1-gallon (≈3.7L) O.Berk™ jar after change of fluid.

Rehydration

Several stages of dehydration were present in the HC at the outset, representing 14.8% of the collection (Table 1). The range of dehydration included partially dehydrated specimens, some covered by mold and bacterium, to extreme cases where dehydration was so severe that the shape of the skeleton was clearly visible. Twenty-two extremely dehydrated specimens were not rehydrated, but rather kept as is, or retaining only the bones or shells. In situations where it was necessary to remove bacterial and mold growth from specimens before rehydrating (Fig. 8), the specimens were re-fixed in 10% buffered formalin after rehydration.

Among the 231 specimens rehydrated, only 47 could be rehydrated without use of a surfactant. Several specimens in which rehydration began in distilled water had to be moved to 3% surfactant solution to reach a desirable condition (Fig. 9). The use of surfactants (Sparkleen or Decon 90) was never the first option, due to possible physiological damage that surfactants may impart to cell structure.

Figure 8. Before (left) and after (right) photos showing the results of mold and bacterial removal, and rehydration of partially dehydrated specimens. The advanced stage of fluid evaporation and microorganism growth can be attributed to a lack of a curatorial staff and an inappropriate enclosure (food jar) and storage conditions. Specimens in similar conditions previously were discarded, although they could have been saved with appropriate curation techniques.

Figure 9. Before (left; top and bottom) and after (right) rehydration of two Crawfish Frogs (*Lithobates areolatus)*, collected on June 7, 1939 in Crawford Co., Kansas. The first rehydration used warm distilled water, but after four days without further improvement the specimens were transferred to 3% Sparkleen solution. The copper-colored liquid observed on the bottom of the jar (left; top and bottom) is the result of rust from the old metal lid, which also increased specimen discoloration.

Spatial Analysis, Distribution and Origin of Herpetological Specimens

The Herpetology Collection houses specimens from 23 U.S. states, including: Alabama, Arizona, Arkansas, California, Colorado, Florida, Indiana, Kansas, Louisiana, Michigan, Mississippi, Missouri, Montana, Nevada, New Mexico, North Carolina, Oklahoma, Oregon, South Carolina, Texas, Utah, Washington and Wisconsin, as well as a few specimens from Mexico (state unknown) and Manitoba, Canada. Specimens from Kentucky, Tennessee and Peru also are among the preserved specimens; however, they were found only recently in the basement of the biology building at PSU and still require curation. They therefore were excluded from the results of this project.

As expected, a large number of collections originated in Kansas (56.6%), Missouri (12.0%), Arkansas (5.1%) and Oklahoma (4.7%), representing 78.6% of the specimens in the HC altogether. California is the third state regarding total number of specimens (6.6%); however, of 108 specimens collected, 98 were tadpoles of the same species collected on the same date and location. A total of 14.4% of the specimens originate from the other 19 U.S. states, Canada and Mexico (Fig. 10). The remaining 7% are specimens that lack locality data, which is the most significant data limitation regarding their value for research.

Specimens collected in Kansas totaled 923 specimens collected from 37 counties, specimens from Missouri totaled 195 specimens collected from 15 counties; followed by California (108 spec., 5 counties), Arkansas (83 spec., 13 counties) and Oklahoma (77 spec., 11 counties). A total of 128 specimens were collected from the remaining 18 U.S. states (Fig. 11).

The wide geographical distribution of specimens highlights the importance of the HC as a repository of species from a wide range of habitats within the continental United States. The large percentage of specimens from the four-state area emphasizes the importance of the collection for understanding of local biodiversity and its conservation (Fig. 12). Some specimens had incomplete locality records, with a simple description of the collecting site but lacking information concerning county or municipality, thus preventing their addition to the county map. Among these were specimens from Arkansas (8 specimens), California (4), Kansas (7), Louisiana (1), Missouri (3), New Mexico (1),

Oklahoma (1), Oregon (2), Texas (4) and Washington (1). These records were recorded as Unknown Location for each state and included on the analysis of total records per state.

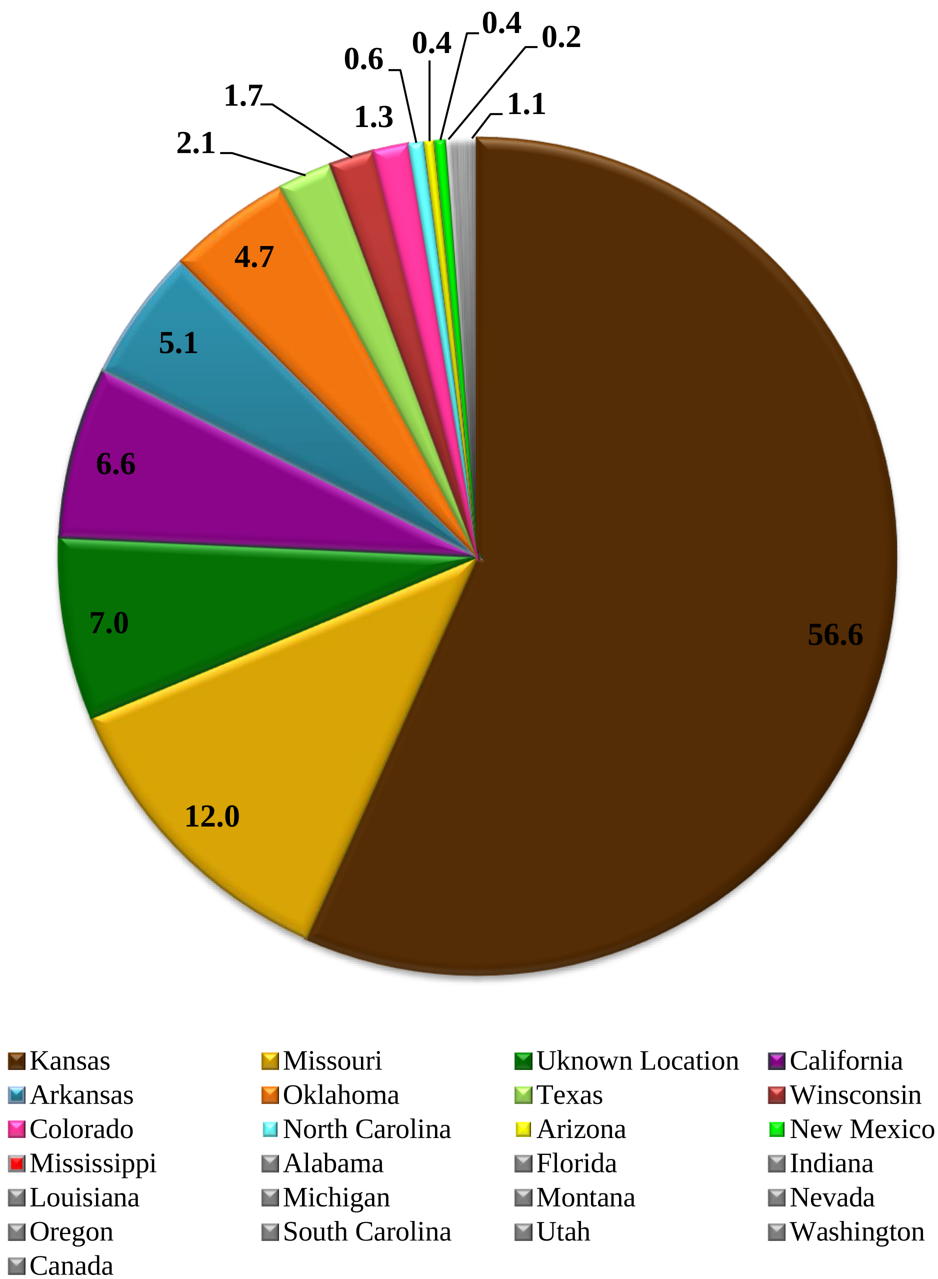

Figure 10. Percentage of specimens collected in each U.S. state, Canada and Mexico. At 7.0%, specimens with Unknown Location are the third most numerous, highlighting a significant limitation of the data in this collection. Specimens from MI, AL, FL, IN, LA, MT, NV, OR, SC, UT, WA, Canada and Mexico were grouped together (gray) representing 1.1% of the specimens in the HC.

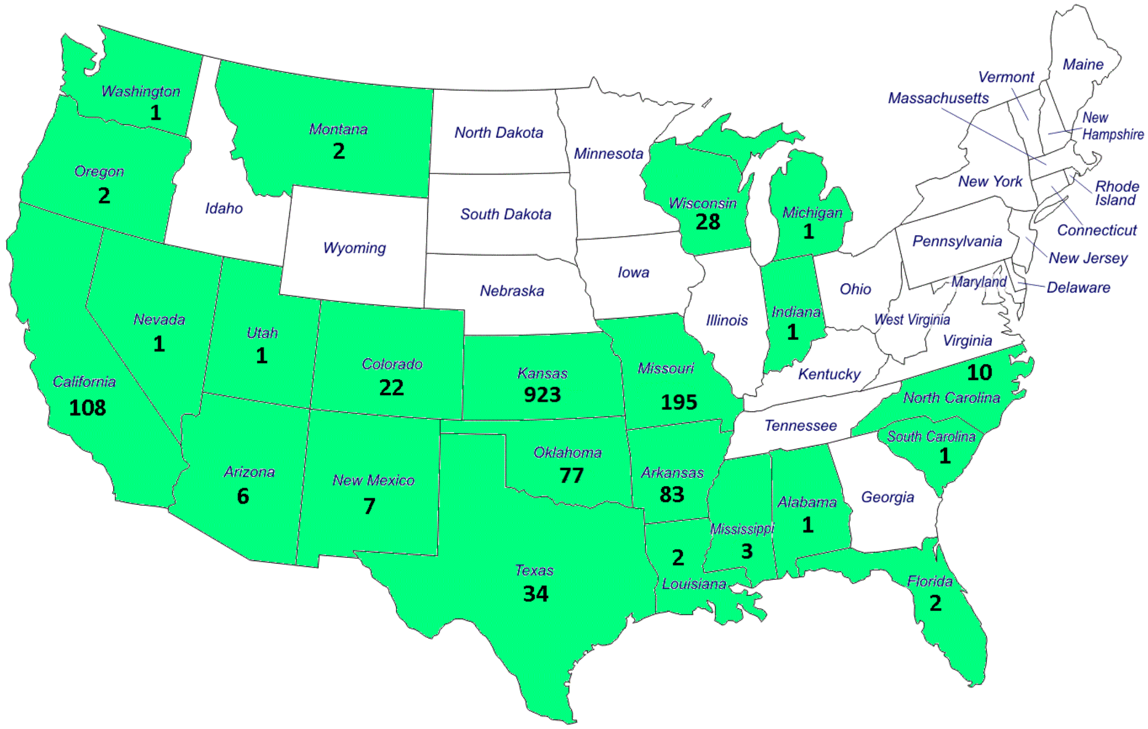

Figure 11. Spatial distribution and origin of HC specimens showing the total specimens collected in each U.S. state. The four-state area (Kansas, Missouri, Oklahoma and Arkansas) is the most thoroughly represented, highlighting the importance of this collection as a repository of local species. **Source:** Adapted by Schneider, N.A. from d-maps.com

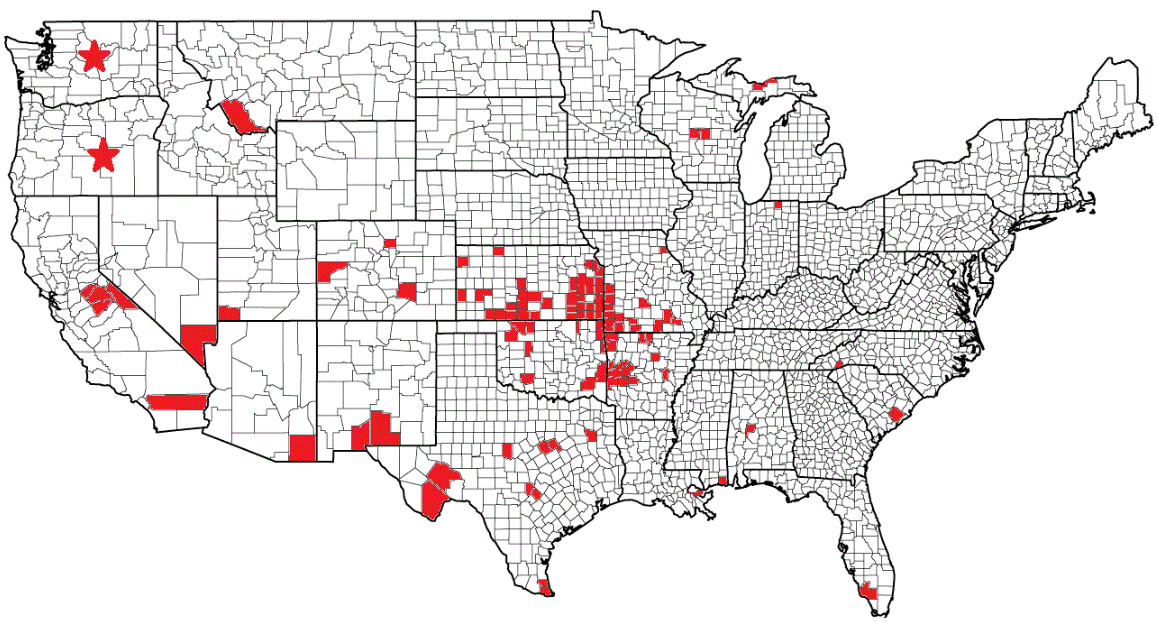

Figure 12. Spatial distribution and origin of specimens by county; highlighting the local collecting pattern at the corner of the four-state region - Kansas, Missouri, Oklahoma and Arkansas. Oregon and Washington states are represented by a star to indicate that there has been collecting on those states, however the counties were not recorded at the time of collections. **Source:** Adapted by Schneider N.A. from worldatlas.com

The results of the spatial analysis indicated that 53.0% (489 specimens) of the preserved specimens from Kansas were collected in Crawford Co., followed by Cherokee Co. with 14.2% (131 specimens) and Bourbon Co. with 13.3% (123 specimens) (Fig. 13 and 15). Neosho County is represented by 3.3% of the specimens (30 specimens); Montgomery (1.8%, 17 specimens); Finney (1.7%, 16); Linn (1.6%, 15); Labette (1.4%, 13); Leavenworth (1.1%, 10); Wilson (0.9%, 8); Unknown County (0.8%, 7) and Miami (0.7%, 6). The remaining 26 counties were grouped by similar percentages and totaled 6.3% (Fig. 13).

A spatial bias in the collections was identified, in that most collecting locations were adjacent to major highways, around or within city limits, and nearby waterways (Fig. 14). Pyke and Ehrlich (2009) and Newbold (2010) discussed spatial bias as one of the most common recurring limitations in collections, given that collectors tend to collect in easily accessible or local hotspots. Crawford Co. had the most complete spatial coverage, but included a few gaps in the western portion of the county.

Collecting efforts at Pittsburg State University have focused mostly on southeast Kansas, due to the location of Pittsburg State University (Crawford Co.) and conservation areas, such as over 50 Wildlife Mined Land Areas (Crawford Co., Cherokee Co.), the Spring River Wildlife Area (Cherokee Co.), and Hollister Wildlife Area (Bourbon Co.), reinforcing the major strength of this collection as a historical record of local biodiversity and collecting activities. Bourbon, Crawford, Cherokee and Labette counties comprise the Cherokee Lowlands ecoregion, and the extreme southeast portion of Cherokee Co. includes a small westward extension of the Ozark Plateau ecoregion. The Cherokee Lowlands and Ozark Plateau present highly distinct types of habitats, which are inhabited by species of amphibians unique to this area, including several represented in the HC: Long-tailed Salamander, Cave Salamander, and Grotto Salamander found on the Ozark Plateau; Pickerel Frog and Eastern Narrow-mouthed Toad found on both physiographic regions.

In addition to the species indicated above, the HC includes 130 specimens from Kansas collected from 30 counties, such as Western Tiger Salamander, Mudpuppy, Great Plains Toad, Woodhouse Toad, Plains Spadefoot, Prairie Rattlesnake, Western Groundsnake, False Map Turtle, and Texas Horned Lizard.

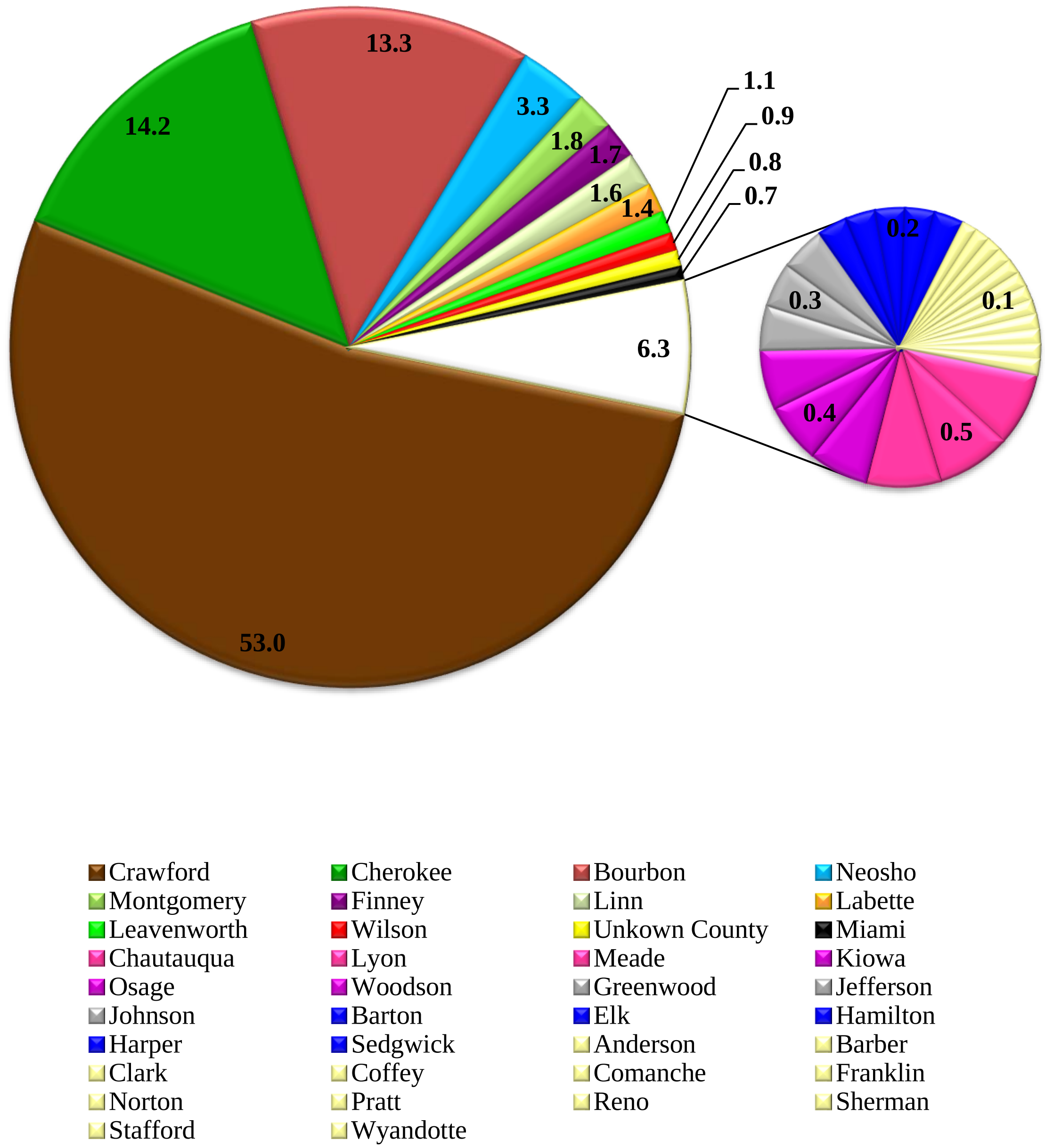

Figure 13. Percentage of specimens collected by county in Kansas. Unknown County (bright yellow) represents specimens that lack county-level information. Given that 26 counties had similar percentages of collecting, they have been grouped (graph on right) representing 6.3% of the specimens originating in Kansas.

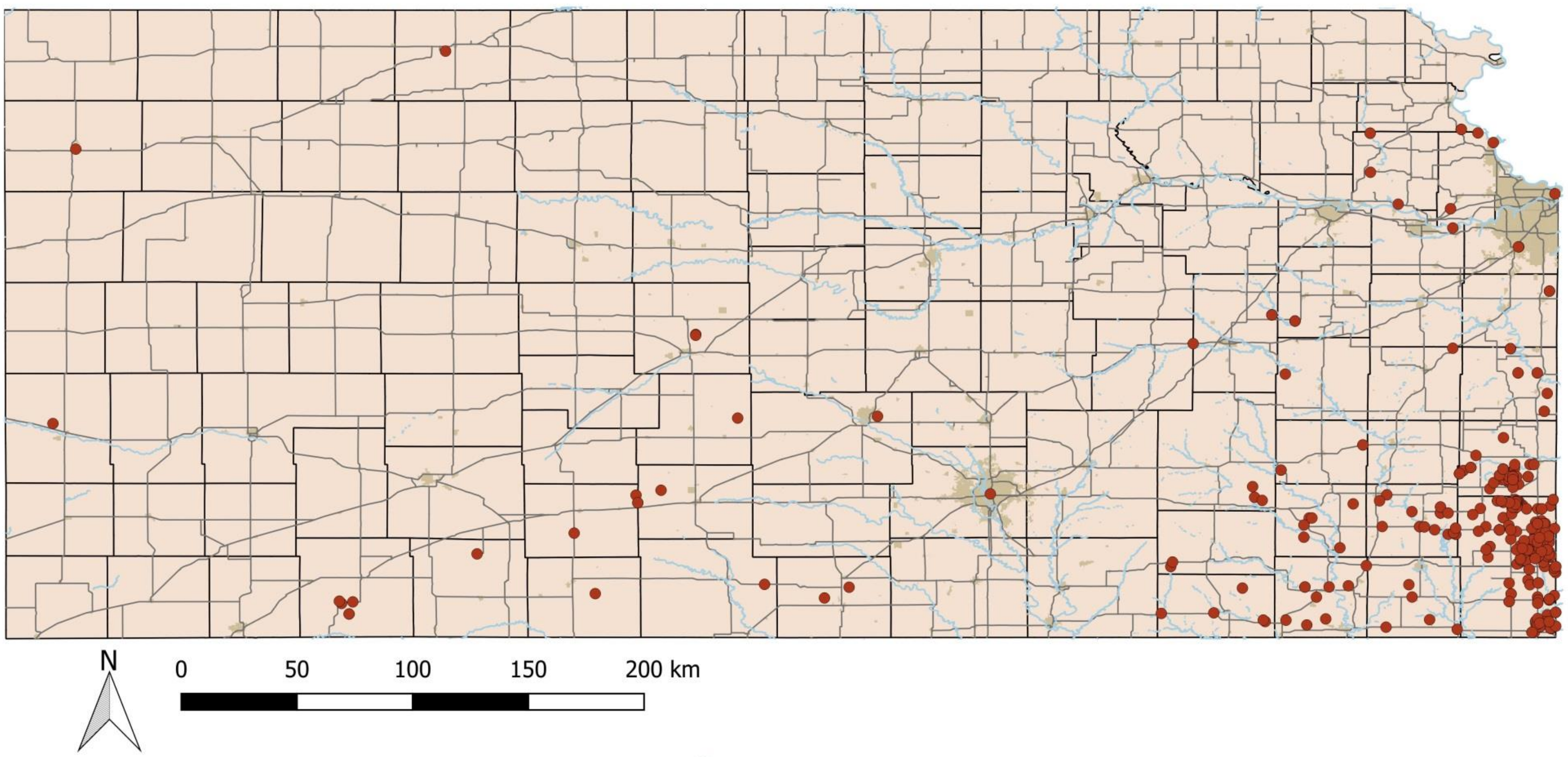

Figure 14. Spatial origin and distribution of herpetological specimens collected in Kansas and preserved at Pittsburg State University. Red dots represent individual specimens collected from 1939 to 2013. Spatial bias can be inferred, given that the majority of specimens were collected along major roads or highways, within or near city limits, or along waterways. **Source:** Schneider, N.A.

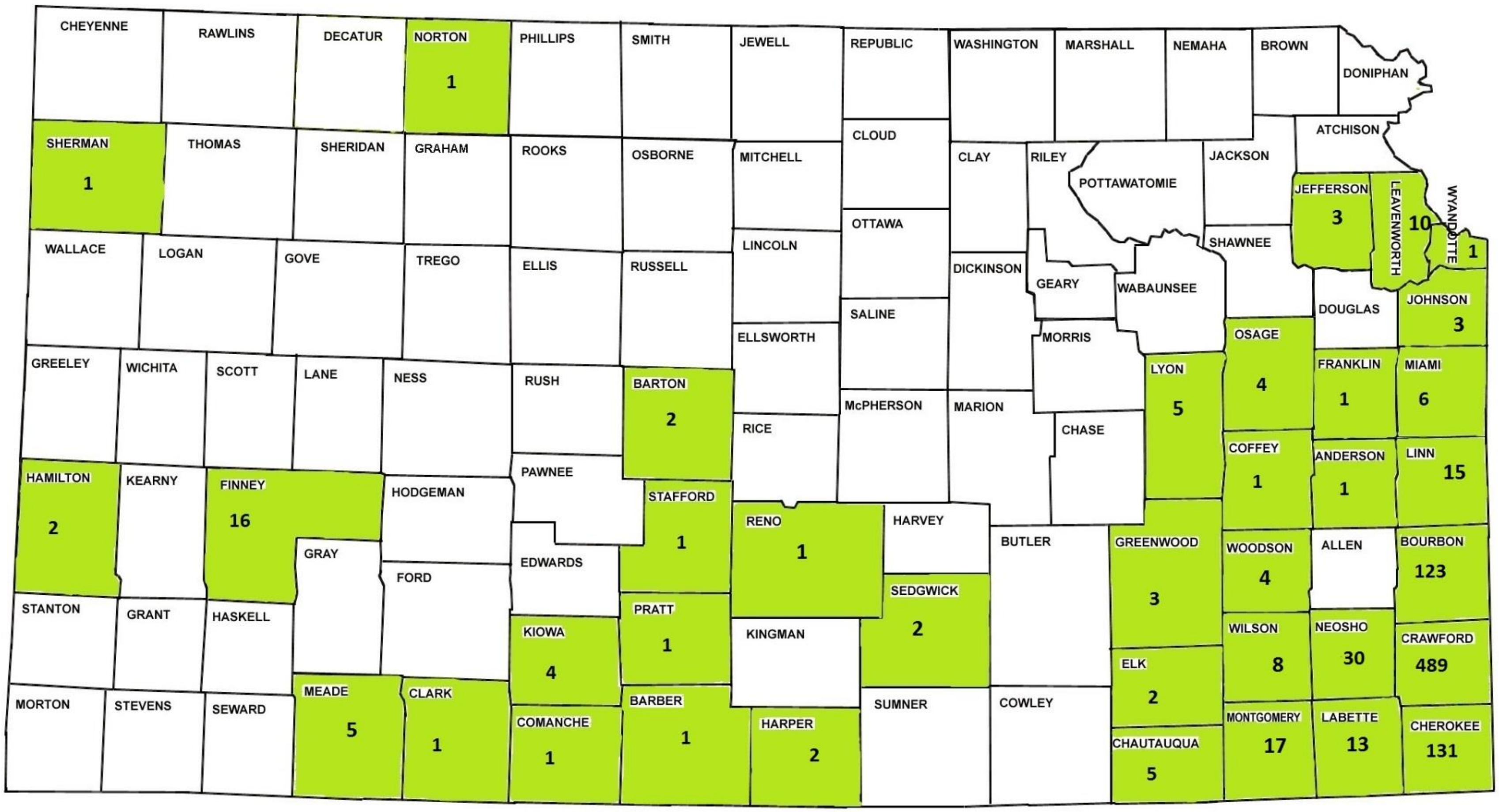

Figure 15. Total number of specimens collected per county from 1939 to 2013 in Kansas housed at PSU. The largest portion of the specimens originated from the southeast counties of Cherokee, Crawford and Bourbon. **Source:** Adapted by Schneider, N.A. from www.kshs.org

Herpetological specimens at PSU have been collected by researchers, professors, students and private collectors. From the mid-1920s to early 1980s, specimens were collected mainly by faculty, researchers, private collectors and herpetology enthusiasts, including former Professor Branley A. Branson, emeritus Professor Dr. James R. Triplett, Dave Johnson, James Bergant, Robert J. Mangile, Robert Jewell, and Tom R. Johnson. During mid-1990s, the private collector Robert J. Mangile donated his collection to Pittsburg State University, which comprises 232 specimens, representing 14.2% of the HC and at least 55 taxa. After 1982, most specimens were collected and preserved by students taking the herpetology class led by Dr. James R. Triplett, comprising approximately 45% of the collection.

Major data limitations for the HC included incomplete locality data, lack of habitat and weather data, and incomplete date (often only month or year). Most of the limitations were resolved after the digitization of field notebooks, providing a way to access data from different collectors altogether.

Temporal Analysis of Herpetological Specimens

The oldest specimen preserved in the HC is a gravid female of *Gastrophryne carolinensis*, collected on July 11, 1926 by Tylor and J.E.H. White, at DeVall Bluff, in Prairie Co., Arkansas. The most recent specimens preserved include *Anaxyrus americanus*, *Diadophis punctatus arnyi*, and *Plestiodon fasciatus*, collected on May 4, 2013 by herpetology student Ethan Blessent at Schermerhorn Park, 1 mi. S of Galena, Cherokee Co., KS. Only sporadic additions have occurred after 2013, including a female of *Haldea striatula* killed by a domestic cat collected in April 2017 by Wayne Bockelman and Megan Corrigan in Pittsburg, Crawford Co., KS. Another addition was a dead juvenile of *Nerodia rhombifer* on July 16, 2017 by Natalia A. Schneider and Fabio Giacomelli at Mined Land Wildlife Area #8, Crawford Co., Pittsburg, KS. These unexpected finds can be of scientific value. For example, *Haldea striatula* is a species in need of conservation in Kansas, and is present in urban environments vulnerable to many anthropogenic threats, including an abundant predatory species appreciated by most people – the domestic cat.

The monthly collecting peak for all catalogued specimens was in April, totaling 589 specimens, followed by May (248), March (224), July (127), June (115), September (61), August (40), February (39), October (22), January and December (3), and November (1). Specimens lacking the month of collection were categorized as Unknown and totaled 159 individuals. The number of individuals collected per month was plotted along a colored seasonal gradient (Fig. 16). As expected, the highest numbers of individuals were collected during the Spring, a period in which herpetofauna are more active due to favorable temperatures and humidity levels, which also coincides with the reproduction period. Following the behavior of the species, April and May are periods of frequent field trips for herpetology classes. Herpetofauna are still highly active in the summer, however the HC experienced a reduction in collecting effort after 1982, after which students became the main providers of specimens.

Fall and Winter pose more challenges for encountering specimens. Given that herpetology classes at PSU are offered only in Spring semester, the majority of preserved specimens collected from June to January were obtained by researchers, herpetology enthusiasts and private collectors. These data helped provide a broader dataset of herpetofauna throughout the year, and added significant coverage of a larger geographical area. More specimens were collected during Winter compared to Fall, especially during February, when herpetology students already were back in classes and actively collecting specimens.

The highest level of activity occurred in 1964, when 178 individuals were collected (Fig. 16), followed by four other years of relatively high levels of collecting: 1967 (134 specimens); 1968 (128), 1997 (119), and 1987 (116). Specimens that did not have information regarding collecting year were grouped as Unknown, totaling 122 individuals. During the 1960’s former Professor Dr. Branley A. Branson taught Herpetology every other year, as did emeritus Professor Dr. James R. Triplett from 1981 to 2013. Both professors led important collecting field excursions. During 1960’s, PSU also received an important donation from private collector Robert J. Mangile, a resident of Frontenac - Kansas

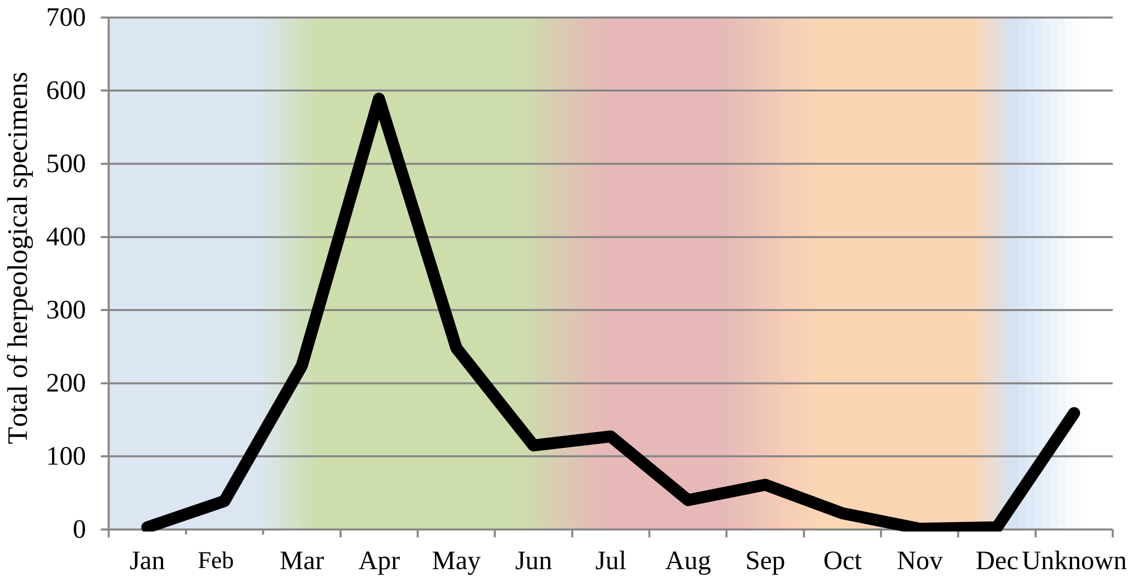

Figure 16. Monthly collecting trends of herpetological specimens, comparing total of individuals collected per month and by the shaded seasonal gradient. Collecting activities peaked in April, with a total of 589 individuals. Vast majority of specimens were collected in the Spring (1,176 individuals; green background), followed by Summer (228; red), Fall (26, orange) and Winter (42; blue). Specimens lacking month of collection were grouped as Unknown, totaling 159 individuals (white background).

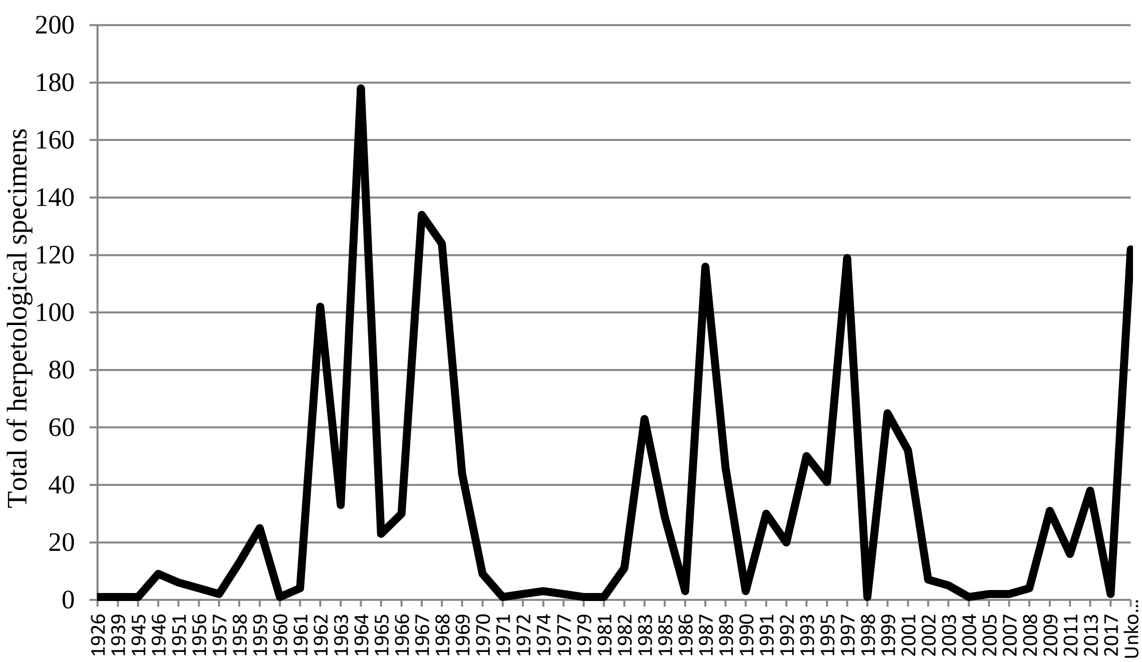

Figure 17. The number of herpetological specimens collected annually. The highest peak was 1964 with 178 individuals, followed by 1967 (134 individuals), 1968 (124), 1997 (119), 1987 (116). Specimens lacking collecting year were grouped as Unknown and totaled 122 individuals. During 1960's and from 1981 to 2013, herpetology classes were offered every other year. Two nearly complete drops-off are observed, from 1971 to 1981 and from 2004 to 2008, which are discussed in the text.

A temporal analysis by decade showed a distinct collecting peak during the 1960s, totaling 673 individuals (Fig. 18). The 1960s had the most comprehensive levels of monthly, with specimens collected each month of the year. Lesser peaks occurred during the 1990s (329 individuals) and the 1980s (269 individuals). The decrease in collecting during 1970's probably was a result of the retirement of former Professor Branley A. Branson and the expansion of The Endangered Species Act in 1973, which established the requirement of collecting permits. After 1982 most collecting coincided with herpetology classes offered during the Spring semester, reflected by the larger number of specimens collected from March through May. Collecting likely declined after the 2000s due to increased restrictions on collecting, given that a larger number of reptiles and amphibians had been federally listed as Threatened or Endangered.

A separate temporal analysis analyzed all specimens collected in Kansas (Fig. 19). It compared the total of reptiles and amphibians collected per decade and per month. As previously observed for the entire HC, the 1960s was the peak decade of collecting in Kansas (315 individuals out of 923), followed by 1990's (256 individuals) and the 1980's (146 individuals). A significant decrease in collecting occurred during the 1970's. Specimens with incomplete date totaled 28. Temporal monthly collecting peaked in April, with a total of 393 individuals, followed by May (145 individuals), and March (142). November was the only month in which no specimen has been collected in Kansas.

