ISSN 1536-7738

Oklahoma Native Plant Record

Journal of the Oklahoma Native Plant Society
Volume 15, December 2015

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P. O. Box 14274
Tulsa, Oklahoma 74159-1274
Volume 15, December 2015

ISSN 1536-7738
http://ojs.library.okstate.edu/osu/

The purpose of ONPS is to encourage the study, protection, propagation, appreciation, and use of the native plants of Oklahoma. Membership in ONPS is open to any person who supports the aims of the Society. ONPS offers individual, student, family, and life memberships.

Cover photo: *Euonymous americana*
(Hearts-a-bustin; Strawberry bush)
by Connie Arnold

https://doi.org/10.22488/okstate.17.100108

Oklahoma Native Plant Record
Volume 15

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Foreword

After 15 years, we are more than pleased with the variety of excellent articles submitted and accepted for publication in the *Oklahoma Native Plant Record*. This year, as most years, together, they meet all ONPS goals.

"*Encouraging the study of native plants.*" We never know how the record of a single study will encourage future research, but we are sure our historic article will be of special value to today's botanists and ecologists studying historic species distributions and environmental changes. In 1934, Ben Osborn may not have been aware of how valuable his list of flowering dates would be to the issue of global warming, but his article, "First Flowering Dates for Central Oklahoma" fills that role. In his preface to that article, Dr. Wayne Elisens contributes the history that puts that data into perspective. Floristic surveys, like that of Black Mesa by Amy Buthod and Bruce Hoagland from the Oklahoma Biological Survey, and descriptions like those in *Forest Structure and Fire History at Lake Arcadia* by Chad King, from the University of Central Oklahoma, make future comparative studies not only possible, but likely.

"Encouraging the protection of native plants." Kudzu (*Pueraria montana*) has long been described as an invasive species, but like many exotic species that have been introduced without thought of how they would interact with native species, it didn't start out that way. Marli Claytor and Karen Hickman from Oklahoma State University summarize the current extent of Kudzu and what might be done to protect our native species.

"Encouraging the propagation of native plants." The risks of monoculture plantings and the benefits of planting multiple species within gardens is the topic of the article by Oklahoma State University's Bonner, Rebek, Cole, Kahn, and Steets. This research is important for landscapers and gardeners because of plant species' effects on arthropod abundance, a main point of Douglas Tallamy's recent presentations at the Society's events in Tulsa and Oklahoma City. Their article provides the data and reasons to heed his advice.

"Encouraging the appreciation of native plants." For enthusiasts and plant lovers, this year we have started a new tradition, by chosing our Critic's Choice Essay from previous "Botanist's Corner" articles published in the *Gaillardia*, the Society's newsletter. This year's essay, by the late Paul Buck, about an often maligned native species, is entitled "Mistletoe, *Phoradendron serotinum*".

"Encouraging the use of native plants." In the past, we have published articles about how Native Americans used native plant species. "Antifungal Activity in Extracts of Plants from Southwestern Oklahoma against *Aspergillus flavus*" shows us how plants can be used for more current medicinal purposes. It is also a great example of research projects that can inspire students who are involved to continue in botany. This year's student research project is from Tahzeeba Frisby and her students at Cameron University in Lawton.

As you can see, articles for all interest groups of our membership (gardeners, academic faculty, landscapers, and enthusiasts) are represented. It is the wide variety of authors who contribute to our journal that helps us bring those many interests together in ways that best promote our goals. Why not consider submitting your manuscript next year? Remember that our editorial board includes a manuscript editor, Dr. Mark Fishbein, who can find help for first time and citizen-scientist authors. Tell us about your ideas and submit your articles early, so we can see that your work gets the most helpful reviews and comments.

Don't forget that The *Oklahoma Native Plant Record* is a professionally reviewed publication, listed globally in the "Directory of Open Access Journals", and our abstracts are indexed in the "Centre for Agricultural Bioscience International", which is based in the U.K.

Sheila Strawn, Managing Editor

PREFACE TO FIRST FLOWERING DATES FOR CENTRAL OKLAHOMA

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Global climate change is predicted to have deleterious effects on human health and welfare including frequency of extreme weather events, sea level rise and coastal flooding, decreased agricultural productivity, fluctuating biotic interactions and range shifts, and altered seasonality and phenology (IPCC 2014). Phenology, the study of cyclic and seasonal natural phenomena such as flowering and animal migrations, is especially important as an indicator of changing climates and ecosystem changes (e.g., Diez et al. 2012). For plants, tracking of first- or peak-flowering events has been a common approach to investigate species' responses to climatic factors. Individuals, organizations, and botanical gardens have recorded flowering times for a wide range of species over many years (Tooke & Battey 2010). Currently, academic as well as citizen scientists are actively engaged in gathering plant phenological data. Schools, online communities, and native plant societies are often involved in phenological tracking activities (e.g., Haggerty and Mazer 2008) by partnering with agencies such as the USA National Phenology Network (www.usanpn.org).

Below is a privately printed but unpublished report of first flowering dates for a variety of species in central Oklahoma from 1927–1929 and 1933. Much of the baseline data was gathered by Lois Gould in 1927–1929 (Gould 1928, 1929a, 1929b) in central Oklahoma as part of a comparative study among 14 midwestern colleges and universities. In 1933, Ben Osborn added observations from Norman and Oklahoma City and organized the compiled data chronologically to provide a 3-year record of flowering phenology by earliest and average flowering date, species name, common name, location, year of observation, and observer (Gould or Osborn). Mr. Osborn typed this report and deposited a copy in the library of the Robert Bebb Herbarium of the University of Oklahoma as "Separate No. 27" where it has remained until this printing. The present report has not been published formally to the best of our knowledge. We hope this summary of first flowering in central Oklahoma in the early twentieth century will assist present-day investigations of the biological effects of climate change by providing a valuable plant phenological benchmark.

Lois H. Gould, daughter of Dr. Charles Gould who was the former director of the Oklahoma Geological Survey, received a B.A. in Botany from the University of Oklahoma in 1930. Her passions included art, plants, and birds. Ms. Gould married the Canadian entomologist Dr. Ralph D. Bird, who taught at the University of Oklahoma from 1929 to 1933. Mrs. (Gould) Bird moved to Canada when her husband accepted a position at a Canadian Federal Entomology Laboratory (Anonymous 1972).

https://doi.org/10.22488/okstate.17.100110

Ben O. Osborn received the first Bachelor's degree in Agricultural Journalism from Oklahoma A&M College (Oklahoma State University) in 1931. He began his career as a copy editor with the Oklahoma Livestock News and was a news and radio script writer for the Oklahoma Agricultural Extension Service. He then embarked on a 36-year career with the USDA as a soil conservationist, information specialist, speechwriter, and editor for the Journal of Soil and Water Conservation (Anonymous 1999). With co-author Elizabeth Barkley, Mr. Osborn published a list of the vascular plants of Pottawatomie County, Oklahoma (Barkley and Osborn 1933).

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FIRST FLOWERING DATES FOR CENTRAL OKLAHOMA

Unpublished Report
Bebb Herbarium
University of Oklahoma
May 1, 1934

Ben Osborn

Keywords: blooming, climate change, historical, phenology

INTRODUCTION

The following list of native and cultivated plants is arranged according to the first recorded date of blooming for each species as observed in Oklahoma County or any of the counties contiguous to it. Most of the records are from Norman in Cleveland County, with a few from Oklahoma City in Oklahoma County and Shawnee in Pottawatomie County.

Where more than one has been recorded, the average is given as the arithmetic average of the dates.

Dates of observations of plants which had attained full bloom, where known, have been eliminated, except where they were the earliest recorded dates for the species. Such dates have been starred (*) in the list.

A plant is considered in bloom when as many as one flower is open and having either stamens shedding pollen or the stigma ready to receive pollen, as indicated by pollen grains adhering to it or the obviously mature condition of the stigma. Exceptionally early bloomings within the shelter of buildings or other barriers are included, but those resulting from artificial heat have been eliminated.

https://doi.org/10.22488/okstate.17.100111

FIRST FLOWERING DATES

Editor's Note: Where nomenclature has been updated using ITIS–Integrated Taxonomic Information Service (http://www.itis.gov), the original name is in brackets [ ]. There are no voucher specimens for this work; therefore, species identifications are provisional. The dates of observations of plants which have attained full bloom are marked with a single asterisk (*). Observations added by Fred Barkley on 6 June 1934, are marked with a double asterisk (**).

JANUARY

1 January
Stellaria media. Average 30 January. Earliest, Oklahoma City, 1933, Osborn.**

7 January
Common dandelion, *Taraxacum officinale*. Average 25 January. Earliest, Norman, 1929, Gould.

19 January
Shepherd's purse, *Capsella bursa-pastoris*. Average 7 February. Earliest, Norman, 1927, Gould.

21 January
Chinese elm, *Ulmus pumila*. Earliest, Oklahoma City, 1933, Osborn.

23 January
American elm, *Ulmus americana*. Average 10 February. Earliest, Oklahoma City, 1933, Osborn.**

28 January
Silver maple, *Acer saccharinum*. Average 8 February. Earliest, Oklahoma City, 1934, Osborn.

30 January
Arbor vitae, *Thuja occidentalis*. Oklahoma City, 1933, Osborn.

FEBRUARY

1 February
Common violet, *Viola sororia* var. *sororia* [=*Viola papilionacea*]. Oklahoma City, 1933, Osborn.

3 February
Spring beauty, *Claytonia virginica*. Average 18 February. Earliest, Norman, 1927, Gould.

5 February
Bush honeysuckle, *Lonicera fragrantissima*. Average 6 February. Earliest, Norman, 1927, Gould.

6 February
Wild pansy, *Viola bicolor* [=*Viola rafinesquii*]. Average 24 February. Earliest, Norman, 1928, Gould.
Virginia rock-cress, *Planodes virginica* [=*Arabis virginica*]. Average 26 February. Earliest, Norman, 1928, Gould.

7 February
Jasminum nudiflorum [=*Jasminum nudum*]. Norman, 1927, Gould.

8 February
Least bluet, *Houstonia pusilla* [=*Houstonia minima*]. Average 27 February. Earliest, Norman, 1927, Gould.

14 February
Goldenbells, *Forsythia suspensa*. Norman, 1927, Gould.

24 February
Carolina whitlow-grass, *Draba reptans* [=*Draba caroliniana*]. Average 2 March. Earliest, Norman, 1927, Gould.
Midland dogtooth-violet, *Erythronium mesochoreum*. Average 6 March. Earliest, Norman, 1927, Gould.
Anemone caroliniana. Average 14 March. Earliest, Norman, 1927, Gould.

25 February
Shortpod whitlow-grass, *Draba brachycarpa*. Average 7 March. Earliest, Norman, 1927, Gould.

28 February
Red cedar, *Juniperus virginiana*. Average 6 March. Earliest, Norman, 1927, Gould.
Thunberg spirea, *Spiraea thunbergii*. Average 7 March. Earliest, Norman, 1927, Gould.

MARCH

3 March
Hoary tansy-mustard, *Descurainia incana* [=*Sisymbrium canescens*]. Average 29 March. Earliest, Norman, 1928, Gould.

4 March
Slippery elm, *Ulmus rubra* [=*Ulmus fulva*]. Average 8 March. Earliest, Norman, 1927, Gould.**

5 March
Chaenomeles japonica. Average 14 March. Earliest, Norman, 1928, Gould.

7 March
Peach, *Prunus persica*. Average 16 March. Earliest, Norman, 1927, Gould.
Androsace occidentalis. Average 12 March. Earliest, Norman, 1927, Gould.

10 March
Pear, *Pyrus communis*. Norman, 1927, Gould.
Shortstalk chickweed, *Cerastium brachypodum*. Average 10 March. Earliest, Norman, 1928, Gould.
Common henbit, *Lamium amplexicaule*. (Open flowers). Average 12 March. Earliest, Norman, 1928, Gould.
Plantainleaf cat's-foot, *Antennaria plantaginifolia*. Average 24 March. Earliest, Norman, 1927, Gould.
Chickasaw plum, *Prunus angustifolia*. Average 17 March. Earliest, Norman, 1927, Gould.**

11 March
Yellow false-garlic, *Nothoscordum bivalve*. Average 19 March. Earliest, Oklahoma City, 1924, Osborn.
Spreading pearlwort, *Sagina decumbens*. Average 26 March. Earliest, Norman, 1928, Gould.

12 March
Box-elder, *Acer negundo*. Average 19 March. Earliest, Norman, 1928, Gould.

15 March
Prunus triloba. Norman, 1927, Gould.
Apricot, *Prunus armeniaca*. Norman, 1927, Gould.
Yellow woodsorrel, *Oxalis stricta*. Average 31 March. Earliest, Norman, 1927, Gould.

16 March
Missouri buffalo currant, *Ribes aureum*. Average 19 March. Earliest, Norman, 1927, Gould.

18 March
Redbud, *Cercis canadensis*. Average 24 March. Earliest, Norman, 1927, Gould.
Tulip, *Tulipa* sp. Oklahoma City, 1934, Osborn.

19 March
Low plum, *Prunus gracilis*. Average 23 March. Earliest, Norman, 1928, Gould.**
Carrotleaf parsley, *Lomatium foeniculaceum* ssp. *daucifolium* [=*Lomatium daucifolium*]. Average 24 March. Earliest, Norman, 1928, Gould.
Missouri violet, *Viola sororia* var. *missouriensis* [=*Viola missouriensis*]. Average 24 March. Earliest, Norman, 1928, Gould. (Reported as *V. papilionacea.*)

20 March
Berberis aquifolium [=*Mahonia aquifolium*]. Norman, 1927, Gould.

22 March
Cottonwood, *Populus deltoides*. Average 26 March. Earliest, Norman, 1928, Gould.

24 March
Lilac, *Syringa x persica* [=*Syringa persica*]. Norman, 1927, Gould.
Common pansy, *Viola tricolor*. Norman, 1927, Gould.
American plum, *Prunus americana*. Norman, 1929, Gould.
Ground-plum, *Astragalus crassicarpus* var. *crassicarpus* [=*Astragalus caryocarpus*]. Average 28 March. Earliest, Norman, 1929, Gould.

25 March
Narrowleaf puccoon, *Lithospermum incisum* [=*Lithospermum angustifolium*]. Average 27 March. Earliest, Norman, 1928, Gould.