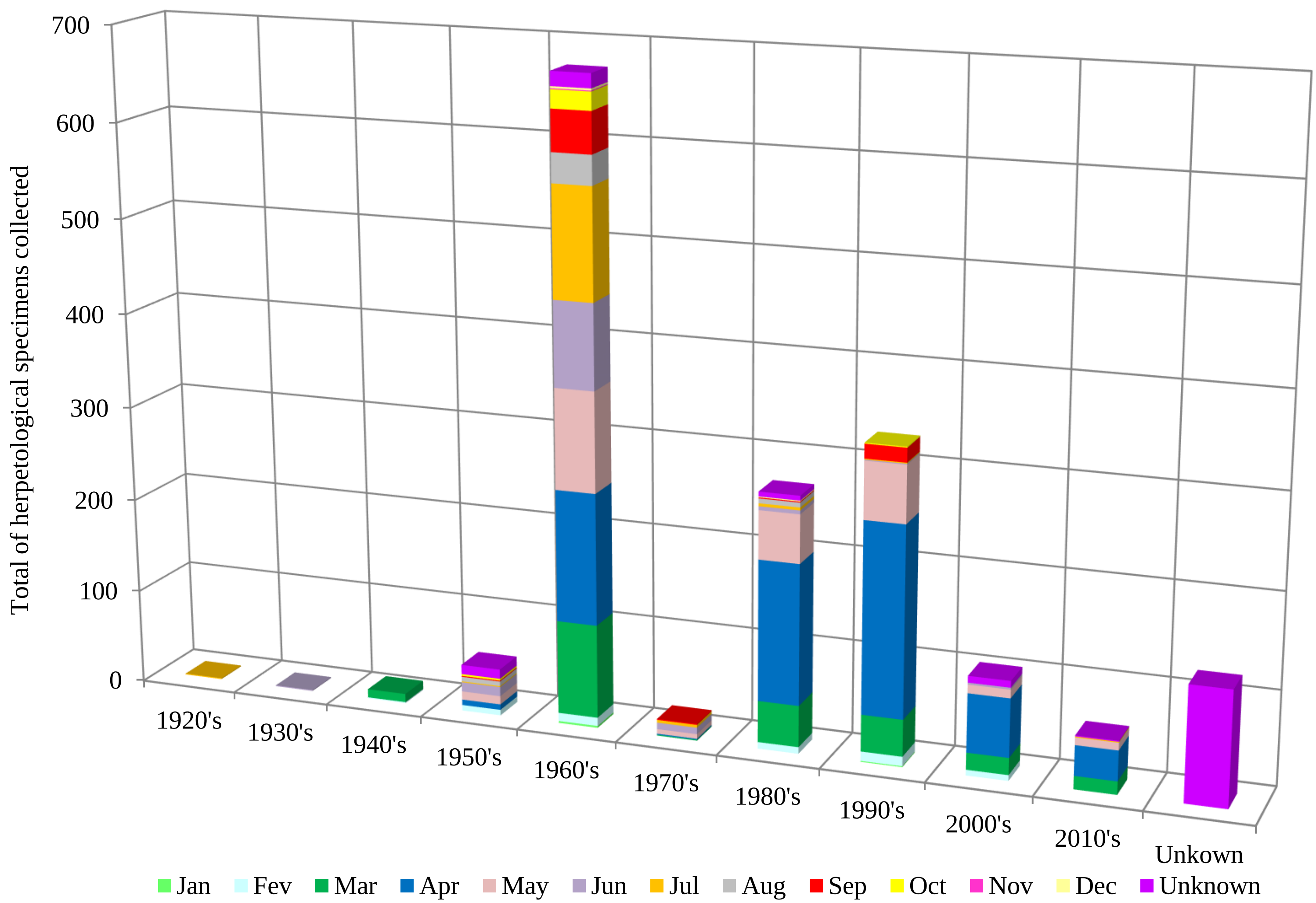

Figure 18. Temporal analysis showin total of herpetological specimens collected by decade and by month. The peak decade was the 1960's, followed by 1990's and 1980's. Specimens with incomplete date were grouped as Unknown, totaling 122 individuals. In the 1960's specimens were collected from all months.

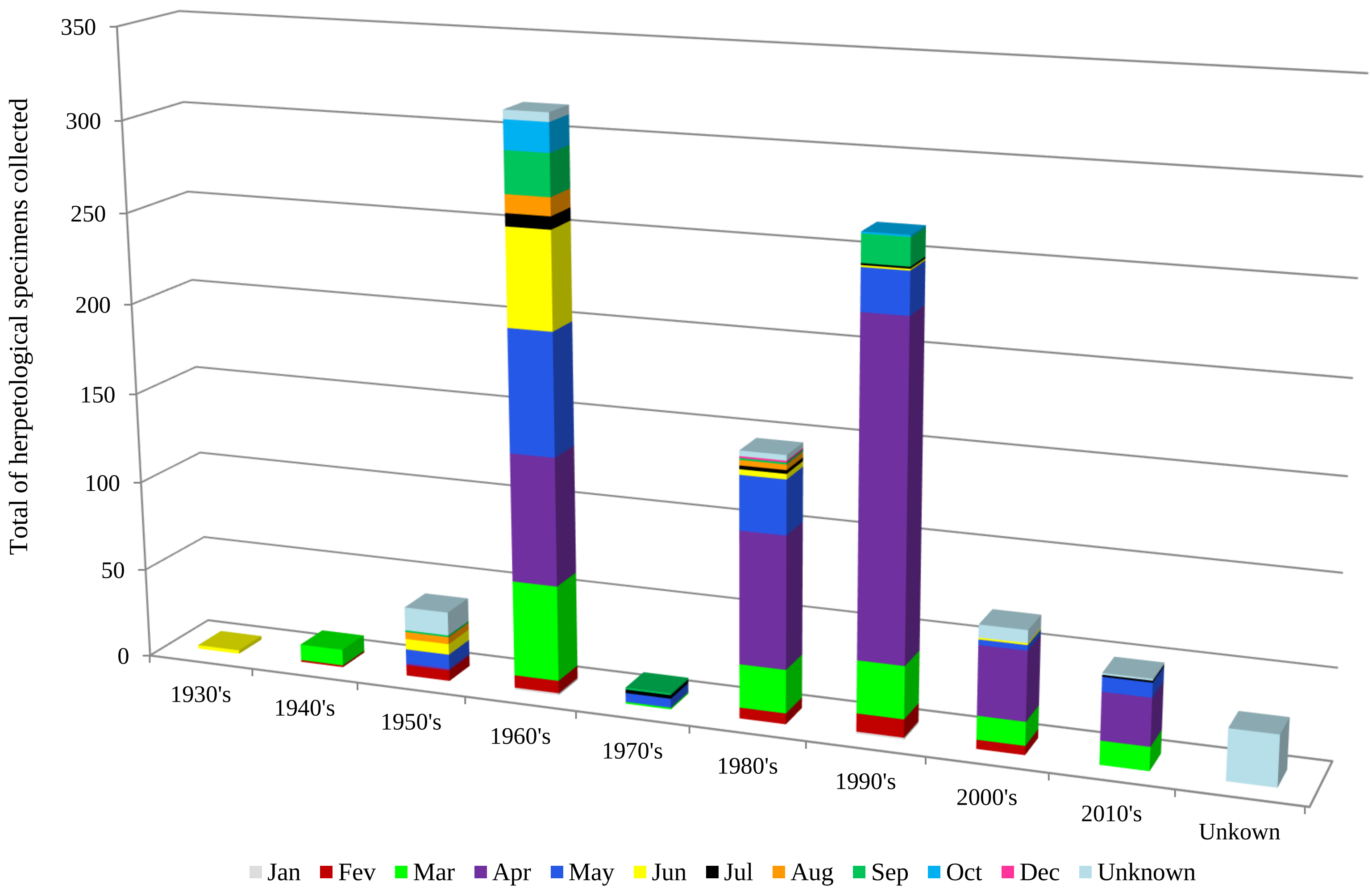

Figure 19. Temporal analysis showing total of herpetological specimens collected by decade and month in Kansas. The peak decade was the 1960's, followed by 1990's and 1980's. Specimens with incomplete dates were grouped as Unknown, totaling 28 individuals. November was the only month without collecting in Kansas.

Summary of Biodiversity in the Herpetology Collection

As of May 2017, the Herpetology Collection housed 181 taxa within five orders: Anura (38 taxa), Caudata (34), Crocodilia (1), Chelonia (18), and Squamata [Lacertilia (55) and Serpentes (55)] (Table 2).

A phylogenetic organization of species and subspecies was created following The Tree of Life Project (Maddison and Schulz, 2007) and Collins *et al.* (2010), including updated taxonomy for both scientific and common names (Crother, 2012; Crother, 2017). The most numerous Anuran is *Spea intermontana* with 98 tadpoles collected at Bodie, Mono Co., California on July 2, 1962. The most numerous Caudata is *Eurycea longicauda melanopleura,* with 38 individuals collected from Arkansas, Missouri, Kansas and Oklahoma. Within Chelonia the most numerous species is *Chyrsemys picta bellii*, with 23 individuals collected in different locations in southeast Kansas. Among Squamata, *Plestiodon fasciatus* is the most numerous Lacertilian totaling 45 individuals, whereas *Diadophis punctatus arnyi* is the most numerous Serpentes with 76 individuals.

Table 2. Phylogenetic list of preserved herpetological specimens at Pittsburg State University including total number of individuals per species.

Kingdom: Animalia
Phylum: Chordata
Superclass: Tetrapoda
  Class: Amphibia
  Subclass: Lissamphibia
  Superorder: Batrachia
    Order: Anura
      Family: Scaphiopodidae
        Scaphiopus couchii - Couch's Spadefoot (1)
        Spea bombifrons - Plains Spadefoot (22)
        Spea intermontana - Great Basin Spadefoot (98)
      Family Bufonidae
        Anaxyrus americanus - American Toad (45)
        Anaxyrus boreas - Western Toad (18)
        Anaxyrus canorus - Yosemite Toad (1)
        Anaxyrus cognatus - Great Plains Toad (2)
        Anaxyrus hemiophrys - Canadian Toad (1)
        Anaxyrus woodhousii - Woodhouse's Toad (11)
        Anaxyrus woodhousii woodhousii - Rocky Mountain Toad (1)

Incilius nebulifer - Gulf Coast Toad (2)
Family Hylidae
Acris blanchardi - Blanchard's Cricket Frog (79)
Acris crepitans - Eastern Cricket Frog (9)
Hyla arenicolor - Canyon Treefrog (1)
Hyla chrysocelis / H. versicolor - Cope's Gray Treefrog / Gray Treefrog (37)
Hyla squirella - Squirrel Treefrog (2)
Pseudacris clarkii - Spotted Chorus Frog (4)
Pseudacris crucifer - Spring Peeper (18)
Pseudacris fouquettei - Cajun Chorus Frog (1)
Pseudacris maculata - Boreal Chorus Frog (46)
Pseudacris triseriata - Western Chorus Frog (7)
Family Ranidae
Lithobates areolatus - Crawfish Frog (4)
Lithobates areolatus circulosus - Northern Crawfish Frog (13)
Lithobates berlandieri - Rio Grande Leopard Frog (1)
Lithobates blairi - Plains Leopard Frog (5)
Lithobates catesbeianus - American Bullfrog (40)
Lithobates clamitans - Green Frog (16)
Lithobates palustris - Pickerel Frog (1)
Lithobates pipiens - Northern Leopard Frog (15)
Lithobates sphenocephalus - Southern Leopard Frog (50)
Lithobates sylvaticus - Wood Frog (2)
Rana boylii - Foothill Yellow-legged Frog (1)
Rana luteiventris - Columbia Spotted Frog (3)
Rana pretiosa - Oregon Spotted Frog (2)
Rana sierrae - Sierra Nevada Yellow-legged Frog (1)
Family Microhylidae
Gastrophryne carolinensis - Eastern Narrow-mouthed Toad (3)
Gastrophryne olivacea - Western Narrow-mouthed Toad (2)
Order: Caudata
Family Cryptobranchidae
Cryptobranchus alleganiensis - Hellbender (3)
Family Siranidae
Siren lacertina - Greater Siren (1)
Family Proteidae
Necturus maculosus - Mudpuppy (3)
Necturus maculosus louisianensis - Red River Mudpuppy (1)
Necturus maculosus maculosus - Common Mudpuppy (1)

Family Rhyacotritonidae

Rhyacotriton olympicus - Olympic Torrent Salamander (1)

Family Salamandridae

Notophthalmus viridescens - Eastern Newt (9)

Notophthalmus viridescens louisianensis - Central Newt (6)

Taricha granulosa - Rough-skinned Newt (2)

Family Plethodontidae

Desmognathus brimleyorum - Ouachita Dusky Salamander (7)

Desmognathus fuscus - Northern Dusky Salamander (6)

Desmognathus monticola - Seal Salamander (3)

Desmognathus quadramaculatus - Black-bellied Salamander (7)

Eurycea longicauda longicauda - Eastern Long-tailed Salamander (2)

Eurycea longicauda melanopleura - Dark-sided Salamander (38)

Eurycea lucifuga - Cave Salamander (19)

Eurycea multiplicata - Many-ribbed Salamander (7)

Eurycea nana - San Marcos Salamander (1)

Eurycea spelaea - Grotto Salamander (7)

Eurycea tynerensis - Oklahoma Salamander (9)

Plethodon albagula - Western Slimy Salamander (4)

Plethodon angusticlavius - Ozark Zigzag Salamander (1)

Plethodon caddoensis - Caddo Mountain Salamander (2)

Plethodon cinereus - Eastern Red-backed Salamander (9)

Plethodon dorsalis - Northern Zigzag Salamander (2)

Plethodon fourchensis - Fourche Mountain Salamander (2)

Plethodon glutinosus - Northern Slimy Salamander (21)

Plethodon ouachitae - Rich Mountain Salamander (5)

Plethodon serratus - Southern Red-backed Salamander (3)

Family Amphiumidae

Amphiuma tridactylum - Three-toed Amphiuma (1)

Family Ambystomatidae

Ambystoma maculatum - Spotted Salamander (4)

Ambystoma mavortium - Western Tiger Salamander (1)

Ambystoma texanum - Small-mouthed Salamander (12)

Ambystoma tigrinum - Eastern Tiger Salamander (4)

Class: Reptilia

Subclass: Anapsida

Order: Testudines

Family: Chelydridae

Chelydra serpentina - Snapping Turtle (10)

Family Kinosternidae

Kinosternon flavescens - Yellow Mud Turtle (5)

Kinosternon subrubrum - Eastern Mud Turtle (1)

Sternotherus odoratus - Eastern Musk Turtle (11)

Family Emydidae

Chrysemys picta - Painted Turtle (2)

Chyrsemys picta bellii - Western Painted Turtle (23)

Chrysemys picta picta - Eastern Painted Turtle (1)

Emydoidea blandingii - Blanding's Turtle (1)

Graptemys pseudogeographica - False Map Turtle (1)

Graptemys pseudogeographica kohnii - Mississippi Map Turtle (2)

Pseudemys concinna - River Cooter (2)

Pseudemys concinna floridana - Coastal Plain Cooter (1)

Terrapene carolina triunguis - Three-toed Box Turtle (17)

Terrapene ornata - Ornate Box Turtle (12)

Trachemys scripta elegans – Red-eared Slider (15)

Family Trionychidae

Apalone ferox - Florida Softshell (1)

Apalone mutica mutica - Midland Smooth Softshell (2)

Apalone spinifera spinifera - Eastern Spiny Softshell (4)

Subclass: Diapsida

Superorder: Crocodylomorpha

Order: Crocodilia

Family Alligatoridae

Caiman crocodilus - Spectacled Caiman (1)

Superorder: Lepidosauria

Order: Squamata

Infraorder: Gekkota

Family Eublepharidae

Eublepharis macularius - Leopard Gecko (1)

Family Gekkonidae

Gecko gecko - Tokay Gecko (1)

Infraorder: Anguimorpha

Family: Anguidae

Elgaria coerulea - Northern Alligator Lizard (1)

Elgaria multicarinata - Southern Alligator Lizard (1)

Infraorder: Sincomorpha

Family Scincidae

Plestiodon anthracinus - Coal Skink (5)

Plestiodon fasciatus - Common Five-lined Skink (45)

Plestiodon laticeps - Broad-headed Skink (12)
Plestiodon obsoletus - Great Plains Skink (38)
Scincella lateralis - Little Brown Skink (33)

Family Teiidae

Aspidoscelis gularis - Common Spotted Whiptail (1)
Aspidoscelis marmorata - Marbled Whiptail (1)
Aspidoscelis sackii - Sack's Spotted Whiptail (1)
Aspidoscelis sexlineata - Six-lined Racerunner (7)
Aspidoscelis sexlineata viridis - Prairie Racerunner (13)
Aspidoscelis tesselata - Common Checkered Whiptail (7)
Aspidoscelis tigris - Tiger Whiptail (5)

Infraorder: Anguimorpha

Family Anguidae

Ophisaurus attenuatus - Slender Glass Lizard (2)
Ophisaurus attenuatus attenuatus - Western Slender Glass Lizard (7)

Infraorder: Iguania

Family Crotaphytidae

Crotaphytus collaris - Eastern Collared Lizard (21)
Gambelia wislizenii - Long-nosed Leopard Lizard (2)

Family Dactyloidae

Anolis carolinensis - Green Anole (2)

Family Phrynosomatidae

Callisaurus draconoides - Zebra-tailed Lizard (2)
Cophosaurus texanus - Greater Earless Lizard (2)
Holbrookia propinqua propinqua - Northern Keeled Earless Lizard (1)
Phrynosoma cornutum - Texas Horned Lizard (6)
Phrynosoma douglasii - Pygmy Short-horned Lizard (1)
Phrynosoma platyrhinos - Desert Horned Lizard (1)
Sceloporus consobrinus - Prairie Lizard (17)
Sceloporus jarrovii - Yarrow's Spiny Lizard (1)
Sceloporus magister - Desert Spiny Lizard (1)
Sceloporus occidentalis - Western Fence Lizard (1)
Sceloporus undulatus - Eastern Fence Lizard (17)
Sceloporus virgatus - Striped Plateau Lizard (4)
Uta stansburiana - Common Side-blotched Lizard (6)
Uta stansburiana stansburiana - Northern Side-blotched Lizard (1)

Suborder: Serpentes

Family Pythonidae

Python reticulatus - Reticulated Python (1)

Family Crotalidae

Agkistrodon contortrix – Eastern Copperhead (9)
Agkistrodon piscivorus - Northern Cottonmouth (4)
Crotalus atrox - Western Diamond-backed Rattlesnake (5)
Crotalus horridus - Timber Rattlesnake (6)
Crotalus viridis - Prairie Rattlesnake (6)
Sistrurus tergeminus - Western Massasauga (8)

Family Natricidae

Haldea striatula - Rough Earthsnake (5)
Nerodia erythrogaster - Plain-bellied Watersnake (23)
Nerodia rhombifer - Diamond-backed Watersnake (8)
Nerodia sipedon - Common Watersnake (8)
Nerodia sipedon sipedon - Northern Watersnake (26)
Regina grahamii - Graham's Crawfish Snake (5)
Storeria dekayi - Dekay's Brownsnake (16)
Storeria dekayi texana - Texas Brownsnake (1)
Storeria occipitomaculata - Red-bellied Snake (4)
Thamnophis elegans - Terrestrial Gartersnake (2)
Thamnophis proximus - Western Ribbonsnake (10)
Thamnophis radix - Plains Gartersnake (1)
Thamnophis sauritus - Eastern Ribbonsnake (1)
Thamnophis sirtalis - Common Gartersnake (27)
Thamnophis sirtalis parietalis - Red-sidded Gartersnake (15)
Thamnophis sirtalis pickeringii - Puget Sound Gartersnake (1)
Thamnophis sirtalis sirtalis - Eastern Gartersnake (2)
Tropidoclonion lineatum - Lined Snake (10)
Virginia valeriae - Smooth Earthsnake (1)

Family Dipsadidae

Carphophis amoenus - Common Wormsnake (10)
Carphophis amoenus amoenus - Eastern Wormsnake (2)
Carphophis vermis - Western Wormsnake (19)
Diadophis punctatus - Ring-necked Snake (8)
Diadophis punctatus arnyi – Prairie Ring-necked Snake (76)
Diadophis punctatus edwardsii - Northern Ring-necked Snake (1)
Diadophis punctatus punctatus - Southern Ring-necked Snake (1)
Diadophis punctatus stictogenys - Mississippi Ring-necked Snake (1)
Heterodon nasicus - Plains Hog-nosed Snake (2)
Heterodon platirhinos - Eastern Hog-nosed Snake (3)

Family Colubridae

Coluber constrictor - North American Racer (17)

Coluber constrictor flaviventris - Eastern Yellow-bellied Racer (20)
Coluber flagellum flagellum - Eastern Coachwhip (3)
Coluber flagellum testaceus - Western Coachwhip (1)
Drymarchon corais corais - Western Indigo Snake (1)
Lampropeltis calligaster - Prairie Kingsnake (26)
Lampropeltis getula - Eastern Kingsnake (1)
Lampropeltis holbrooki - Speckled Kingsnake (11)
Lampropeltis triangulum – Eastern Milksnake (8)
Lampropeltis zonata - California Mountain Kingsnake (1)
Opheodrys aestivus - Rough Greensnake (13)
Opheodrys vernalis - Smooth Greensnake (2)
Pantherophis emoryi - Great Plains Ratsnake (3)
Pantherophis obsoletus - Western Ratsnake (18)
Pituophis catenifer - Gophersnake (1)
Pituophis catenifer sayi - Bullsnake (11)
Sonora semiannulata - Western Groundsnake (1)
Tantilla gracilis - Flat-headed Snake (10)
Tantilla nigriceps - Plains Black-headed Snake (1)

Conservation of Taxa in the Herpetology Collection

Modern scientific collecting is not among the major causes in the decline of herpetofauna species. In fact, collections preserve specimens for a future in which some may become extinct. Thus, continuous but judicious specimen collecting is crucial if we are to understand species and diversity change over time (Winker *et al.*, 1991; Remsen, 1995; Rocha *et al.*, 2014; Warren, 2015).

Fourteen of 188 taxa preserved in the Herpetology Collection are listed on the IUCN Red List as Endangered, Vulnerable or Near-Threatened species; another nine are listed as Least Concern but with decreasing populations. The population data from 42 taxa have not yet been assessed, and 26 taxa need updating over conservation and population status (Appendix C). Of the 14 species mentioned previously, three are Endangered, four are Vulnerable, and seven are Near Threatened. Endangered species include:

Anaxyrus canorus – Yosemite Toad, with population decreasing likely due to chytridiomycosis, airborne contaminants, and livestock grazing (Hammerson *et al.*, 2004); the population trend and conservation status in need of updating.

Rana sierra - Sierra Nevada Yellow-legged Frog, with population decreasing mainly due to predation by introduced non-native fish species, but also due to red-leg disease, chytridiomycosis and airborne agrochemicals (Hammerson, 2008).

Emydoidea blandingii - Blanding's Turtle, with population decreasing due to road mortality and collection for trade; it is the second most common bycatch species of the commercial trapping of Snapping Turtles (van Dijk and Rhodin, 2011).

Species listed as Vulnerable include:

Rana pretiosa - Oregon Spotted Frog, with population decreasing mainly due to introduced American Bullfrog; however, impacts from introduced fish and loss of habitat have been recorded (Hammerson and Pearl, 2004), and population trends and conservation status need updating.

Eurycea nana - San Marcos Salamander, vulnerable due to poor water quality as a result of urbanization and agricultural practices (Hammerson and Chippindale, 2004); population trends and conservation status in need of updating.

Plethodon fourchensis - Fourche Mountain Salamander, has been threatened previously by deforestation but habitat improvement has been recorded; its population trends are unknown and its conservation status needs updating (Hammerson, 2004a).

Rhyacotriton olympicus - Olympic Torrent Salamander, with population decreasing due to increases in mean annual temperature and sedimentation from deforestation (Hammerson, 2004b).

Near Threatened species include: *Lithobates areolatus* (Crawfish Frog); *Rana boylii* (Foothill Yellow-legged Frog); *Cryptobranchus alleganiensis* (Hellbender); *Eurycea tynerensis* (Oklahoma Salamander); *Plethodon caddoensis* (Caddo Mountain Salamander); *Plethodon ouachitae* (Rich Mountain Salamander); and *Terrapene ornata* (Ornate Box Turtle). All except *Plethodon caddoensis* and *P. ouachitae,* are experiencing decreases in population, mainly due to habitat loss and degradation and road

mortality. They all need updating over population trends and conservation status. The latter two are listed as Near Threatened, however population trends are unknown for *P. caddoensis* but are currently stable for *P. ouachitae.*

Biodiversity levels in Kansas of the Herpetological Collection

Biodiversity and conservation data were analyzed separately for specimens collected in Kansas. A total of 81 taxa were collected throughout the state (Table 3) belonging to four orders: Anura (14 taxa), Caudata (8), Chelonia (14), Squamata (Lacertilia with 11 taxa, and Serpentes with 34 taxa). The most numerous specimens were *Acris blanchardi* (78 specimens), *Diadophis punctatus* (74), *Plestiodon obsoletus* (38), *Chrysemys picta bellii* (23), and *Ambystoma texanum* (8).

Of the 81 taxa, five are Threatened, one is Endangered, six are in need of conservation (i.e. SINC) and four are species in need of information (i.e. SINI) (KDWPT, 2017; Taggart, 2017) (Appendix C). The Endangered species *Eurycea spelaea* is known only from caves and springs in the extreme southeast portion of Kansas in the Ozark Plateau ecoregion. Two threatened species are restricted to the southeast corner of Cherokee County, these being *Gastrophryne carolinensis* – Eastern Narrow-mouthed Toad, and *Eurycea longicauda melanopleura* - Dark-sided Salamander; the other three taxa are found at the extreme east portion, mostly restricted to the counties that border Missouri: *Lithobates clamitans* – Green Frog, *Notophthalmus viridescens louisianensis* - Central Newt, and *Plestiodon laticeps* – Broad-headed Skink. Species in need of conservation are *Lithobates areolatus* – Crawfish Frog, *Crotalus horridus* – Timber Rattlesnake, *Haldea striatula* – Rough Earthsnake, *Heterodon nasicus* - Plains Hog-nosed Snake, *Heterodon platirhinos* - Eastern Hog-nosed Snake, and *Virginia valeriae* – Smooth Earthsnake.

Table 3. Phylogenetic list of herpetological specimens collected in Kansas preserved at the Herpetology Collection at Pittsburg State University including total number of individuals per species.

Kingdom: Animalia
Phylum: Chordata
Superclass: Tetrapoda
    Class: Amphibia
    Subclass: Lissamphibia
    Superorder: Batrachia
        Order: Anura
            Family: Scaphiopodidae
                Spea bombifrons - Plains Spadefoot (19)
            Family Bufonidae
                Anaxyrus americanus - American Toad (29)
                Anaxyrus cognatus - Great Plains Toad (2)
                Anaxyrus woodhousii - Woodhouse's Toad (5)
            Family Hylidae
                Acris blanchardi - Blanchard's Cricket Frog (78)
                Hyla chrysocelis / H. versicolor - Cope's Gray Treefrog / Gray Treefrog (11)
                Pseudacris maculata - Boreal Chorus Frog (40)
            Family Ranidae
                Lithobates areolatus circulosus - Northern Crawfish Frog (13)
                Lithobates blairi - Plains Leopard Frog (3)
                Lithobates catesbeianus - American bullfrog (34)
                Lithobates clamitans - Green Frog (1)
                Lithobates sphenocephalus - Southern Leopard Frog (48)
            Family Microhylidae
                Gastrophryne carolinensis - Eastern Narrow-mouthed Toad (1)
        Order: Caudata
            Family Proteidae
                Necturus maculosus - Mudpuppy (1)
                Necturus maculosus maculosus - Common Mudpuppy (1)
            Family Salamandridae
                Notophthalmus viridescens louisianensis - Central Newt (3)
            Family Plethodontidae
                Eurycea longicauda melanopleura - Dark-sided Salamander (3)
                Eurycea spelaea - Grotto Salamander (1)
            Family Ambystomatidae
                Ambystoma mavortium - Western Tiger Salamander (1)
                Ambystoma texanum - Small-mouthed Salamander (8)
                Ambystoma tigrinum - Eastern Tiger Salamander (1)

Class: Reptilia
Subclass: Anapsida
Order: Testudines
Family: Chelydridae
Chelydra serpentina - Snapping Turtle (8)
Family Kinosternidae
Kinosternon flavescens - Yellow Mud Turtle (1)
Sternotherus odoratus - Eastern Musk Turtle (9)
Family Emydidae
Chyrsemys picta bellii - Western Painted Turtle (23)
Chrysemys picta picta - Eastern Painted Turtle (1)
Graptemys pseudogeographica - False Map Turtle (1)
Graptemys pseudogeographica kohnii - Mississippi Map Turtle (1)
Pseudemys concinna - River Cooter (2)
Terrapene carolina triunguis - Three-toed Box Turtle (17)
Terrapene ornata - Ornate Box Turtle (10)
Trachemys scripta elegans – Red-eared Slider (14)
Family Trionychidae
Apalone ferox - Florida Softshell (1)
Apalone mutica mutica - Midland Smooth Softshell (2)
Apalone spinifera spinifera - Eastern Spiny Softshell (3)
Subclass: Diapsida
Superorder: Lepidosauria
Order: Squamata
Infraorder: Gekkota
Family Eublepharidae
Eublepharis macularius - Leopard Gecko (1)
Infraorder: Sincomorpha
Family Scincidae
Plestiodon anthracinus - Coal Skink (4)
Plestiodon fasciatus - Common Five-lined Skink (36)
Plestiodon laticeps - Broad-headed Skink (6)
Plestiodon obsoletus - Great Plains Skink (38)
Scincella lateralis - Little Brown Skink (20)
Family Teiidae
Aspidoscelis sexlineata viridis - Prairie Racerunner (12)
Infraorder: Anguimorpha
Family Anguidae
Ophisaurus attenuatus attenuatus - Western Slender Glass Lizard (7)

Infraorder: Iguania

Family Crotaphytidae

Crotaphytus collaris - Eastern Collared Lizard (16)

Family Phrynosomatidae

Phrynosoma cornutum - Texas Horned Lizard (2)

Sceloporus consobrinus - Prairie Lizard (15)

Suborder: Serpentes

Family Pythonidae

Python reticulatus - Reticulated Python (1)

Family Crotalidae

Agkistrodon contortrix – Eastern Copperhead (6)

Crotalus horridus - Timber Rattlesnake (5)

Crotalus viridis - Prairie Rattlesnake (5)

Sistrurus tergeminus - Western Massasauga (8)

Family Natricidae

Haldea striatula - Rough Earthsnake (5)

Nerodia erythrogaster - Plain-bellied Watersnake (17)

Nerodia rhombifer - Diamond-backed Watersnake (7)

Nerodia sipedon sipedon - Northern Watersnake (22)

Regina grahamii - Graham's Crawfish Snake (5)

Storeria dekayi - Dekay's Brownsnake (15)

Thamnophis proximus - Western Ribbonsnake (9)

Thamnophis sirtalis - Common Gartersnake (22)

Thamnophis sirtalis parietalis - Red-sidded Gartersnake (11)

Tropidoclonion lineatum - Lined Snake (10)

Virginia valeriae - Smooth Earthsnake (1)

Family Dipsadidae

Carphophis vermis - Western Wormsnake (17)

Diadophis punctatus - Ring-necked Snake (3)

Diadophis punctatus arnyi – Prairie Ring-necked Snake (74)

Heterodon nasicus - Plains Hog-nosed Snake (2)

Heterodon platirhinos - Eastern Hog-nosed Snake (2)

Family Colubridae

Coluber constrictor - North American Racer (15)

Coluber constrictor flaviventris - Eastern Yellow-bellied Racer (19)

Coluber flagellum flagellum - Eastern Coachwhip (3)

Coluber flagellum testaceus - Western Coachwhip (1)

Lampropeltis calligaster - Prairie Kingsnake (19)

Lampropeltis holbrooki - Speckled Kingsnake (8)

Lampropeltis triangulum - Milksnake (6)

Opheodrys aestivus - Rough Greensnake (12)
Pantherophis emoryi - Great Plains Ratsnake (2)
Pantherophis obsoletus - Western Ratsnake (17)
Pituophis catenifer sayi - Bullsnake (10)
Sonora semiannulata - Western Groundsnake (1)
Tantilla gracilis - Flat-headed Snake (9)

New County Records of Reptiles and Amphibians in Kansas

The location records of all specimens collected in Kansas were cross-referenced to data available at the Kansas Herpetological Atlas and at VertNet (2016) – an online database for biological collections worldwide. The comparison resulted in 16 new county records for Kansas, including:

- *Apalone spinifera spinifera* (Crawford Co., 1987) (Fig. 38);
- *Apalone mutica mutica* (Neosho Co., 2 specimens, 1967) (Fig. 38);
- *Crotalus viridis* (Reno Co., 1972) (Fig. 47);
- *Heterodon nasicus* (Crawford Co., 1968) (Fig. 59);
- *Heterodon platirhinos* (Crawford Co., 1965) (Fig. 59);
- *Kinosternon flavescens* (Crawford Co., 1968) (Fig. 33);
- *Plestiodon anthracinus* (Crawford Co., 1967) (Fig. 39);
- *Plestiodon anthracinus* (Bourbon Co., 1999) (Fig. 39);
- *Plestiodon laticeps* (Lyon Co., 1997) (Fig. 39);
- *Pseudemys concinna* (Crawford Co., 2 specimens, 1967 and 1982) (Fig. 33);
- *Regina grahamii* (Crawford Co., 1967) (Fig. 51);
- *Sceloporus consobrinus* (Crawford Co., 1 specimen 1958, 2 spec. 1997, 1 spec 2009) (Fig. 46);
- *Sceloporus consobrinus* (Miami Co., 2 specimens, 1995) (Fig. 46);
- *Sternotherus odoratus* (Chautauqua Co., 1995) (Fig. 38);
- *Tropidoclonion lineatum* (Comanche Co., 1981) (Fig. 56);
- *Virginia valeriae* (Bourbon Co., 1992) (Fig. 49);

Three species are classified as Species In Need of Conservation (*H. nasicus, H. platirhinos* and *V. valeriae)*, and one is classified as Threatened (*P. laticeps*), reiterating the importance of natural history collections' data for better documenting and

understanding local patterns of biodiversity, as well as the importance of digitizing and publishing collections' data so that they are available globally.

Besides new county records, the HC at PSU will provide the Kansas Herpetological Atlas and similar databases with additional records to enhance the composition of species distribution maps and biodiversity data. For example, *Ambystoma texanum* previously had only one record from Crawford Co., but seven additional new location records are at HC. As another example, *Chrysemys picta bellii* had only two records for Crawford Co., but the HC has 20 new records to add. For *Coluber constrictor*, the six records for Crawford Co. will be joined with 19 new records at HC. Finally, 14 additional records of *Nerodia sipedon sipedon* will be added to the three previous records for Crawford Co.

New length records for Amphibians and Reptiles

As specimens are prepared for preservation some standard body measurements should be recorded. One specimen of Three-toad Box Turtle (*Terrapene carolina triunguis*) had a notation on its tag indicating it was a large specimen. Measuring 156 mm, the female was 31 mm larger than the average size (Powell *et al.,* 2016), and according to records available (Sullivan and Roth, 1989; Collins *et al.*, 2010; UMMZ, 2015a, b, c), it possibly is second largest specimen ever recorded on collection databases (Kansas Herpetofaunal Atlas and VertNet). After this discovery, all larger specimens of each species were re-measured to check for new length records. Six new length records were found for the state of Kansas, and four new national length records (Table 4).

Table 4. New length records for Amphibians and Reptiles in Kansas

Species	Location	Total Length (mm)	Kansas Size Record (mm)	U.S. Size Record (mm)
Acris blanchardi	Crawford Co., KS	35	33	38
Lithobates areolatus circulosus	Crawford Co., KS	223	122	122
Lithobates catesbeianus	Crawford Co., KS	275	185	220
Lithobates sphenocephalus	Crawford Co., KS	222	87	127
Coluber flagellum	Meade Co., KS	1860	1822	2590
Lampropeltis c. calligaster	Bourbon Co., KS	1427	1324	1430
Tropidoclonion lineatum	Jefferson Co., KS	672	446	544
Haldea striatula	Crawford Co., KS	307	290	324
Sceloporus consobrinus	Cherokee Co., KS	173	165	191
Sternotherus odoratus	Chautauqua Co., KS	124	114	150

Temporal and Spatial Distribution of Amphibians and Reptiles in Kansas

To use distributional data much beyond state or county-level distribution maps, georeferencing each specimen is necessary. The more accurate a locality description the better; however, like most historical collections prior to approximately the mid to late 1980s, specimens in the HC lack GPS or UTM locations. Instead, they mostly have a description of the collecting location using cardinal coordinates along with miles, or use the American surveying system of Township, Range and Section. (The latter is virtually unintelligible outside of the United States in the absence of a method of conversion to more standard units.) Unfortunately, collectors occasionaly were lax in their descriptions (as was more commonly accepted at the time), and only indicated which County and State the specimen was collected, thus creating limitations of spatial data. Such limitations sometimes can be corrected by the description of the collecting site, or by having access to the collector's field notebook or publications.

Fortunately, less than 10% of Kansas specimens preserved at the HC had serious levels of spatial data limitation, allowing for valuable spatiotemporal maps of preserved specimens to be generated (Figs. 20 – 66). For locations that only had Township, Range and Section (TRS henceforth), the collecting error radius average was approximately 800

m. For locations that included TRS, along with cardinal coordinates and distance in miles from a point, the collecting error radius average varied between approximately 60 m and 400 m. The error radius for GPS locations averaged approximately 70 m.

A strong spatial bias was identified among the HC collections. As noted above for Figure 13, specimens were collected mostly near cities and along roads. This is understandable, given frequent time and funding limitations. Even so and with the spatial bias, the maps can be an important source for aiding future monitoring studies, and for research projects for prioritizing study sites that have not been thoroughly surveyed. Additionally, Winker (2004) points out that few collections of urban and suburban specimens exist mostly due to the fact that biodiversity science generally samples from natural areas, not urban environments. Considering the levels of urbanization in today's world it is likely that the value of collections like these is probably quite high.

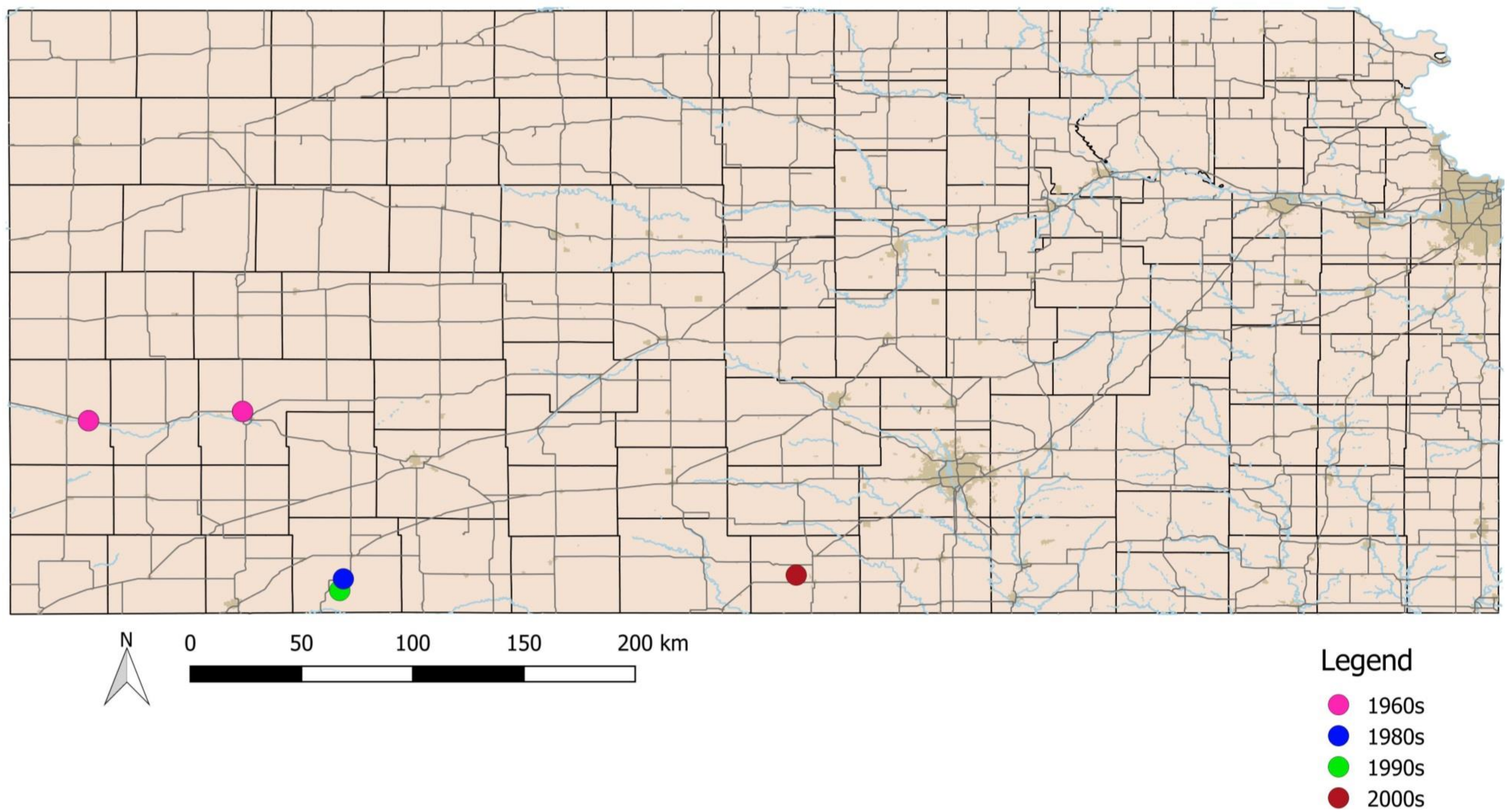

Figure 20. Spatial and temporal distribution of collecting sites of *Spea bombifrons* (Plains Spadefoot).

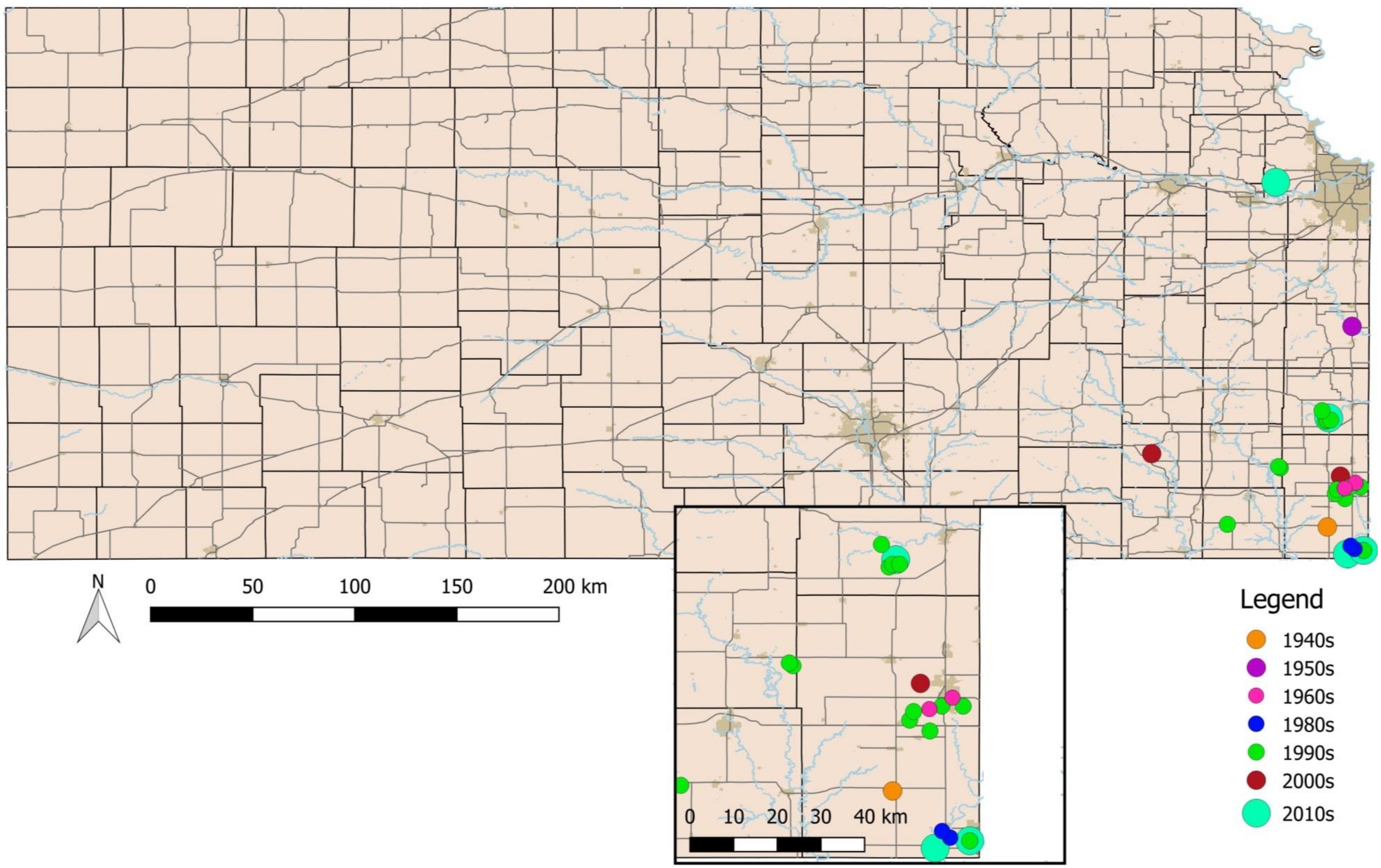

Figure 21. Spatial and temporal distribution of collecting sites of *Anaxyrus americanus* (American Toad).

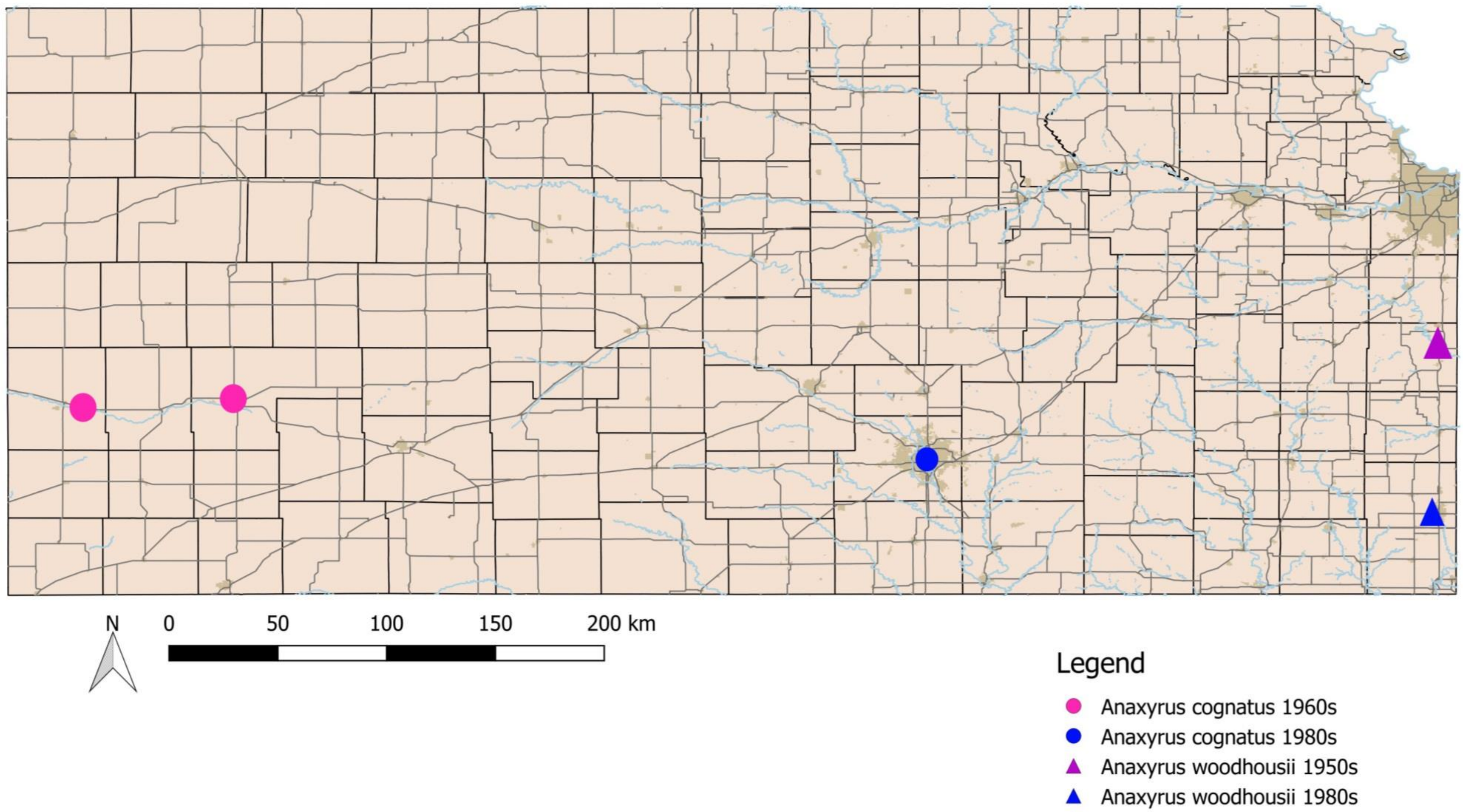

Figure 22. Spatial and temporal distribution of collecting sites of *Anaxyrus cognatus* (Great Plains Toad) and *Anaxyrus woodhousii* (Woodhouse's Toad).

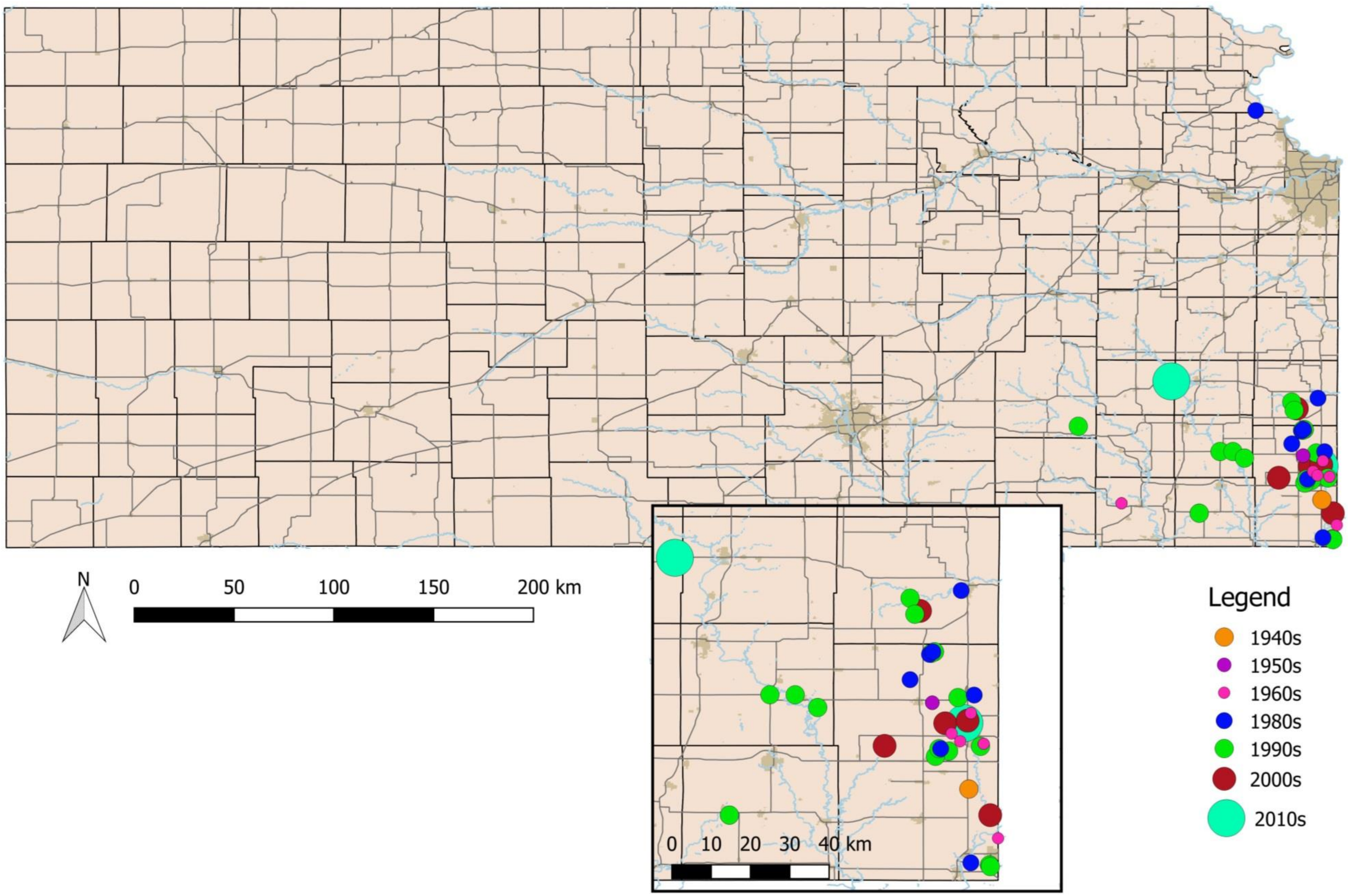

Figure 23. Spatial and temporal distribution of collecting sites of *Acris blanchardi* (Blanchard's Cricket Frog).

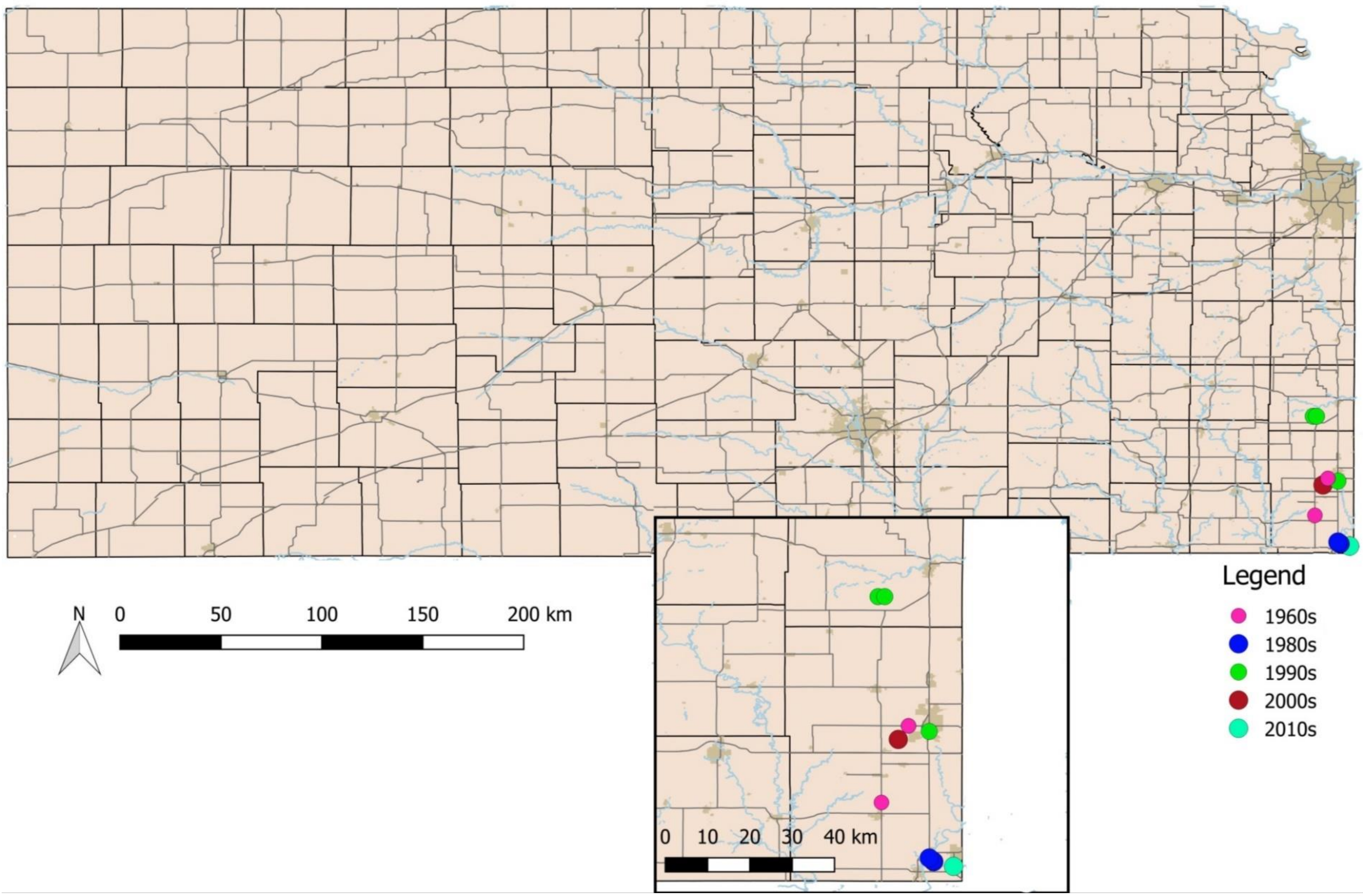

Figure 24. Spatial and temporal distribution of collecting sites of *Hyla chrysocelis / Hyla versicolor* (Cope's Gray Treefrog / Gray Treefrog).

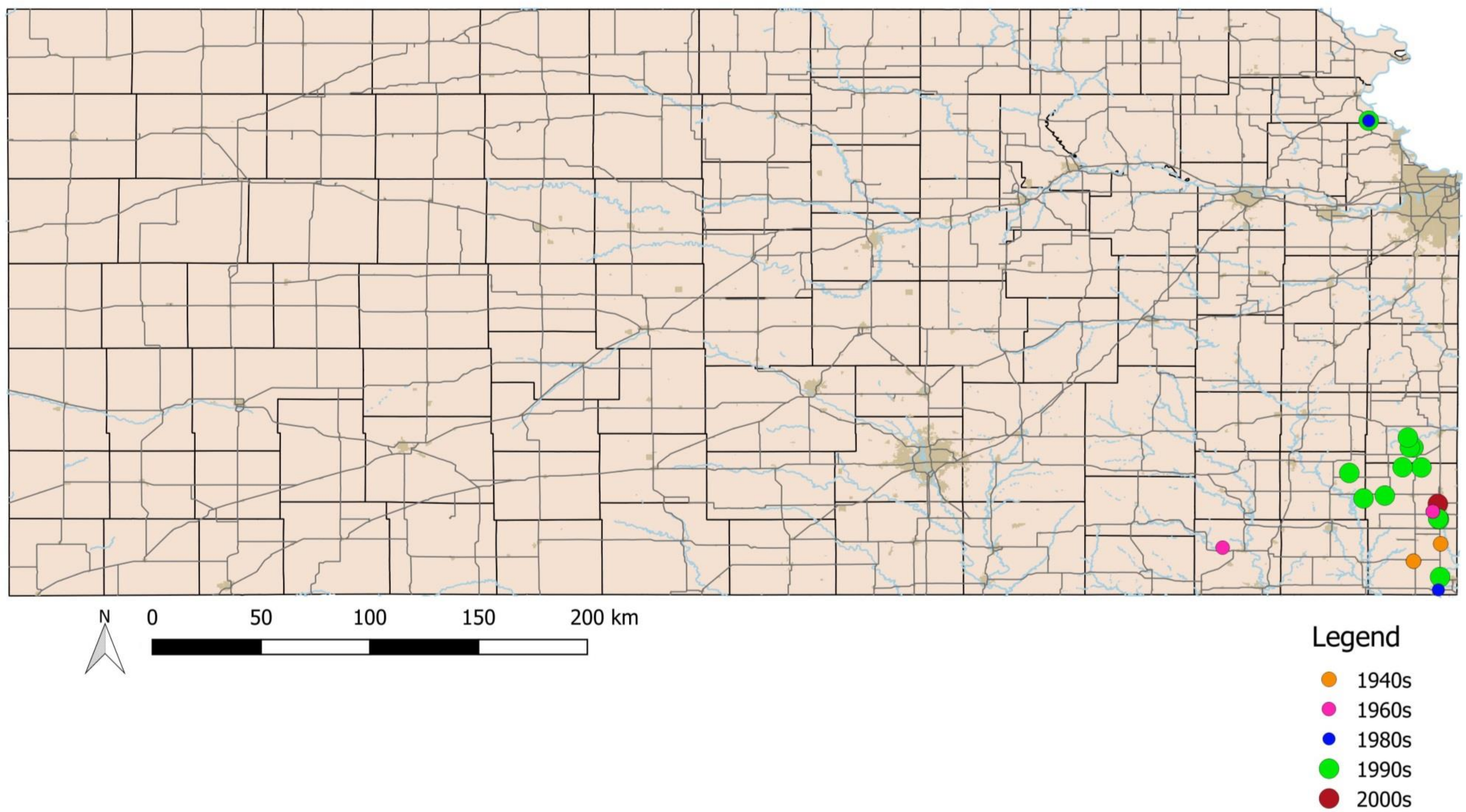

Figure 25. Spatial and temporal distribution of collecting sites of *Pseudacris maculata* (Boreal Chorus Frog).

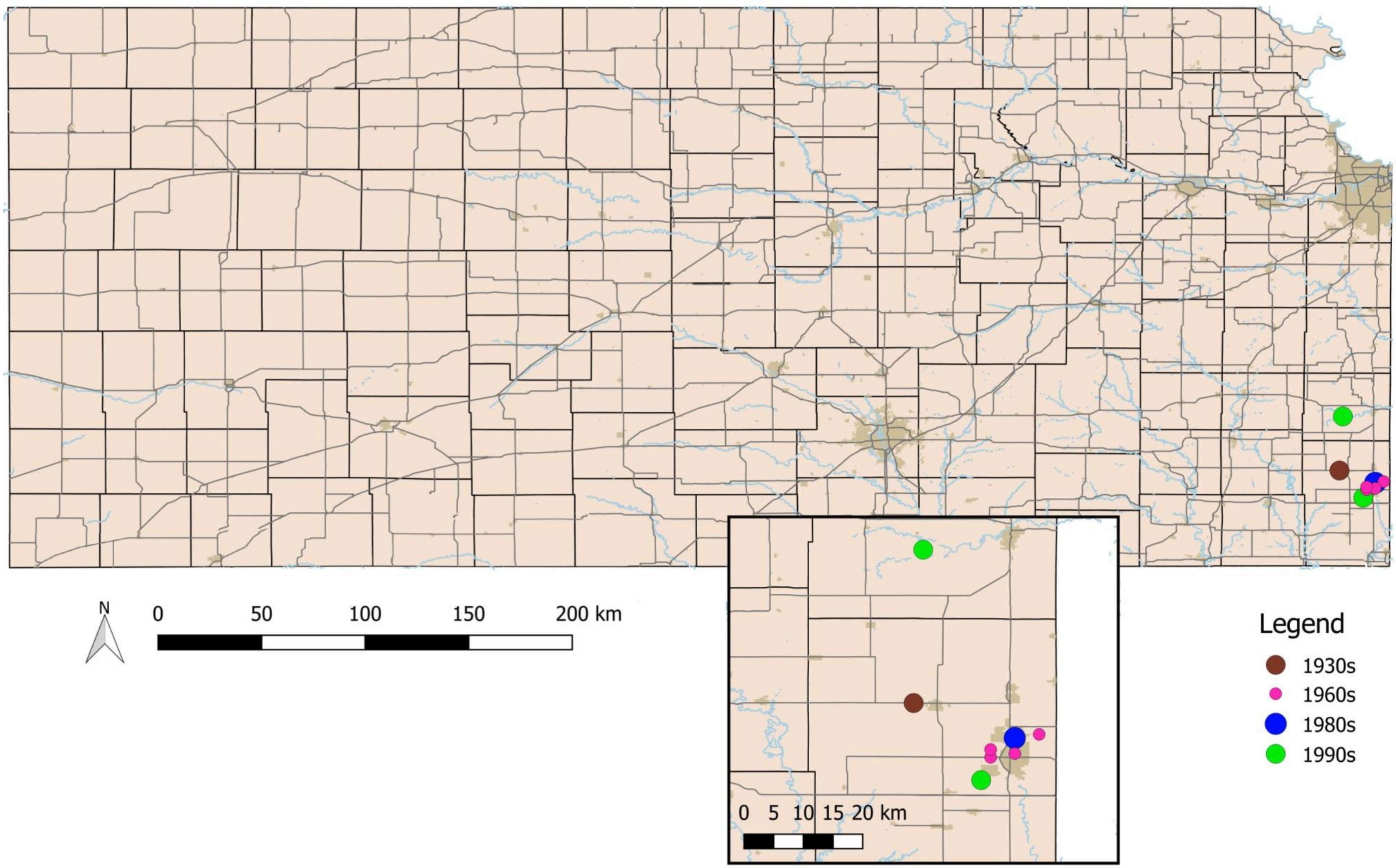

Figure 26. Spatial and temporal distribution of collecting sites of *Lithobates areolatus circulosus* (Northern Crawfish Frog). *Lithobates a. circulosus* is a species in need of conservation in Kansas.

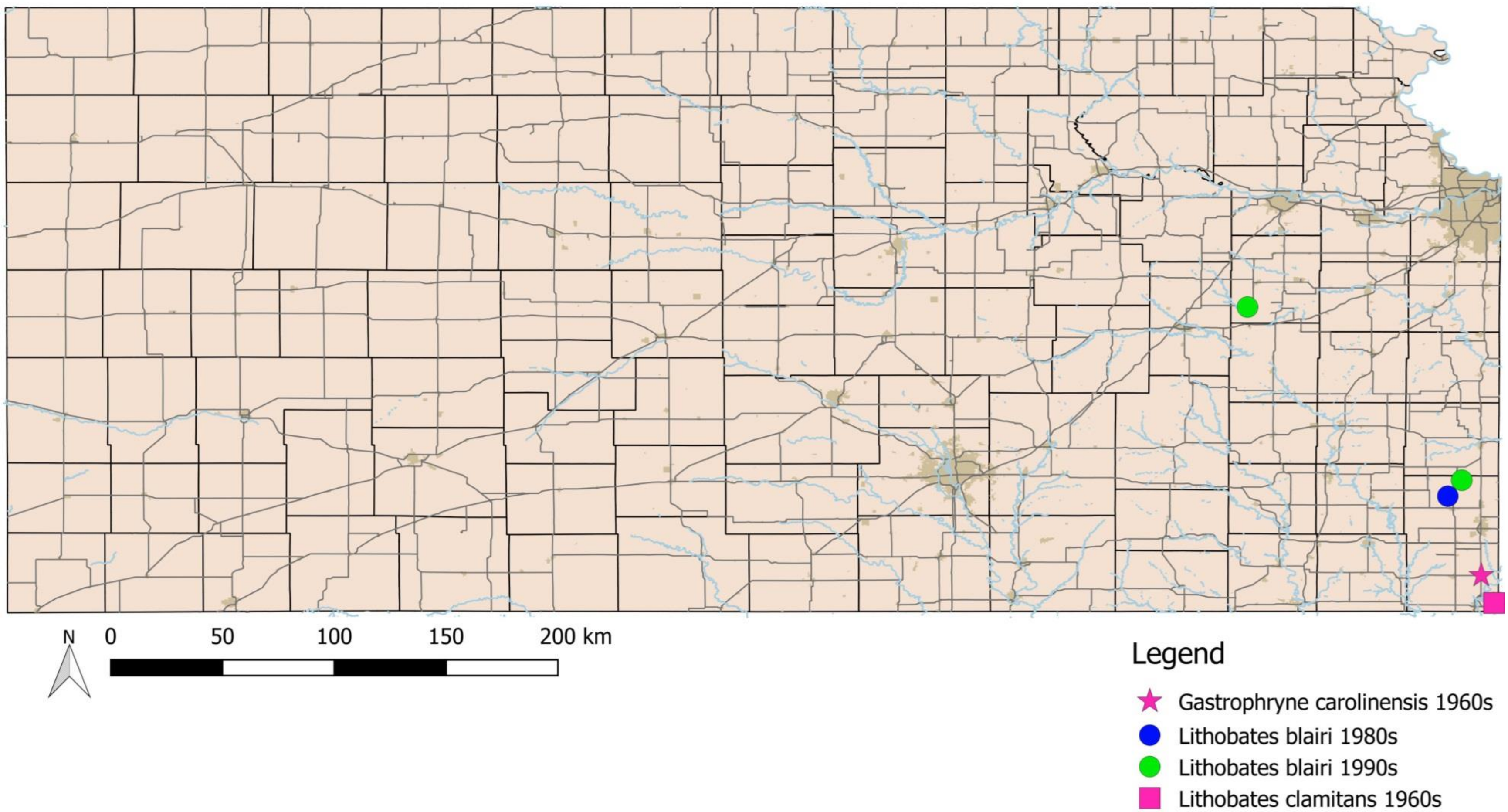

Figure 27 Spatial and temporal distribution of collecting sites of *Lithobates blairi* (Plains Leopard Frog), *Lithobates clamitans* (Green Frog), and *Gastrophryne carolinensis* (Eastern Narrow-mouthed Toads). *Lithobates clamitans* and G. *carolinensis* are Threatened in Kansas.

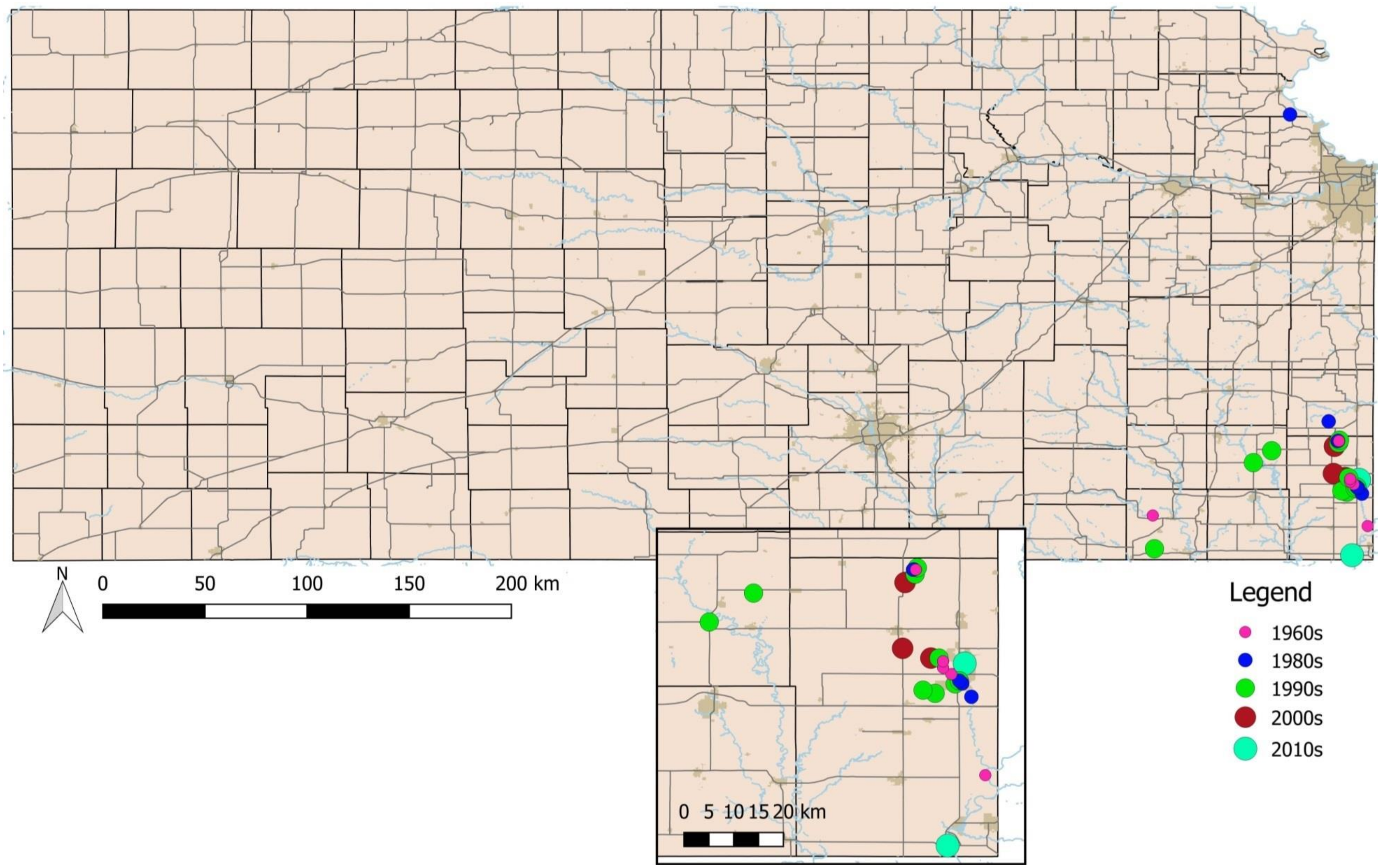

Figure 28. Spatial and temporal distribution of collecting sites of *Lithobates catesbeianus* (American Bullfrog).

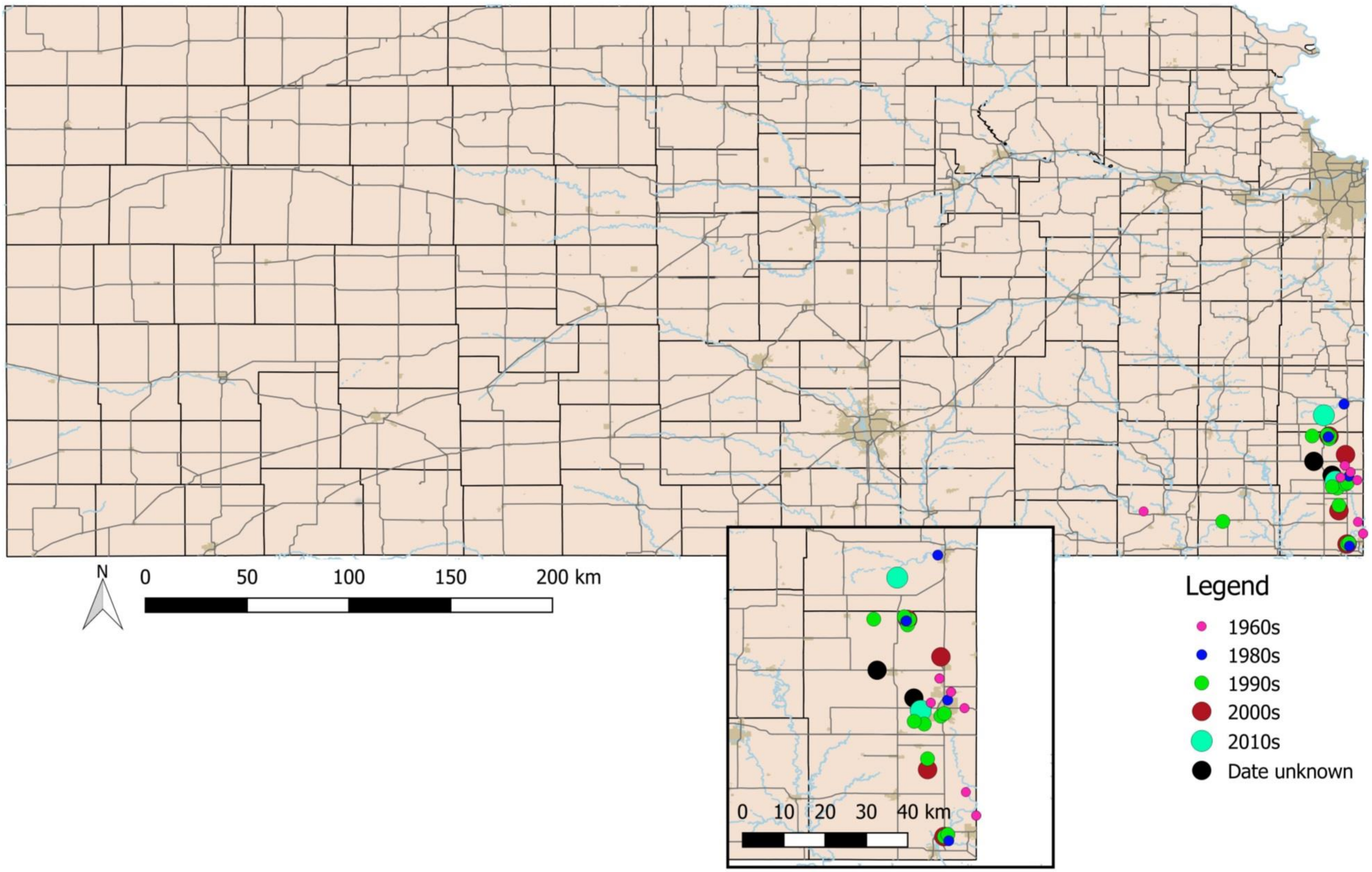

Figure 29. Spatial and temporal distribution of collecting sites of *Lithobates sphenocephalus* (Southern Leopard Frog).