27 March
Lonicera tatarica. Norman, 1927, Gould.
Lonicera morrowii. Norman, 1927, Gould.

28 March
Ash, *Fraxinus* sp. Oklahoma City, 1933, Osburn.

29 March
Eastern hackberry, *Celtis occidentalis*. Norman, 1928, Gould.
Maclura pomifera. Norman, 1928, Gould.
Krigia caespitosa [=*Scrinia oppositifolia*]. Average 9 April. Earliest, Norman, 1929, Gould.
Purple poppy-mallow, *Callirhoe involucrata*. Average 13 April. Earliest, Norman, 1928, Gould.

30 March
White mulberry, *Morus alba*. Norman, 1928, Gould.
Purslane speedwell, *Veronica peregrina*. Average 2 April. Earliest, Norman, 1928, Gould.
Androstephium coeruleum. Average 5 April. Earliest, Norman, 1928, Gould.
Red mulberry, *Morus rubra*. Average 9 April. Earliest, Norman, 1928, Gould.

31 March
Chaenomeles speciosa [=*Chaenomeles lagenaria*]. Norman, 1927, Gould.
Prairie false-dandelion, *Nothocalais cuspidata* [=*Agoseris cuspidata*]. Average 2 April. Earliest, Norman, 1927, Gould.

APRIL

1 April
Apple, *Malus pumila* [=*Pyrus malus*]. Norman, 1927, Gould.
Prunus cerasus. Norman, 1927, Gould.
Scarlet strawberry, *Fragaria virginiana*. Norman, 1927, Gould.
Thunberg barberry, *Berberis thunbergii*. Norman, 1927, Gould.
Common lilac, *Syringa vulgaris*. Average 2 April. Earliest, Norman, 1927, Gould.
Stout blue-eyed grass, *Sisyrinchium angustifolium* [=*Sisyrinchium gramineum*]. Average 3 April. Earliest, Norman, 1928, Gould.
Tall dock, *Rumex altissimus*. Average 13 April. Earliest, Oklahoma City, 1933, Osborn.

2 April
Caragana arborescens. Norman, 1927, Gould.
Colutea arborescens. Norman, 1927, Gould.
White ash, *Fraximus americana*. Average 6 April. Earliest, Norman, 1927, Gould.
Philadelphia fleabane, *Erigeron philadelphicus*. Average 3 April. Earliest, Norman, 1928, Gould.
Blackjack oak, *Quercus marilandica*. Average 4 April. Earliest, Norman, 1928, Gould.
Prairie ragwort, *Packera plattensis* [=*Senecio plattensis*]. Average 8 April. Earliest, Norman, 1928, Gould.
Violet wood-sorrel, *Oxalis violacea*. Average 9 April. Earliest, Oklahoma City, 1933, Osborn.
Cutleaf evening-primrose, *Oenothera laciniata*. Average 9 April. Earliest, Norman, 1929, Gould.
Roundleaf moneyflower, *Mimulus glabratus* var. *jamesii*. Average 21 April. Earliest, Norman, 1928, Gould.

3 April
Comptonia peregrina [=*Myrica asplenifolia*]. Norman, 1927, Gould.

4 April
Ovalleaf bladderpod, *Physaria ovalifolia* ssp. *ovalifolia* [=*Lesquerella ovalifolia*]. Norman, 1929, Gould.
Spurge, *Euphorbia* sp. Oklahoma City, 1933*, Osborn.

5 April
Ellisia nyctelea. Norman, 1927, Gould.
Smooth yellow violet, *Viola pubescens* var. *scabriuscula [=Viola scabriuscula*]. Norman, 1927, Gould.
Virginia pepper-grass, *Lepidium virginicum*. Norman, 1929, Gould.
Poncirus trifoliata. Norman, 1927, Gould.
Juglans cinera. Norman, 1927, Gould.
Celtis laevigata [=*Celtis mississippiensis*]. Norman, 1927, Gould.
Mousetail, *Myosurus minimus*. Average 8 April. Earliest, Norman, 1929, Gould.
Dewberry, *Rubus flagellaris* [=*Rubus villosus*]. Average 14 April. Earliest, Norman, 1927, Gould.
Spiny sow-thistle, *Sonchus asper*. Average 15 April. Earliest, Norman, 1928, Gould.

6 April
Sycamore, *Platanus occidentalis*. Norman, 1929, Gould.

7 April
Broussonetia papyrifera. Norman, 1929, Gould.
Slender bladderpod, *Physaria gracilis* [=*Lesquerella gracilis*]. Norman, 1929, Gould.

8 April
Plantago elongata. Norman, 1927, Gould.
Wild columbine, *Aquilegia canadensis*. Norman, 1927, Gould.
Spiraea vanhoutei. Norman, 1927, Gould.
Tamarisk, *Tamarix gallica*. Average 9 April. Earliest, Norman, 1927, Gould.
Large wild-onion, *Allium canadense* var. *mobilense* [=*Allium mutabile*]. Average 12 April. Earliest, Norman, 1927, Gould.
Blue toadflax, *Nuttallanthus canadensis* [=*Linaria canadensis*]. Average 16 April. Earliest, Norman, 1927, Gould.
Wooly yarrow, *Achillea millefolium* [=*Achillea lanulosa*]. Average 23 April. Earliest, Norman, 1927, Gould.
Meadow garlic, *Allium canadense*. Average 23 April. Earliest, Oklahoma City, 1933, Osborn.

9 April
Corn speedwell, *Veronica arvensis*. Norman, 1929, Gould.
Lonicera flava. Norman, 1927, Gould.
Black willow, *Salix nigra*. Average 12 April. Earliest, Oklahoma City, 1933, Osborn.
Bracted false-indigo, *Baptisia bracteata*. Average 14 April. Earliest, Oklahoma City, 1933, Osborn.
Western plantain, *Plantago virginica*. Average 14 April. Earliest, Norman, 1927, Gould.

10 April
Black-haw, *Viburnum plicatum* [=*Viburnum tomentosum*]. Norman, 1927, Gould.
Western crab-apple, *Malus ioensis* [=*Pyrus ioensis*]. Norman, 1927, Gould.
Aquilegia coerulea. Norman, 1927, Gould.

Spreading chervil, *Chaerophyllum procumbens*. Average 13 April. Earliest, Oklahoma City, 1933, Osborn.
Goose-grass, *Galium aparine*. Average 14 April. Earliest, Oklahoma City, 1933, Osborn.

11 April
Tropaeolum majus. Norman, 1927, Gould.
Robinia hispida. Norman, 1927, Gould.
Salix alba [=*Salix vitellina*]. Norman, 1927, Gould.
Western tansy-mustard, *Descurainia incisa* ssp. *incisa* [=*Sisymbrium incisum*]. Norman, 1928, Gould.
Vaillant's goose-grass, *Galium aparine* [=*Galium aparine baillantii*]. Average 12 April. Earliest, Norman, 1928, Gould.
Tetraneuris linearifolia [=*Actinea linearifolia*]. Average 14 April. Earliest, Norman, 1928, Gould.

12 April
Viburnum opulus [=*Viburnum opulus sterile*]. Norman, 1927, Gould.
Black medick, *Medicago lupulina*. Average 6 May. Earliest, Norman, 1927, Gould.

13 April
Fragrant sumac, *Rhus aromatica* [=*Rhus canadensis*]. Norman, 1927, Gould.
Kerria japonica. Norman, 1927, Gould.
Frost grape, *Vitis vulpina* [=*Vitis cordifolia*]. Average 10 April. Norman, 1928, Gould.

14 April
Alyssum alyssoides. Norman, 1927, Gould.
Crataegus calpodendron [=*Crataegus globosa*]. Norman, 1927, Gould.
Black-haw, *Virburnum prunifolium*. Norman, 1927, Gould.
Leafystem false-dandelion, *Pyrrhopappus carolinianus*. Norman, 1927, Gould.
Corydalis aurea ssp. *occidentalis*. Average 19 April. Earliest, Norman, 1927, Gould.
Black locust, *Robinia pseudoacacia*. Average 20 April. Earliest, Norman, 1927, Gould.
Beaked corn-salad, *Valerianella radiata*. Average 22 April. Earliest, Norman, 1927, Gould.
Pecan, *Carya illinoiensis*. Average 27 April. Earliest, Norman, 1927, Gould.

15 April
Calycanthus floridus. Norman, 1927, Gould.
Pepper-grass, *Lepidium apetalum*. Average 15 April. Earliest, Norman, 1929, Gould, and Oklahoma City, 1933, Osborn.
Silverleaf nightshade, *Solanum elaeagnifolium*. Average 29 April. Earliest, Oklahoma City, 1933, Osborn.

16 April
Western crab apple, *Pyrus ioensis*. Norman, 1927, Gould.**
Cryptanthe fendleri. Average 20 April. Earliest, Norman, 1927, Gould.
Post oak, *Quercus stellata*. Average 17 April. Earliest, Norman, 1927, Gould.
Western daisy, *Astranthium integrifolium* ssp. *integrifolium* [=*Bellis integrifolia*]. Average 20 April. Earliest, Norman, 1927, Gould.
Red oak, *Quercus rubra* [=*Quercus borealis*]. Norman, 1929, Gould.

17 April
Rhodotypos scandens [=*Rhodotypos kerrioides*]. Norman, 1927, Gould.
Yellow oak, *Quercus muhlenbergii*. Norman, 1929, Gould.
Cotoneaster gaballei. Norman, 1927, Gould.
Cotoneaster horizonatlis. Norman, 1927, Gould.
Southern black-haw, *Viburnum rufidulum*. Average 19 April. Earliest, Norman, 1929, Gould.
Burr oak, *Quercus macrocarpa*. Average 23 April. Earliest, Norman, 1929, Gould.

18 April
Nandina domestica. Norman, 1927, Gould.
Aronia melanocarpa. Norman, 1927, Gould.
Carolina cranebill, *Geranium carolinianum*. Average 27 April. Earliest, Norman, 1928, Gould.

19 April
Phacelia hirsuta. Norman, 1927, Gould.
Eleagnus angustifolia. Norman, 1927, Gould.
Deutzia scabra. Norman, 1927, Gould.
Stemless loco-weed, *Oxytropis lambertii*. Average 24 April. Earliest, Norman, 1927, Gould.**
Small skullcap, *Scutellaria parvula*. Average 25 April. Earliest, Norman, 1927, Gould.

20 April
Rosa rubiqinosa [=*Rosa eglanteria*]. Norman, 1927, Gould.

21 April
Hoary puccoon, *Lithospermum canescens*. Norman, 1929, Gould.
Light poppy-mallow, *Callirhoe alcaeoides*. Oklahoma City, 1934, Osborn.

22 April
White clover, *Trifolium repens*. Oklahoma City, 1934, Osborn.
Nuttall onion, *Allium drummondii* [=*Allium nuttallii*]. Norman, 1928, Gould.
True water-cress, *Nasturtium officinale* [=*Radicula nasturium-aquaticum*]. Norman, 1929, Gould.
Western spiderwort, *Tradescantia occidentalis*. Average 24 April. Earliest, Norman, 1929, Gould.
Poison oak, *Toxicodendron pubescens* [=*Rhus toxicodendron*]. Average 29 April. Earliest, Norman, 1929, Gould.
Horsenettle, *Solanum carolinense*. Average 3 May. Earliest, 1929, Gould.

23 April
Smallflower verbena, *Glandularia bipinnatifida* var. *bipinnatifida* [=*Verbena bipinnatifida*]. Norman, 1938, Gould.
Rock sandwort, *Minuartia tenella* [=*Arenaria stricta*]. Norman, 1928, Gould.
Petioled wild four-o-clock, *Mirabilis nyctaginea* [=*Oxybaphus nyctagineus*]. Average 1 May. Earliest, Norman, 1929, Gould.

24 April
Alfalfa, *Medicago sativa*. Average 1 May. Earliest, Norman, 1929, Gould.
Curly dock, *Rumex crispus*. Average 3 May. Earliest, Norman, 1929, Gould.

25 April
Red-haw, *Crataegus* sp. Average 25 April. Norman, 1928–29, Gould.
Rough false-dandelion, *Pyrrhopappus grandiflorus* [=*Pyrrhopappus scaposus*]. Average 26 April. Earliest, Norman, 1929, Gould.
Blue false-indigo, *Baptisia australis*. Average 27 April. Earliest, Norman, 1929, Gould.
Chaetopappa asteroides. Average 29 April. Earliest, Norman, 1928, Gould.
Black walnut, *Juglans nigra*. Average 30 April. Earliest, Norman, 1928, Gould.
Wild parsnip, *Pastinaca nativa*. Average 1 May. Earliest, Norman, 1928, Gould.
Hairy puccoon, *Lithospermum caroliniense* [=*Lithospermum gmelinī*]. Average 3 May. Earliest, Norman, 1928, Gould.

26 April
Vetch, *Vicia* sp. (cultivated). Oklahoma City, 1934, Osborn.
Basket oak, *Quercus michauxii*. Norman, 1928, Gould.

27 April
Common greenbriar, *Smilax rotundifolia*. Norman, 1929, Gould.
Virginia willow, *Itea virginica*. Norman, 1929, Gould.
Yellow sweet-clover, *Melilotus officinalis*. Average 4 May. Earliest, Oklahoma City, 1934*, Osborn.

28 April
Spreading fleabane, *Erigeron divergens*. Average 3 May. Earliest, Norman, 1927, Gould.
Serrateleaf evening-primrose, *Oenothera serrulata*. Average 1 May. Earliest, Norman, 1927, Gould.

29 April
Skunk bush, *Rhus aromatica* [=*Rhus canadensis trilobatus*]. Norman, 1928, Gould.
Purple milkweed, *Asclepias purpurascens*. Norman, 1928, Gould.
Bluntleaf milkweed, *Asclepias amplexicaulis*. Average 5 May. Earliest, Norman, 1929, Gould.
Wavyleaf gaura, *Oenothera sinuosa* [=*Gaura sinuata*]. Average 2 May. Earliest, Norman, 1929, Gould.**

30 April
Reflexed spiderwort, *Tradescantia ohiensis* [=*Tradescantia reflexa*]. Norman, 1927, Gould.
Mexican sandbur, *Tribulus terrestris*. Average 30 April. Earliest, Norman, 1928, Gould.
Cutleaf bayless gaillardia, *Gaillardia suavis*. Average 5 May. Earliest, Oklahoma City, 1933, Osborn.
Oblongleaf milkweed, *Asclepias viridis* [=*Asclepiodora viridis*]. Average 6 May. Earliest, Oklahoma City, 1933, Osborn.

MAY

1 May
Vetch, *Vicia tetrasperma*. Norman, 1928, Gould.
Small Venus'-lookingglass, *Triodanis perfoliata* ssp. *biflora* [=*Specularia biflora*]. Norman, 1928, Gould.
Prairie larkspur, *Delphinium carolinianum* [=*Delphinium penardi*]. Norman, 1928, Gould.
Sand grape, *Vitis rupestris*. Norman, 1928, Gould.
American mistletoe, *Phoradendron serotinum* ssp. *serotinum* [=*Phoradendron flavescens*]. Norman, 1928, Gould.