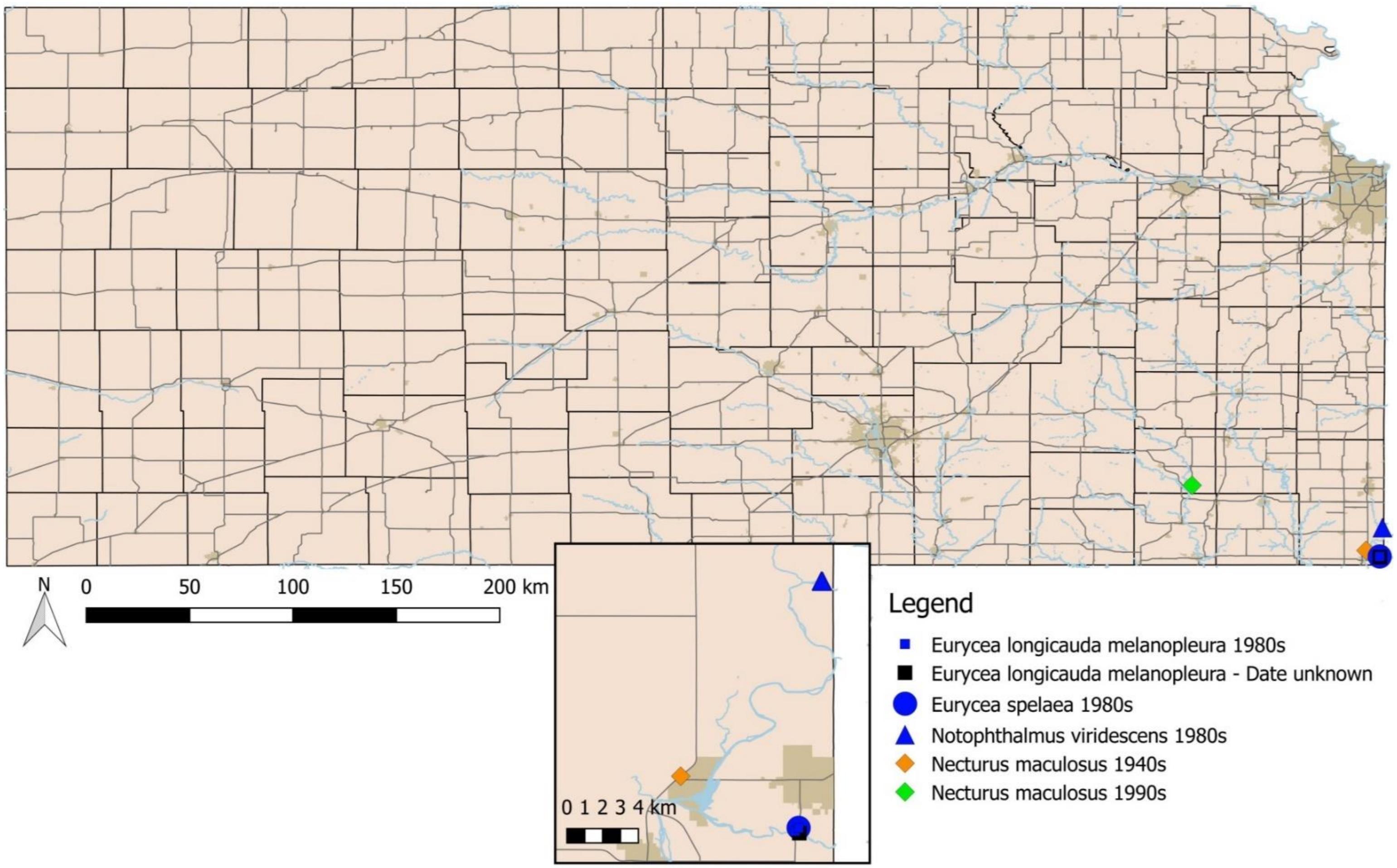

Figure 30. Spatial and temporal distribution of collecting sites of *Necturus maculosus* (Mudpuppy), *Notophthalmus viridescens louisianensis* (Central Newt), *Eurycea longicauda melanopleura* (Dark-sided Salamander), and *Eurycea spelaea* (Grotto Salamander). *Eurycea l. melanopleura* is Threatened, *E. spaleae* is Endangered, *N. viridescens* is Threatened, and *N. maculosus* is a species in need of information in Kansas.

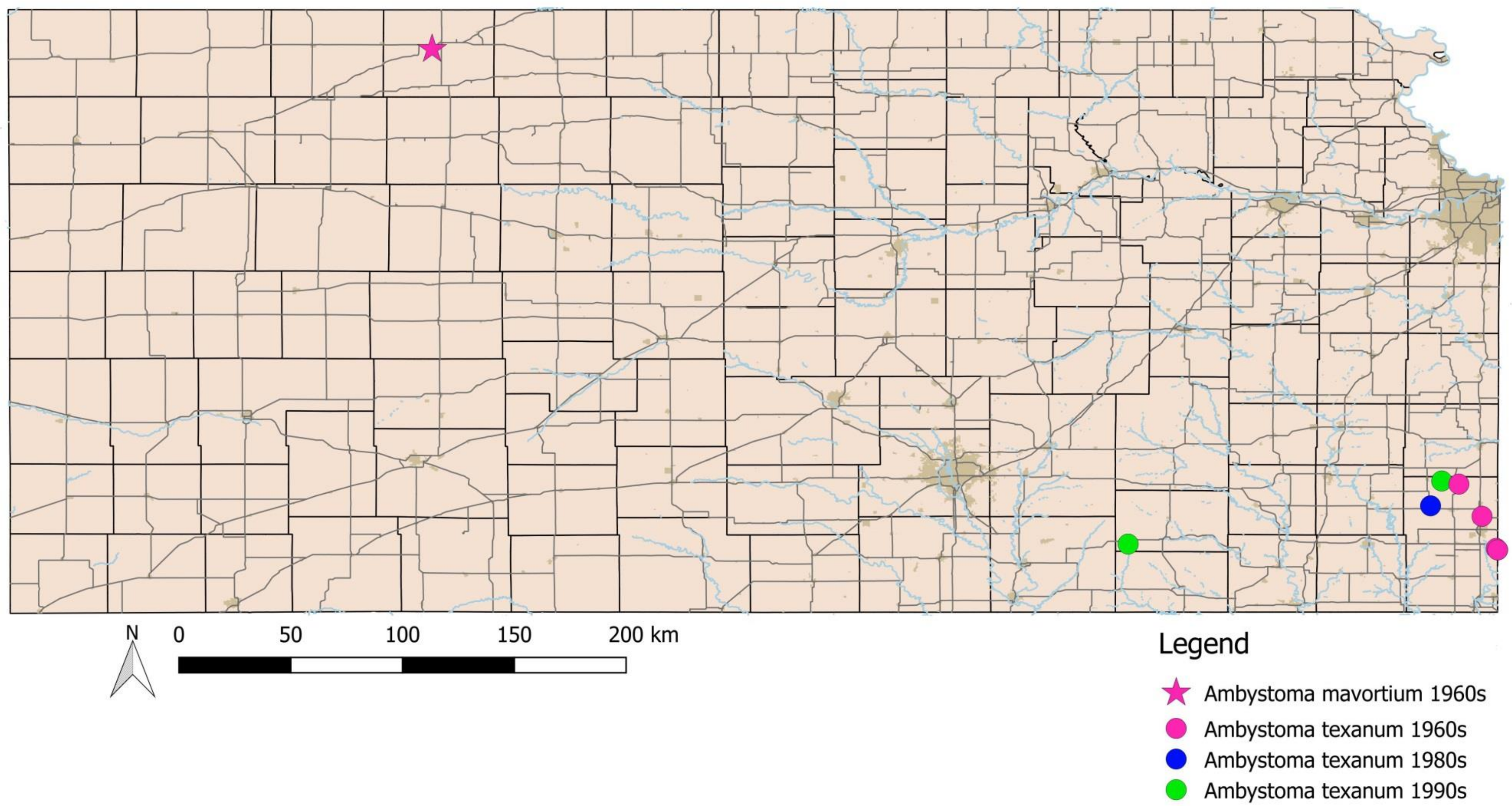

Figure 31. Spatial and temporal distribution of collecting sites of *Ambystoma mavortium* (Western Tiger Salamander) and *Ambystoma texanum* (Small-mouthed Salamander).

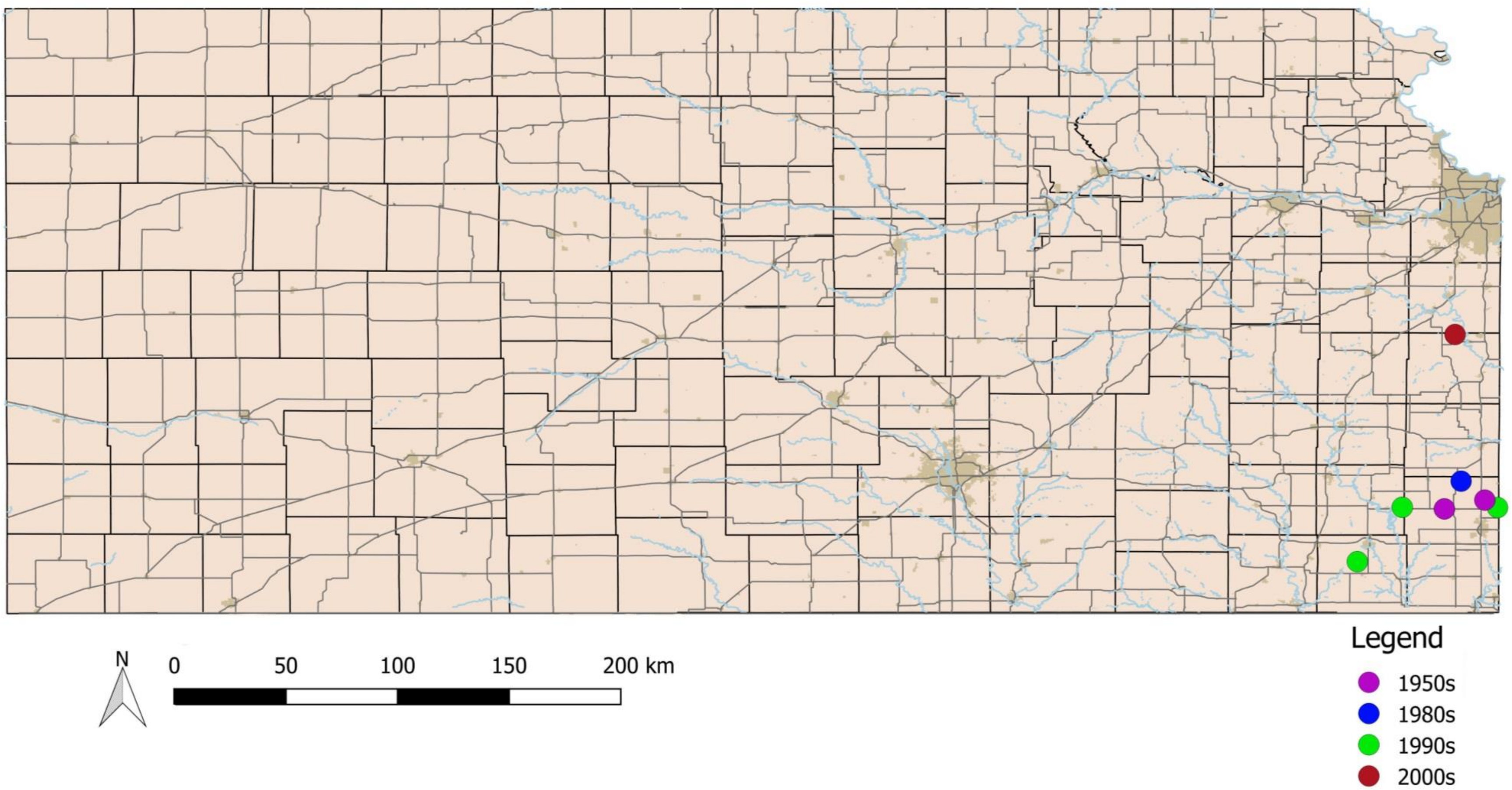

Figure 32. Spatial and temporal distribution of collecting sites of *Chelydra serpentina* (Snapping Turtle).

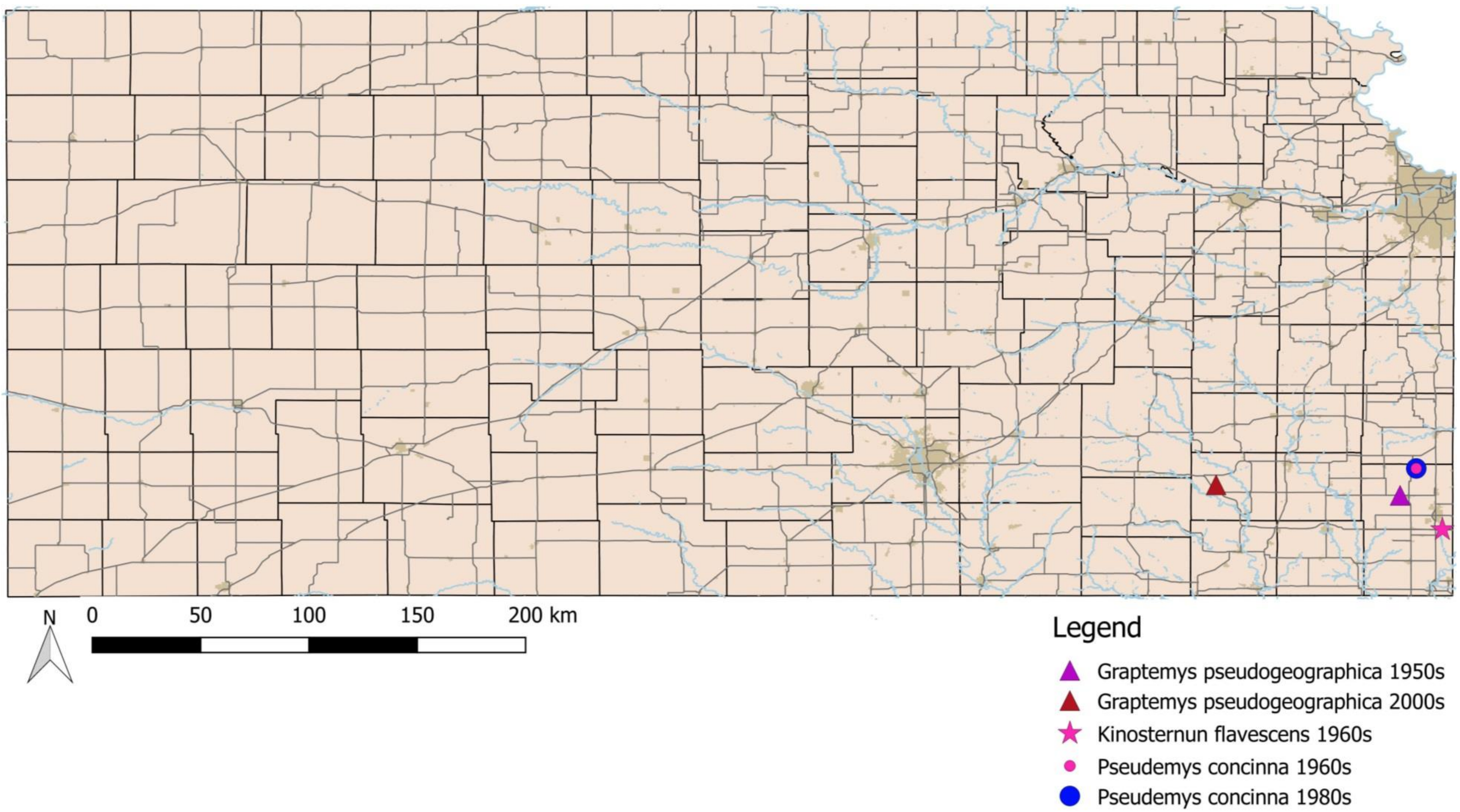

Figure 33. Spatial and temporal distribution of collecting sites of *Kinosternon flavescens* (Yellow Mud Turtle), *Graptemys pseudogeographica* (False Map Turtle), and *Pseudemys concinna* (River Cooter).

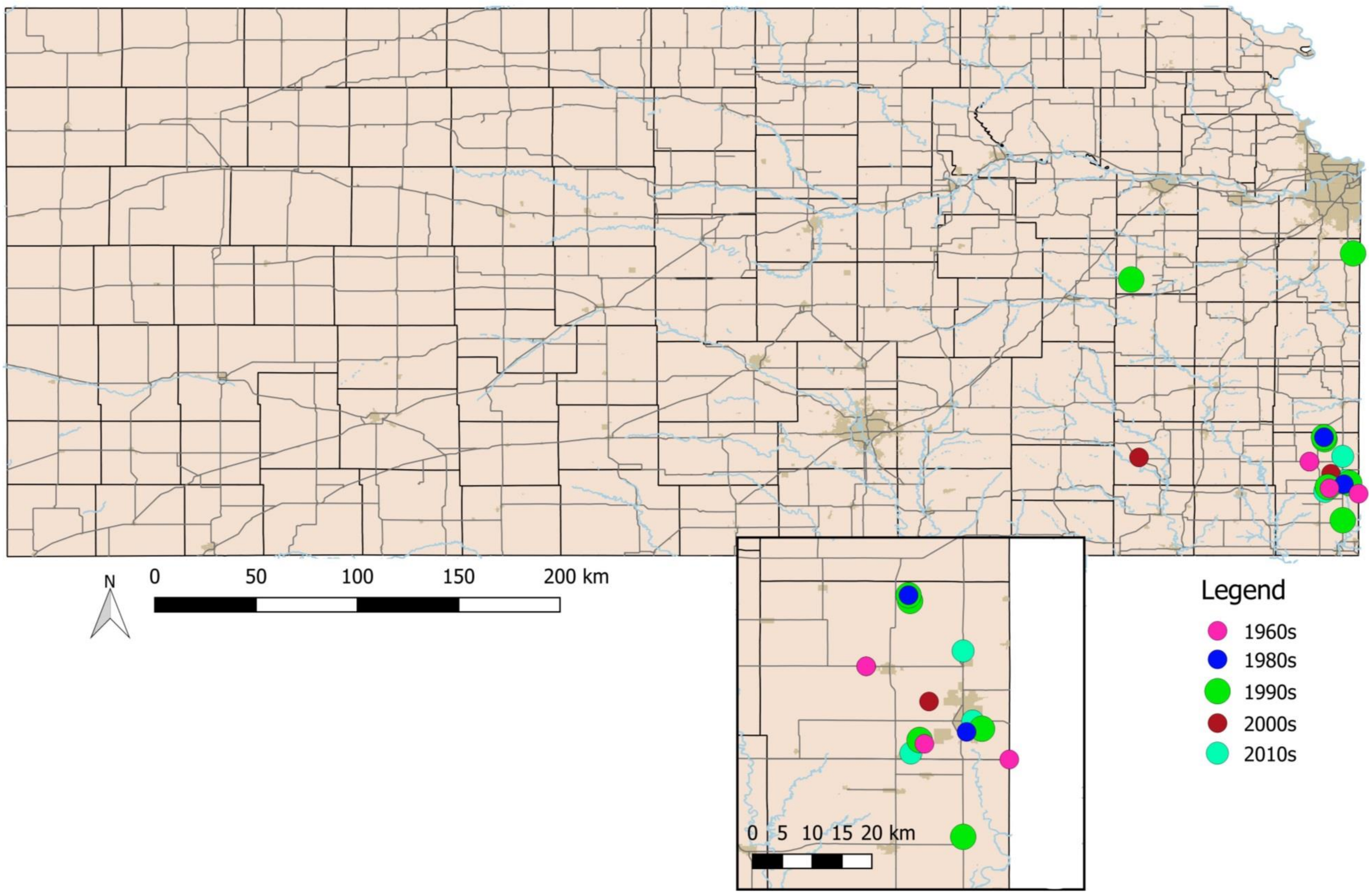

Figure 34. Spatial and temporal distribution of collecting sites of *Chyrsemys picta bellii* (Western Painted Turtle).

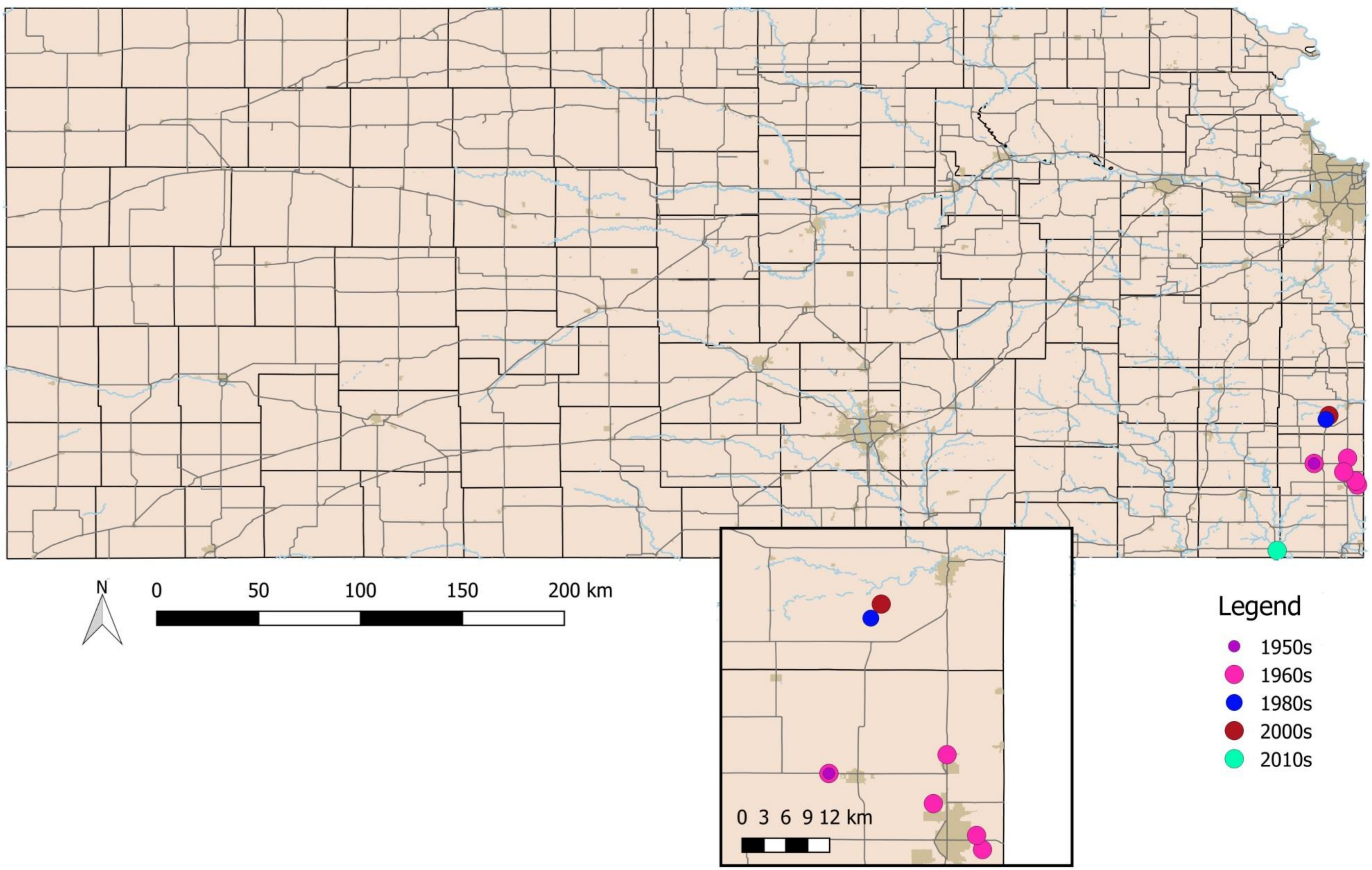

Figure 35. Spatial and temporal distribution of collecting sites of *Terrapene ornata* (Ornate Box Turtle).

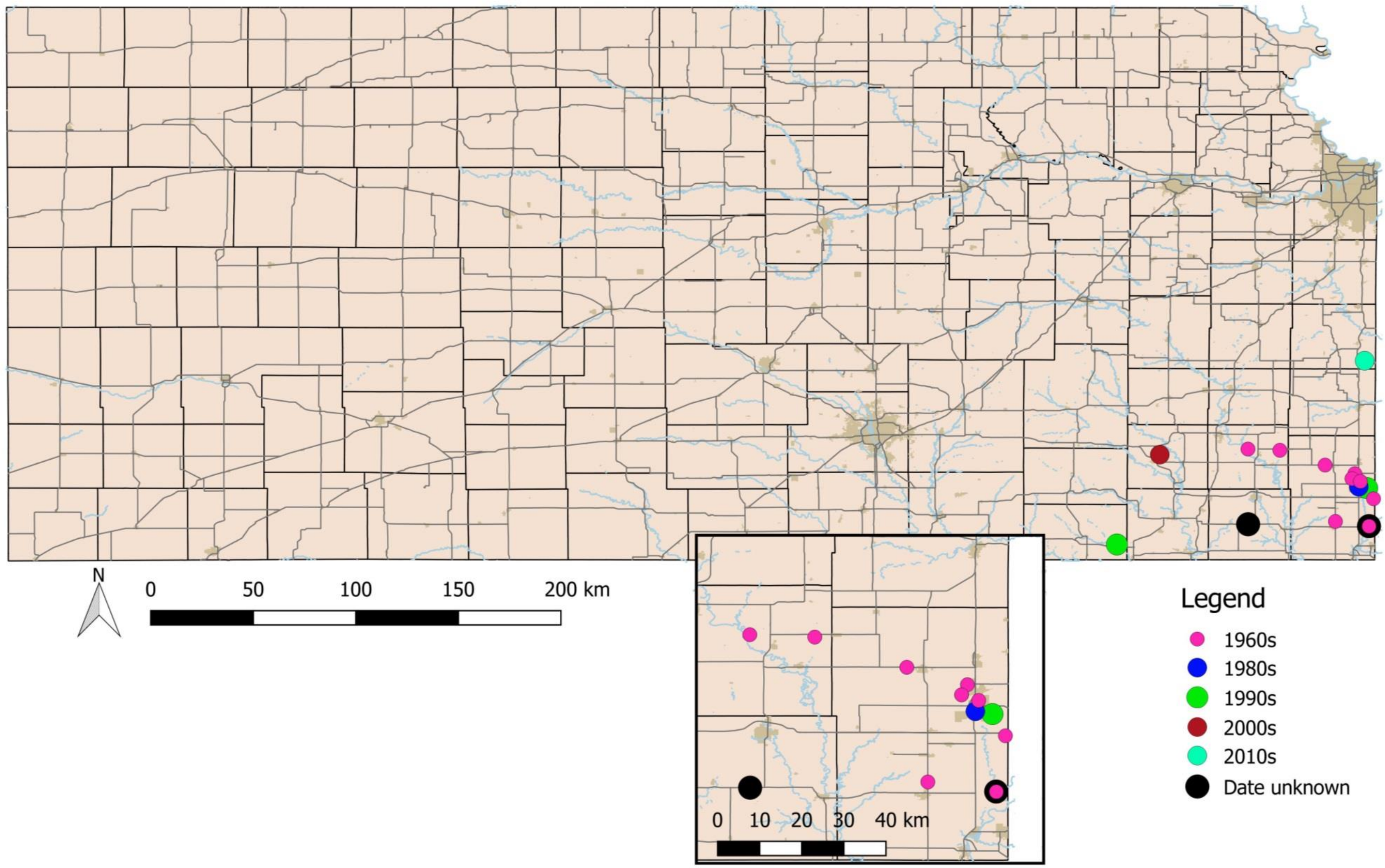

Figure 36. Spatial and temporal distribution of collecting sites of *Terrapene carolina triunguis* (Three-toed Box Turtle).

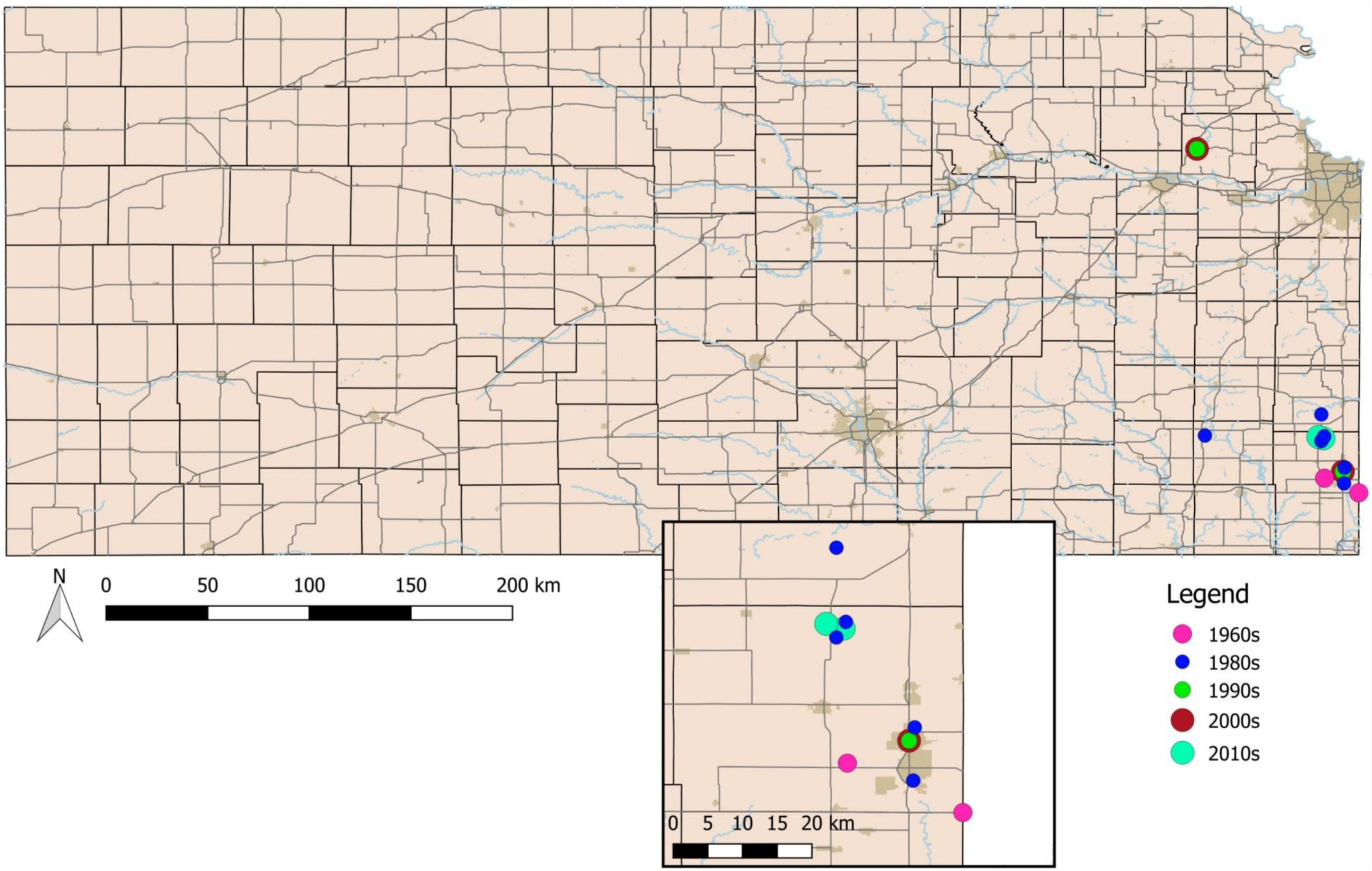

Figure 37. Spatial and temporal distribution of collecting sites of *Trachemys scripta elegans* (Red-eared Slider).

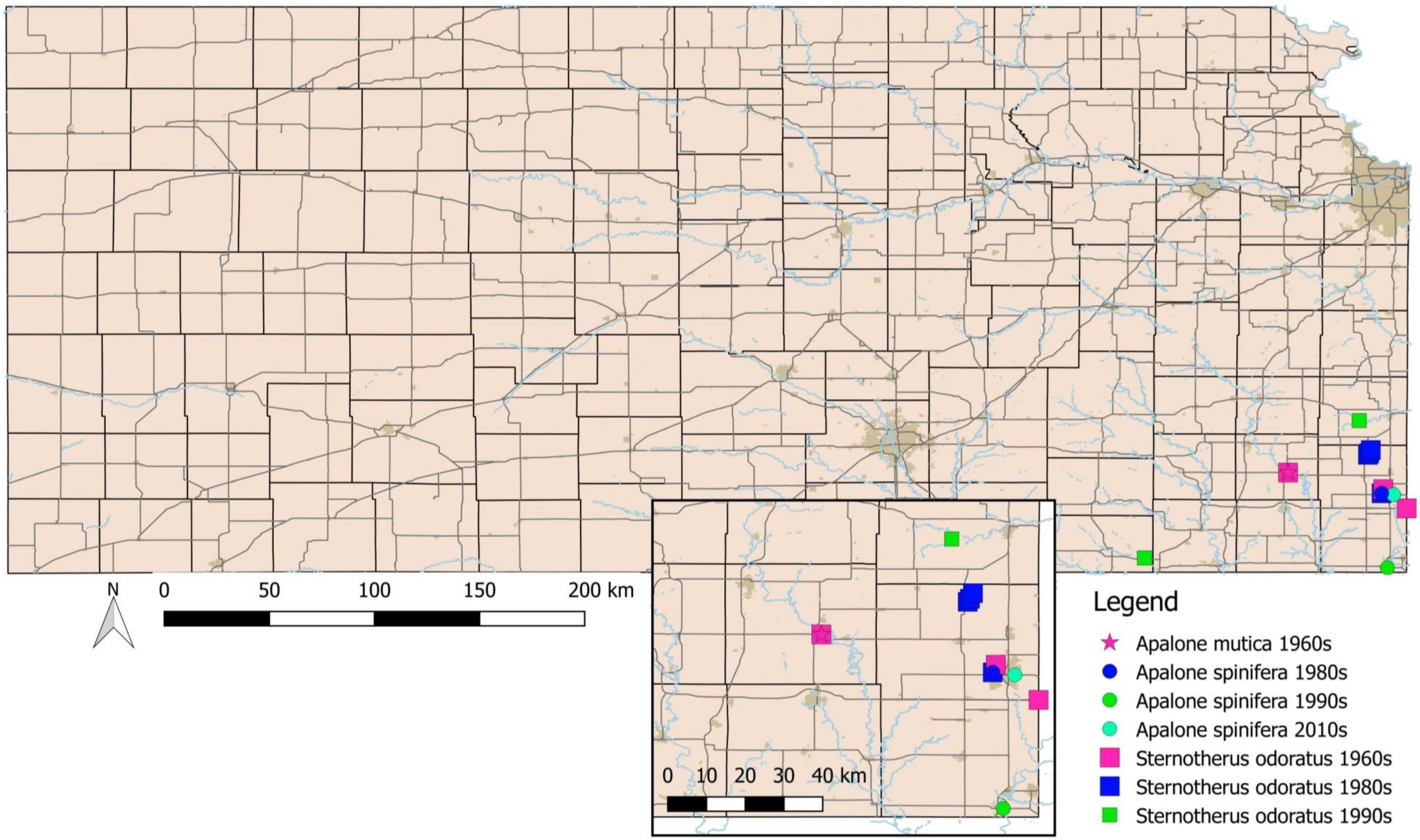

Figure 38. Spatial and temporal distribution of collecting sites of *Apalone mutica* (Midland Smooth Softshell), *Apalone spinifera* (Eastern Spiny Softshell) and *Sternotherus odoratus* (Eastern Musk Turtle).

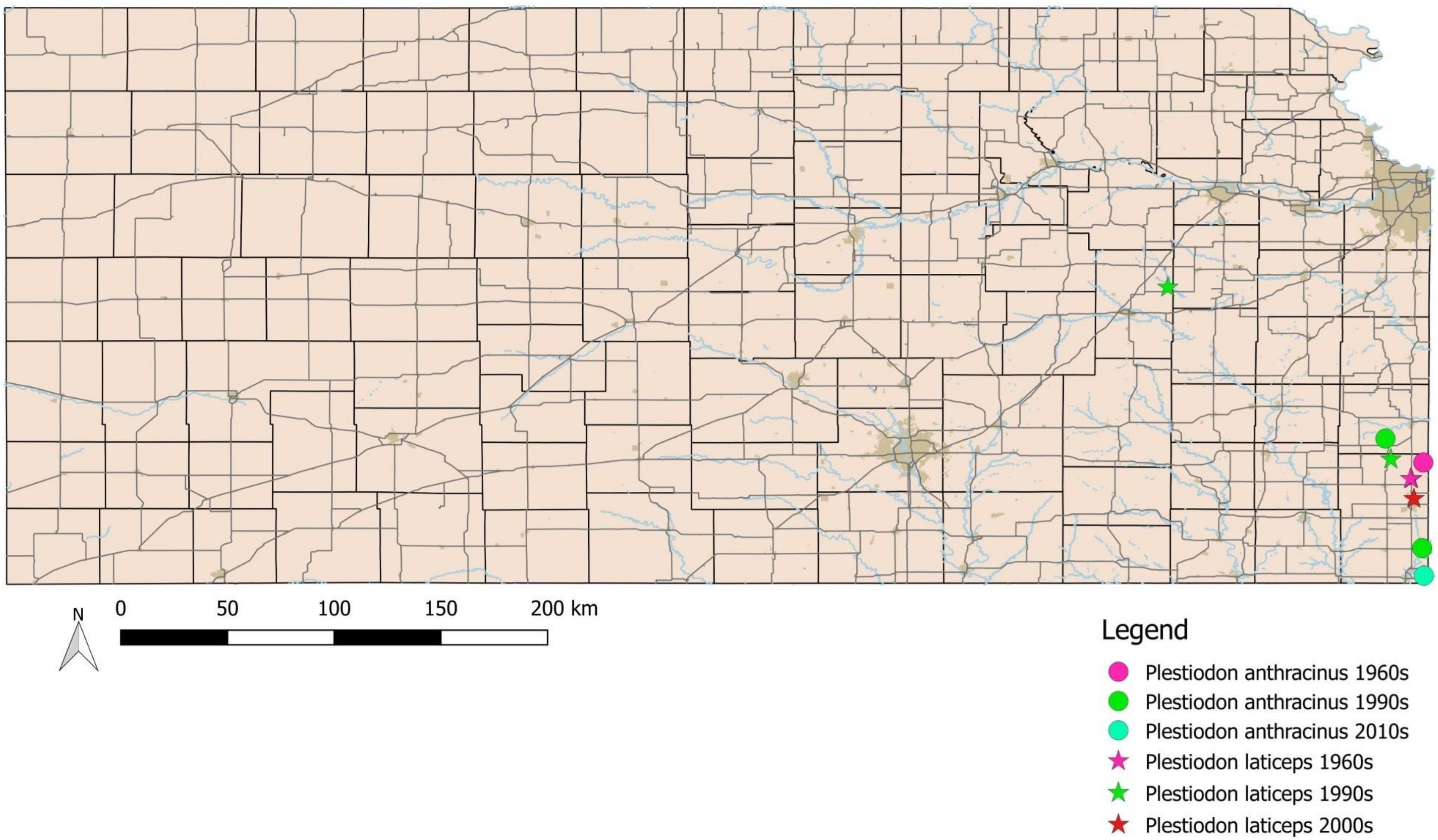

Figure 39. Spatial and temporal distribution of collecting sites of *Plestiodon anthracinus* (Coal Skink), and *Plestiodon laticeps* (Broad-headed Skink). *Plestiodon laticeps* is a Threatened in Kansas.