2 May
Sleepy catchfly, *Silene antirrhina*. Average 6 May. Earliest, Norman, 1929, Gould.
Venus'-lookingglass, *Specularia perfoliata*. Average 12 May. Earliest, Norman, 1928, Gould.

3 May
Salsify, *Tragopogon porrifolius*. Norman, 1928, Gould.
River locust, *Amorpha fruticosa*. Average 5 May. Earliest, Norman, 1927, Gould.
Scarlet gaura, *Oenothera suffrutescens* [=*Gaura coccinea*]. Norman, 1927, Gould.
Downy phlox, *Phlox pilosa*. Average 4 May. Norman, 1929, Gould.
Calystegia sepium ssp. *angulata* [=*Convolvulus repens*]. Average 6 May. Earliest, Norman, 1929, Gould.
Smooth soapweed, *Yucca glauca*. Average 7 May. Earliest, Norman, 1929, Gould.
Large beardtongue, *Penstemon cobaea*. Average 7 May. Earliest, Norman, 1927, Gould.
Slender beardtongue, *Penstemon gracilis*. Average 2 May. Earliest, Norman, 1929, Gould.
Missouri evening-primrose, *Oenothera macrocarpa* ssp. *macrocarpa* [=*Oenothera missouriensis*]. Average 8 May. Earliest, Norman, 1929, Gould.
Whorled tickseed, *Coreopsis verticillata*. Average 8 May. Earliest, Norman, 1927, Gould.
Spermolepis echinata. Average 12 May. Earliest, Norman, 1927, Gould.
Spurge nettle, *Cnidoscolus urens* var. *stimulosus* [=*Jatropha stimulosa*]. Average 12 May. Earliest, Norman, 1929, Gould.
Hymenopappus scabiosaeus [=*Hymenopappus carolinensis*]. Average 13 May. Earliest, Norman, 1929, Gould.

4 May
Hairy bedstraw, *Galium pilosum*. Norman, 1929, Gould.
Delphinium carolinianum [=*Delphinium virescens*]. Norman, 1929, Gould.
Psoralidium tenuiflorum [=*Psoralea tenuiflora*]. Norman, 1929, Gould.

5 May
Galium tricornutum [=*Galium tricorne*]. Norman, 1928, Gould.
Spinyleaf catbriar, *Smilax bona-nox*. Average 9 May. Earliest, Norman, 1927, Gould.
Showy evening-primrose, *Oenothera speciosa*. Average 6 May. Earliest, Norman, 1927, Gould.
Largeflower flax, *Linum rigidum*. Average 7 May. Earliest, Norman, 1929, Gould.
Sweet-scented grape, *Vitus vulpina*. Average 8 May. Earliest, Norman, 1927, Gould.
Sensitive-briar, *Mimosa microphylla* [=*Schrankia uncinata*]. Average 11 May. Earliest, Norman, 1927, Gould.

6 May
Longstalk green-briar, *Smilax pseudo-china*. Norman, 1928, Gould.
Pediomelum digitatum [=*Psoralea digitata*]. Norman, 1928, Gould.
Kentucky coffee-tree, *Gymnocladus dioica*. Norman, 1928, Gould.

7 May
Polygala senega. Average 15 May. Earliest, Norman, 1927, Gould.
Virginia ground-cherry, *Physalis virginiana*. Norman, 1927, Gould.
Dwarf morning-glory, *Evolvulus nuttallianus* [=*Evolvulus argenteus*]. Average 10 May. Earliest, Norman, 1927, Gould.

8 May
Bracted plantain, *Plantago aristata*. Norman, 1928, Gould.
Honey locust, *Gledtisia triacanthos*. Norman, 1928, Gould.
Rib-grass, *Plantago lanceolata*. Norman, 1928, Gould.

9 May
Western catalpa, *Catalpa speciosa*. Norman, 1929, Gould.
Catalpa, *Catalpa bignonioides* [=*Catalpa catalpa*]. Norman, 1927, Gould.
American vetch, *Vicia americana*. Norman, 1927, Gould.**
American elder, *Sambucus nigra* ssp. *canadensis* [=*Sambucus canadensis*]. Average 13 May. Earliest, Norman, 1929, Gould.
White sweet-clover, *Melilotus albus*. Average 14 May. Earliest, Norman, 1927, Gould.
Roughleaf dogwood, *Cornus asperifolia*. Average 16 May. Earliest, Norman, 1927, Gould.

12 May
Potentilla norvegica [=*Potentilla monspeliensis*]. Norman, 1927, Gould.
Purple lemon-mint, *Monarda citriodora* [=*Monarda dispersa*]. Average 23 May. Earliest, Norman, 1927, Gould.

13 May
Queen's delight, *Stillingia sylvatica*. Norman, 1928, Gould.
Rabbit tobacco, *Diaperia prolifera* [=*Evax prolifera*]. Norman, 1928, Gould.
Canada moonseed, *Menispermum candense*. Norman, 1928, Gould.
Bristly greenbriar, *Smilax tamnoides* [=*Smilax hispida*]. Norman, 1928, Gould.

14 May
Low ground-cherry, *Physalis pumila*. Norman, 1928, Gould.
Low hairy ground-cherry, *Physalis pubescens*. Norman, 1928, Gould.
Largeflower tickseed, *Coreopsis grandiflora*. Oklahoma City, 1933*, Osborn.
Pediomelum cuspidatum [=*Psoralea cuspidata*]. Average 17 May. Earliest, Norman, 1927, Gould.
Bank-bur, *Krameria lanceolata*. Average 21 May. Earliest, Norman, 1927, Gould.

15 May
Dogbane, *Apocynum cannabinum*. Norman, 1929, Gould.
Showy gaillardia, *Gaillardia puchella*. Norman, 1928, Gould.
Green dragon, *Arisaema dracontium*. Average 17 May. Earliest, Norman, 1929, Gould.

16 May
Dwarf verbena, *Glandularia pumila* [=*Verbena pumila*]. Norman, 1928, Gould.

17 May
Ground ivy, *Nepeta cataria*. Norman, 1928, Gould.
Purple cone-flower, *Echinacea purpurea* [=*Brauneria purpurea*]. Average 21 May. Earliest, Norman, 1928, Gould.
Field bindweed, *Convolvulus arvensis*. Norman, 1928, Gould.

19 May
Decumbent milkweed, *Asclepias asperula* [=*Asclepiodora decumbens*]. Norman, 1928, Gould.
Tumble mustard, *Sisymbrium altissimum*. Norman, 1929, Gould.
Engelmannia peristenia [=*Engelmannia pinnatifida*]. Norman, 1929, Gould.
Persimmon, *Disopyros virginiana*. Norman, 1929, Gould.

20 May
Phlox maculata. Norman, 1927, Gould.
Leafy white prickly-poppy, *Argemone polyanthemos* [=*Argemone intermedia*]. Average 22 May. Earliest, Norman, 1927, Gould.

21 May
Smooth Solomon's seal, *Polygonatum biflorum* [=*Polygonatum commutatum*]. Norman, 1928, Gould.
Bluntleaf spurge, *Euphorbia spathulata* [=*Euphorbia obtusata*]. Norman, 1928, Gould.
Low dwarf mallow, *Malva neglecta* [=*Malva rotundifolia*]. Norman, 1928.
Climbing bittersweet, *Celastrus scandens*. Norman, 1928, Gould.
Prairie sunflower, *Helianthus petiolaris*. Average 24 May. Earliest, Norman, 1928.

22 May
Spermolepis inermis [=*Spermolepis patens*]. Norman, 1928, Gould.

23 May
Longhead coneflower, *Ratibida columnifera* [=*Lepachys columnaris*]. Average 29 May. Earliest, Norman, 1927, Gould.

24 May
Wild sweet-pea, *Tephrosia virginiana*. Average 25 May. Earliest, Norman, 1928, Gould.

25 May
Ruellia strepens. Average 26 May. Earliest, Norman, 1929, Gould.

26 May
Intermediate bush-clover, *Lespedeza simulata*. Norman, 1927, Gould.
Snow-on-the-mountain, *Euphorbia marginata*. Norman, 1928, Gould.
Lead plant, *Amorpha canescens*. Norman, 1927, Gould.
Claspingleaf coneflower, *Rudbeckia amplexicaulis*. Norman, 1927, Gould.
Smooth sumac, *Rhus glabra*. Average 28 May. Earliest, Norman, 1927, Gould.
Black-eyed Susan, *Rudbeckia hirta*. Earliest, Norman, 1927, Gould.

27 May
Denseflower water-willow, *Justicia americana* [=*Dianthera americana*]. Norman, 1929, Gould.**

28 May
Lateflower talinum, *Phemeranthus calycinus* [=*Talinum calycinum*]. Norman, 1928, Gould.
Smallflower talinum, *Phemeranthus parviforus* [=*Talinum parviflora*]. Norman, 1928, Gould
Buffalo burr, *Solanum rostratum*. Norman, 1928, Gould.

30 May
Butterfly weed, *Asclepias tuberosa*. Norman, 1929, Gould.

31 May
Sabatia campestris. Norman, 1927, Gould.
Yellow gaillardia, *Gaillardia pinnatifida*. Norman, 1928, Gould.

JUNE

1 June
Gold tickseed, *Coreopsis tinctoria*. Norman, 1927, Gould.

FOREST STRUCTURE AND FIRE HISTORY AT LAKE ARCADIA, OKLAHOMA COUNTY, OKLAHOMA (1820–2014)

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Keywords: fire history, Cross Timbers, dendroecology

ABSTRACT

Evidence indicates that the structure of Oklahoma Cross Timbers forests are in transition due to changing climate, land-use patterns, and fire suppression efforts. However, only a handful of studies have addressed the history of fire across the Oklahoma Cross Timbers landscape. This research adds to the body of literature by studying the contemporary forest structure and fire history at Lake Arcadia in Oklahoma County, Oklahoma. Results demonstrate that post oak (*Quercus stellata* Wangenh.) and blackjack oak (*Q. marilandica* Münchh.), two common species in Oklahoma Cross Timbers, dominate the forest. However, several mesophytic tree species are found in the overstory as well as the sapling layer of the forest. A total of 25 fire events (mean fire interval = 4.14 years) were documented during the 20th century using fire-scar analysis of *Q. stellata* trees and remnant wood (stumps, snags, recently dead trees). High fire frequencies in the early to mid-20th century corresponded to the recruitment of *Q. stellata* and *Q. marilandica*. Wet conditions (PDSI > 0) during the late 20th century and no fires after 1985 corresponded to the recruitment of non-oak, mesophytic species at the study site. The results of this study suggest that changes in fire frequency and moisture availability are contributing to changes in tree density and species composition at the study site.

INTRODUCTION

Fire has long been recognized as an important driver of forest dynamics (Pyne 1982). In eastern North America, fire was a likely contributor to the development and sustainment of oak (*Quercus* spp.) forests (Abrams 1992). Anthropogenic fire likely played a role in promoting upland oak forests, as well as changes in these forests (Guyette et al. 2002). Several upland *Quercus* species benefit from and are adapted to frequent surface fires for their regeneration and recruitment in forests (Abrams 1992). However, fire suppression during the 20th century has led to increasing densities of fire-sensitive, mesophytic tree species and a decline in *Quercus* density (Nowacki and Abrams 2008).

Understanding the frequency of historic fires has an important role in explaining changes to contemporary forests. The result is a rich history of studies of fire history across the eastern United States (Shumway et al. 2001; Guyette et al. 2006; McEwan et al. 2007; King and Muzika 2014; Muzika et al. 2015; among others). One of the common patterns found in these studies is that surface fires were often frequent events prior to Euro-American settlement of the area and that fire remained frequent during early Euro-American settlement prior to fire suppression efforts in the early and mid-20th century. Several interacting factors likely contributed to changing fire frequencies in eastern North American forests, including human density, topography, drought, and

https://doi.org/10.22488/okstate.17.100112

climate change (Guyette et al. 2002; McEwan et al. 2011).

Recently, research on forest structure and dynamics in Oklahoma Cross Timbers forests and savannas has highlighted the increase in fire-sensitive tree density and decrease in *Quercus* density since the 1950s attributed to drought and fire suppression efforts (DeSantis et al. 2011). Fire history studies in the Oklahoma Cross Timbers have demonstrated frequent fires prior to Euro-American settlement and a continued presence of fire on the landscape into the mid-20th century (Shirakura 2003; Clark et al. 2007; Stambaugh et al. 2009; DeSantis et al. 2010a; Allen and Palmer 2011).

This research adds to the growing body of literature of forest dynamics and fire history in the Oklahoma Cross Timbers. Preliminary investigation of the Arcadia Conservation Education Area in northeast Oklahoma County revealed the presence of fire scarred trees and remnant wood indicative of historic fires at the site. This research had two objectives: 1) describe the contemporary forest structure by analyzing species composition, density, basal area, and age structure in the overstory and sapling layers of the forest and 2) relate the forest structure to the frequency of historic fires using dendrochronology.

METHODS

Study Site

Lake Arcadia is an approximately 736 ha recreational and water supply lake located in northeastern Oklahoma County. The Army Corps of Engineers constructed the lake beginning in 1980 with the lake pool filling by 1987. The study site was located on the south side of the lake at the Arcadia Conservation Education Area (ACEA) (35°37'29"N, 97°23'16"W). The ACEA is an approximately 226 ha area administered by the Oklahoma Department of Wildlife Conservation since 1996; prescribed fire is not utilized at the site (D. Griffith, Area Manager, pers. comm.). Mean annual temperature is 15.63°C, and mean annual precipitation is 91.4 cm. Annual precipitation is bimodal with the greatest amounts of precipitation during May-June and September-October (Oklahoma Climatological Survey, www.mesonet.org). Soils in this area are classified as Stephenville-Darnell-Niotaze, characterized by shallow sandy to loamy soils (Dominick 2003; Carter and Gregory 2008). Elevations at the study site range from 308.5 m at the lake edge to 323.4 m at the southern boundary of the ACEA.

Preliminary investigation revealed fire scarred trees and remnant wood within a 43 ha area of the ACEA. The focus of this research was within the 43 ha area to study the fire history and forest composition and structure.

Forest Composition, Age Structure, and Radial Growth

Stand structure data were collected on twenty 0.04 ha fixed-area plots located randomly within the 43 ha study area. Within each plot, the diameter at breast height (DBH) of all overstory trees (DBH >10 cm) was measured, and trees were identified to species. For each species in the overstory, estimates of relative density (trees/ha), relative dominance (basal area/ha), and relative importance were calculated to describe the contemporary composition of the forest overstory. Increment cores were collected at 30 cm above the ground from two to four of the largest overstory trees per plot for estimates of age structure and radial growth at the study site. Tree selection was based on the development of the longest tree-ring chronology for the site which can limit age structure interpretation. A total of 71 increment cores were collected from ACEA.

Two 0.01 ha fixed-area subplots were established in each 0.04 ha overstory plot to analyze the species composition and density of saplings (DBH <10 cm, >1.37 m height).

Saplings were identified to species and counted within each subplot. Cross-sections of one to two saplings were collected from paired subplots to study the age structure and radial growth of saplings.

Increment cores were returned to the University of Central Oklahoma where they were mounted and sanded with progressively finer sandpaper (80-grit to 1200-grit) in order to see individual tree-ring boundaries and cellular structure (Stokes and Smiley 1996). Cross-dating procedures were used to confidently assign calendar years to each tree-ring on an increment core. Individual ring-width measurements, to the nearest 0.01mm, were collected on each sample using a Velmex TA Unislide System (Velmex, Inc., Bloomfield, NY), binocular microscope, and J2X measurement software (Voortech Consulting, Holderness, NH). Tree-ring series measurements were compared graphically, using the list method (Yamaguchi 1991), and statistically using the program COFECHA (Holmes 1983; Grissino-Mayer 2001a). Following cross-dating and assignment of calendar years to each tree-ring, pith dates were recorded for age estimation at coring height and tree cohort establishment at the study site. In the event that the pith was missed in an increment core, the methods of Duncan (1989) were used to estimate the number of tree-rings missed to the pith of the tree.

Fire History

Cross-sections were collected selectively from *Q. stellata* remnant wood to study the fire history of the site. *Quercus stellata* has been used successfully for fire history studies in Oklahoma Cross Timbers (Clark et al. 2007; Stambaugh et al. 2009; DeSantis et al. 2010b; Allen and Palmer 2011). The analysis approach of Guyette and Stambaugh (2004) was used to identify fire scars in *Q. stellata*. In their study, fire scars were identified based on bark fissure patterns, common in oak species (Smith and Sutherland 2001), and scarring that occurs across multiple samples during the same year.

A total of 21 samples exhibited scarring associated with surface fires, including 13 recently dead *Q. stellata*, two saplings that demonstrated fire scars, and six snags. Three samples could not be successfully cross-dated. All samples were sanded with progressively finer sandpaper (80-grit to 1200-grit). Ring-widths for each remnant sample were measured using the Velmex TA Unislide System (Velmex, Inc., Bloomfield, NY), binocular microscope, and J2X measurement software (Voortech Consulting, Holderness, NH). Based on cross-dating, calendar years for each tree-ring on fire-scarred samples were assigned using correlation analysis with a master tree-ring chronology created from 39 cross-dated *Q. stellata* tree-ring series from the study site.

Calendar years were assigned to each identified fire scar on a sample. A fire chronology was created based on all fire scars for analysis of fire frequency (mean fire interval) and fire severity (fire years in which >25% samples were scarred) using the program FHX2 (Grissino-Mayer 2001b). Superposed epoch analysis (Grissino-Mayer 2001b) was used to test the association of fire year and drought. Instrumental Palmer Drought Severity Index (Palmer 1965) data for the time period 1895 to 2013 from Oklahoma Climate Region 5 were used to associate fire year and drought. An average was calculated for Reconstructed Palmer Drought Severity Index (Cook et al. 2004) for gridpoint 178 and 179 for purposes of comparing drought and growth of trees prior to 1895. Reconstructed Palmer Drought Severity Indices are reconstruction models based on the association of instrumental Palmer Drought Severity Indices and regional tree-ring chronologies (Cook et al. 1999).

RESULTS

A total of nine species were identified in the overstory at Lake Arcadia. *Quercus stellata* was the most dominant species, but *Q. marilandica* had the highest density. Overall, these two species accounted for 88% of the basal area and 68% of the overstory tree density at the study site. Two *Celtis* species (*C. laevigata* Willd.; *C. occidentalis* L.) combined had the third highest relative tree density (15.1%) and relative dominance (5.51%) (Table 1).

Table 1 Overstory (DBH >10 cm) statistics and sapling (DBH <10 cm, >1.37 m height) density at Lake Arcadia, Oklahoma County, Oklahoma. Relative importance value = (relative density + relative dominance)/2.

Species	Trees/ha	Relative Density	Basal Area (m^2/ha)	Relative Dominance	Relative Importance Value	Sapling Density (stems/ha)
Quercus stellata	95	29.2	14.5	70.1	49.7	576
Quercus marilandica	126	38.8	3.66	17.7	28.3	480
Celtis laevigata	28	8.62	0.71	3.47	6.04	192
Juniperus virginiana	25	7.69	0.78	3.77	5.73	384
Celtis occidentalis	21	6.46	0.42	2.05	4.25	1056
Ulmus rubra	15	4.62	0.50	2.41	3.51	192
Sideroxylon lanuginosum	7	2.15	0.06	0.27	1.21	---
Ulmus americana	4	1.23	0.03	0.12	0.68	96
Sapindus drummondii	4	1.23	0.01	0.05	0.64	---
Cornus drummondii	---	---	---	---	---	384
Cercis canadensis	---	---	---	---	---	192
Quercus muehlenbergii	---	---	---	---	---	96
Celtis reticulata	---	---	---	---	---	96
Total	**325**	**100**	**20.6**	**100**	**100**	**3744**

A total of 11 species was found in the sapling layer (see Table 1). The sapling layer was rather dense (3,744 stems/ha). *Celtis occidentalis* had the highest sapling density (1,056 stems/ha), and the three *Celtis* species accounted for 35% of the sapling density at Lake Arcadia. Approximately 78% of the overstory tree species was also found in the sapling layer; the exceptions were *Sapindus drummondii* Hook & Arn. and *Sideroxylon*

lanuginosum Michx. Two species, that have the potential of growing up to the existing overstory, were identified in the sapling layer but were not found in the overstory (*C. reticulata* Torr.; *Q. muehlenbergii* Engelm.).

A total of 137 samples from nine species was collected for analysis of forest age structure, radial growth, and fire history at Lake Arcadia. *Quercus stellata* and *Q. marilandica* accounted for 71% (n = 97) of the samples. Increment cores were collected from *Q. stellata* (n = 39), *Q. marilandica* (n = 16), *S. drummondii* (n = 1), *Ulmus americana* (L.) (n = 1), *U. rubra* (Muhl.) (n = 3), *S. lanuginosum* (n = 2), *C. laevigata* (n = 3), and *C. occidentalis* (n = 6). Sapling cross-sections were collected from *Q. stellata* (n = 5), *Q. marilandica* (n = 16), *S. lanuginosum* (n = 2), *C. laevigata* (n = 8), *C. occidentalis* (n = 10), and *Juniperus virginiana* (L.) (n = 4) for estimates of sapling age and radial growth. A total of 21 *Q. stellata* samples exhibited scarring associated with surface fires. Two *Q. stellata* saplings exhibited fire scars.

The largest diameter tree in our study plots was a *Q. stellata* that measured 67.5 cm DBH. The oldest tree was a *Q. stellata* that was 193 years old (1821–2014). However, only 23.3% of *Q. stellata* trees dated prior to the 20th century (Fig. 1). The oldest *Q. marilandica* in our study plots was 108 years old (1906–2014). *Q. stellata* demonstrated continuous recruitment beginning in the 1880s, with the 1910s having the recruitment of a large cohort (see Fig. 1). *Q. marilandica* also demonstrated continuous recruitment during the early and mid-20th century. The oldest non-*Quercus* individual in the overstory was a *C. occidentalis* that was 62 years old (1952–2014). The age structure of the non-*Quercus* species in the overstory (*S. drummondii*, *U. americana*, *U. rubra*, *S. lanuginosum*, *C. laevigata*, *C. occidentalis*) indicated continuous recruitment beginning in the 1950s and peaking during the 1980s (see Fig. 1).

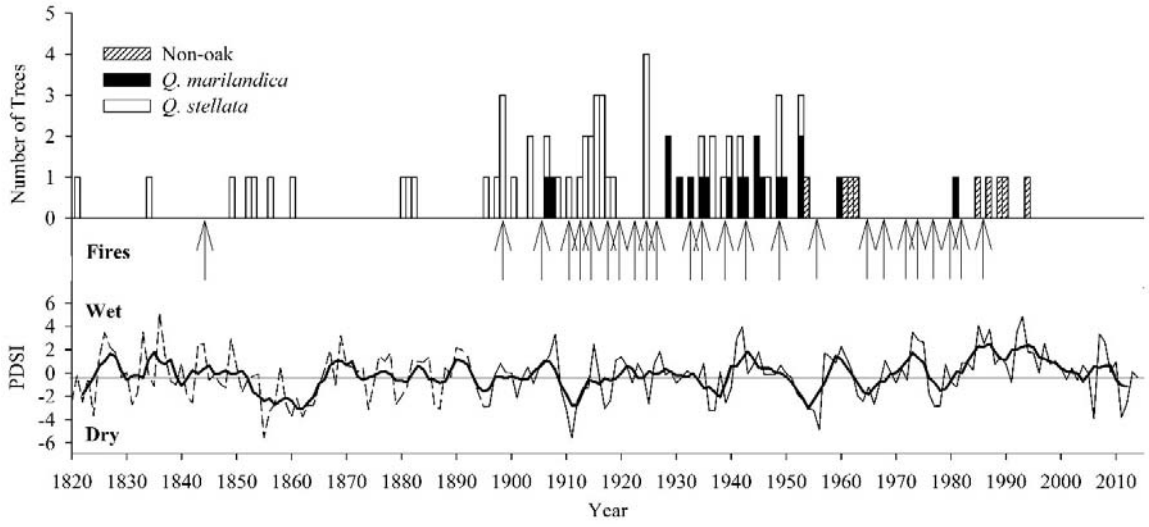

Figure 1 Age structure of *Q. stellata*, *Q. marilandica*, and non-oak species. Non-oak species include *S. lanuginosum, C. laevigata, C. occidentalis*, *S. drummondii*, *U. americana*, *U. rubra*. Arrows indicate the year of a fire. Bottom graph represents reconstructed (dashed line) and instrumental (full line) Palmer Drought Severity Index (PDSI) for central Oklahoma.

The oldest sapling in the understory of the study plots was a *Q. marilandica* that was 62 years old (1952–2014). Approximately 49% (n = 19) of non-oak saplings recruited during the 1980s (Fig. 2). Establishment of non-oak species appeared to correspond to periods of above-average PDSI following the 1960s (see Figs. 1, 2).

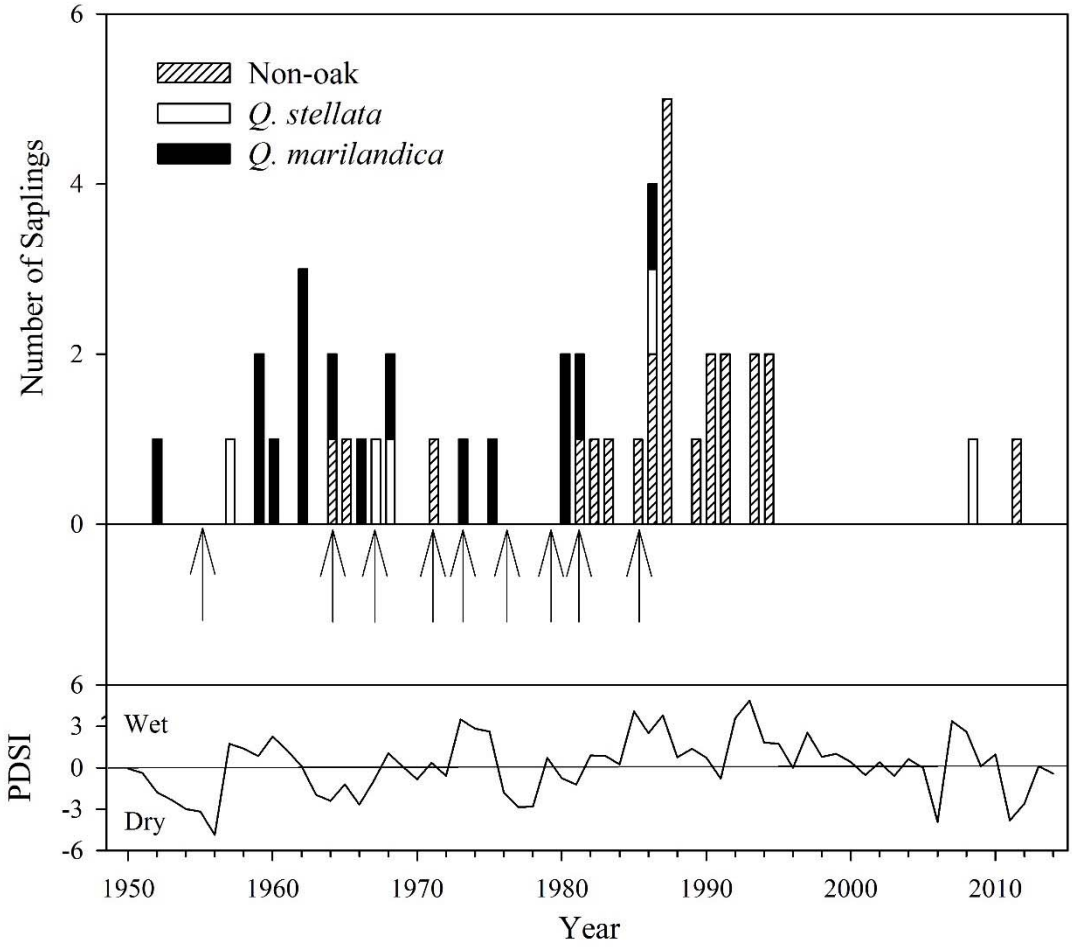

Figure 2 Age structure of *Q. stellata*, *Q. marilandica*, and non-oak saplings at Lake Arcadia. Non-oak species includes *S. lanuginosum, C. laevigata, C. occidentalis*, and *J. virginiana*. Arrows represent years of fire. Bottom graph is the instrumental PDSI for central Oklahoma (1952-2014).