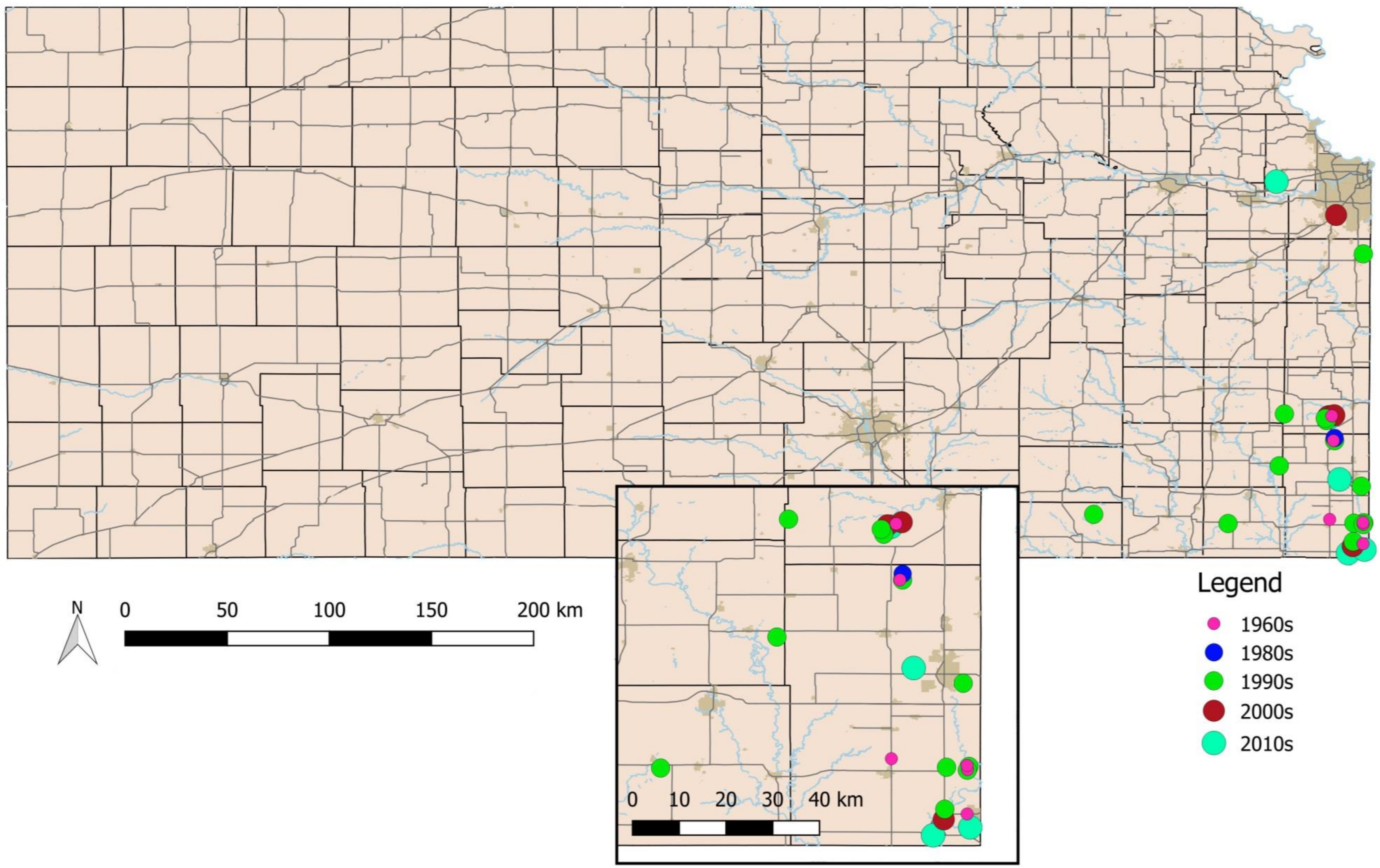

Figure 40. Spatial and temporal distribution of collecting sites of *Plestiodon fasciatus* (Common Five-lined Skink).

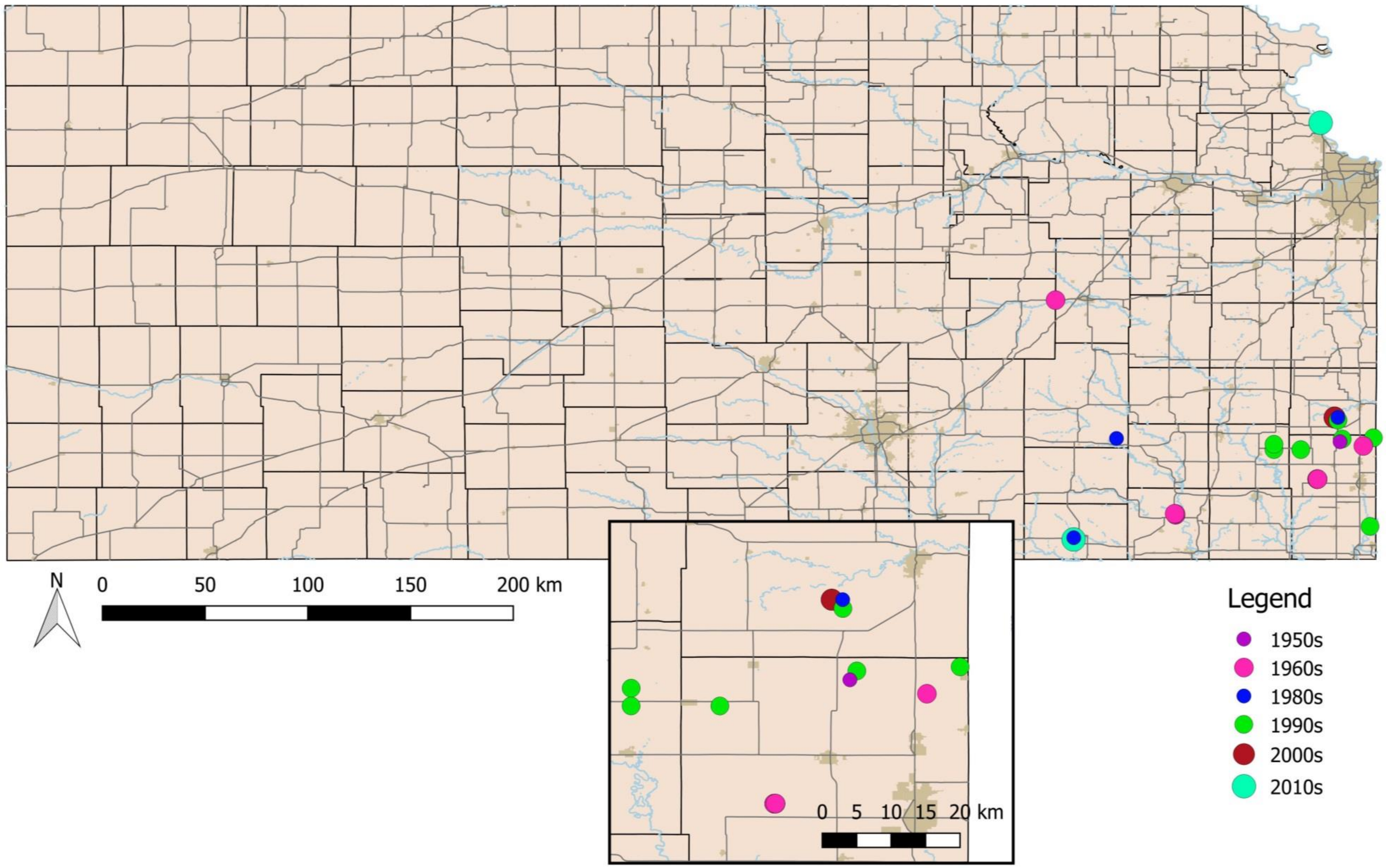

Figure 41. Spatial and temporal distribution of collecting sites of *Plestiodon obsoletus* (Great Plains Skink).

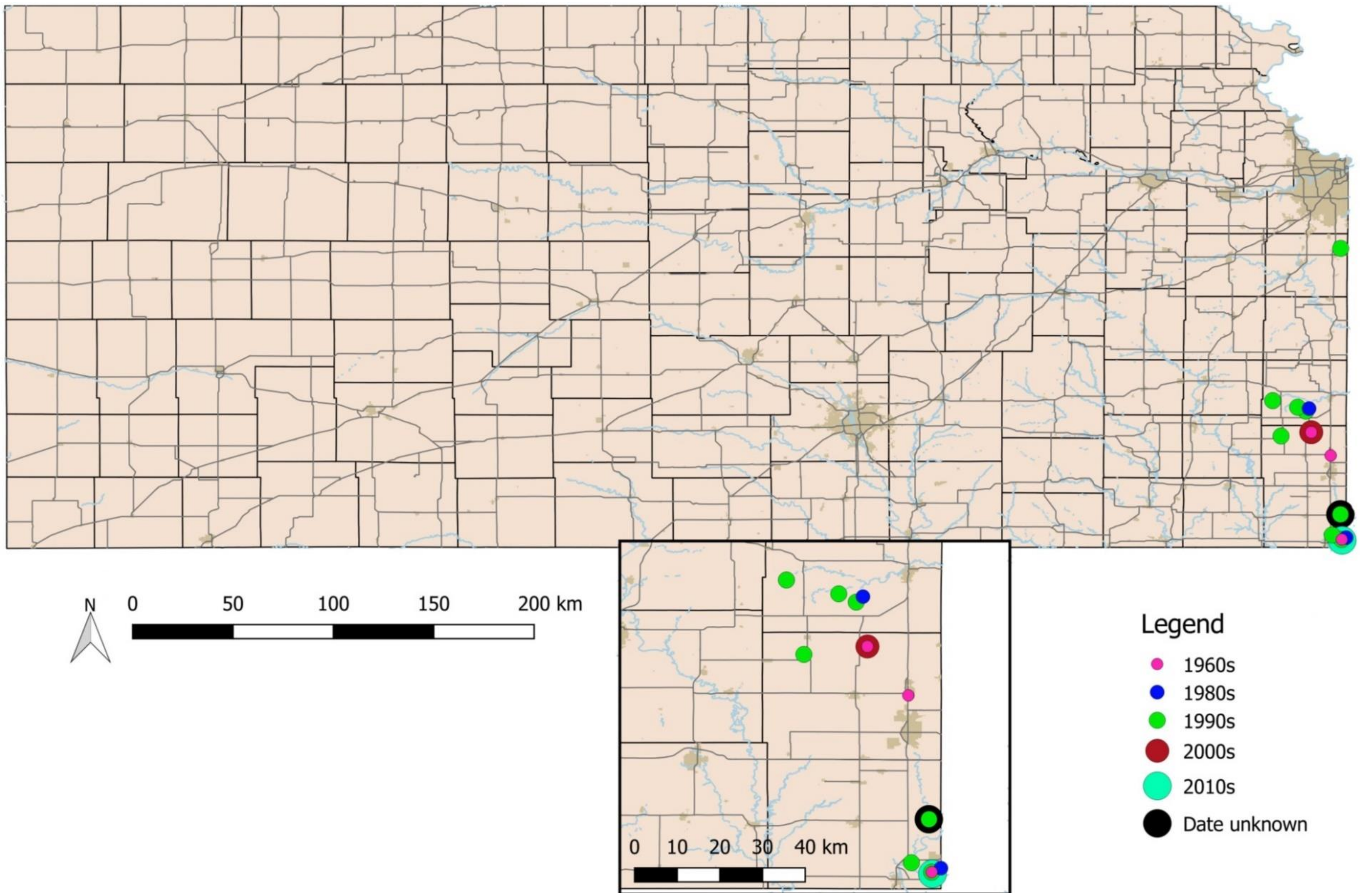

Figure 42. Spatial and temporal distribution of collecting sites of *Scincella lateralis* (Little Brown Skink).

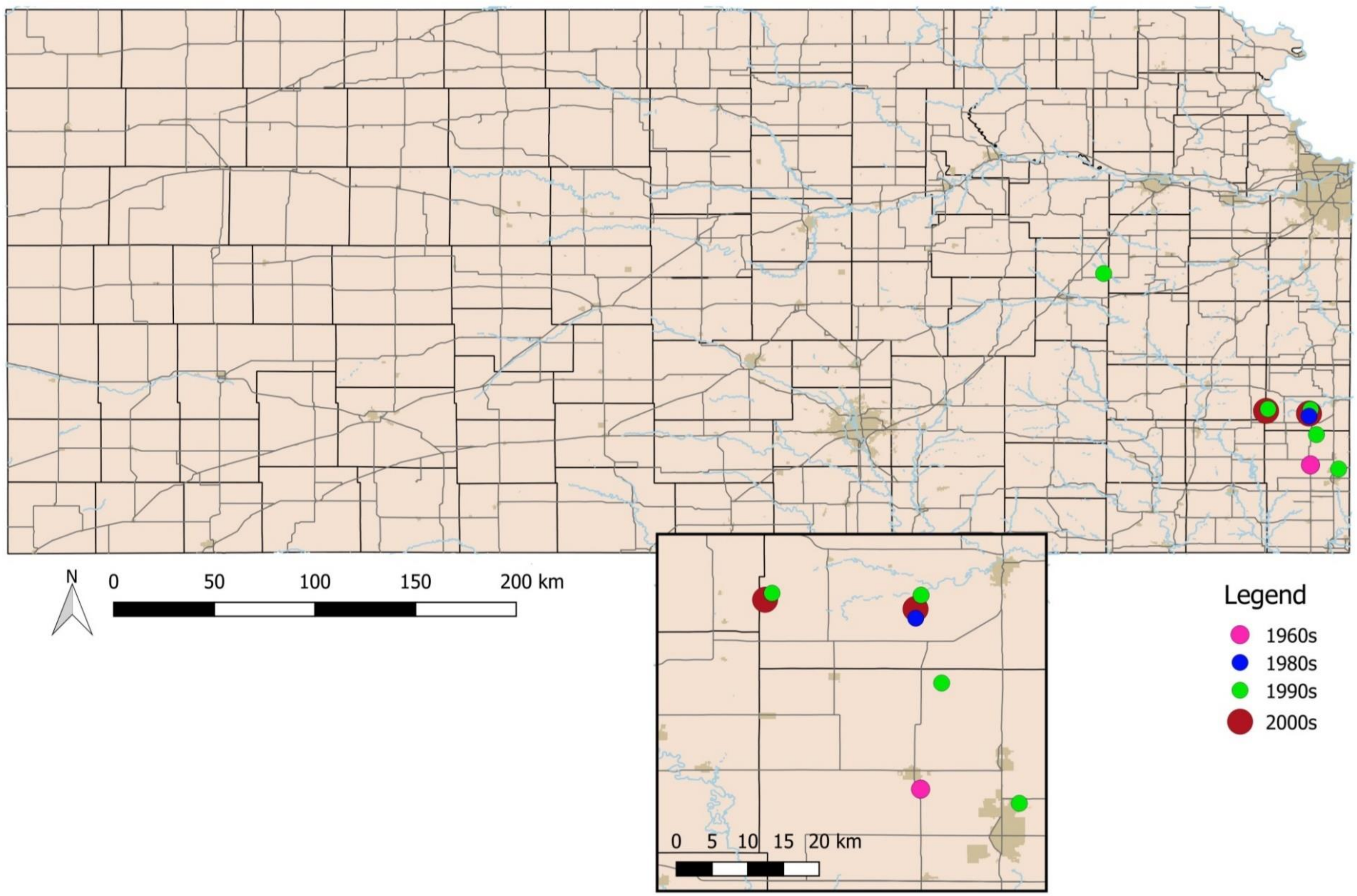

Figure 43. Spatial and temporal distribution of collecting sites of *Aspidoscelis sexlineata viridis* (Prairie Racerunner).

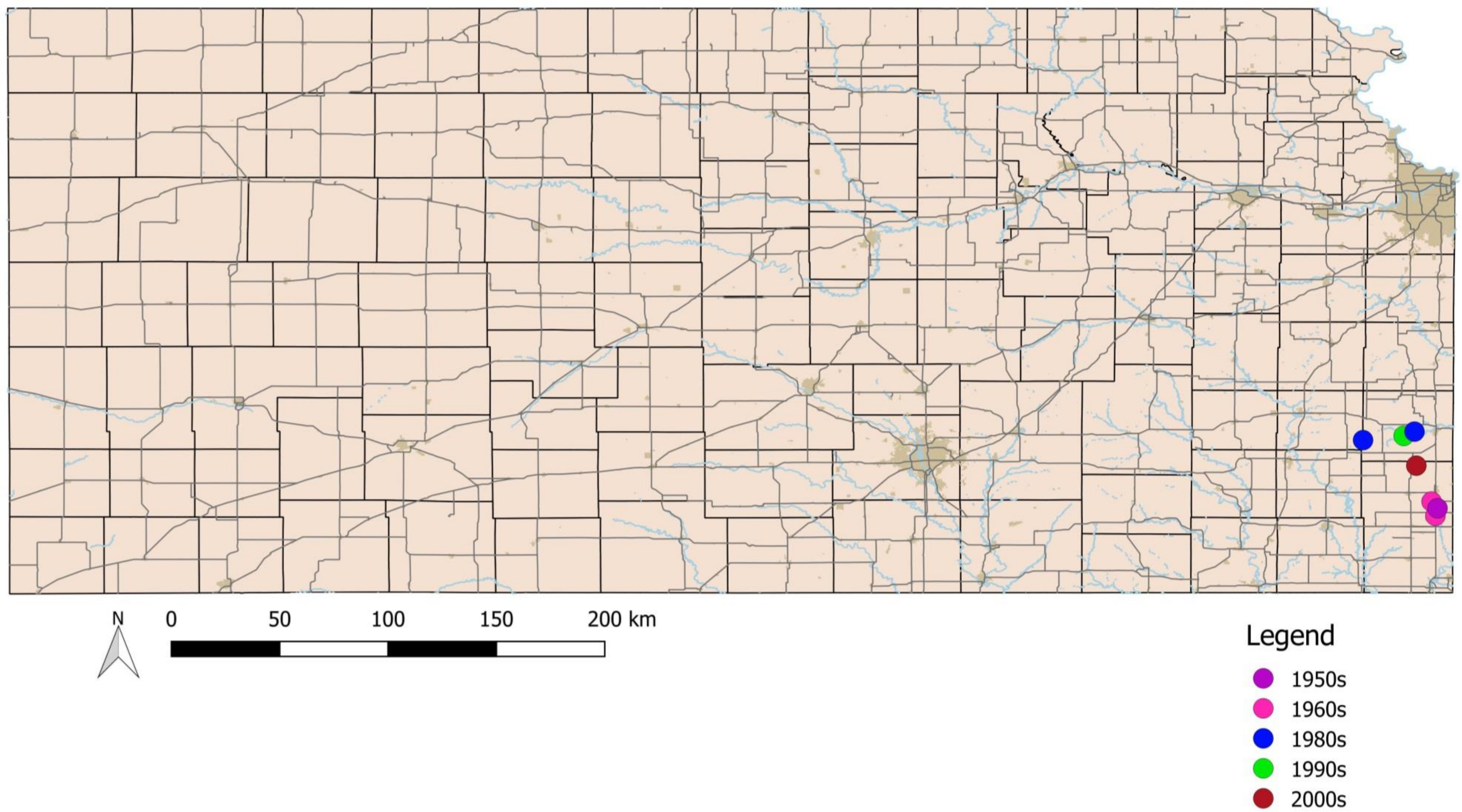

Figure 44. Spatial and temporal distribution of collecting sites of *Ophisaurus attenuatus attenuatus* (Western Slender Glass Lizard).

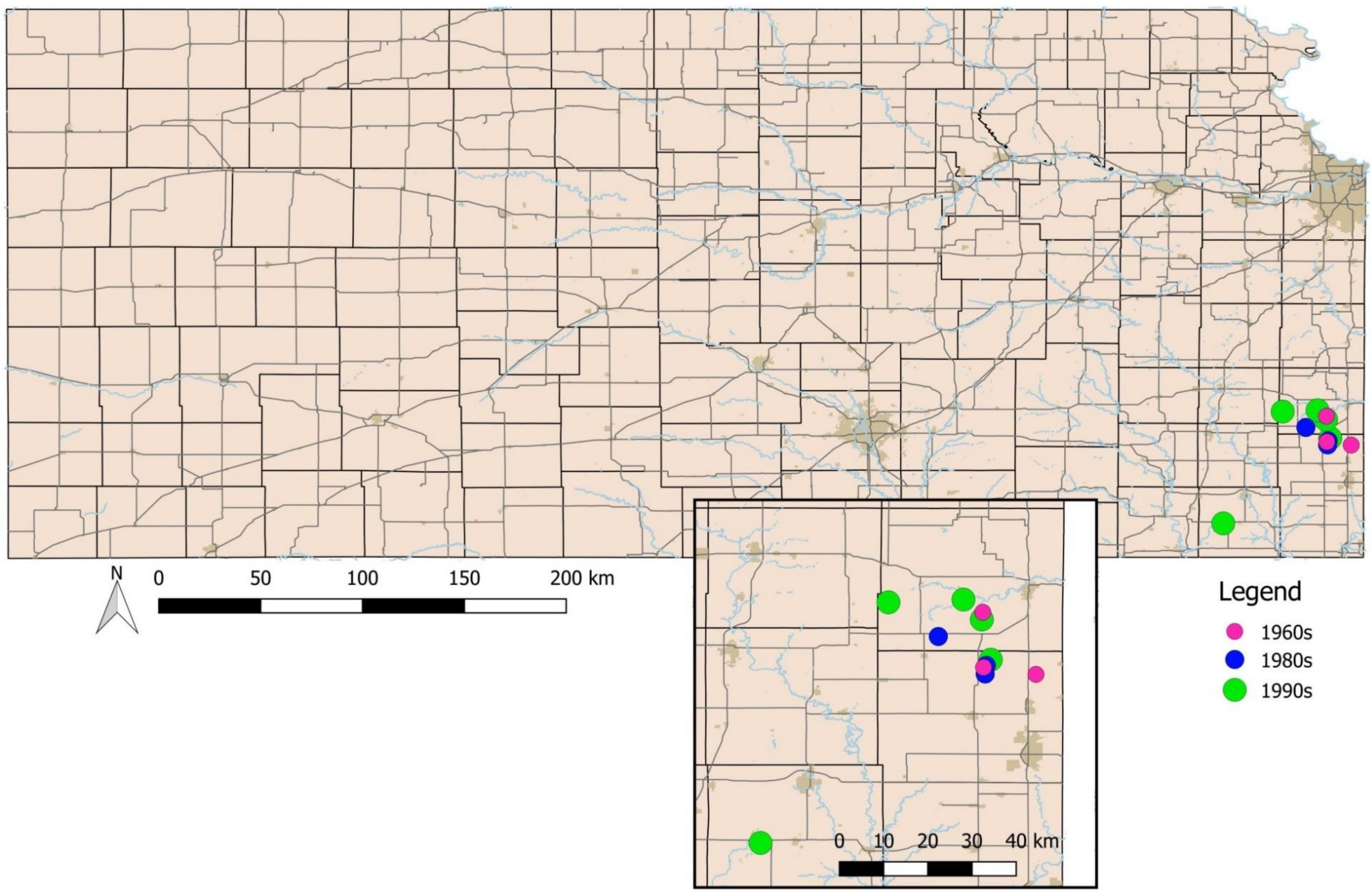

Figure 45. Spatial and temporal distribution of collecting sites of *Crotaphytus collaris* (Eastern Collared Lizard).

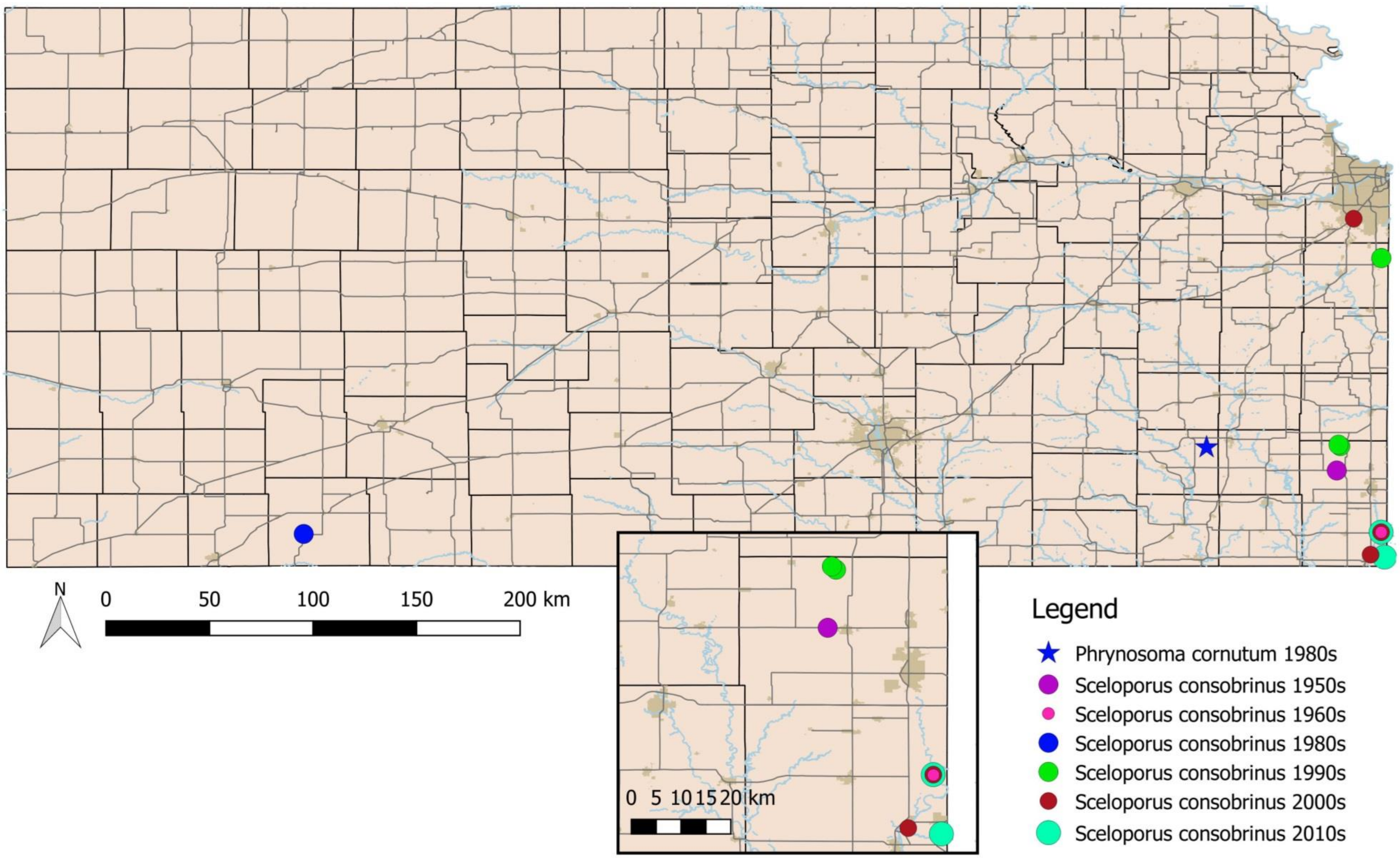

Figure 46. Spatial and temporal distribution of collecting sites of *Phrynosoma cornutum* (Texas Horned Lizard), and *Sceloporus consobrinus* (Prairie Lizard). *Phrynosoma cornutum* is a species in need of information in Kansas.

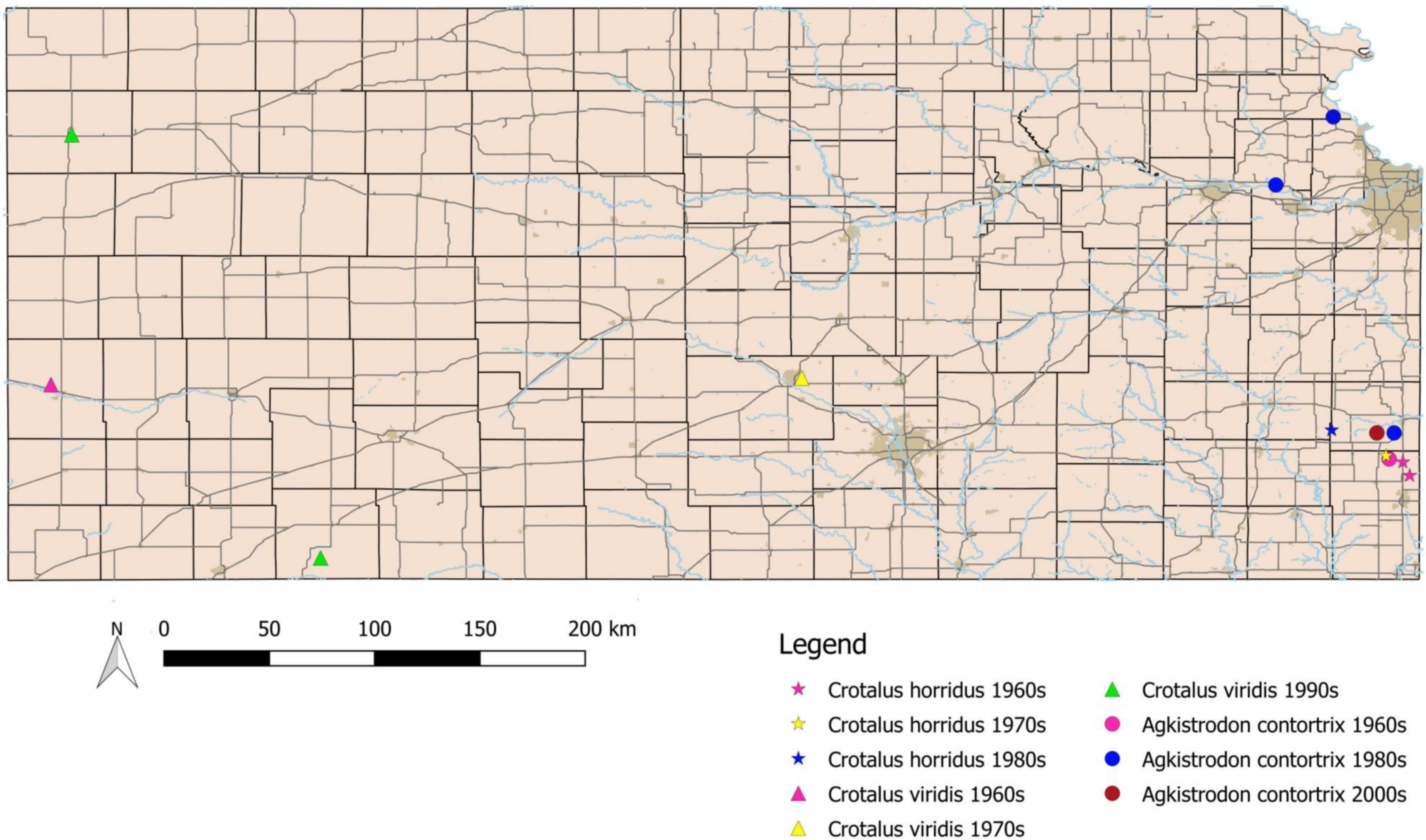

Figure 47. Spatial and temporal distribution of collecting sites of *Agkistrodon contortrix* (Eastern Copperhead), *Crotalus horridus* (Timber Rattlesnake), and *Crotalus viridis* (Prairie Rattlesnake). *Crotalus horridus* is a species in need of conservation in Kansas.

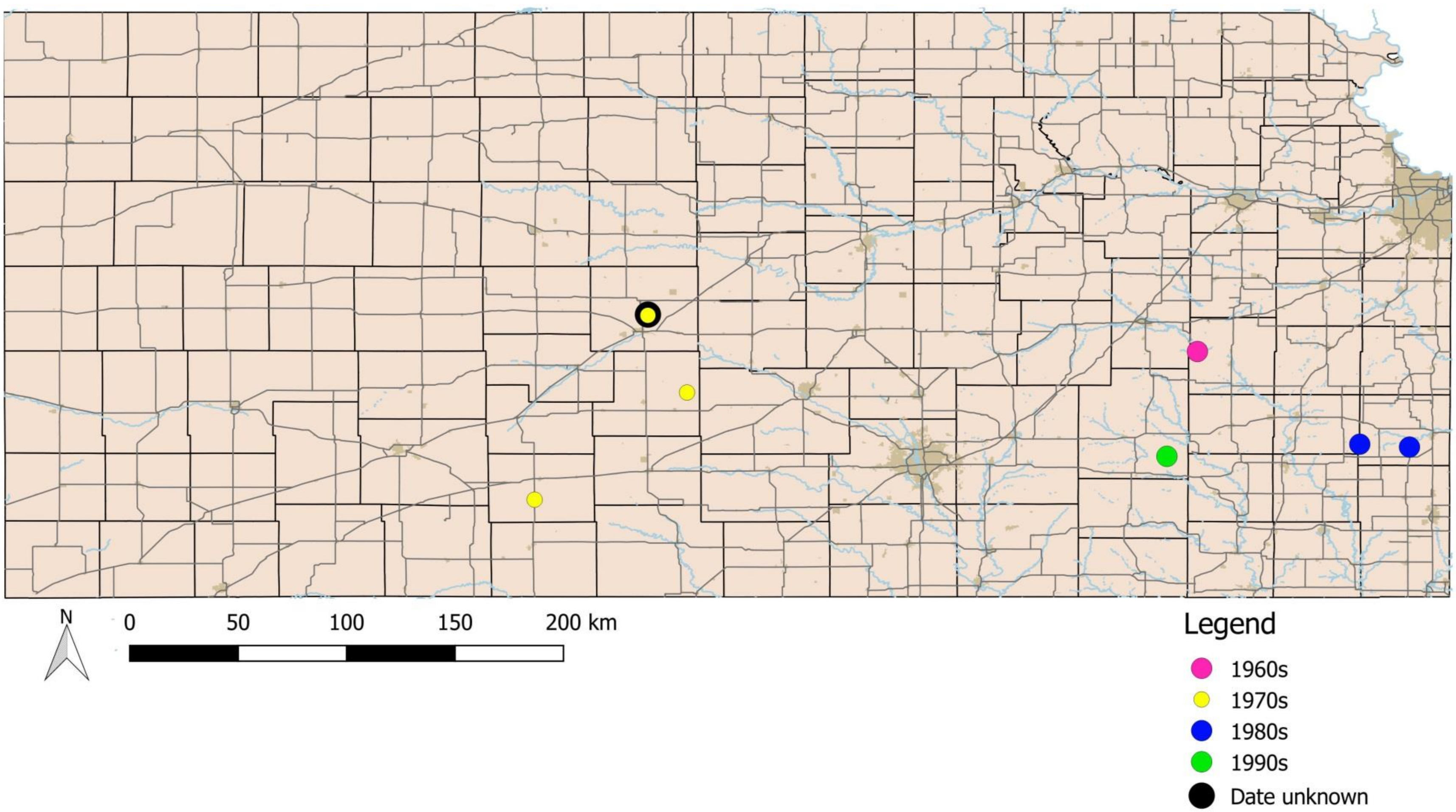

Figure 48. Spatial and temporal distribution of collecting sites of *Sistrurus tergeminus* (Western Massasauga).

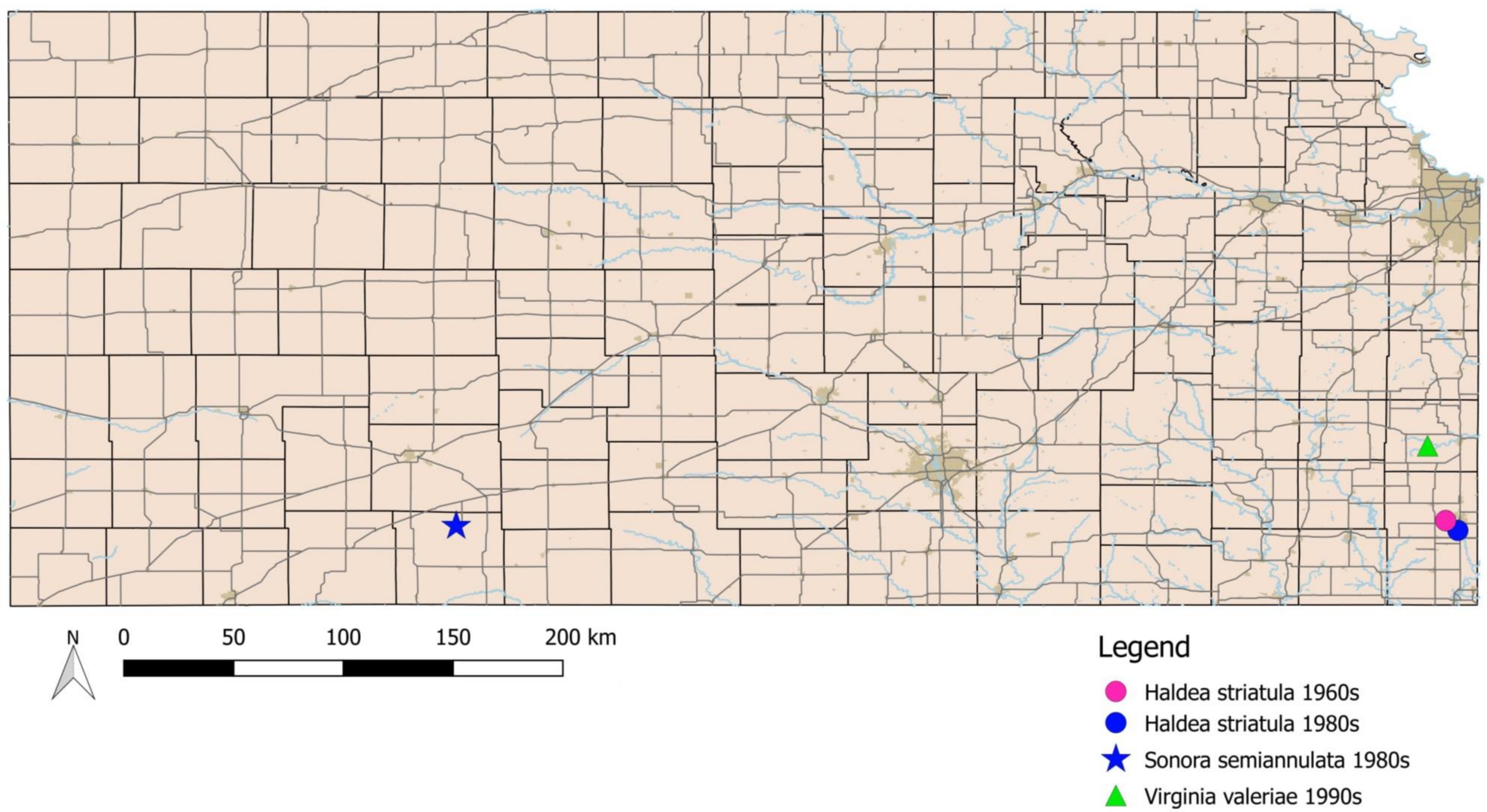

Figure 49. Spatial and temporal distribution of collecting sites of *Haldea striatula* (Rough Earthsnake), *Virginia valeriae* (Smooth Earthsnake), and *Sonora semiannulata* (Western Groundsnake). *Haldea striatula* and *V. valeriae* are species in need of conservation in Kansas.