Fifty-one fire scars were identified and dated, that occurred from 25 different fire events (Fig. 3). The earliest fire occurred in 1844 with a range of fire years from 1844 to 1985. However, the 1844 fire scar was not used in any of the fire analyses due to a low sample depth during that time period it and being represented on only one sample. Approximately 29.7% of years 1898 to 1985 had a fire. The most severe fire years (based on percentage of trees scarred) included 1898 (33.3%), 1912 (55.6%), 1922 (41.7%), and 1955 (41.7%). The mean fire interval (MFI) for all fires from 1898 to 1985 was 4.14 years (SD ± 2.22, range 2–9 years). Superposed epoch analysis was conducted to test the association between drought and any fire year. Results indicated no significant association between any fire year (1898 to 1985) and drought (Fig. 4). Severe fires

during 1912 and 1955 were associated with extreme drought conditions (PDSI ≤ -2.99). For the period 1898 to 1985, 13 fires occurred during dry conditions (PDSI < 0), and 11 fires occurred during wet conditions (PDSI > 0).

DISCUSSION

Changes in historic fire regimes and land-use patterns often lead to changes in forest structure and composition. In the Cross Timbers region of Oklahoma, changes in forest structure and composition are apparent in terms of increasing tree density, particularly increases in fire sensitive tree species (DeSantis et al. 2010a, 2011). The result of changing historic forest dynamics is the "mesophication" (Nowacki and Abrams 2008) of Cross Timbers forests. This study demonstrates the continued dominance of *Q. stellata* and *Q. marilandica* in the overstory of this Cross Timbers forest. However, this study also highlights the effect of a changing fire regime on forest structure at the study site.

Total basal area for this study is similar to other studies across multiple sites in the Oklahoma Cross Timbers (DeSantis et al. 2010a, 2011) and Arkansas Cross Timbers (Bragg et al. 2012). DeSantis et al. (2010a) demonstrate increases in non-oak basal area and tree density across multiple sites in Oklahoma between the 1950s and 2000s. This study shows that *Celtis* species collectively make up the third most important group at the study site (see Table 1). The two *Celtis* species, *Juniperus virginiana* and *Ulmus* species, in this study along with the other non-oak species are sensitive to fire as seedlings and saplings (Coladonato 1992, 1993; Sullivan 1993; Anderson 2003; Gucker 2011). Generally, only the most severe fires will kill overstory trees of these species.

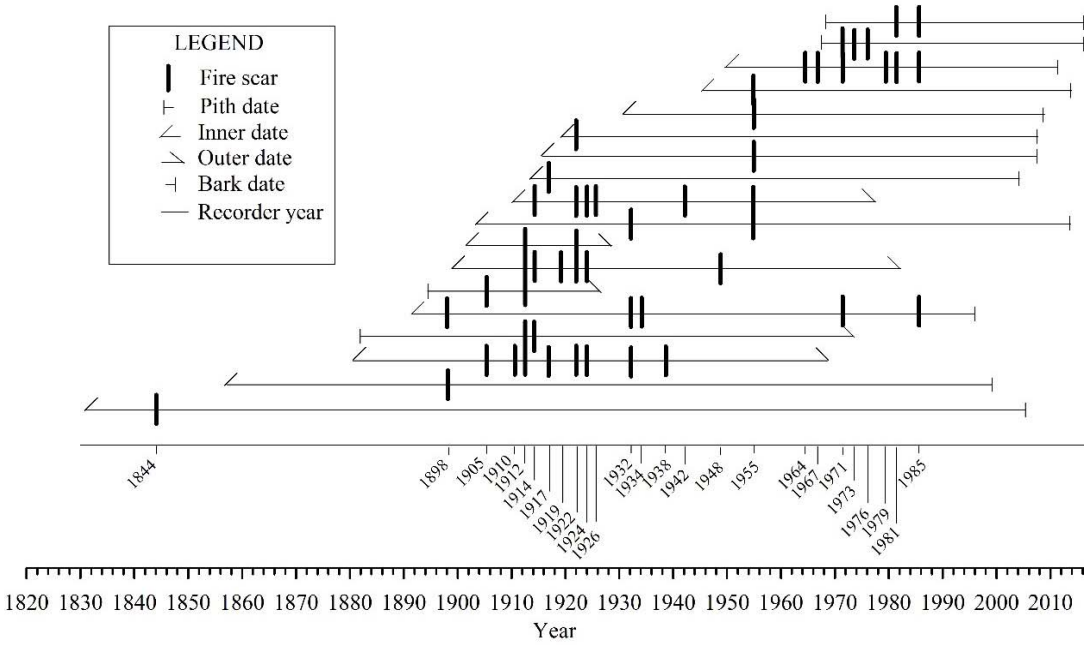

Figure 3 Fire history graph for Lake Arcadia in northeastern Oklahoma County, Oklahoma. Horizontal lines represent the length of the tree-ring record for each sample (n = 18). Vertical dashes represent the year of a fire scar in each tree-ring record. The composite fire chronology is represented by the fire years.

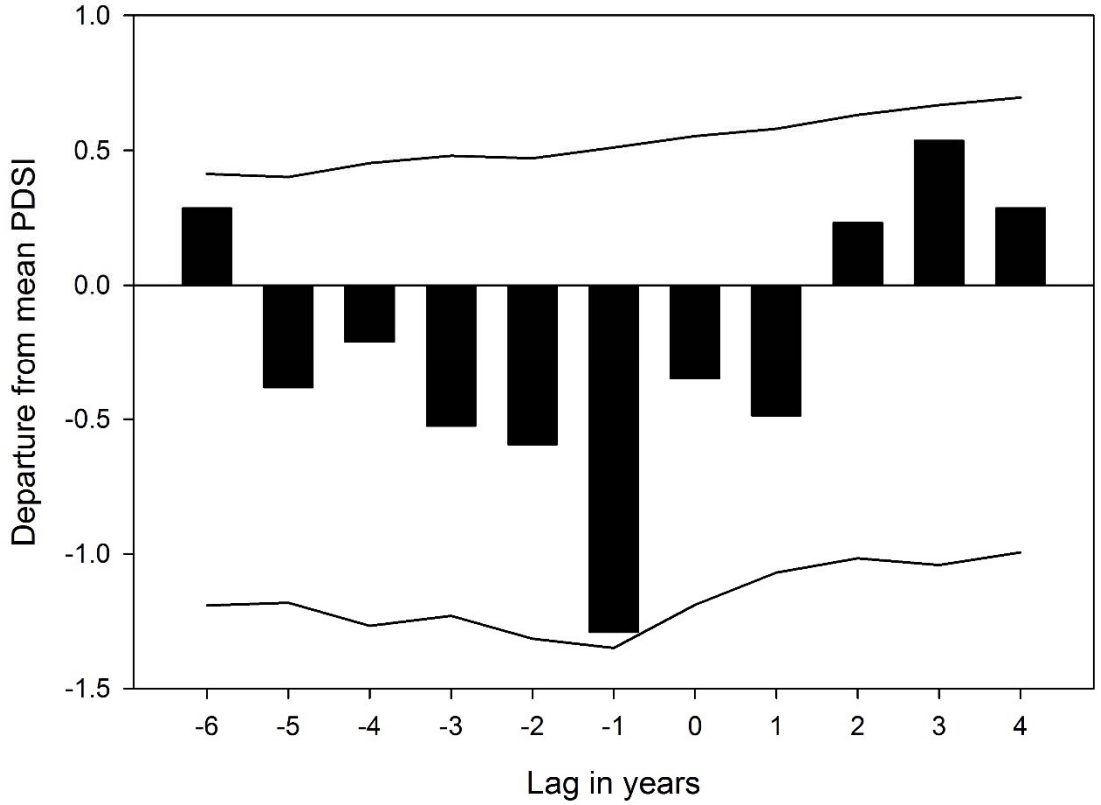

Figure 4 Superposed epoch analysis for all fires from 1898 to 1985 compared to PDSI (drought). This program analyzes the relationship between any fire year and drought (Grissino-Mayer 2001b). Year "0" is the year of any fire year; Year "-1" indicates the departure from the mean PDSI one year prior to any fire year. Horizontal lines represent 95% confidence interval based on 1000 Monte Carlo simulations.

This study highlights the recruitment of a large number of non-oak trees during the 1980s (see Figs. 1, 2). There are two factors which likely contributed to this recruitment. The last fire that was documented at Lake Arcadia occurred in 1985 (see Fig. 3). Additionally, following the drought during the late 1970s and early 1980s in the central Oklahoma region was an 18 year period (1982–2000) of above-average PDSI (see Figs. 1, 2). This 18 year period along with no fires after 1985 likely contributed to recruitment of these non-oak species. The data also show recruitment of non-oak trees following the 1950s drought (see Figs. 1, 2).

DeSantis et al. (2011) found increases in species recruitment following drought during the 1950s and decreases in *Quercus* recruitment associated with fire suppression. Clark et al. (2007) indicated increased recruitment of *J. virginiana* during fire free periods and increased recruitment of *Quercus* species following frequent fires. The results of this study also suggest that fire-free periods (between 1955 and 1964; post-1985) (see Fig. 3) contributed to non-oak recruitment at Lake Arcadia. The 1964 and 1967 fires are represented on only one sample, which may suggest that these fires were of low severity and had little effect on non-oak recruitment during this time

period. Recruitment of *Q. stellata* during the early 20th century occurred during high fire frequency (1905–1926, MFI = 2.62 years). Following 1926, the number of fires declined to seven in a 38 year period (1926–1964). The current *Q. marilandica* overstory recruited during the mid-20th century period, which coincided with surface fires.

The fire frequency at Lake Arcadia (MFI = 4.14 years) is within the range of other studies in the Cross Timbers and other mixed-species forests of Oklahoma. DeSantis et al. (2010b) in Okmulgee County reported an MFI equal to 2.7 years for the time period 1750 to 2005. When considering a similar time period to this study, they report an MFI of 2 years. Clark et al. (2007) indicated a range of fire frequency between 2.5 and 6 years (1770–2002) based on the aspect of the forest stand at sites in Osage County. Allen and Palmer (2011) report an MFI for all fires of 2.3 years (1729–2005) at a different site in Osage County. Stambaugh et al. (2009) at the Wichita Mountains National Wildlife Refuge found an MFI of 4.7 years for all fires between 1722 and 2001. At the Nickel Family Nature and Wildlife Preserve in northeastern Oklahoma, Stambaugh et al. (2013) found a fire frequency of 2.6 years in a mixed oak-pine (*Quercus-Pinus*) forest. Masters et al. (1995) in a study of fire history in McCurtain County reported a mean fire interval of 3.8 years.

Comparing the association between drought and fire year revealed no significant association at Lake Arcadia (see Fig. 4). This result is similar to other studies in the Oklahoma Cross Timbers (Allen and Palmer 2011; DeSantis et al. 2010b; Stambaugh et al. 2009) and contrary to that reported by Clark et al. (2007). Three of four severe fire years (1898, 1912, 1955) coincided with below average PDSI (drought) conditions. The exception was the 1922 severe fire year which coincided with above average PDSI.

In all previous studies of fire history in the Oklahoma Cross Timbers, fires were found to be frequent events prior to Euro-American settlement (<1890) and after Euro-American settlement (>1890). There is a noticeable lack of fires between 1844 and 1898 (see Fig. 3). There are several possible explanations for the absence of fire scars. Not every fire which occurs at a site will result in the formation of a fire scar. Most remnant samples had only heartwood present that often resulted in too few tree-rings to accurately cross-date. Decomposition of the heartwood was also a common feature of the trees at Lake Arcadia that possibly resulted in the loss of fire scars that were present during the mid and late 19th century. However, even with the limited temporal scope of the fire history, this study demonstrates frequent fires at the Lake Arcadia area during the 20th century.

CONCLUSIONS

Fire was likely an important factor that sustained the dominance of *Quercus* species in upland forests (Abrams 1992). While this study has some limitations, it does highlight *Quercus* recruitment coincided with frequent fires during the 20th century. Changes in fire frequency after 1985 and fire-free periods promoted non-oak recruitment in the understory, similar to other studies in the Oklahoma Cross Timbers (Clark et al. 2007) and across other upland *Quercus* forests in the eastern United States (Abrams 1992). Studies of fire history are important for understanding forest development, the historical role of humans on the landscape, and the development of management guidelines for sites which utilize prescribed fire. This study adds to the growing knowledge of historic fire frequency in the Oklahoma Cross Timbers. Fires were frequent events that shaped the historic Cross Timbers, and often the high frequencies continued into the mid and late 20th century. Comparatively, the number of

studies that have specifically addressed fire history in the Oklahoma Cross Timbers is limited. Other sites should be selected and studied to further expand the knowledge of historic fire on the Cross Timbers landscape.

ACKNOWLEDGEMENTS

I would like to thank Justin Cheek and Shey Ramsey at the University of Central Oklahoma for their assistance in the field and laboratory. I would like to graciously thank Daniel Griffith at the Arcadia Conservation Education Area for permission to collect samples at the study site. Thanks to two anonymous reviewers for constructive comments and suggestions on an earlier version of this manuscript. This research was supported by the Office of Research and Grants at the University of Central Oklahoma.

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INTERPLANTING FLORAL RESOURCE PLANTS WITH VEGETABLE PLANTS ENHANCES BENEFICIAL ARTHROPOD ABUNDANCE IN A HOME GARDEN

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Keywords: cowpea, herbivory, native ornamentals, natural enemies, pollinators, polyculture, tomato

ABSTRACT

We examined whether interplanting vegetable and ornamental flowering plants reduces herbivory and enhances photosynthetic rate, plant growth, natural enemy abundance, and pollinator visitation relative to monoculture plantings. We found no evidence of physiological or growth costs due to growth in polyculture. Herbivore damage to plants did not differ with planting regime. Natural enemies occurred in greater abundance in polycultures compared to monocultures. Pollinator diversity was enhanced in some polyculture plots. We suggest that interplanting vegetable and flowering ornamental plants at small spatial scales may improve plant health and reproduction through natural pest control and a diversified pollinator pool.

INTRODUCTION

Habitat manipulation strategies regulate pest populations in managed landscapes by enhancing the abundance of arthropod predators and parasitoids (natural enemies) by provisioning additional plant-based resources (i.e., nectar, pollen, alternative prey, or shelter) (Rebek et al. 2005, 2006; Fiedler et al. 2008). These same strategies may also have beneficial effects for pollinator abundance and diversity due to an increased abundance of flowering plants in the managed landscape (Tuell et al. 2008). A common habitat manipulation strategy that often benefits natural enemies and pollinators in managed landscapes is the use of polycultures, the cultivation of multiple plant species together.

Relative to most monoculture plantings, polycultures offer beneficial arthropods (i.e., natural enemies and pollinators) greater floral resources (i.e., nectar and pollen rewards) throughout the growing season, alternative prey, and increased habitat structure and availability of nesting sites (Andow and Risch 1985; Andow 1991; Landis et al. 2000, 2005; Hooks and Johnson 2003). Polycultures may also provide improved microhabitats for plants and arthropods, such as increased shade and protection from wind, relative to most monocultures (Andow 1991; Landis et al. 2000). In addition to enhancing beneficial

https://doi.org/10.22488/okstate.17.100113

arthropod abundance, polycultures often support lower pest arthropod abundance than monocultures (Kloen and Altieri 1990; Nicholls and Altieri 2004; Ponti et al. 2007; Isaacs et al. 2009).

At small spatial scales, such as those of home gardens, polycultures are a particularly attractive alternative to cultivation techniques that require heavy pesticide applications to control pest arthropods. Home gardeners commonly use pesticides to control pest arthropods (Sadof et al. 2004); in the United States, 16% of all insecticides applied annually are used in residential gardens and lawns (U. S. EPA 2011). Widespread residential pesticide use poses significant threats to human health and the environment by increasing the incidence of pesticide poisonings (Pimentel et al. 1992; U. S. EPA 2009), reducing stream and ground water quality (Cohen 2010), and killing non-target organisms (e.g., insect pollinators, aquatic fauna) (Johansen and Mayer 1990; Pimentel et al. 1992; Relyea 2009). Effective alternatives to residential pesticide applications are needed to improve safety, minimize effects on non-target organisms, and reduce environmental contamination.

To date, most studies of polyculture techniques have examined the role of plant-based resources for natural enemy ecology and in regulating natural enemy populations (Fiedler et al. 2008). What remains less well studied is whether planting polyculture gardens of vegetable and flowering ornamental plants has other beneficial effects for garden crops. We hypothesized that plants grown in polycultures will have higher rates of pollinator visitation as well as higher abundance of natural enemies relative to monoculture plantings. With an increase in natural enemy abundance (Landis et al. 2000), we hypothesized that plants grown in polycultures will experience reduced rates of herbivory compared to monoculture plantings. Herbivore damage is known to adversely affect photosynthesis and plant growth (Crawley 1997; Zangerl et al. 2002). Thus, if growing plants in polycultures reduces herbivory, then we hypothesized that plants in polycultures will have higher rates of photosynthesis and growth relative to monoculture plantings. Accordingly, the first objective of this study was to examine whether polycultures of vegetable and flowering ornamental plants reduce herbivory relative to monocultures. Our second objective was to examine whether polycultures of vegetable and ornamental plants enhance photosynthetic rate and growth relative to monocultures. Our third objective was to examine whether polycultures enhance pollinator visitation and pollinator diversity relative to monoculture plantings. Our fourth objective was to examine whether polycultures enhance natural enemy abundance relative to monocultures.

MATERIALS AND METHODS

Garden Design

We conducted this study at The Botanic Garden at Oklahoma State University (Stillwater, OK; 36°07'08.6" N, 97°06'04.5" W) from April 23, 2009, to September 1, 2009. Seven plant species were included in the study. Four native, commonly cultivated ornamental species were largeflower tickseed (*Coreopsis grandiflora* Hogg ex Sweet 'Early Sunrise'), purple coneflower *(Echinacea purpurea* (L.) Moench), blanketflower (*Gaillardia* x *grandiflora* Van Houtte 'Arizona Sun'), and goldenrod (*Solidago* sp. 'Wichita Mountains'). Three commonly cultivated vegetable species were cilantro (*Coriandrum sativum* L.), tomato (*Solanum lycopersicum* L. 'Mountain Fresh Plus'), and cowpea (*Vigna unguiculata* (L.) Walp 'Early Scarlet'). We chose vegetable species that would be typical of an Oklahoma or southern U.S. home garden (Hillock and Simons 2002). While tomato and cowpea are self-fertile, visitation by insects, primarily bees, improves

reproductive success (Free 1993). All flowering plants included in our study provide nectar and pollen to beneficial arthropods, and the chosen species overlap in blooming period, ensuring a continuous supply of floral resources.

We used a randomized complete block design consisting of four blocks of nine experimental plots each. Plots within each block were randomly assigned to one of nine planting treatments. Each plot measured 1 m x 2 m and was separated from other plots by a 1 m mulched border. All plots and borders were kept free of weeds by hand pulling. All plots were composed of native soil (Norge loam, fine-silty, mixed, thermic Udic Paleustolls) and were provided supplemental water by drip irrigation. Plants were not fertilized, as adequate plant mineral nutrients were available from fertilization of previous trials. The nine planting treatments included monocultures of each of the plant species (seven plots) and two different polycultures to add more generality to our results.

One polyculture consisted of largeflower tickseed, goldenrod, cilantro, and tomato (one plot; 'Polyculture One'), and the other polyculture consisted of purple coneflower, blanketflower, goldenrod, and cowpea (one plot; 'Polyculture Two'). Monocultures of the four ornamental species were planted with 18 plants/plot on April 23–24, 2009, using established nursery stock. Tomato monocultures were planted on April 25, 2009, using established nursery stock and included two plants/plot; plants were centered in each plot and spaced 60 cm apart within the row. We seeded the monocultures of cilantro on April 25, 2009, at a density of 240 seeds per 1 m row, with six rows per plot. Monocultures of cowpea were seeded on May 22, 2009, at a density of 20 seeds per 1 m row, with two rows per plot. We later thinned cowpea to 10 plants per 1 m row where stands permitted. Within Polyculture One plots, we planted six largeflower tickseed, three goldenrod, two tomatoes, and seeded one row of cilantro at a density of 240 seeds per 1 m row. We planted goldenrod and largeflower tickseed on April 23–24, 2009. We planted tomatoes and seeded cilantro on April 25, 2009.

Within Polyculture Two plots, we planted three purple coneflower, three blanketflower, three goldenrod, and seeded two rows of cowpea at a density of 20 seeds per 1 m row. We later thinned cowpeas to 10 plants per row where stands permitted. We planted cowpeas on May 22, 2009, and purple coneflower, blanketflower, and goldenrod on April 23–24, 2009. The plant species were sown at densities recommended by the Oklahoma Cooperative Extension Service. Different species were not planted on the same date because 1) a planting date of April 25 was too early for cowpea, which was direct-seeded and requires warm soils for proper germination; and 2) ornamental plants (purple coneflower, blanketflower, and goldenrod) were planted at later dates as a result of plant availability. We did not observe any shading of later-planted species by those planted earlier.

Cilantro and goldenrod did not establish in monoculture or polyculture. In addition, several plots of the other plant species did not establish well. Thus, our analyses included four plots of largeflower tickseed, three plots of purple coneflower, four plots of blanketflower, three plots of tomato, two plots of cowpea, two plots of Polyculture One, and three plots of Polyculture Two.

Herbivory

To determine whether planting regime influenced rates of herbivory, we quantified leaf damage on two plants of each species per plot twice during the growing season (June and July). For each plant, we estimated herbivore damage on one standard module per plant (Turcotte et al. 2014) by counting the total number of leaves and the number of leaves with

herbivore damage on the module (i.e., branch, rosette). The plant module varied for each species based on plant morphology; we counted all of the leaves on purple coneflower, only the leaves of the basal rosette on blanketflower, the leaves of one stem on largeflower tickseed, and the leaves of one lower branch on tomato and cowpea. For these same plants, we then recorded the number of leaves on the module damaged by herbivory. We calculated percent of damaged leaves for each plant as the total number of damaged leaves divided by the total number of leaves per module. We quantified herbivore damage on plants rather than inventorying herbivores, as herbivore damage represents a more comprehensive temporal perspective on herbivory in these plots; however, common herbivores in these crops included aphids (family Aphididae), tomato hornworms (*Manduca quinquemaculata*), flea beetles (family Chrysomelidae, tribe Alticini), squash bugs (*Anasa tristis*), cucumber beetles (*Acalymma* sp. and *Diabrotica* sp.), and spider mites (family Tetranychidae).

Plant Height and Photosynthetic Measurements

To determine whether planting regimes (i.e., monoculture versus polyculture) affected traits related to plant health, we quantified height to the nearest centimeter and measured light-saturated photosynthetic rate using an infrared gas analyzer (LI-6400, LI-COR, Inc.; Lincoln, NE). Photosynthetic rate was quantified for one newly expanded leaf from each of two plants per species per plot. We recorded these measurements twice during the growing season (July and August). Each month, all measurements were taken within a three-day period between 09:00–13:00 CDST on sunny days. We standardized leaf chamber conditions with a temperature of 30°C, photosynthetically active radiation (PAR) at 1500 μmol $m^{-2}s^{-1}$, and CO_2 concentration of 400 ppm. Because calculations of photosynthetic rate are based in part on leaf surface area, we collected leaves that did not fill the entire leaf chamber and later determined leaf surface area using image analysis software (ImageJ, National Institutes of Health Freeware; Bethesda, MD).

Pollinator Abundance and Composition

Throughout the summer, we observed insect visitation to flowers within our experimental plots during 15 min observation periods. Observations were limited to sunny days when the wind was calm. Throughout the summer, observation times varied throughout the day (between 07:00–17:00 CDST) to capture a wider diversity of insect visitors. On a given day, we rotated observations among experimental plots (Kearns and Inouye 1993). During each 15 min observation period, we recorded insect visitation at the flower or inflorescence level, recording visits to all open flowers or inflorescences on several plants within each experimental plot. Observations of tomato and cowpea were conducted at the flower level; whereas, observations of all other plant species were conducted at the inflorescence level. Within a plot, we observed as many flowers or inflorescences on as many plants as was possible at one time, including simultaneous observations of several plant species in polycultures. We recorded floral visitors from four insect orders: beetles (Coleoptera); wasps, honey bees, bumble bees, and small-bodied bees (Hymenoptera); true flies (Diptera); and butterflies (Lepidoptera).

We observed monoculture plots for a total of 7.75 h over the course of the experiment. Total duration of observations varied among species in monocultures (tomato, 1.25 h; cowpea, 1 h; purple coneflower, 1.5 h; largeflower tickseed, 1.5 h; blanketflower, 2.5 h). We observed polyculture plots for a total of 9 h over the

course of the experiment. As with monocultures, the total duration of observations varied among species in polycultures (tomato, 1.25 h; cowpea, 1.75 h; purple coneflower, 1.5 h; largeflower tickseed, 2.5 h; blanketflower, 3 h).

Natural Enemy Abundance and Composition

We sampled natural enemies using 7.5 x 13 cm yellow sticky cards (Hoback et al. 1999) every two weeks throughout the growing season, for a total of seven sample dates over the course of the experiment. Around mid-morning on selected days, two sticky cards per plot were placed 1 m above ground level on stakes and left for 48 h. We used a compound stereomicroscope to identify and sort specimens from the sticky cards into twelve groups of arthropods: spiders (order Araneae), rove beetles (family Staphylinidae), lady beetles (family Coccinellidae), hover flies (family Syrphidae), tachinid flies (family Tachinidae), minute pirate bugs (family Anthocoridae), nabid bugs (family Nabidae), other predators, parasitic wasps, other wasps, bees, and other pollinators. Arthropods were sorted and identified to family level and/or functional group (e.g., parasitic wasps) for comparison among plots. We defined total natural enemy abundance as the sum of individuals of all arthropod classes found on the sticky cards, excluding bees and other pollinators from the total. As yellow sticky cards are not effective at sampling the pollinator community (Kearns and Inouye 1993), we did not analyze the bee/other pollinator data gathered from the sticky cards.

Flowering Phenology

For comparison with natural enemy abundance, we recorded flowering phenology weekly as the number of open flowers (tomato and cowpea) or inflorescences (purple coneflower, blanketflower, and largeflower tickseed) for three plants per species per plot.

Statistical Analyses

To examine whether planting regime (monoculture versus polyculture), species, or their interaction influenced percent of leaves with herbivore damage, height, or light-saturated photosynthetic rate, we performed separate repeated measures analyses of variance (ANOVA) for each variable (PROC GLM, SAS Institute; Cary, NC). The model for these analyses included block as a random effect and planting regime, plant species, and their interaction as fixed effects. Prior to analysis, we tested all response variables for normality and found that the variables met ANOVA assumptions without data transformation.

We performed *G*-tests (Zar 1999) separately for each plant species to determine whether pollinators under- or over-visited flowers (or inflorescences) on plants grown in monoculture relative to those in polyculture. Visits to flowers (or inflorescences) were considered independent events and the unit of sampling was individual flowers (or inflorescences) within a plot. Observations of tomato and cowpea were performed at the flower level. Analyses of members of Asteraceae (purple coneflower, blanketflower, and largeflower tickseed) were performed at the inflorescence level. For each 15 min observation period, we calculated visitation rate per plant species as the total number of visitors divided by the total number of flowers (or inflorescences). Visitation rate was not normally distributed, even after data transformation; thus, we performed non-parametric Kruskal-Wallis tests separately for each plant species (PROC NPAR1WAY, SAS Institute; Cary, NC) to determine whether planting regime influenced total visitation rate of all floral visitors.

To determine whether planting regime, sampling date, or their interaction

influenced total abundance of natural enemies, we performed repeated measures ANOVA (PROC MIXED, SAS Institute; Cary, NC). The model for this analysis included block as a random effect and planting regime, sampling date, and their interaction as fixed effects. When we detected a significant main effect of species or a significant interaction between plant species and planting regime, we used Tukey post hoc comparisons to test for differences among means. To determine whether there was a relationship between total natural enemy abundance and flowering phenology (i.e., number of open flowers) of each plant species over the course of the experiment, we used a non-parametric Spearman rank correlation (PROC CORR, SAS Institute; Cary, NC).

RESULTS

Herbivory

Leaf damage did not differ between plants grown in monoculture versus those grown in polyculture gardens across the growing season ($F_{1,15} = 0.71$, $P = 0.41$) (Fig. 1; monoculture vs. polyculture mean leaf damage ± standard error (SE): 16.12% ± 2.27% vs. 9.46% ± 2.49% leaves damaged). All species responded similarly in terms of leaf damage to planting regime across the growing season ($F_{4,15} = 0.69$, $P = 0.61$). As expected, leaf damage differed across plant species ($F_{5,15} = 29.41$, $P < 0.0001$; see Fig. 1).

Plant Height and Photosynthetic Measurements

As expected, plant species differed in height and photosynthetic rate (Height: $F_{4,16} = 49.90$, $P < 0.0001$; Photosynthetic rate: $F_{4,15} = 31.69$, $P < 0.0001$) (Fig. 2). Plant height and photosynthetic rate were not significantly affected by planting regime (i.e., monoculture versus polyculture) (analyses not shown; all $P > 0.10$ for planting regime effect) and all species responded similarly in terms of these traits to planting regime (analyses not shown; all $P > 0.10$ for interaction term).