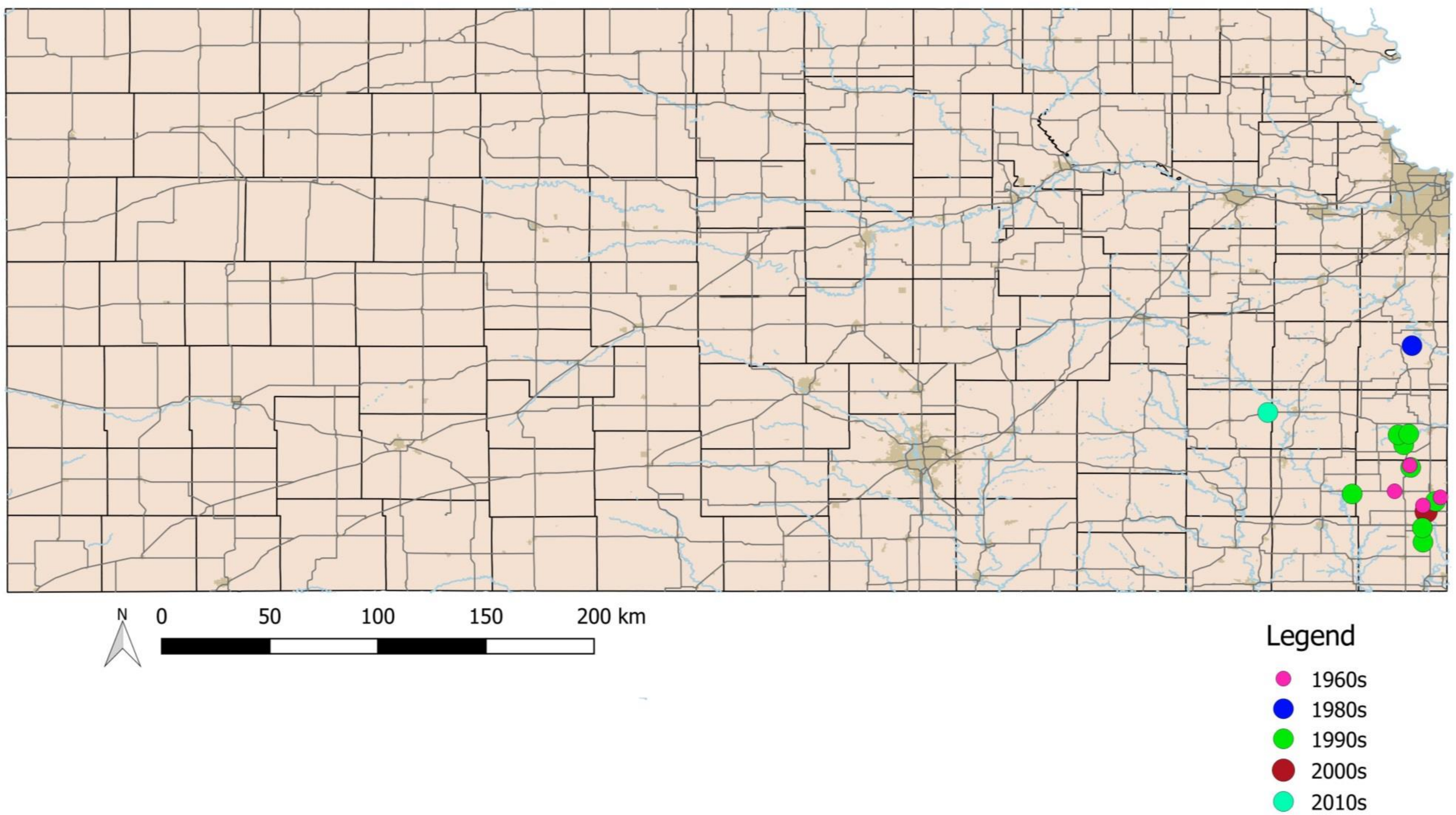

Figure 50. Spatial and temporal distribution of collecting sites of *Nerodia erythrogaster* (Plain-bellied Watersnake).

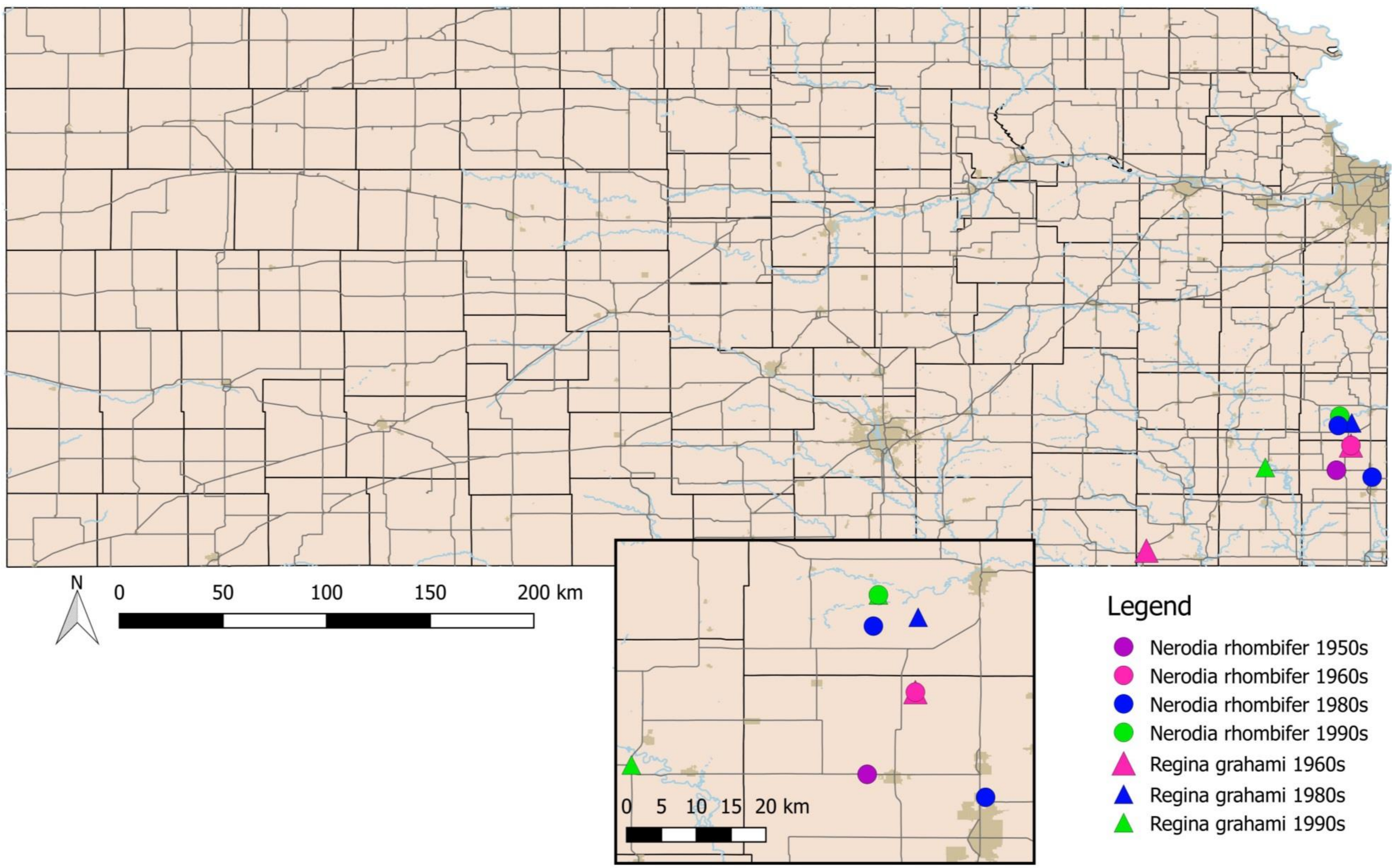

Figure 51. Spatial and temporal distribution of collecting sites of *Nerodia rhombifer* (Diamond-backed Watersnake) and *Regina grahamii* (Graham's Crawfish Snake).

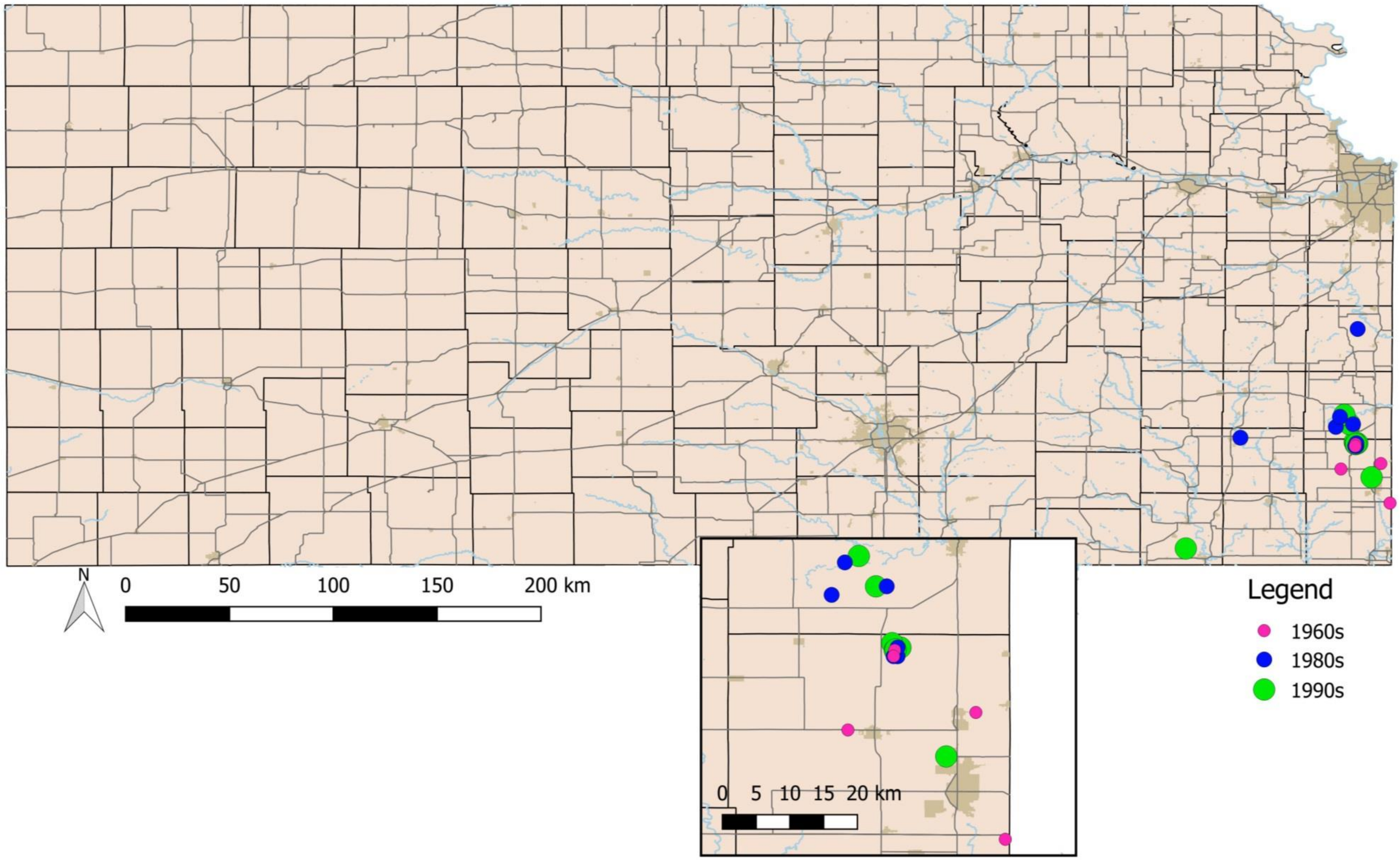

Figure 52. Spatial and temporal distribution of collecting sites of *Nerodia sipedon sipedon* (Northern Watersnake).

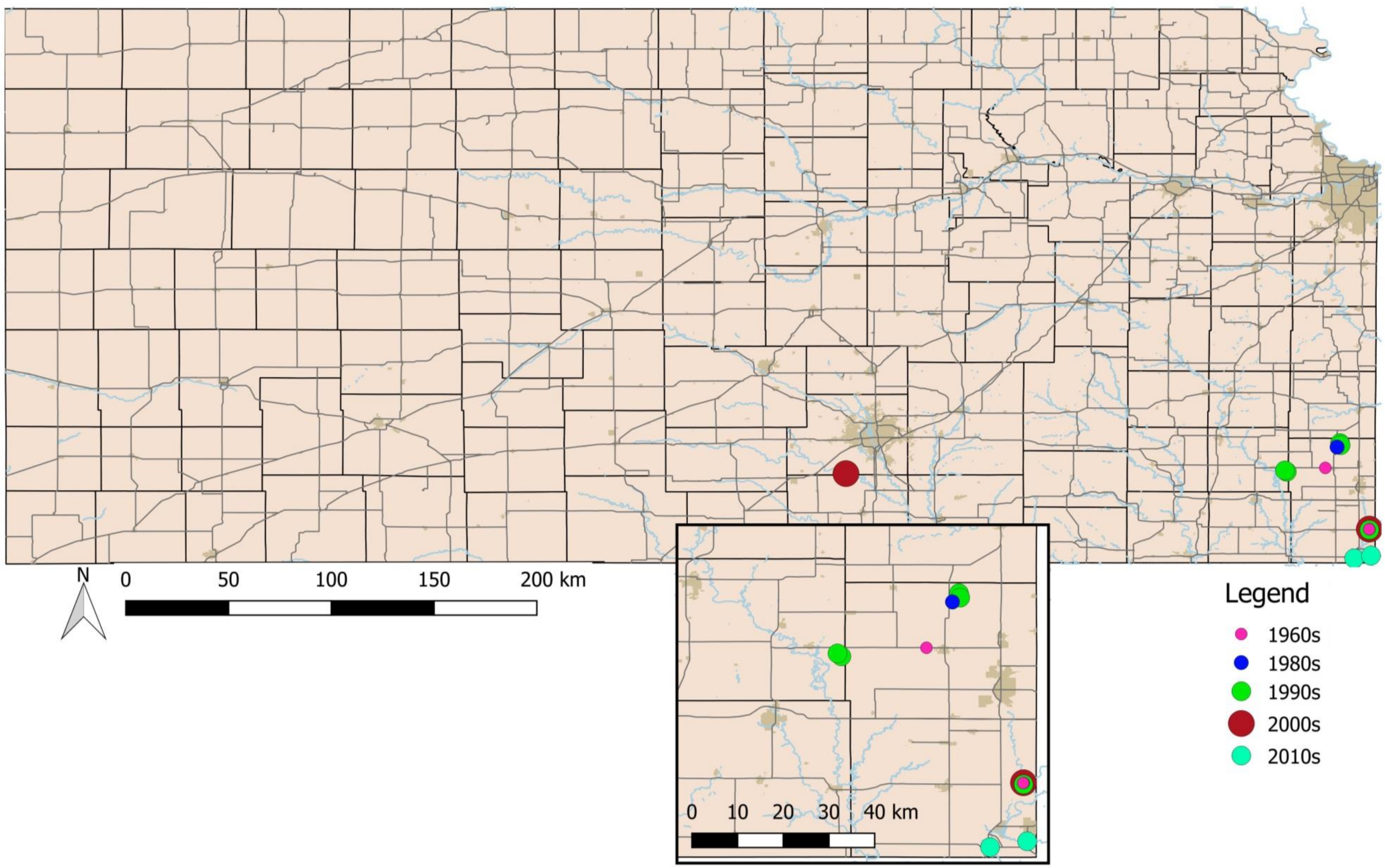

Figure 53. Spatial and temporal distribution of collecting sites of *Storeria dekayi* (Dekay's Brownsnake).

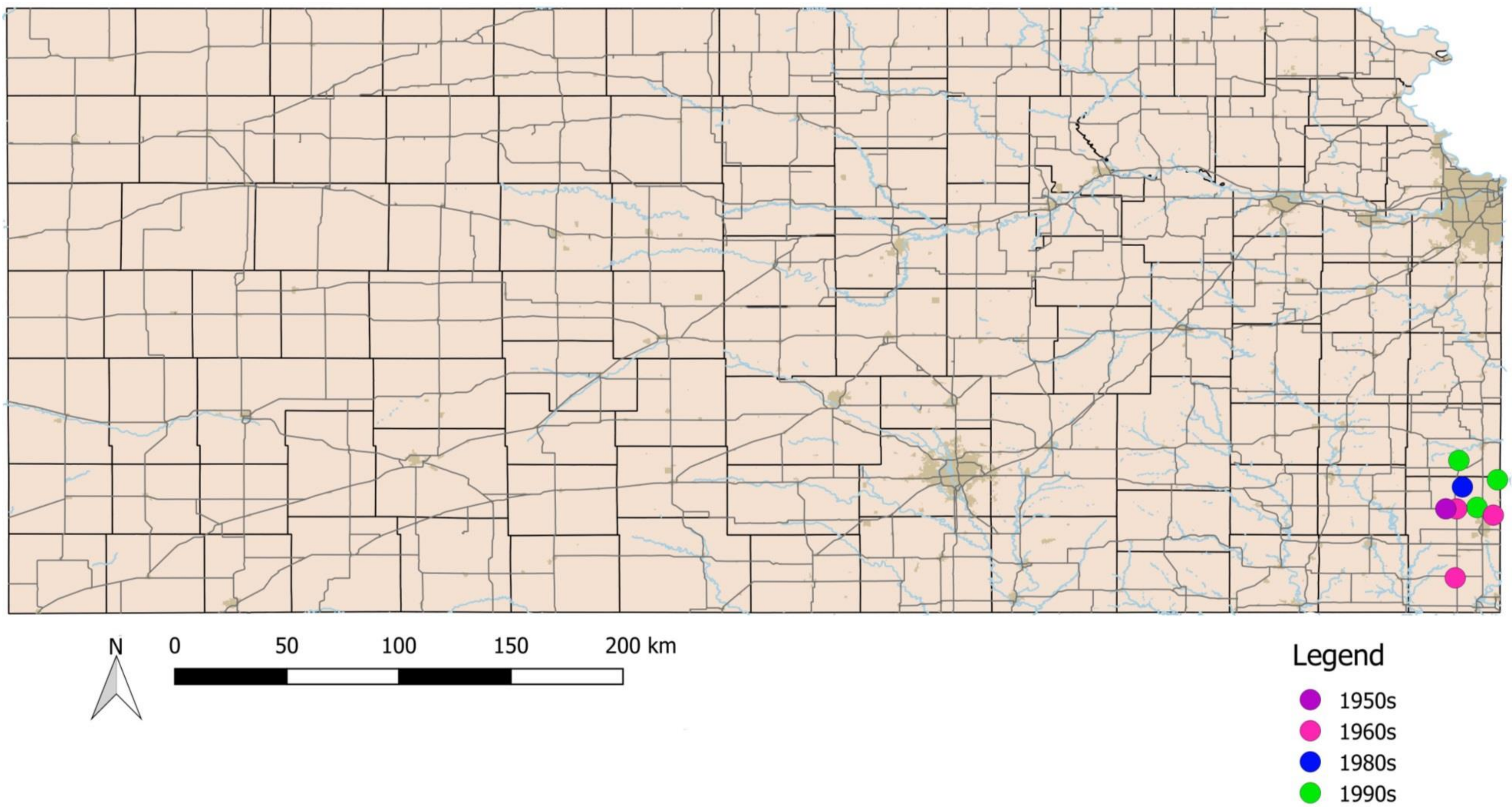

Figure 54. Spatial and temporal distribution of collecting sites of *Thamnophis proximus* (Western Ribbonsnake).

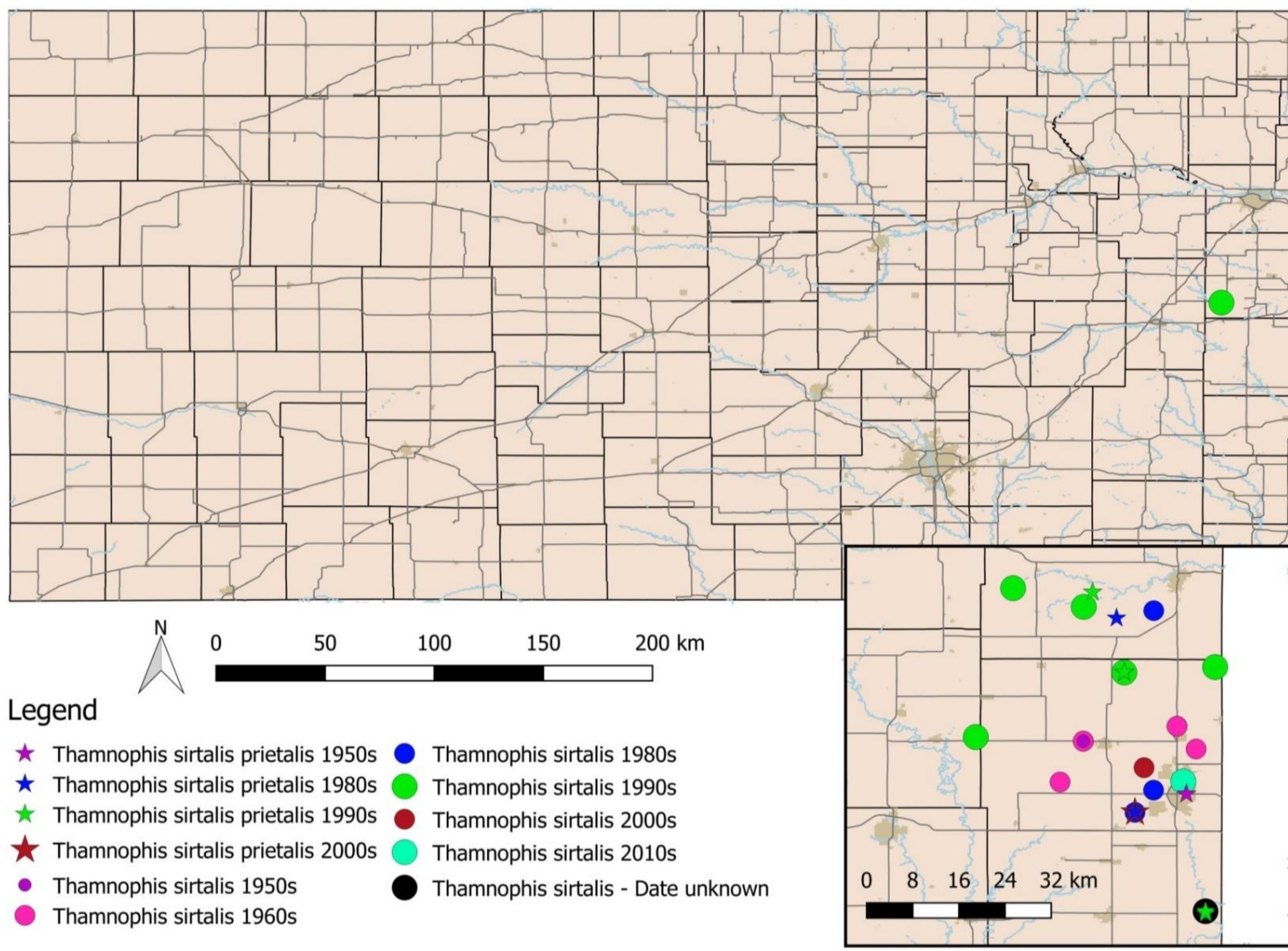

Figure 55. Spatial and temporal distribution of collecting sites of *Thamnophis sirtalis* (Common Gartersnake) and *Thamnophis sirtalis parietalis* (Red-sidded Gartersnake).

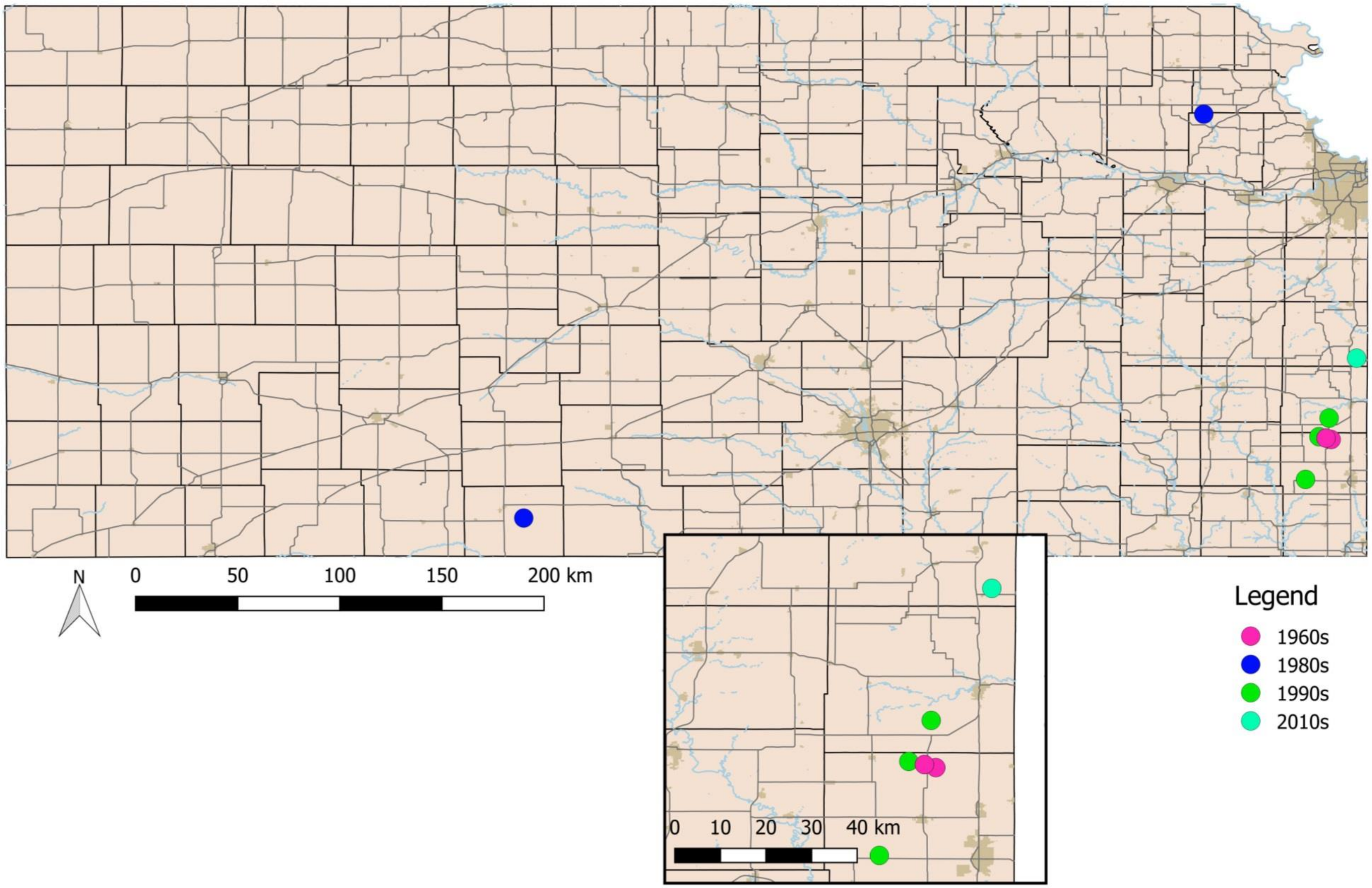

Figure 56. Spatial and temporal distribution of collecting sites of *Tropidoclonion lineatum* (Lined Snake).

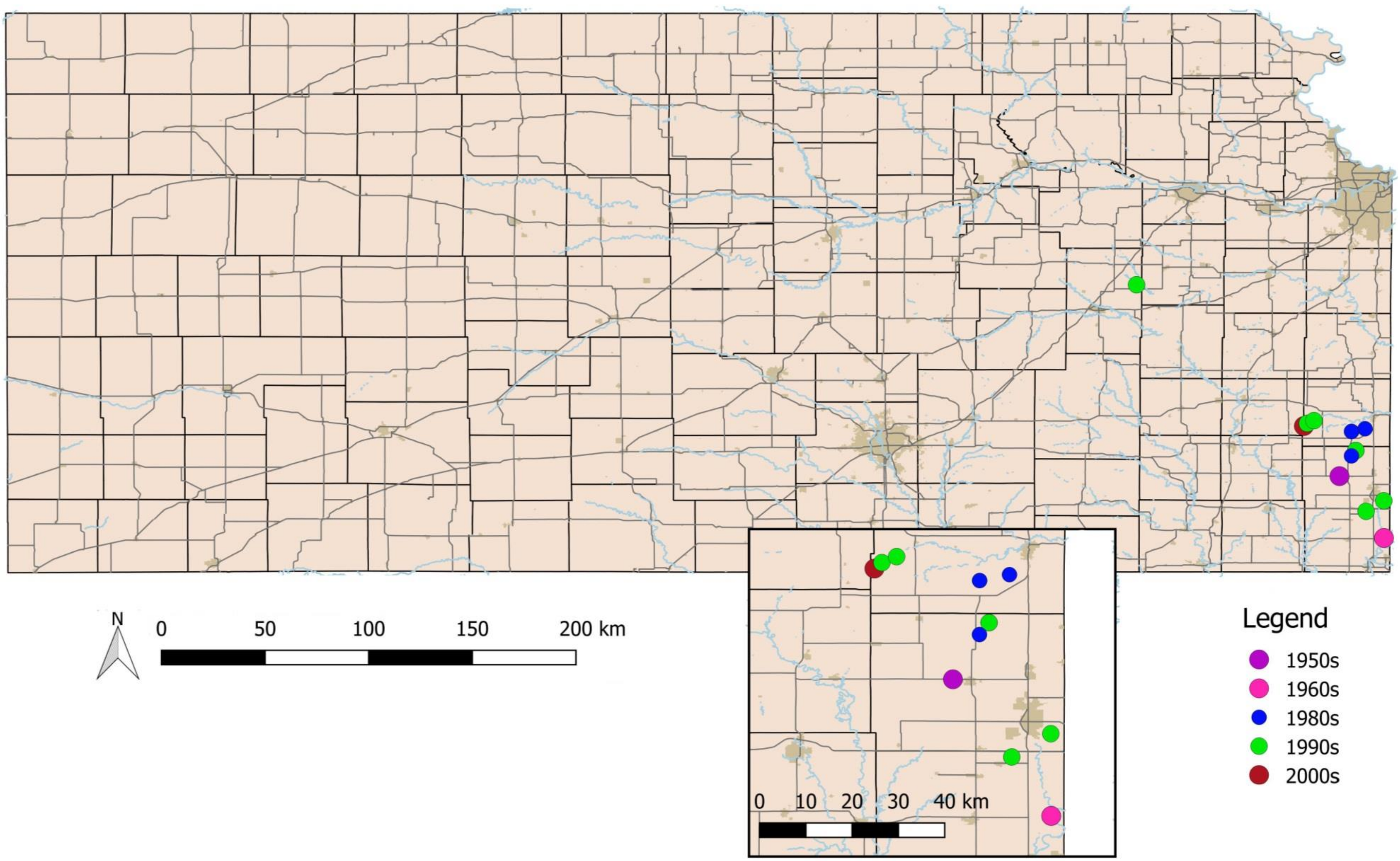

Figure 57. Spatial and temporal distribution of collecting sites of *Carphophis vermis* (Western Wormsnake).

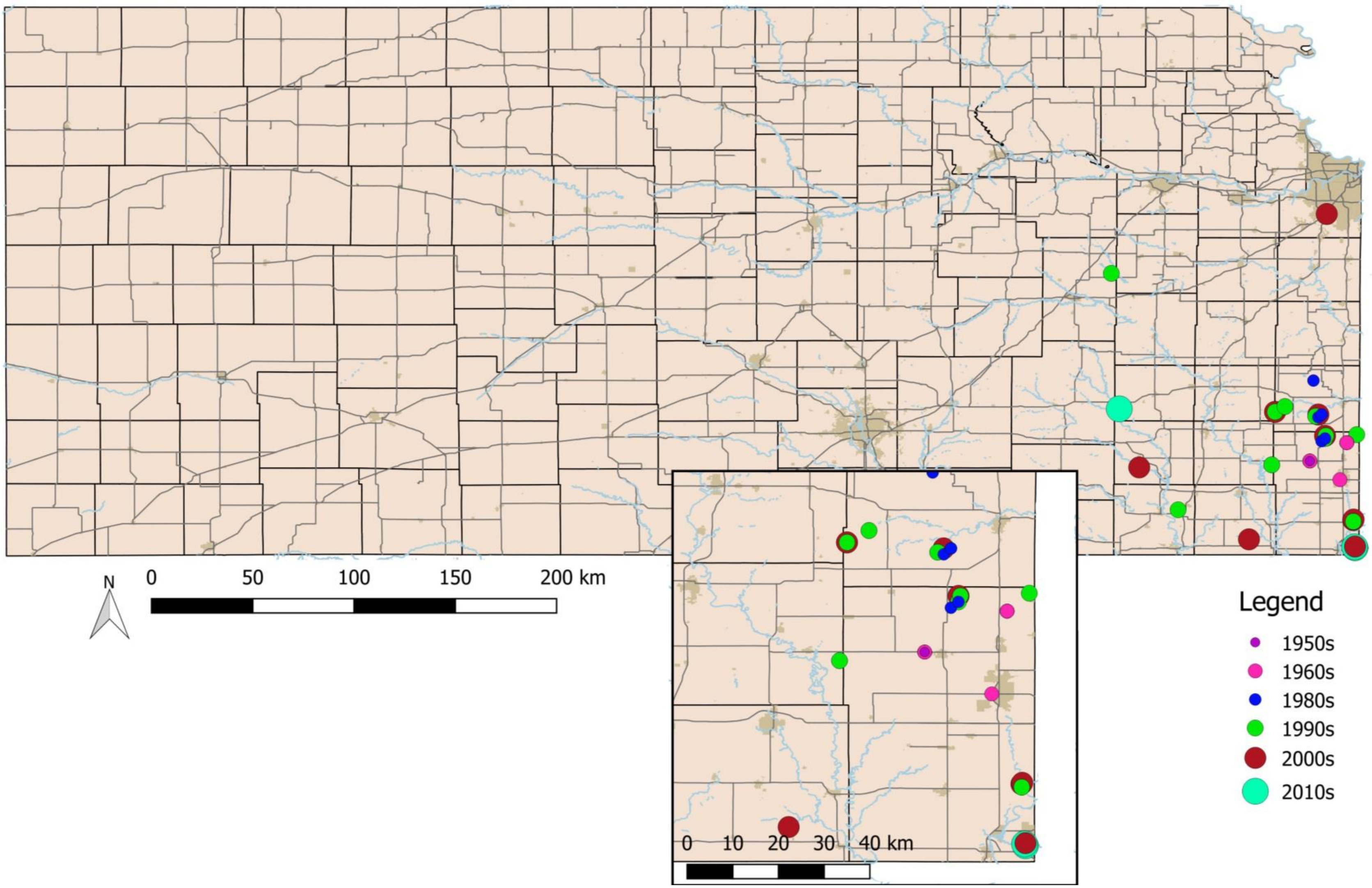

Figure 58. Spatial and temporal distribution of collecting sites of *Diadophis punctatus arnyi* (Prairie Ring-necked Snake).

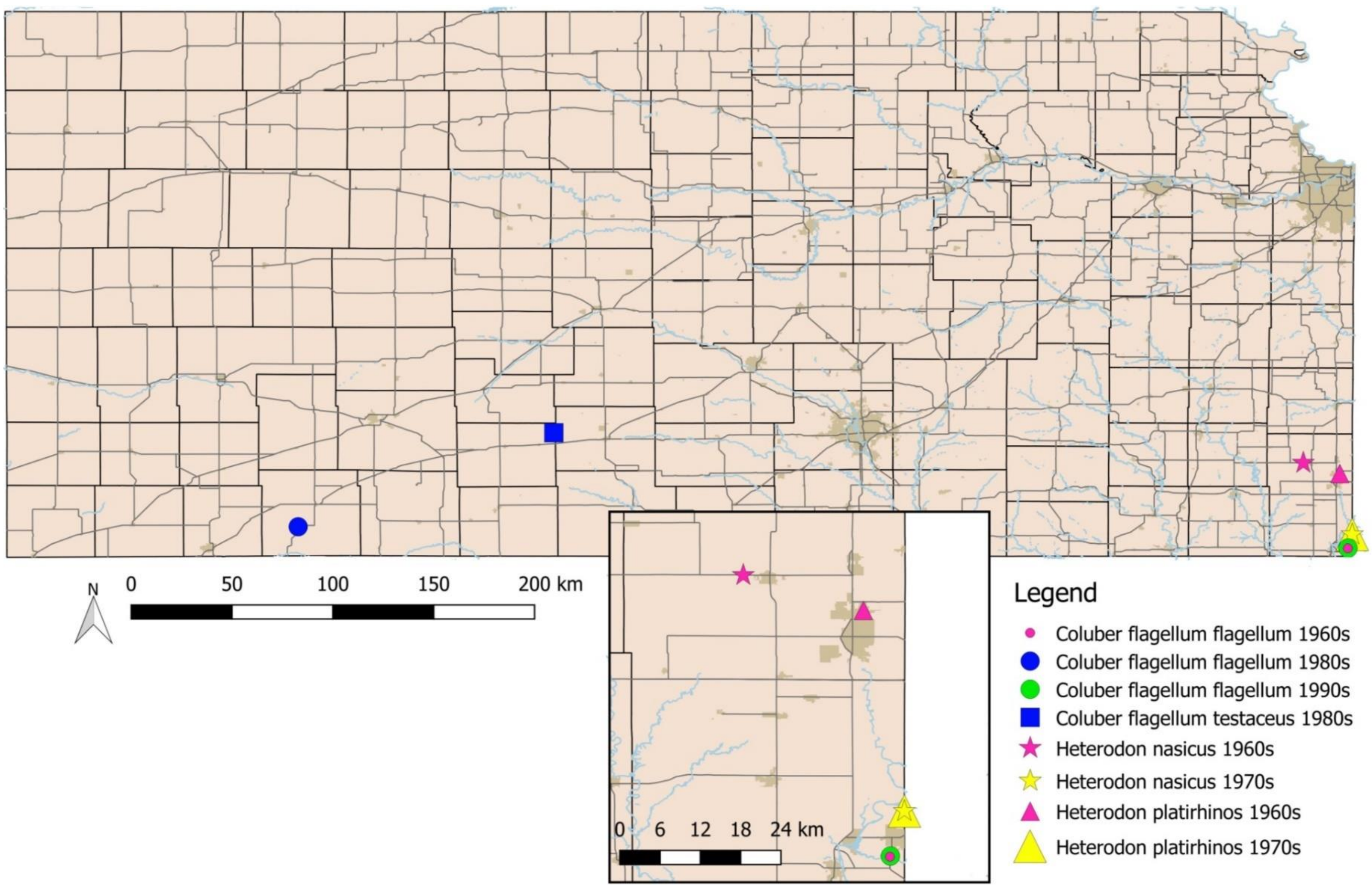

Figure 59. Spatial and temporal distribution of collecting sites of *Heterodon nasicus* (Plains Hog-nosed Snake), *Heterodon platirhinos* (Eastern Hog-nosed Snake), *Coluber flagellum flagellum* (Eastern Coachwhip), and *Coluber flagellum testaceus* (Western Coachwhip). *Heterodon nasicus* and *H. platirhinos* are species in need of conservation in Kansas.

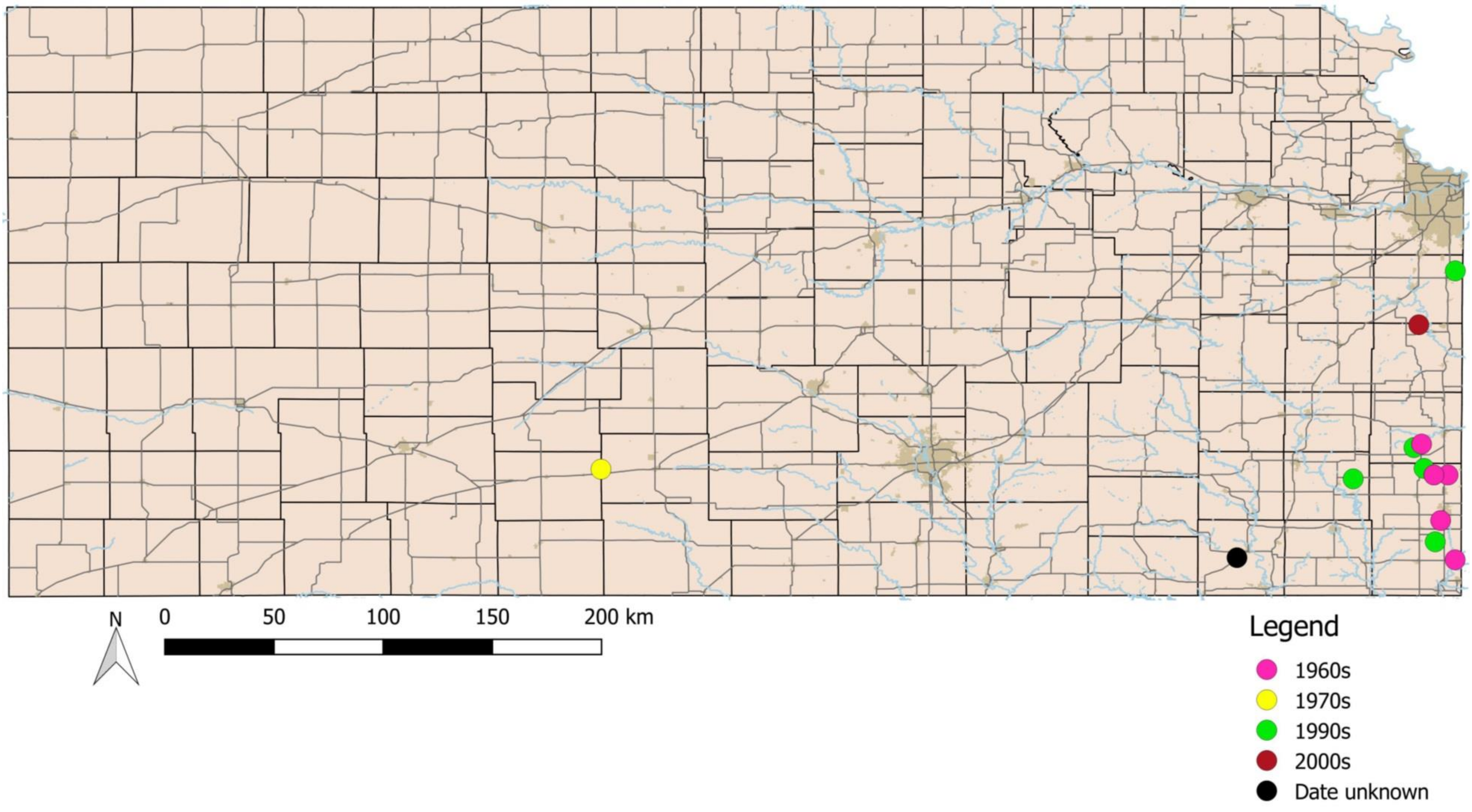

Figure 60. Spatial and temporal distribution of collecting sites of *Coluber constrictor* (North American Racer).

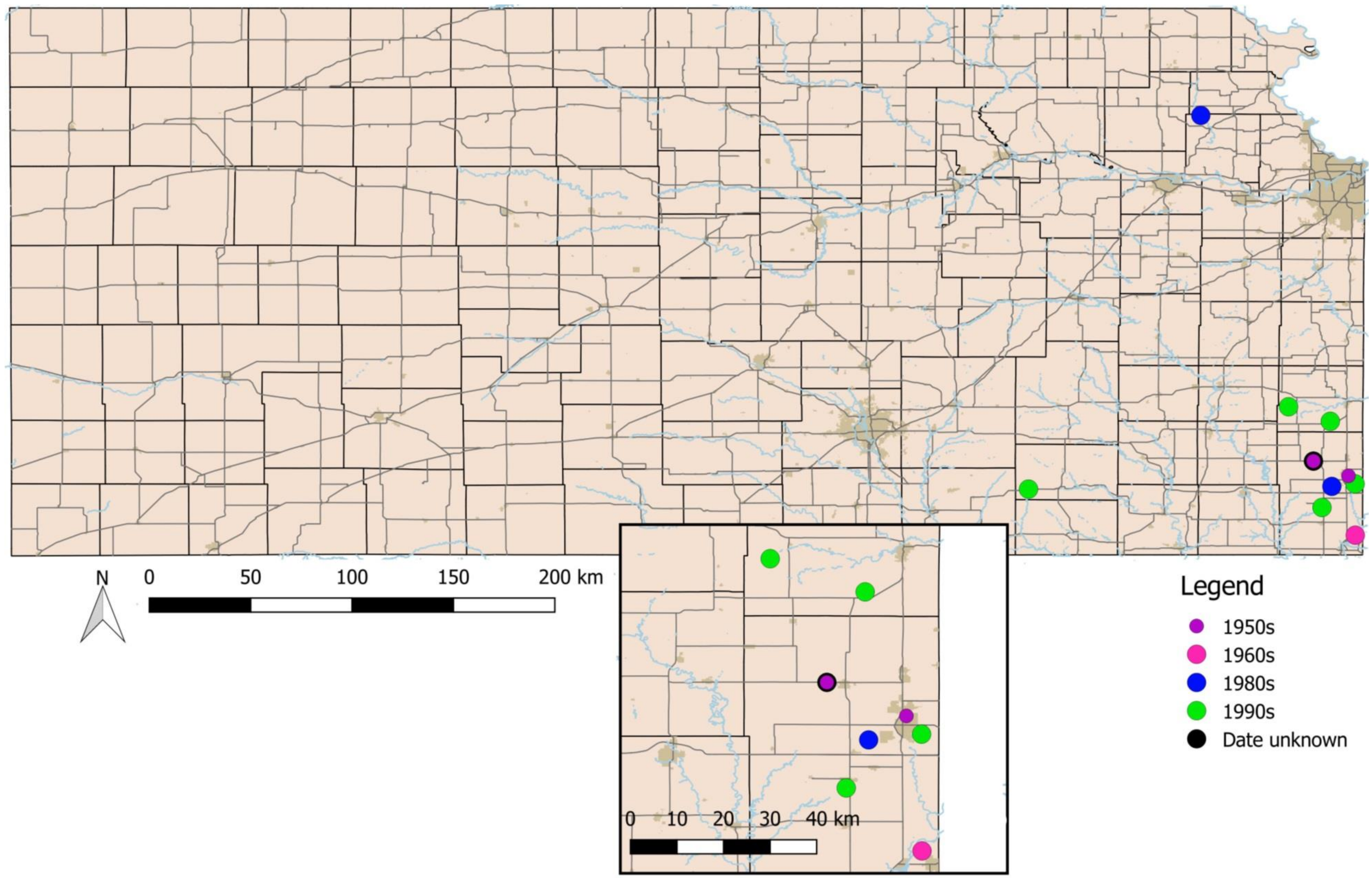

Figure 61. Spatial and temporal distribution of collecting sites of *Coluber constrictor flaviventris* (Eastern Yellow-bellied Racer).

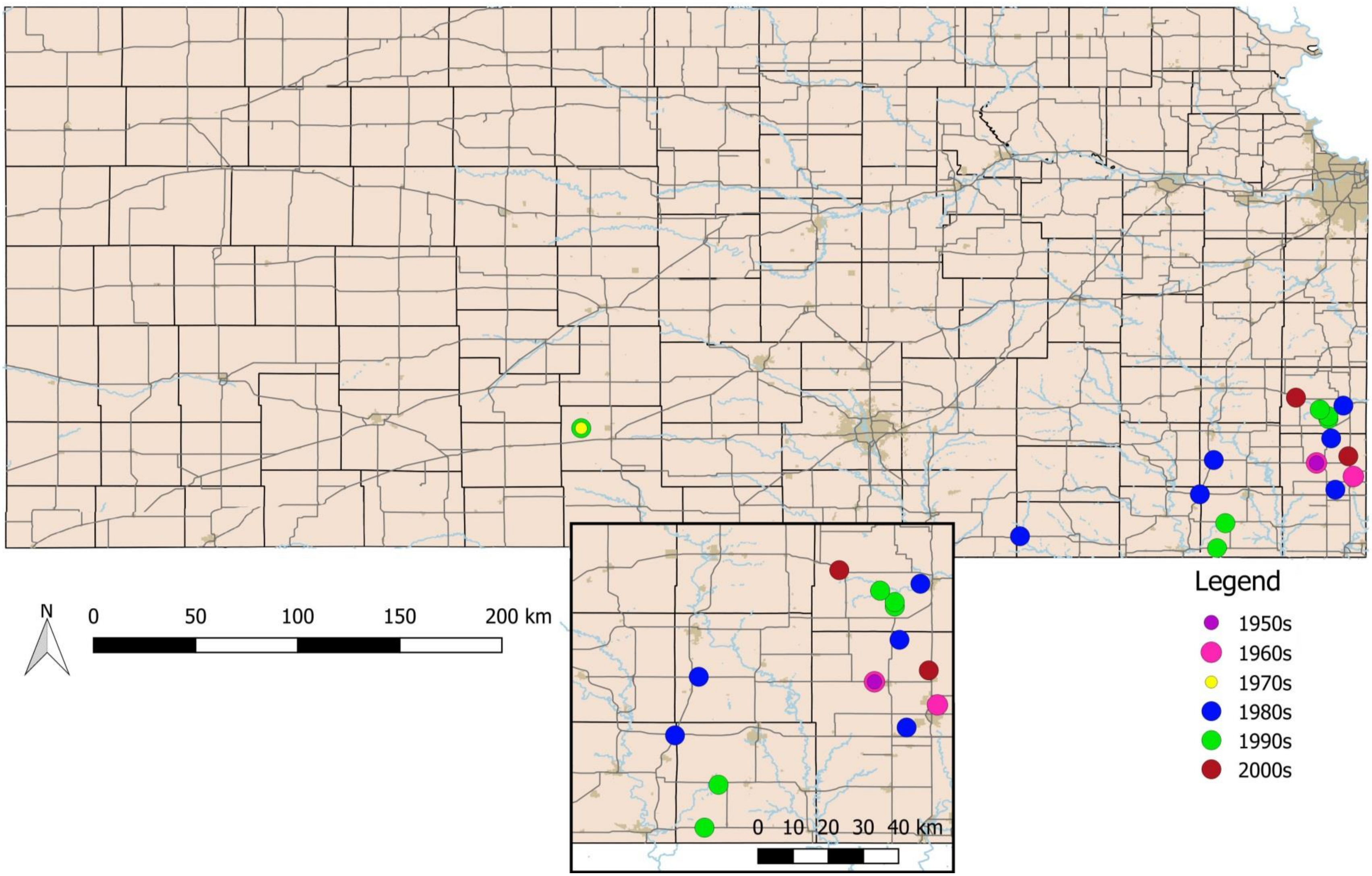

Figure 62. Spatial and temporal distribution of collecting sites of *Lampropeltis calligaster* (Prairie Kingsnake).

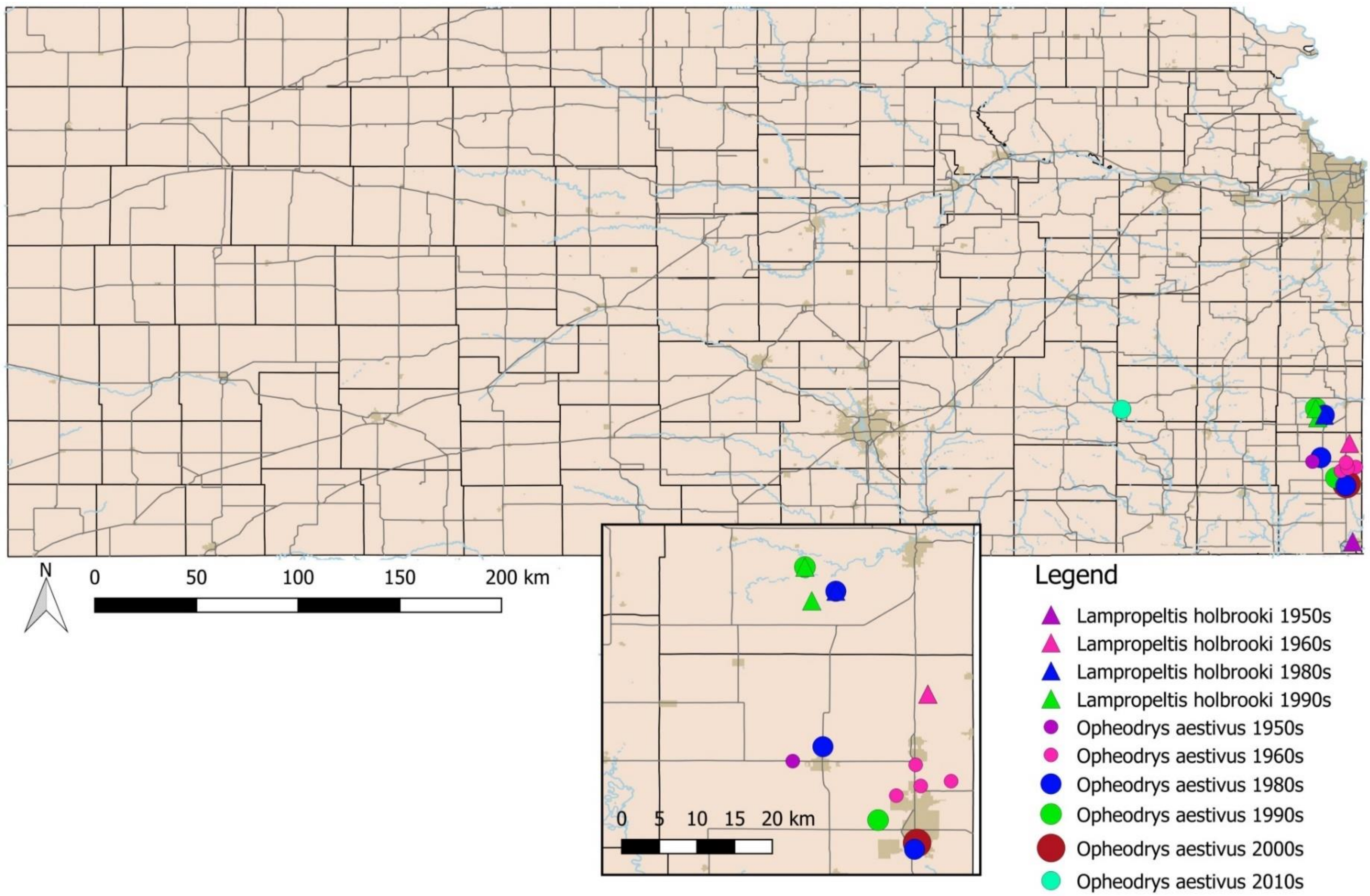

Figure 63. Spatial and temporal distribution of collecting sites of *Lampropeltis holbrooki* (Speckled Kingsnake) and *Opheodrys aestivus* (Rough Greensnake).

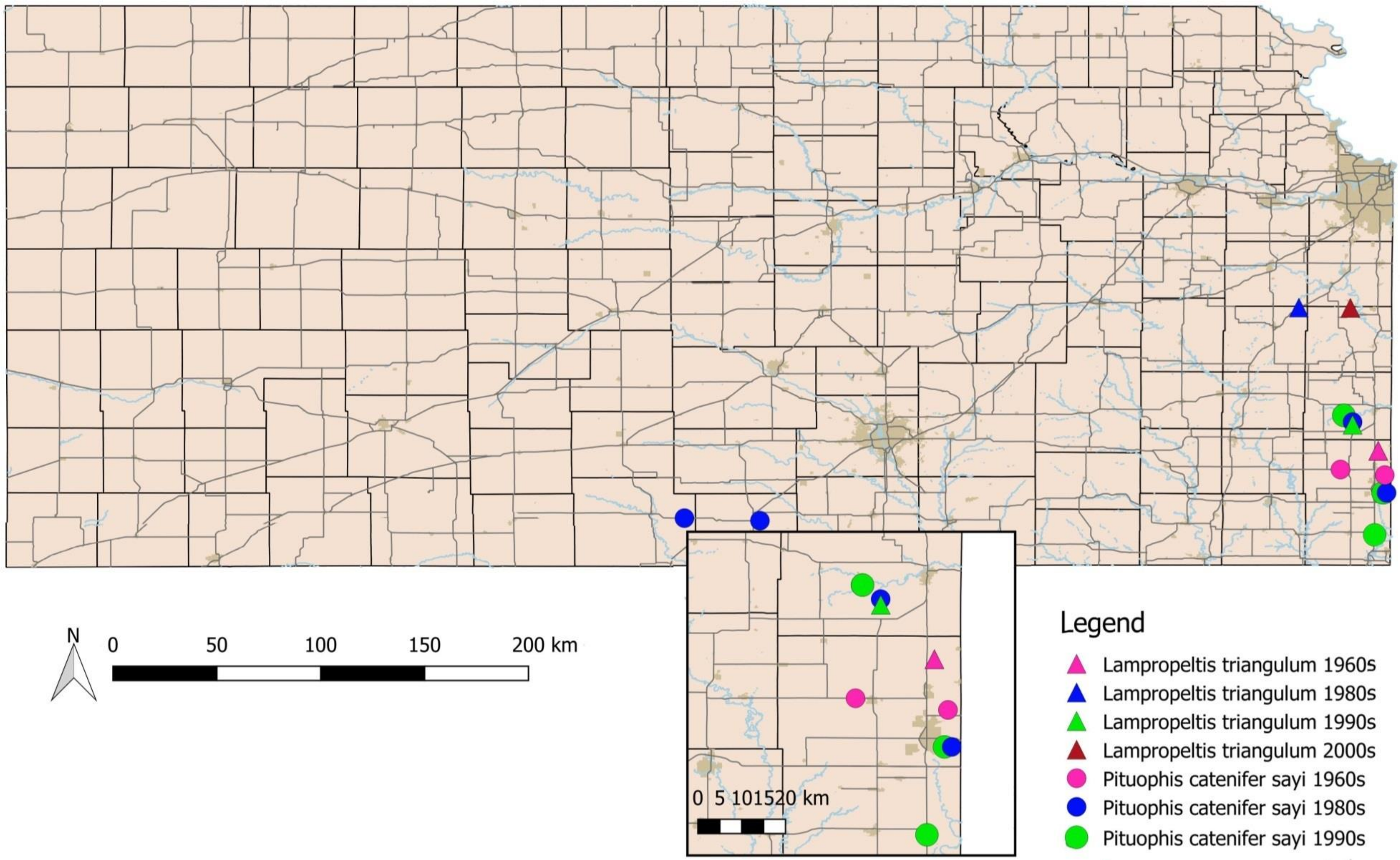

Figure 64. Spatial and temporal distribution of collecting sites of *Lampropeltis triangulum* (Milksnake) and *Pituophis catenifer sayi* (Bullsnake).

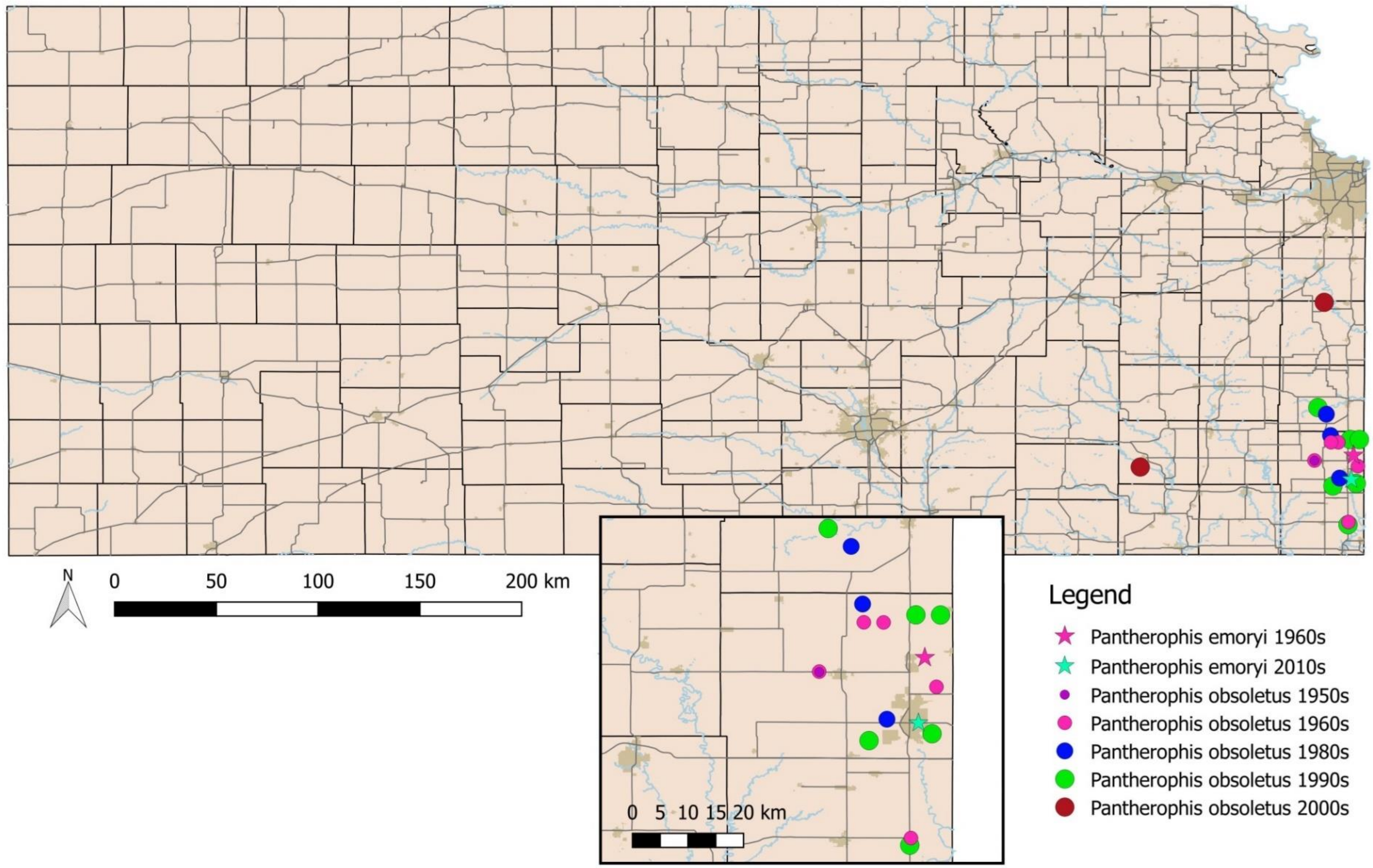

Figure 65. Spatial and temporal distribution of collecting sites of *Pantherophis emoryi* (Great Plains Ratsnake) and *Pantherophis obsoletus* (Western Ratsnake).

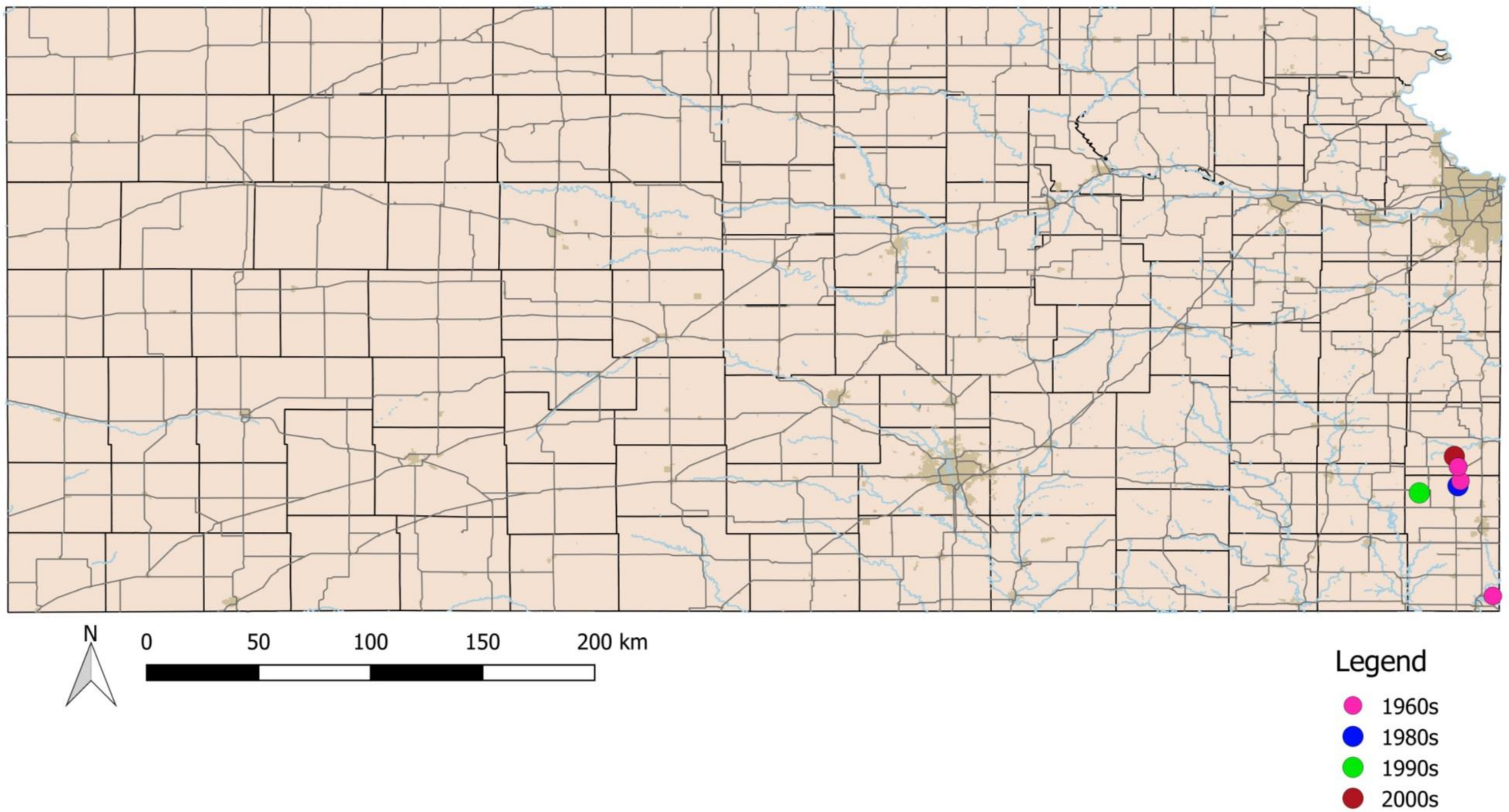

Figure 66. Spatial and temporal distribution of collecting sites of *Tantilla gracilis* (Flat-headed Snake).

Herpetology Teaching Collection

The 471 specimens without scientific data were moved to the teaching collection, which presently includes 87 taxa (Table 5).

Table 5. Phylogenetic list of herpetological specimens preserved at the Herpetology Teaching Collection at Pittsburg State University including total number of individuals per species.

Kingdom: Animalia
Phylum: Chordata
Superclass: Tetrapoda
  Class: Amphibia
  Subclass: Lissamphibia
  Superorder: Batrachia
    Order: Anura
      Family: Scaphiopodidae
        Spea bombifrons – Plains Spadefoot (1)
      Family Bufonidae
        Anaxyrus americanus – American Toad (15)
        Anaxyrus microscaphus – Arizona Toad (33)
        Anaxyrus punctatus – Red-spotted Toad (2)
        Anaxyrus terrestris – Southern Toad (29)
        Anaxyrus woodhousii - Woodhouse's Toad (9)
      Family Hylidae
        Acris blanchardi - Blanchard's Cricket Frog (23)
        Hyla chrysocelis / H. versicolor - Cope's Gray Treefrog / Gray Treefrog (3)
        Hyla squirella – Squirrel Tree Frog (12)
        Pseudacris clarkii - Spotted Chorus Frog (2)
        Pseudacris crucifer – Spring Peeper (1)
        Pseudacris ocularis – Little Grass Frog (4)
        Pseudacris triseriata – Western Chorus Frog (3)
      Family Ranidae
        Lithobates areolatus – Crawfish Frog (5)
        Lithobates berlandieri – Rio Grande Leopard Frog (2)
        Lithobates blairi – Plains Leopard Frog (5)
        Lithobates catesbeianus – American Bullfrog (14)
        Lithobates pipiens – Northern Leopard Frog (6)
        Lithobates sphenocephalus - Southern Leopard Frog (22)
    Order: Caudata
      Family Salamandridae
        Notophthalmus viridescens louisianensis - Central Newt (8)

Notophthalmus viridescens viridescens – Eastern Newt (4)
Family Ambystomatidae
Ambystoma opacum - Marbled Salamander (8)
Ambystoma texanum – Small-mouthed Salamander (8)
Ambystoma tigrinum – Eastern Tiger Salamander (7)
Family Plethodontidae
Eurycea aquatica – Brownback Salamander (1)
Eurycea longicauda melanopleura - Dark-sided Salamander (10)
Eurycea lucifuga – Cave Salamander (11)
Eurycea quadridigitata – Dwarf Salamander (1)
Plethodon albagula - Western Slimy Salamander (6)
Plethodon caddoensis – Caddo Mountain Salamander (2)
Plethodon kiamichi - Kiamichi Slimy Salamander (6)
Plethodon ouachita - Rich Mountain Salamander (4)
Plethodon serratus - Southern Red-backed Salamander (1)
Class: Reptilia
Subclass: Anapsida
Order: Testudines
Family: Chelydridae
Chelydra serpentina – Snapping Turtle (6)
Family Kinosternidae
Kinosternon arizonense – Arizona Mud Turtle (1)
Kinosternon flavescens – Yellow Mud Turtle (1)
Sternotherus odoratus - Eastern Musk Turtle (3)
Family Emydidae
Chyrsemys picta bellii - Western Painted Turtle (11)
Graptemys geographica – Northern Map Turtle (3)
Pseudemys concinna – River Cooter (2)
Terrapene carolina triunguis – Three-toad Box Turtle (6)
Terrapene ornata – Ornate Box Turtle (6)
Trachemys scripta elegans – Red-eared Slider (16)
Family Trionychidae
Apalone ferox – Florida Softshell (1)
Apalone spinifera spinifera - Eastern Spiny Softshell (20)
Subclass: Diapsida
Superorder: Crocodylomorpha
Order: Crocodilia
Family Alligatoridae
Caiman crocodilus – Spectacled Caiman (1)

Superorder: Lepidosauria

Order: Squamata

Infraorder: Gekkota

Family Eublepharidae

Coleonyx sp. – Gecko (1)

Infraorder: Sincomorpha

Family Scincidae

Plestiodon fasciatus – Common Five-lined Skink (22)

Plestiodon laticeps – Broad-headed Skink (3)

Plestiodon obsoletus – Great Plains Skink (6)

Plestiodon septentrionalis septentrionalis – Northern Prairie Skink (1)

Scincella lateralis – Little Brown Skink (5)

Family Teiidae

Aspidoscelis sexlineata – Six-lined Racerunner (4)

Infraorder: Anguimorpha

Family Anguidae

Ophisaurus attenuates – Slender Glass Lizard (8)

Infraorder: Iguania

Family Crotaphytidae

Crotaphytus collaris – Eastern Collard Lizard (5)

Family Phrynosomatidae

Sceloporus consobrinus - Prairie Lizard (4)

Uta stansburiana – Common Side-blotched Lizard (3)

Family Corytophanidae

Basiliscus vittatus – Brown Basilisk (1)

Suborder: Serpentes

Family Boidae

Boa constrictor - Red-tailed Boa (1)

Family Crotalidae

Agkistrodon contortrix – Eastern Copperhead (3)

Crotalus horridus – Timber Rattlesnake (4)

Sistrurus tergeminus – Western Massassauga (1)

Family Natricidae

Nerodia erythrogaster – Plain-bellied Watersnake (2)

Nerodia rhombifer – Diamond-backed Watersnake (2)

Nerodia sipedon sipedon – Common Watersnake (3)

Regina grahamii – Graham's Crawfish Snake (5)

Storeria dekayi - Dekay's Brownsnake (2)

Thamnophis proximus - Western Ribbonsnake (2)

Thamnophis radix – Plains Gartersnake (1)
Thamnophis sirtalis – Common Gartersnake (9)
Thamnophis sirtalis parietalis – Red-sided Gartersnake (2)
Tropidoclonion lineatum – Lined Snake (1)
Family Dipsadidae
Carphophis vermis – Western Wormsnake (7)
Diadophis punctatus arnyi - Prairie Ring-necked Snake (2)
Diadophis punctatus punctatus – Southern Ring-necked Snake (1)
Heterodon nasicus – Plains Hog-nosed Snake (1)
Family Colubridae
Coluber constrictor – North American Racer (4)
Coluber constrictor flaviventris – Eastern Yellow-bellied Racer (3)
Coluber schotti – Schott's Whipsnake (1)
Lampropeltis calligaster - Prairie Kingsnake (4)
Lampropeltis holbrooki – Speckled Kingsnake (5)
Lampropeltis triangulum – Eastern Milksnake (1)
Opheodrys aestivus – Rough Green Snake (5)
Pantherophis obsoletus – Western Ratsnake (4)
Pituophis catenifer – Gophersnake (1)
Pituophis catenifer sayi – Bullsnake (1)
Pituophis melanoleucus – Eastern Pinesnake (1)

Most specimens had been curated during the previous assessment for the research portion of the HC; however, an additional 123 specimens required further curatorial work including a change of fluids and rehydration (Fig. 67). Approximately 42 specimens were preserved in formalin (which were later transferred to 70% ethanol), 22 were in unknown preservatives, 10 specimens were rehydrated, and the remaining 49 specimens needed new fluids due to low levels of preservative or the presence of rust from metal lids.

After specimen identifications were reviewed, they were grouped by species into individual jars (Figs. 68 and 69). The jars and the boxes containing them were labeled only with the catalogue number. A cross-reference list was created containing the catalogue number, species name and total number of specimens per jar. Specimens in the teaching collection were not tagged individually, as their purpose is to help students learn to identify species.

The herpetology teaching collection also includes approximately 32 freeze-dried turtle specimens, which required immediate curation and pest control (Fig. 70).

Figure 67. Results of inappropriate specimen enclosures: dehydration and rust. Several specimens in the teaching collection had extensive curatorial needs, including rehydration and changes of fluid, mainly due to being preserved in formalin or other unknown preservatives, evaporation of preservative of the solution, or presence of rust. Rust, as seen on the picture above, can cause extreme deterioration to the lid, jar and especially to the specimen, altering the preservative solution and specimen coloration.

Figure 68. Specimen identifications were verified using dichotomous keys (Altig and McDiarmid, 2015; Collins *et al*., 2010; ; Powell *et al*., 2012) **and field guides** (Collins, 1993; Powell *et al.*, 2016; Stebbins, 2003). Afterwards the specimens were grouped by species, curated, and placed in new collection jars.

Figure 69. Herpetology teaching collection specimens after curation, preserved in 70% ETOH, stored on O.Berk© jars and polypropelene containers. From left to right: Rough Green Snake (*Opheodrys aestivus*), Graham's Crawfish Snake (*Regina grahamii*), Western Massassauga (*Sistrurus tergeminus*), Ornate Box Turtle (*Terrapene ornata*), and Red-eared Slider (*Trachemys scripta*).

Figure 70. Before (left) and after curation (right) and pest control of freeze-dried specimens. A. Specimens had been stored for over a decade in a humid basement inside a cardboard box, resulting in deterioration through humidity and proliferation of insect pests. The yellow dust is the result of dermestid beetles feeding on the dried specimens. Some specimens had severe damage, including loss of entire limbs. **B.** After curation and pest control, specimens were identified, grouped by species, and stored in archival cardboard boxes linned with ethafoam.

Databasing and Initial Digitization of the Herpetology Collection, Field Notebooks and Teaching Collection

The HC database (Appendix D) provides easy access to data, which was essential for the data analysis carried out in this project. It now can be shared digitally and several copies have backed-up in different locations at PSU. Backups are important, because the laboratory does not have its own computer; and is the base work for publishing the data on online databases (e.g. VertNet). Field Notebooks (Appendix E) and the Teaching Collection also were databased.

The next step priority should be completing digitization by transfering the HC data into Darwin Core standards, which were created to facilitate the sharing of biological information worldwide by provided a standardized glossary of terms to be used in columns, fields, and attributes of the database (Wieczorek *et al.*, 2015). Along with the publication of the dataset, it will be necessary to have a collection website or a webpage [within the institution's website] providing information about the institution, the collection, and specimens available, as well as a link to the online database(s) where collection`s data is stored (Appendix F).

Digitization commonly includes imaging specimens. However, this is an extremely time consuming task that required photographing specimens from several angles to capture all identifying characters, editing photos, and publishing high-quality images (Appendix G). Thus, photography of HC specimens (e.g., Fig. 71) thus far has been to illustrate the results of the present project, for research presentations (e.g., posters), and to enhance the quality of the website and blog (Appendix F).

Figure 71. Specimen imaging: edited image showing dorsal and ventral sides of a Bullsnake (*Pituophis catenifer sayi*). Specimen collected at Hollister Wildlife Area, Bourbon Co., Kansas on April 19, 1987 by Doug Whiteaker. Nomenclature updated from *Pituophis melanoleucos sayi* to *P. catenifer sayi*. Specimen is a male, measuring 1,405 mm.

CHAPTER IV

CONCLUSIONS

This study has demonstrated that herpetological specimens stored in poor curatorial conditions for over 40 years can, to a large degree, be effectively conserved and returned to modern curatorial standards through informed and systemic application of best curatorial practices. Without the curation and digitization of Pittsburg State University's herpetological collection as carried out in this project, there would be no ability to ask biological questions or test hypotheses of their taxonomy or ecological distribution. As such, steady curation of specimens through time is of fundamental importance if the full complement of biological data is to be accessible to researchers. The databasing of the hand-written catalogue into a Excel database, which this project included, has been completed. These data will be uploaded to the internet soon, after which they will be accessible to any researcher in the world. The results of this project will open new possibilities of research on specimens within a collection that rarely if ever had been a primary source of biodiversity data. It also provides protocols and further steps required for improved curation and management.