Pollinator Abundance and Composition

Of the two vegetable plant species (cowpea and tomato) for which we conducted pollinator observations, we only recorded floral visitors to cowpeas; we observed no insects visiting tomato flowers. Coleoptera were observed more frequently on flowers of cowpea plants grown in polyculture than those grown in monoculture ($G = 3.85$, $P < 0.05$), but the opposite pattern occurred for Diptera ($G = 47.3$, $P < 0.0001$). The pattern of visitation to cowpeas by Hymenoptera and by other floral visitors did not differ significantly with planting regime (all $P > 0.05$). Insects from three orders (i.e., Coleoptera, Diptera, and Hymenoptera) were observed visiting cowpea flowers in monoculture gardens; whereas, we observed insects from four orders (i.e., Coleoptera, Diptera, Hymenoptera, and Lepidoptera) visiting cowpea flowers in polyculture plantings (Fig. 3A). Thus, the diversity of floral visitors to cowpea was greater in polycultures compared to monocultures (see Fig. 3A).

Hymenoptera were observed more frequently visiting inflorescences of purple coneflower plants in polycultures than those in monoculture ($G = 18.6$, $P < 0.001$), but the opposite pattern was found for Coleoptera ($G = 4.1$, $P < 0.05$). Visitation by Lepidoptera and Diptera to purple coneflower did not differ significantly between planting regimes ($P > 0.05$; Fig. 3B). Lepidoptera were observed more frequently visiting inflorescences of blanketflower plants grown in polyculture than those in monoculture ($G = 3.94$, $P < 0.05$). The proportion of visits to inflorescences by Coleoptera, Diptera, and Hymenoptera did not differ significantly between blanketflower planting regimes (all $P > 0.05$; Fig. 3C). The proportion of visits

to inflorescences by four taxa (Coleoptera, Diptera, Hymenoptera, and Lepidoptera) did not differ significantly between largeflower tickseed planting regimes (all $P > 0.05$; Fig. 3D).

Although total visitation rate across all insect orders did not differ significantly between planting regimes for any plant species (purple coneflower: $X^2_1 = 0.0064$, $P > 0.10$; largeflower tickseed: $X^2_1 = 0.6433$, $P > 0.10$; blanketflower: $X^2_1 = 0.1491$, $P > 0.10$; cowpea: $X^2_1 = 3.0$, $P = 0.0833$), cowpea growing in monocultures tended to experience a higher visitation rate compared to cowpeas grown in polycultures (Fig. 4).

Natural Enemy Abundance and Composition

Total abundance of natural enemies was significantly higher in polycultures compared to monocultures ($F_{7,102} = 4.34$, $P = 0.0003$; mean natural enemies/plot/sampling date for monocultures vs. polycultures ± SE: 25.78 ± 1.24 vs. 28.31 ± 2.84). In addition, Polyculture Two yielded 30% higher natural enemy abundance than Polyculture One. Parasitic wasps were by far the most common group of natural enemies in all planting regimes (Fig. 5). Total abundance of natural enemies differed across the season ($F_{6,102} = 68.08$, $P < 0.0001$); natural enemy abundance was highest in late spring, rapidly declined in mid-June, rebounded in mid-July, and then decreased for the remainder of the growing season (Fig. 6). There was a significant interaction between planting regime and sampling date for natural enemy abundance ($F_{42,102} = 1.53$, $P = 0.0434$)

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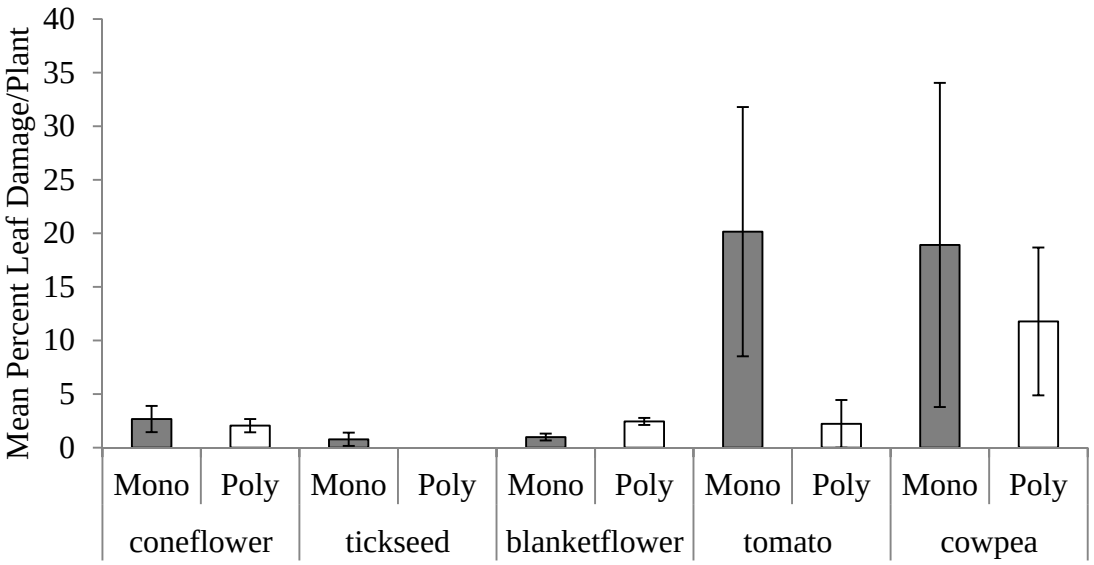

Figure 1A Mean herbivory (± 1 SE) of purple coneflower, largeflower tickseed, blanketflower, tomato, and cowpea grown in monoculture (solid bar) and polyculture (open bar), during June 2009, in Stillwater, Oklahoma.

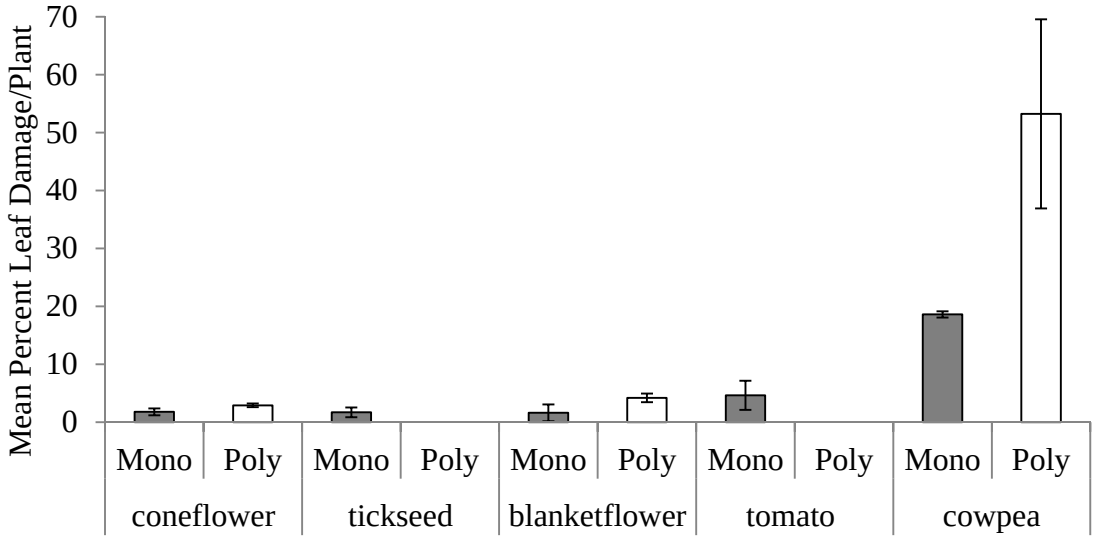

Figure 1B Mean herbivory (± 1 SE) of purple coneflower, largeflower tickseed, blanketflower, tomato, and cowpea grown in monoculture (solid bar) and polyculture (open bar), during July 2009, in Stillwater, Oklahoma.

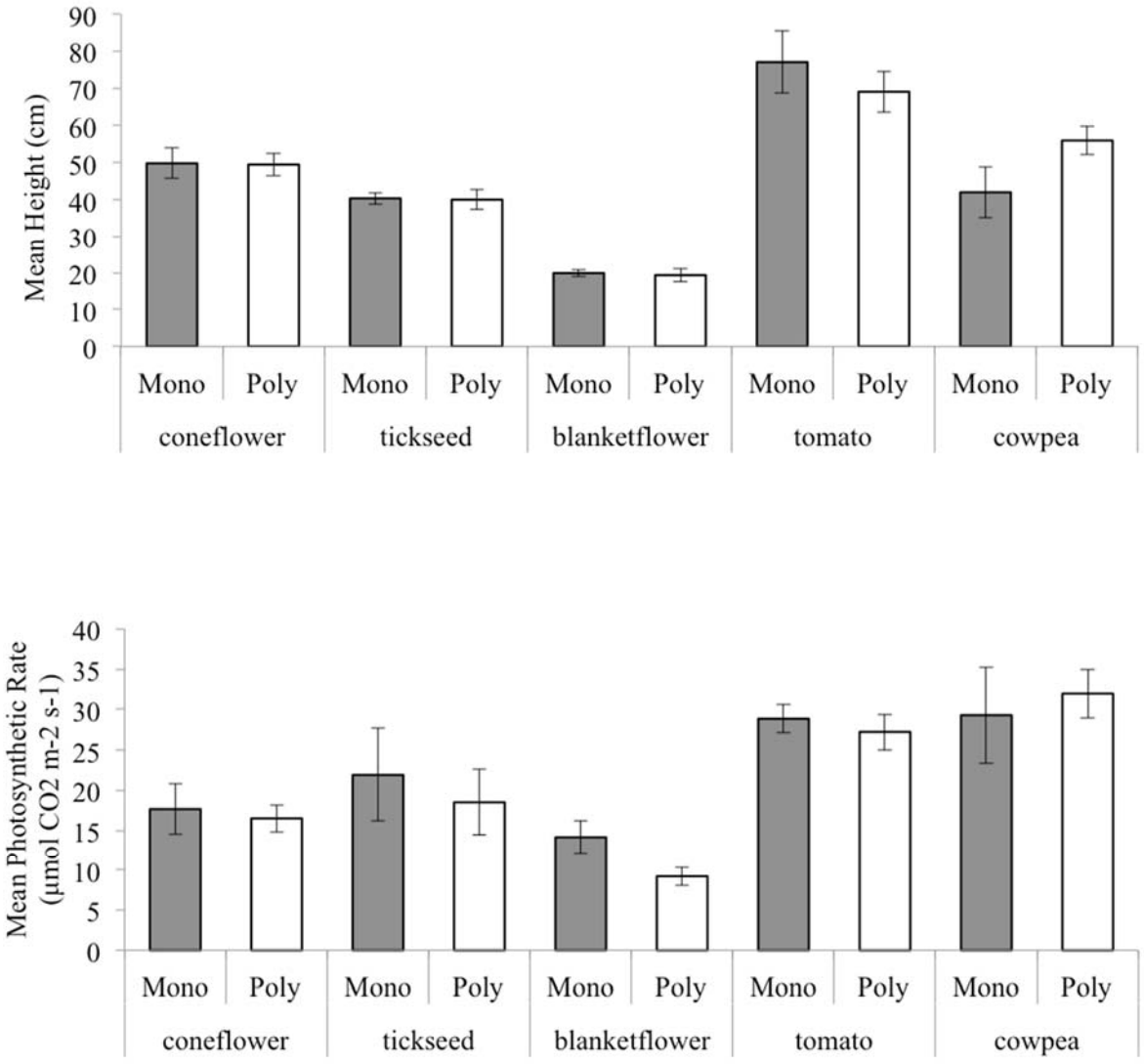

Figure 2 Mean plant height and light-saturated photosynthetic rate (± 1 SE) of ornamentals (coneflower, tickseed, and blanketflower) and vegetables (tomato and cowpea) grown in monoculture (solid bar) and polyculture (open bar), during July 2009, in Stillwater, Oklahoma. August data not shown.

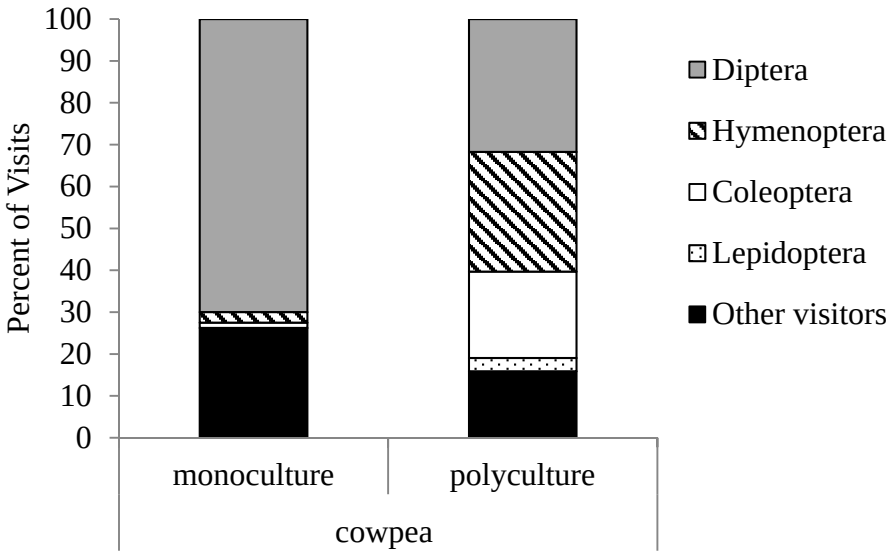

Figure 3A Percent of floral visits from four insect orders to cowpea grown in monoculture and polyculture, April 23–September 1, 2009, in Stillwater, Oklahoma.