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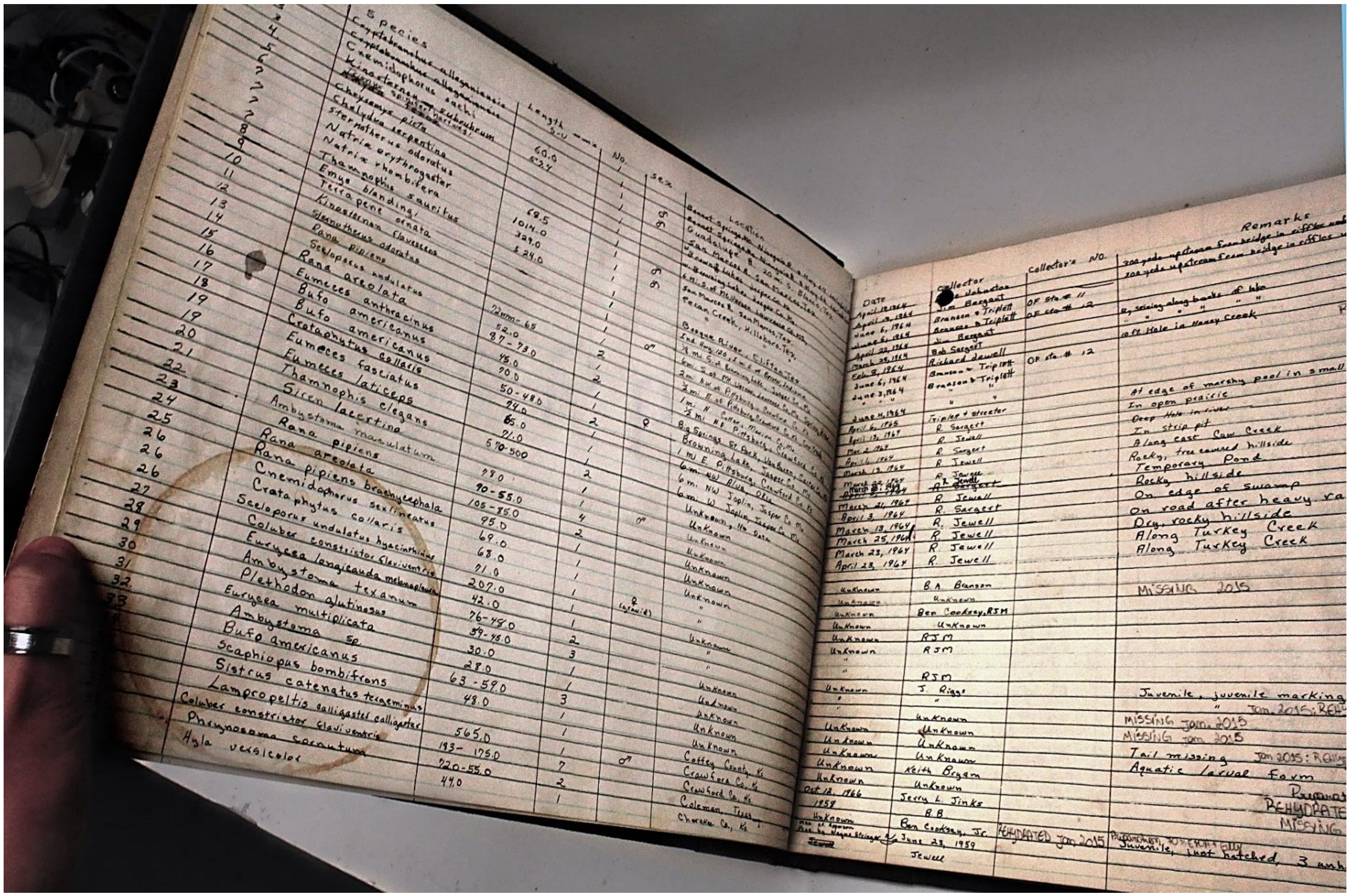
APPENDIX

Appendix A. The first engraved record of a Cabinet of Curiosities. Entitled *Dell'Historia Naturale*, engraved by Ferrante Imperato in 1599, it is a detailed record of Imperato's museum, the disposition of the specimens preserved and the presence of visitors. **Source:** Wikimedia Commons

Appendix B. Herpetology Collection's Handwritten Catalogue. The catalogue contains data for over 600 specimens, including jar number, species, total number of individuals, measurments, sex, collecting location, collecting remarks, collector, date, fluid preparation and curatorial notes. Changes in fluid preparation, curation activity and loans were updated.



Appendix C. Conservation Status and Population Trends of Reptile and Amphibians Preserved at the Herpetology Collection at Pittsburg State University, Kansas. Conservation status abbreviations: LC – Least Concern, NT – Near threatened, TN – Threatened, EN – Endangered, SINI – Species in need of information, and SINC – Species in need of conservation. **Sources:** RedList (IUCN, 2017) and Kansas Herpetofaunal Atlas (Taggart, 2017).

ORDER	SPECIES	CONSERVATION STATUS, POPULATION TREND RedList	CONSERVATION Kansas
ANURA	*Acris blanchardi*	Not yet assessed by RedList	
	Acris crepitans	LC - stable, needs updating	
	Anaxyrus americanus	LC – stable	
	Anaxyrus boreas	LC – decreasing	
	Anaxyrus canorus	EN - decreasing, needs updating	
	Anaxyrus cognatus	LC - stable	
	Anaxyrus hemiophrys	LC - stable	
	Anaxyrus woodhousii	LC - stable, needs updating	
	Anaxyrus woodhousii woodhousii	Not yet assessed by RedList	
	Gastrophryne carolinensis	LC - stable	TN
	Gastrophryne olivacea	LC - stable, needs updating	
	Hyla arenicolor	LC - stable	
	Hyla chrysocelis / Hyla versicolor	LC - stable	
	Hyla squirella	LC - stable, needs updating	
	Incilius nebulifer	LC - stable, needs updating	
	Lithobates areolatus	NT - decreasing, needs update	SINC
	Lithobates areolatus circulosus	Not yet assessed by RedList	
	Lithobates berlandieri	LC - stable	
	Lithobates blairi	LC - decreasing	
	Lithobates catesbeianus	LC - increasing	
	Lithobates clamitans	LC - stable	TN
	Lithobates palustris	LC - stable	SINI
	Lithobates pipiens	LC - decreasing, needs update	

ORDER	SPECIES	CONSERVATION STATUS, POPULATION TREND RedList	CONSERVATION Kansas
ANURA	*Lithobates sphenocephalus*	LC - stable	
	Lithobates sylvaticus	LC - stable	
	Pseudacris clarkii	LC - stable	
	Pseudacris crucifer	LC - stable	SINC
	Pseudacris fouquettei	LC - stable	
	Pseudacris maculata	LC - stable	
	Pseudacris triseriata	LC - decreasing	
	Rana boylii	NT - decreasing, needs update	
	Rana luteiventris	LC - decreasing	
	Rana pretiosa	VU - decreasing, needs update	
	Rana sierrae	EN - decreasing	
	Scaphiopus couchii	LC - stable, needs updating	
	Spea bombifrons	LC - stable	
	Spea intermontana	LC - stable	
CAUDATA	*Ambystoma maculatum*	LC - stable	
	Ambystoma mavortium	LC - stable	
	Ambystoma texanum	LC - stable	
	Ambystoma tigrinum	LC - stable	SINI
	Amphiuma tridactylum	LC - stable, needs updating	
	Cryptobranchus alleganiensis	NT - decreasing, needs update	
	Desmognathus brimleyorum	LC - stable, needs updating	
	Desmognathus fuscus	LC - stable, needs updating	
	Desmognathus monticola	LC - stable, needs updating	
	Desmognathus quadramaculatus	LC - stable	
	Eurycea longicauda longicauda	Not yet assessed by RedList, *E. longicauda* is LC - stable	
	Eurycea longicauda melanopleura	Not yet assessed by RedList, *E. longicauda* is LC - stable	TN
	Eurycea lucifuga	LC - unknown	EN

ORDER	SPECIES	CONSERVATION STATUS, POPULATION TREND RedList	CONSERVATION Kansas
CAUDATA	*Eurycea multiplicata*	LC - stable, needs updating	
	Eurycea nana	VU - stable, needs update	
	Eurycea spelaea	LC - unknown, needs updating	EN
	Eurycea tynerensis	NT - decreasing, needs update	
	Necturus maculosus	LC - stable	SINI
	Necturus maculosus louisianensis	Not yet assessed by RedList	SINI
	Necturus maculosus maculosus	Not yet assessed by RedList	SINI
	Notophthalmus viridescens	LC - stable	
	Notophthalmus viridescens louisianensis	Not yet assessed by RedList	TN
	Plethodon albagula	LC - stable	
	Plethodon angusticlavius	LC - stable	
	Plethodon caddoensis	NT - unknown, needs update	
	Plethodon cinereus	LC - stable	
	Plethodon dorsalis	LC - stable	
	Plethodon fourchensis	VU - unkwonw, needs update	
	Plethodon glutinosus	LC - stable	
	Plethodon ouachitae	NT - stable, needs update	
	Plethodon serratus	LC - stable	
	Rhyacotriton olympicus	VU - decreasing, needs update	
	Siren lacertina	LC - unknown	
	Taricha granulosa	LC - stable	
CHELONIA	*Apalone ferox*	LC - unknown	
	Apalone mutica mutica	Not yet assessed by RedList, *A. mutica* is LC - unknown	
	Apalone spinifera spinifera	Not yet assessed by RedList, *A. spinifera* is LC - stable	
	Chelydra serpentina	LC - stable	
	Chrysemys picta	LC - stable	
	Chrysemys picta bellii	Not yet assessed by RedList	

ORDER	SPECIES	CONSERVATION STATUS, POPULATION TREND RedList	CONSERVATION Kansas
CHELONIA	*Chrysemys picta picta*	Not yet assessed by RedList	
	Emydoidea blandingii	EN - decreasing	
	Graptemys pseudogeographica	LC - unknown	
	Graptemys pseudogeographica kohnii	Not yet assessed by RedList	
	Kinosternon flavescens	LC - unknown	
	Kinosternon subrubrum	LC - unknown	
	Pseudemys concinna	LC - unknown	
	Pseudemys concinna floridana	Not yet assessed by RedList	
	Sternotherus odoratus	LC - stable	
	Terrapene triunguis	Not yet assessed by RedList	
	Terrapene ornata	NT - decreasing	
	Trachemys scripta elegans	Not yet assessed by RedList, *T. scripta* is LC - stable	
CROCODILIA	*Caiman crocodilus*	LC - needs updating	
LACERTILIA	*Anolis carolinensis*	LC - stable	
	Aspidoscelis gularis	LC - stable	
	Aspidoscelis marmorata	Not yet assessed by RedList	
	Aspidoscelis sackii	Not yet assessed by RedList	
	Aspidoscelis sexlineata	LC - stable	
	Aspidoscelis sexlineata viridis	Not yet assessed by RedList	
	Aspidoscelis tesselata	LC - stable	
	Aspidoscelis tigris	LC - stable	
	Collisaurus draconoides	Not yet assessed by RedList	
	Cophosaurus texanus	LC - stable	
	Crotaphytus collaris	LC - stable	
	Elgaria coerulea	LC - stable	
	Elgaria multicarinata	LC - decreasing	
	Eublepharis macularius	LC - unknown	

ORDER	SPECIES	CONSERVATION STATUS, POPULATION TREND RedList	CONSERVATION Kansas
LACERTILIA	*Gambelia wislizenii*	LC - stable	
	Gecko gecko	Not yet assessed by RedList	
	Holbrookia propinqua propinqua	Not yet assessed by RedList, *H. propinqua* is LC - stable	
	Ophisaurus attenuatus	LC - stable	
	Ophisaurus attenuatus attenuatus	Not yet assessed by RedList	
	Phrynosoma cornutum	LC - stable	SINI
	Phrynosoma douglasii	LC - stable	
	Phrynosoma platyrhinos	LC - stable	
	Plestiodon anthracinus	LC - stable	
	Plestiodon fasciatus	LC - stable	
	Plestiodon laticeps	LC - stable	TN
	Plestiodon obsoletus	LC - stable	
	Sceloporus consobrinus	LC - stable	
	Sceloporus jarrovii	Not yet assessed by RedList	
	Sceloporus magister	LC - stable	
	Sceloporus occidentalis	LC - stable	
	Sceloporus undulatus	LC - stable	
	Sceloporus virgatus	LC - stable	
	Scincella lateralis	LC - stable	
	Uta stansburiana	LC - stable	
	Uta stansburiana stansburiana	Not yet assessed by RedList	
SERPENTES	*Agkistrodon contortrix*	LC - stable	
	Agkistrodon piscivorus	LC - stable	SINI
	Carphophis amoenus	LC - stable	
	Carphophis amoenus amoenus	Not yet assessed by RedList	
	Carphophis vermis	LC - stable	
	Coluber constrictor	LC - stable	

ORDER	SPECIES	CONSERVATION STATUS, POPULATION TREND RedList	CONSERVATION Kansas
SERPENTES	*Coluber constrictor flaviventris*	Not yet assessed by RedList	
	Coluber flagellum flagellum	Not yet assessed by RedList	
	Coluber flagellum testaceus	Not yet assessed by RedList	
	Crotalus atrox	LC - stable	
	Crotalus horridus	LC - decreasing	SINC
	Crotalus viridis	LC - stable	
	Diadophis punctatus	LC - stable	
	Diadophis punctatus arnyi	Not yet assessed by RedList	
	Diadophis punctatus edwardsii	Not yet assessed by RedList	
	Diadophis punctatus punctatus	Not yet assessed by RedList	
	Diadophis punctatus stictogenys	Not yet assessed by RedList	
	Drymarchon corais corais	Not yet assessed by RedList	
	Heterodon nasicus	LC - stable	SINC
	Heterodon platirhinos	LC - stable	SINC
	Lampropeltis calligaster	LC - stable	
	Lampropeltis getula	LC - stable	
	Lampropeltis holbrooki	Not yet assessed by RedList	
	Lampropeltis triangulum	Not yet assessed by RedList	
	Lampropeltis zonata	LC - decreasing	
	Nerodia erythrogaster	LC - stable	
	Nerodia rhombifer	LC - stable	
	Nerodia sipedon	LC - stable	
	Nerodia sipedon sipedon	Not yet assessed by RedList	
	Opheodrys aestivus	LC - stable	
	Opheodrys vernalis	LC - stable	
	Pantherophis emoryi	LC - stable	
	Pantherophis obsoletus	LC - stable	

ORDER	SPECIES	CONSERVATION STATUS, POPULATION TREND RedList	CONSERVATION Kansas
SERPENTES	*Pituophis catenifer*	LC - stable	
	Pituophis catenifer sayi	Not yet assessed by RedList	
	Python reticulatus	Not yet assessed by RedList	
	Regina grahamii	LC - unknown	
	Sistrurus catenatus tergeminus	LC - unknown	
	Sonora semiannulata	LC - stable	
	Storeria dekayi	LC - stable	
	Storeria dekayi texana	Not yet assessed by RedList	
	Storeria occipitomaculata	LC - stable	SINC
	Tantilla gracilis	LC - decreasing	
	Tantilla nigriceps	LC - stable	
	Thamnophis elegans	LC - stable	
	Thamnophis proximus	LC - stable, needs updating	
	Thamnophis radix	LC – stable	
	Thamnophis sauritus	LC – stable	
	Thamnophis sirtalis	LC – stable	
	Thamnophis sirtalis parietalis	Not yet assessed by RedList	
	Thamnophis sirtalis pickeringii	Not yet assessed by RedList	
	Thamnophis sirtalis sirtalis	Not yet assessed by RedList	
	Tropidoclonion lineatum	LC – stable	
	Haldea striatula	LC – stable	SINC
	Virginia valeriae	LC – stable	SINC

Appendix D. Example of the digital version of the herpetology catalogue. It contains data taken from the original, handwritten accessions catalogue, including specimen tags of newly catalogued specimens and field notebooks.

	A	B	C	D	E	F	G	H
1	Catalogued JAR #	Species (specimen tag ID)	Species (nomenclature updated 2017)	Taxonomy Change (See Remarks for justification)	Common Name (nomeclature updated 2012)	Total specimen	Length (mm)	Sex
2	1	*Cryptobranehus alleganiensis*	*Cryptobranchus alleganiensis*		Hellbender	1		
3	2	*Cryptobranehus alleganiensis*	*Cryptobranchus alleganiensis*		Hellbender	1		
4	3	*Cnemidophorus sachi*	*Aspidoscelis sackii*		Sack's Spotted Whiptail	1	60	♂
5	3	*Kinosternon subrubrum*	*Kinosternon subrubrum*		Eastern Mud Turtle	1	57.4	♂
6	4	*Trionyx spinifer hartwegi*	*Apalone spinifera spinifera*		Eastern Spiny Sooftshell	1		
7	5	*Chrysemys picta*	*Chrysemys picta*		Painted Turtle	1		
8	6	*Chelydra serpentina*	*Chelydra serpentina*		Snapping Turtle	1		
9	7	*Natrix erythrogaster*	*Nerodia erythrogaster*		Plain-bellied Watersnake	1	1014	♂
10	7	*Natrix rhombifera*	*Nerodia rhombifer*		Diamond-backed Watersnake	1	329	
11	7	*Sternotherus odoratus*	*Sternotherus odoratus*		Eastern Musk Turtle	1	68.5	♂
12	7	*Thamnophis sauritus*	*Thamnophis sauritus*		Eastern Ribbonsnake	1	524	
13	8	*Emys blandingi*	*Emydoidea blandingii*		Blanding's Turtle	1		
14	9	*Terrapene ornata*	*Terrapene ornata*		Ornate Box Turtle	1		
15	10	*Kinosternon flavescens*	*Kinosternon flavescens*		Yellow Mud Turtle	1		♂
16	11	*Sternotherus odoratus*	*Sternotherus odoratus*		Eastern Musk Turtle	1		
17	12	*Rana pipiens*	*Lithobates pipiens*	*Lithobates sphenocephalus*	Southern Leopard Frog	2	72 / 65	
18	13	*Sceloporus undulatus*	*Sceloporus undulatus*		Eastern Fence Lizard	1	52	
19	14	*Rana areolata*	*Lithobates areolatus*	*Lithobates areolatus circulosus*	Northern Crawfish Frog	2	87 / 73	
20	15	*Eumeces anthracinus*	*Plestiodon anthracinus*		Coal Skink	1	45	
21	16	*Bufo americanus*	*Anaxyrus americanus*		American Toad	1	70	♀
22	17	*Bufo americanus*	*Anaxyrus americanus*		American Toad	2	50 / 48	
23	18	*Crotaphytus collaris*	*Crotaphytus collaris*		Eastern Collared Lizard	1	74	
24	19	*Eumeces fasciatus*	*Plestiodon fasciatus*		Common Five-lined Skink	1	65	
25	19	*Eumeces laticeps*	*Plestiodon laticeps*		Broad-headed Skink	1	71	

	State	County	City	Location	Twp & Rng Coordinat	Latitute	Longitude	GPS Coordinates	Date	Year	Collector
2	MISSOURI	Lacledo	Bannet Springs	Bannet Springs, MO. Nianguia Road of Hwy 64. Lacledo Co.					April 19, 1964	1964	Dave Johnston
3	MISSOURI	Lacledo	Bannet Springs	Bannet Springs, MO. Nianguia Road of Hwy 64. Lacledo Co.					April 19, 1964	1964	Jim Bergant
4	TEXAS	Blanco	Blanco	Blanco, TX. Guadalupe Road. 20 mi S. of Blanco.					June 6, 1964	1964	Branley A. Branson & James R. Triplett
5	TEXAS	Hays	San Marcos	San Marcos, TX. San Marcos Road.					June 6, 1964	1964	Branley A. Branson & James R. Triplett
6	MISSOURI	Jasper		Jasper Co., MO. W of Browning Lakes.					April 22, 1964	1964	Jim Bergant
7	MISSOURI	Jasper		Jasper Co., MO. W of Browning Lakes.					March 28, 1964	1964	Robert Sargent
8	MISSOURI	Lawrence		Lawrence Co., MO. 6 mi S of Mt. Vernon.					February 8, 1964	1964	Richard Dewell
9	TEXAS	Hill	Hillsboro	Hillsboro, TX. Pecan Creek.					June 3, 1964	1964	Branley A. Branson & James R. Triplett
10	TEXAS	Hill	Hillsboro	Hillsboro, TX. Pecan Creek.					June 3, 1964	1964	Branley A. Branson & James R. Triplett
11	TEXAS	Hays	San Marcos	San Marcos, TX. San Marcos Road.					June 6, 1964	1964	Branley A. Branson & James R. Triplett
12	TEXAS	Bosque	Clifton	Clifton, TX. Bosque River.					June 4, 1964	1964	Branley A. Branson & James R. Triplett
13	INDIANA	Elkhart	Bristol	Bristol, IN. Indiana Hwy 120, 1 mi E of Bristol.					April 6, 1965	1965	James R. Triplett + Roger Streeter
14	MISSOURI	Jasper		Jasper Co., MO. 1/8 mi S of Browning Lakes.					April 13, 1964	1964	Robert Sargent
15	MISSOURI	Lawrence		Spring River Wildlife Area, 6 mi S of Mt. Vernon, Lawrence Co., MO					March 2, 1964	1964	Robert Jewell
16	KANSAS	Crawford	Pittsburg	Pittsburg, KS. 2 mi NW of Pittsburg, Crawford Co.		37.4403733	-94.7416628	37.4403733, -94.7416628	April 1, 1964	1964	Robert Sargent
17	KANSAS	Crawford	Pittsburg	Pittsburg, KS. 1/2 mi E of Pittsburg, Crawford Co. Cow Creek		37.4109934	-94.6499428	37.4109934, -94.6499428	March 13, 1964	1964	Robert Jewell
18	ARKANSAS	Marion	Cotter	Cotter, AR. 1 mi N of Cotter, Marion Co.					March 22, 1964	1964	Robert Jewell
19	KANSAS	Crawford	Pittsburg	Pittsburg, KS. 1/2 mi NE of Pittsburg, Crawford Co.		37.4183788	-94.6958093	37.4183788, -94.6958093	March 28, 1964	1964	Robert Jewell
20	MISSOURI	Carter	Van Buren	Big Spring Park. 4 mi S of Van Buren, Missouri					March 21, 1964	1964	Robert Jewell
21	MISSOURI	Jasper		Jasper Co., MO. W of Browning Lakes.					April 3, 1964	1964	Robert Sargent
22	KANSAS	Crawford	Pittsburg	Pittsburg, KS. 1 mi E of Pittsburg, Crawford Co.		37.4109432	-94.6867628	37.4109432, -94.6867628	March 13, 1964	1964	Robert Jewell
23	OKLAHOMA	Woods	Alva	Alva, OK. 6 mi NW of Alva.					March 25, 1964	1964	Robert Jewell
24	MISSOURI	Jasper	Joplin	Joplin, MO. 6 mi NW of Joplin, Jasper Co.					March 23, 1964	1964	Robert Jewell
25	MISSOURI	Jasper	Joplin	Joplin, MO. 6 mi NW of Joplin, Jasper Co.					April 23, 1964	1964	Robert Jewell

	Preparation	Preparation Changes	Notes	Remarks	Entry Date	Cataloguing Staff
2	10% formalin		CROCK #6 / Missing 2015	300 yards upstream from bridge in riffles under rocks	1967	James R. Triplett
3	10% formalin		CROCK #6 / Missing 2015	300 yards upstream from bridge in riffles under rocks	1967	James R. Triplett
4	70% EtOH + Glycerine	Refilled Jan. 2015 70% EtOH / Prep. Changed Jul. 2015 70% ETOH		Verify range (supposed to occur only in México), supposedly collected about 180 mi from México / Sta.	1967	James R. Triplett
5	70% EtOH + Glycerine	Refilled Jan. 2015 70% EtOH / Prep. Changed Jul. 2015 70% ETOH		Sta. #11	1967	James R. Triplett
6	10% formalin		CROCK #6 / Missing 2015	By seining along banks of lake	1967	James R. Triplett
7	10% formalin		CROCK #6 / Missing 2015	By seining along banks of lake	1967	James R. Triplett
8	10% formalin		CROCK #6 / Missing 2015	10 ft hole in Honey Creek	1967	James R. Triplett
9	70% EtOH + Glycerine	Rehyd. Jan. 2015: H_2O+surfactant. Prep.: 70% EtOH+Gly / Prep. Changed Jul. 2015 70% ETOH	Rehydrated Jan. 2015		1967	James R. Triplett
10	70% EtOH + Glycerine	Rehyd. Jan. 2015: H_2O+surfactant. Prep.: 70% EtOH+Gly / Prep. Changed Jul. 2015 70% ETOH	Rehydrated Jan. 2015		1967	James R. Triplett
11	70% EtOH + Glycerine	Rehyd. Jan. 2015: H_2O+surfactant. Prep.: 70% EtOH+Gly / Prep. Changed Jul. 2015 70% ETOH	Rehydrated Jan. 2015		1967	James R. Triplett
12	70% EtOH + Glycerine	Rehyd. Jan. 2015: H_2O+surfactant. Prep.: 70% EtOH+Gly / Prep. Changed Jul. 2015 70% ETOH	Rehydrated Jan. 2015		1967	James R. Triplett
13	10% formalin		CROCK #6 / Missing 2015	At edge of marshy pool in small stream	1967	James R. Triplett
14	70% EtOH + Glycerine			In open prairie	1967	James R. Triplett
15	70% EtOH + Glycerine	Refilled Jan. 2015 70% EtOH		Deep hole in river	1967	James R. Triplett
16	70% EtOH + Glycerine			In strip pit	1967	James R. Triplett
17	70% EtOH + Glycerine	Refilled Jan. 2015 70% EtOH		Along E Cow Creek	1967	James R. Triplett
18	70% EtOH + Glycerine	Refilled Jan. 2015 70% EtOH		Rocky, tree covered hillside	1967	James R. Triplett
19	70% EtOH	Refilled Jan. 2015 70% EtOH / Prep. Changed Jul. 2015 70% ETOH		Temporary pond / Based on range this specimen belong	1967	James R. Triplett
20	70% EtOH + Glycerine	Refilled Jan. 2015 70% EtOH		Rocky hillside	1967	James R. Triplett
21	70% EtOH + Glycerine	Refilled Jan. 2015 70% EtOH		On edge of swamp	1967	James R. Triplett
22	70% EtOH + Glycerine			On road after heavy rain	1967	James R. Triplett
23	70% EtOH + Glycerine	Refilled Jul. 2015 70%EtOH		Dry, rocky hillside	1967	James R. Triplett
24	70% EtOH + Glycerine			Along Turkey Creek	1967	James R. Triplett
25	70% EtOH + Glycerine			Along Turkey Creek	1967	James R. Triplett

Appendix E. Example of the digital version of herpetology field notebooks. All field notebooks were databased in Excel, with the following showing a sample of data from field notebooks from 2011.

	Species Name	Common Name	talIndividualsColl	TotalAdults(	TotalJuvCol	TotalIndiviualsIDinFi	State	County	City	
2	*Acris crepitans blanchardi*	Blanchard's Cricket Frog	1			1	MISSOURI	Barton		
3	*Acris crepitans blanchardi*	Blanchard's Cricket Frog	2				KANSAS	Linn	Pleasanton	
4	*Acris crepitans blanchardi*	Blanchard's Cricket Frog				<15	KANSAS	Bourbon		Hc
5	*Acris crepitans blanchardi*	Blanchard's Cricket Frog	1				KANSAS	Woodson		
6	*Acris crepitans blanchardi*	Blanchard's Cricket Frog				<15	KANSAS	Leavenworth		
7	*Acris crepitans blanchardi*	Blanchard's Cricket Frog				3	OKLAHOMA	Delaware		
8	*Acris crepitans crepitans*	Northern Cricket Frog				4	TEXAS	Jasper		
9	*Agkistrodon contortrix*	Copperhead				1	KANSAS	Bourbon		Hc
10	*Agkistrodon contortrix*	Copperhead	1				KANSAS	Woodson		
11	*Anolis carolinensis*	Green Anole				1	ARKANSAS	Polk		(
12	*Bufo americanus*	American Toad				3	ARKANSAS	Benton		
13	*Bufo americanus*	American Toad				<7	KANSAS	Bourbon		Hc
14	*Bufo americanus*	American Toad				<7	KANSAS	Leavenworth		
15	*Bufo americanus*	American Toad				7	ARKANSAS	Scott		
16	*Carphophis vermis*	Western Worm Snake				2	KANSAS	Leavenworth		
17	*Chelydra serpentina*	Snapping Turtle				1	KANSAS	Chautauqua		
18	*Chelydra serpentina*	Snapping Turtle				1	KANSAS	Linn		
19	*Chelydra serpentina*	Snappin Turtle				1	KANSAS	Woodson		
20	*Chrysemys picta*	Northern Painted Turtle				1	KANSAS	Leavenworth		
21	*Coluber constrictor*	Eastern Racer				1	KANSAS	Leavenworth		
22	*Coluber constrictor flaviventris*	Eastern Yellowbelly Racer				1	KANSAS	Chautauqua		
23	*Crotalus horridus*	Timber Rattlesnake				3	KANSAS	Chautauqua		

Herpetology Field Notebooks [Compatibility Mode] - Excel

A43 | Graptemys geographica

	Location	GeoLocation	TwnRanSe	Date	Year	PrimaryCollector	OtherCollectors
2	Barton Co., Missouri	N 37°65'41.25"; W 94°15'31.00"		April 7, 2011	2011	Greg Chapman	
3	At Mine Creek. Pleasanton, Linn Co., Kansas	N 38 10.032; W 94 39.511		March 21, 2011	2011	Kyle Moodie	
4	Hollister Wildlife Area; 0.93 mi E of Elm Creek Reservoir, Bourbon Co., Kansas	N 37°46'00.37"; W 94°49'54.79"		April 6, 2011	2011	Logan Martin	Herp Class 2011
5	Private property 0.25 mi W of Piqua, Woodson Co., Kansas	N 37°55'13.80"; W 95°32'30.30"		April 10, 2011	2011	Logan Martin	
6	Fort Leavenworth Militiray Base, Leavenworth Co., Kansas	N 39°22'05.01"; W 94°55'01.05"		April 23, 2011	2011	Logan Martin	Kyle Moodie, Dillon Knopp
7	3.5 mi SW of Jay, Oklahoma, just off East 441 Road	N 36°22'04.39"; W 94°48'44.46"		April 30, 2011	2011	Logan Martin	Herp Class 2011
8	Angelina National Forest, Boykin Springs area, Jasper Co., Texas	N 31°02'00.63"; W 94°16'50.87"		March 20, 2011	2011	Logan Martin	
9	Hollister Wildlife Area; 0.93 mi E of Elm Creek Reservoir, Bourbon Co., Kansas	N 37°46'00.37"; W 94°49'54.79"		April 6, 2011	2011	Logan Martin	Herp Class 2011
10	Private property 0.76 mi E of Toronto, Woodson Co., Kansas	N 37°47'59.19"; W 95°56'07.71"		April 9, 2011	2011	Logan Martin	
11	Cossatot River River State Park; 7.25 mi E of Vandervoort, Arkansas	N 34°22'43.55"; W 94°14'14.92"		March 19, 2011	2011	Logan Martin	
12	Ozark National Forest, Benton Co., Arkansas	N 36°07'32.08"; W 94°23'53.89"		March 18, 2011	2011	Logan Martin	
13	Hollister Wildlife Area; 0.93 mi E of Elm Creek Reservoir, Bourbon Co., Kansas	N 37°46'00.37"; W 94°49'54.79"		April 6, 2011	2011	Logan Martin	Herp Class 2011
14	Fort Leavenworth Militiray Base, Leavenworth Co., Kansas	N 39°22'05.01"; W 94°55'01.05"		April 23, 2011	2011	Logan Martin	Kyle Moodie, Dillon Knopp
15	Buck Knob Mountain; 17.80 mi NE of Mena, Scott Co., Arkansas	N 34°41'27.13"; W 93°56'33.82"		April 30, 2011	2011	Logan Martin	Herp Class 2011
16	Fort Leavenworth Militiray Base, Leavenworth Co., Kansas	N 39°22'05.01"; W 94°55'01.05"		April 23, 2011	2011	Logan Martin	Kyle Moodie, Dillon Knopp
17	Red Buffalo Ranch, Chautauqua Co., Kansas	N 37°0'9.70"; W 96°17'25.56"		April 9, 2011	2011	Greg Chapman	Rylan, Peyton, Scott Amos
18	Linn Co., Kansas	N 38 54.95; W 94 31.94		May 7, 2011	2011	Kyle Moodie	
19	Private property 0.25 mi W of Piqua, Woodson Co., Kansas	N 37°55'13.80"; W 95°32'30.30"		April 10, 2011	2011	Logan Martin	
20	Fort Leavenworth Militiray Base, Leavenworth Co., Kansas	N 39°22'05.01"; W 94°55'01.05"		April 23, 2011	2011	Logan Martin	Kyle Moodie, Dillon Knopp
21	Fort Leavenworth Militiray Base, Leavenworth Co., Kansas	N 39°22'05.01"; W 94°55'01.05"		April 23, 2011	2011	Logan Martin	Kyle Moodie, Dillon Knopp
22	Red Buffalo Ranch, Chautauqua Co., Kansas	N 37°6'25.59"; W 96°15'23.20"		April 10, 2011	2011	Greg Chapman	Rylan, Peyton, Scott Amos
23	Red Buffalo Ranch, Chautauqua Co., Kansas	N 37°8'50.26"; W 96°15'29.87"		April 9, 2011	2011	Greg Chapman	Rylan, Peyton, Scott Amos

1983 | 1987 | 1989 | 1991 | 1993 | 1995 | 1997 | 1999 | 2001 | 2007 | 2009 | 2011 | 2013 | blank

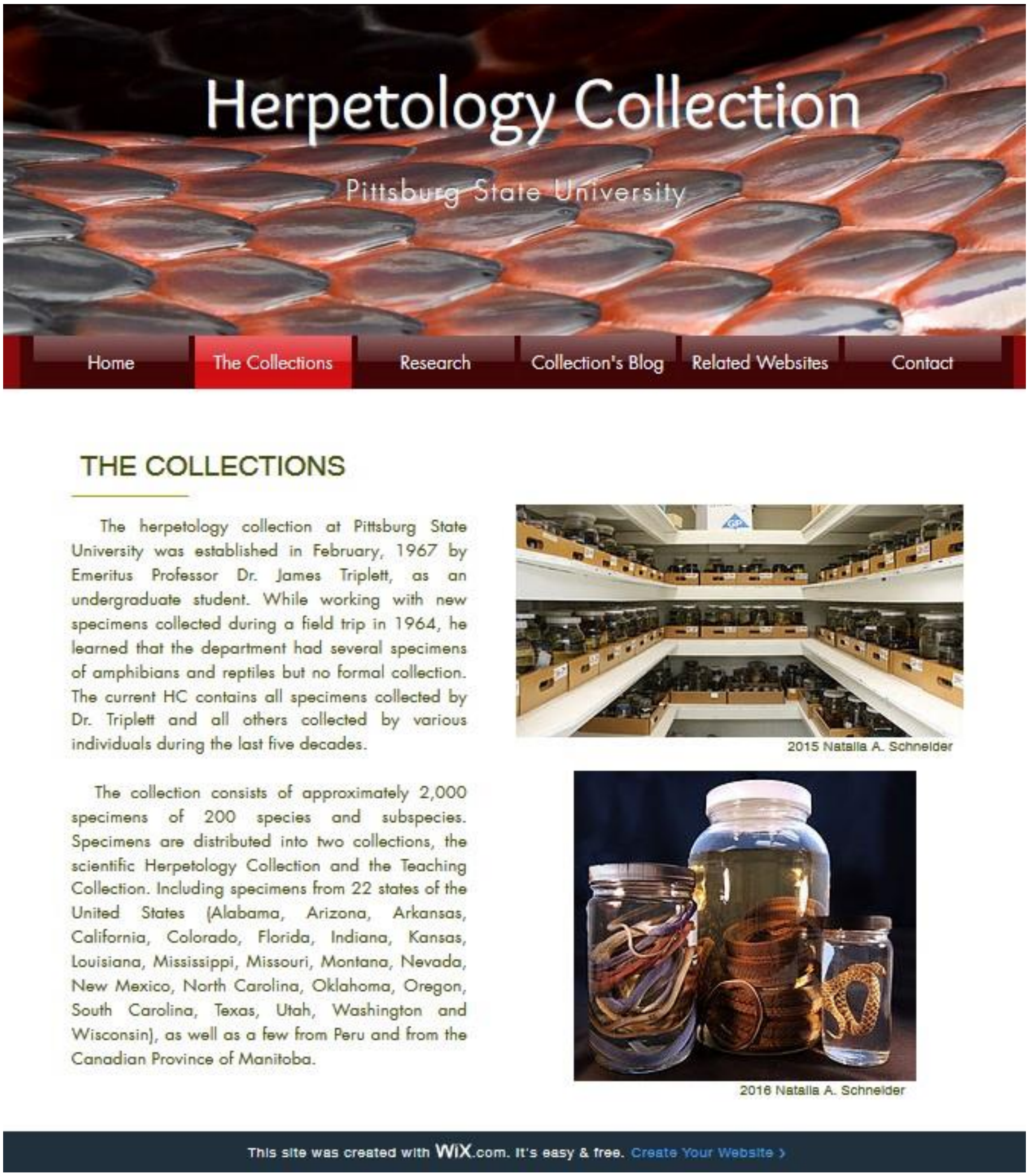
Herpetology Field Notebooks [Compatibility Mode] - Excel

U9 | Rocky ground and permanent shallow pond in wooded area

	Q	R	S	T	U	V
1	SpecimenCatalogued	TotalSpecCatalog	Jar#	nPhoto_onFiel	CollectingSiteNotes	Curator Notes
2	N/A			Y		
3	Y	2	212	N	Rocky stream bank	**Jar #212** contains one specimen under collector Kyle Moodie
4	N/A			N	Rocky ground and permanent shallow pond in wooded area	
5	Y	1	265	N	Pond surrounded by wooded area and roadside	**Jar #265** contains one specimen under collector Logan Martin
6	N/A			N	Various, ponds, streams, grasslands, wooded area, many under	
7	N/A			N	Under rocks in small stream, puddles of water	
8	N/A			Y	Stream in forested area	
9	N/A			N	Rocky ground and permanent shallow pond in wooded area	
10	N			N	Pond surrounded by wooded area and roadside	
11	N/A			Y	Rock piles and fallen trees in forested area	
12	N/A			Y	Permanent shallow pond in forested area	
13	N/A			N	Rocky ground and permanent shallow pond in wooded area	
14	N/A			N	Various, ponds, streams, grasslands, wooded area, many under	
15	N/A			N	High elevation under rocks and logs along slope	
16	N/A			N	Various, ponds, streams, grasslands, wooded area, many under	
17	N/A			N		
18	N/A			Y	Ditch near open pasture and creek	Coordinates provided by collector point to Missouri, not Linn Co., Kansas as indicated on location.
19	N/A			Y	Pond surrounded by wooded area and roadside	
20	N/A			N	Various, ponds, streams, grasslands, wooded area, many under	
21	N/A			Y	Various, ponds, streams, grasslands, wooded area, many under	
22	N/A			N		
23	N/A			Y		

1983 | 1987 | 1989 | 1991 | 1993 | 1995 | 1997 | 1999 | 2001 | 2007 | 2009 | 2011 | 2013 | blank

Appendix F. Website and blog developed for the Herpetology Collection at Pittsburg State University. A side project was carried out to create a sample website and a blog for the HC which included information about the Herpetology Collection and Teaching Collection, research done at the laboratory, basic collection data, and other relevant information. Both webpages were createad using free domains. The website can be accessed at http://herpichcollections.wixsite.com/psuherpetologycoll/



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The blog summarizes information about in the collection. It covers more detail compared to the website for certain aspects of the history of the collection and curatorial activities. The blog can be accessed at https://wetcollectionspsu.blogspot.com/

Appendix G. Specimen imaging at the Herpetology Collection. The main purpose of imaging specimens was to use for the website, blog, research presentations (e.g., posters) and to illustrate the results of the present project. Photography was done using a Canon Rebel T6 mounted on a Promaster Copy Stand (A), or within a portable studio shadow box (D). Individual specimens were photographed on the copy stand (B) and jars in the shadow box (E). Images were edited on Adobe Photoshop CC 2015 to clean the background, adjust lighting, and add scale bars (C and F).


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1 CM