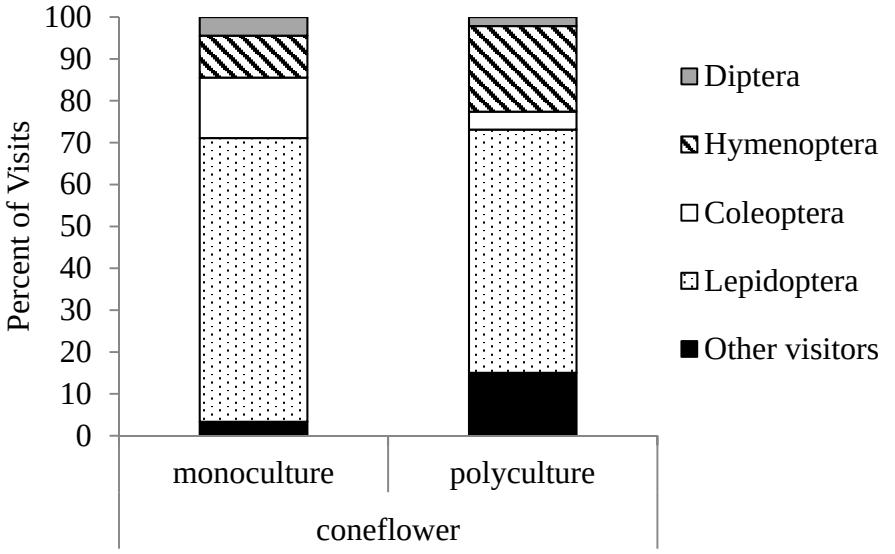

Figure 3B Percent of floral visits from four insect orders to purple coneflower grown in monoculture and polyculture, April 23–September 1, 2009, in Stillwater, Oklahoma.

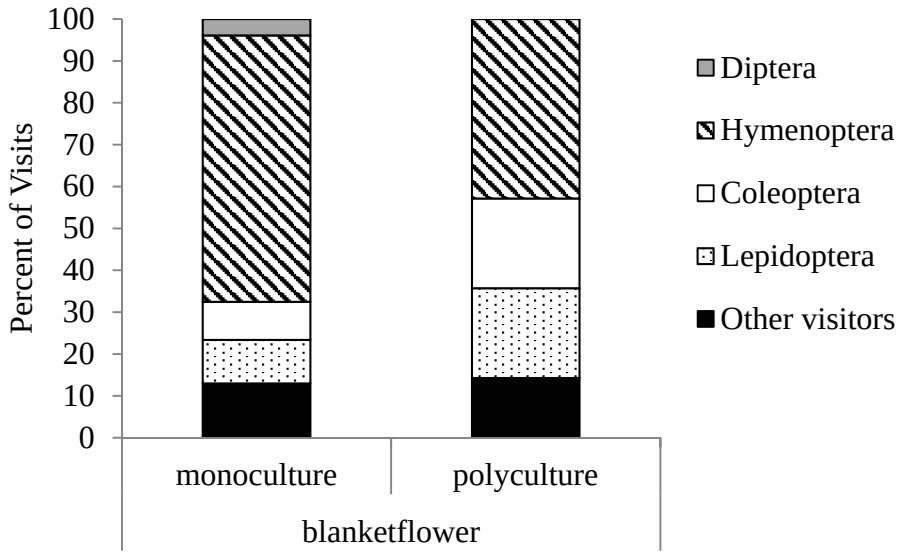

Figure 3C Percent of floral visits from four insect orders to blanketflower grown in monoculture and polyculture, April 23–September 1, 2009, in Stillwater, Oklahoma.

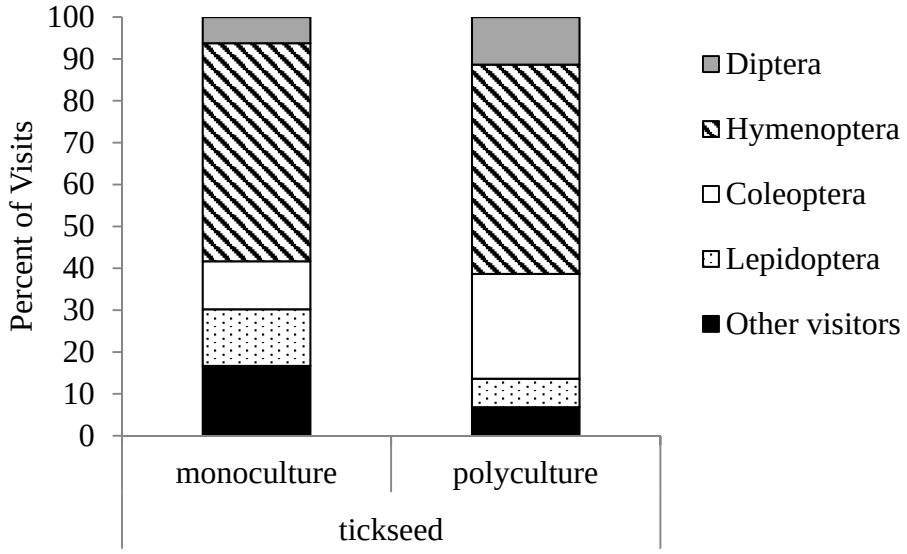

Figure 3D Percent of floral visits from four insect orders to largeflower tickseed grown in monoculture and polyculture, April 23–September 1, 2009, in Stillwater, Oklahoma.

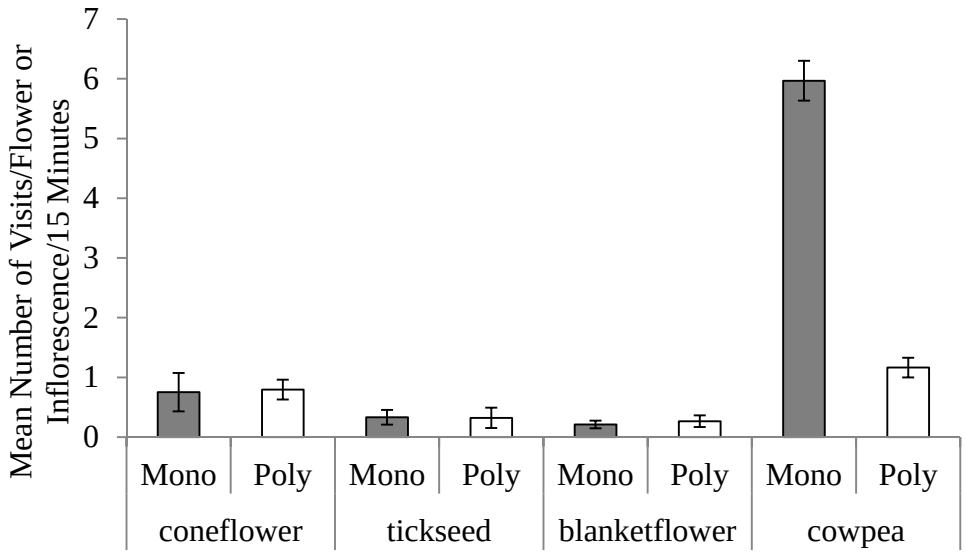

Figure 4 Mean visitation rate (± 1 SE) of floral visiting insects to purple coneflower, largeflower tickseed, blanketflower, and cowpea grown in monoculture and polyculture, April 23–September 1, 2009, in Stillwater, Oklahoma.

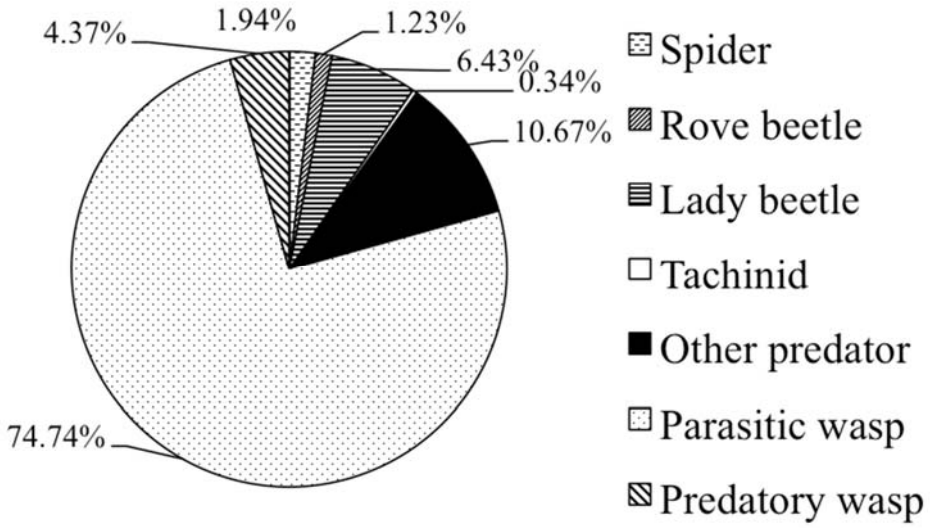

Figure 5A Percent abundance of seven natural enemy groups sampled using yellow sticky cards across all monocultures, April 23–August 25, 2009, in Stillwater, Oklahoma.

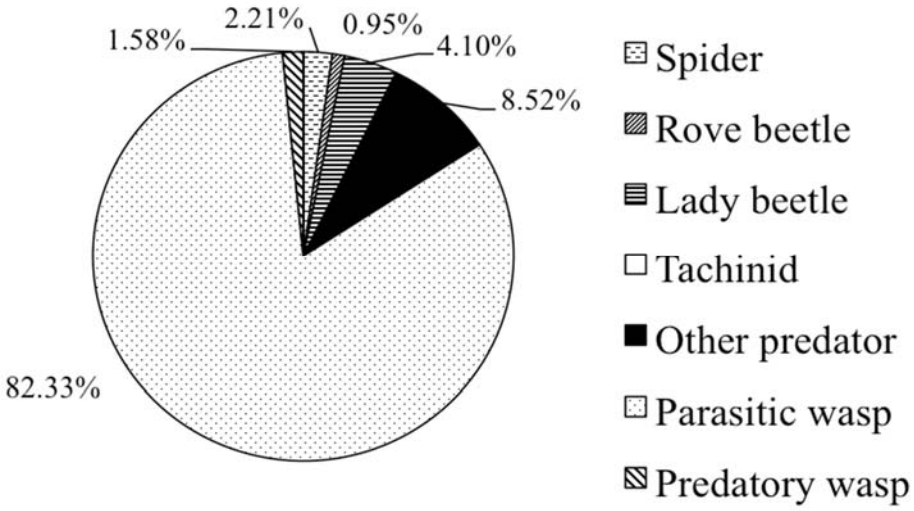

Figure 5B Percent abundance of seven natural enemy groups sampled using yellow sticky cards across Polyculture One, April 23–August 25, 2009, in Stillwater, Oklahoma.

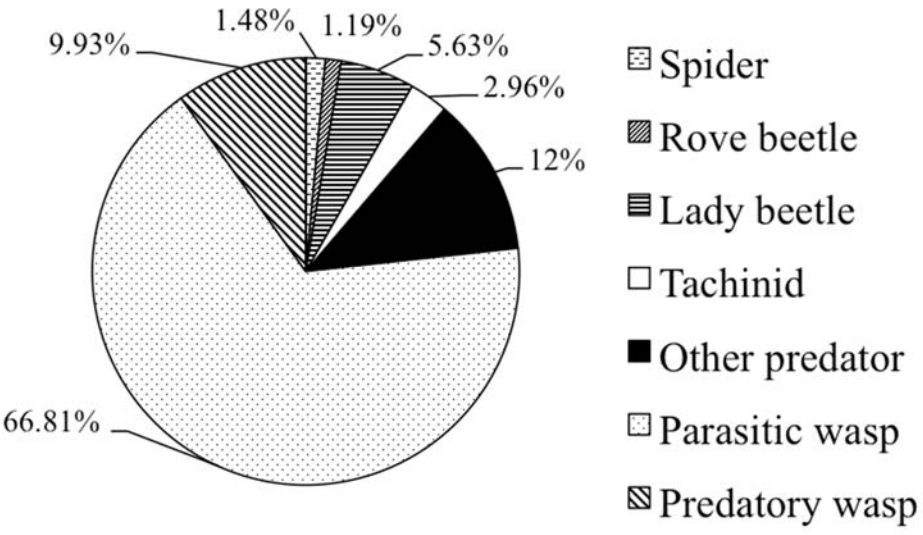

Figure 5C Percent abundance of seven natural enemy groups sampled using yellow sticky cards across Polyculture Two, April 23–August 25, 2009, in Stillwater, Oklahoma.

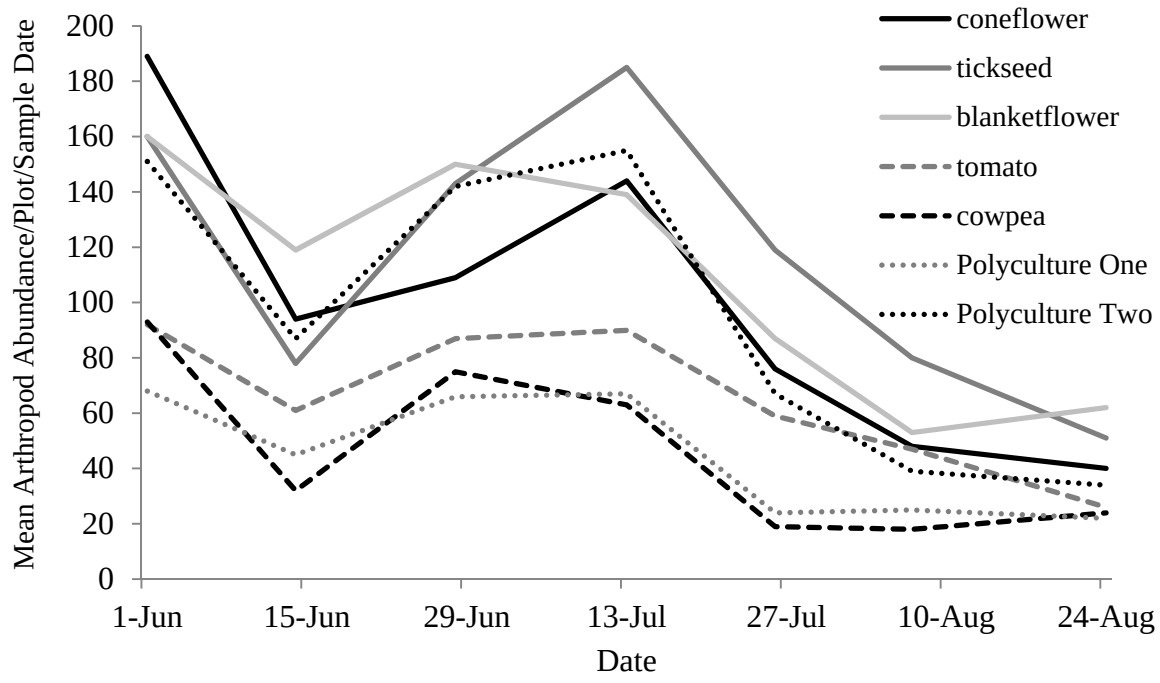

Figure 6 Mean total natural enemy abundance per plot for each plant species grown in monoculture, Polyculture One, and Polyculture Two, June 2–August 25, 2009, in Stillwater, Oklahoma.

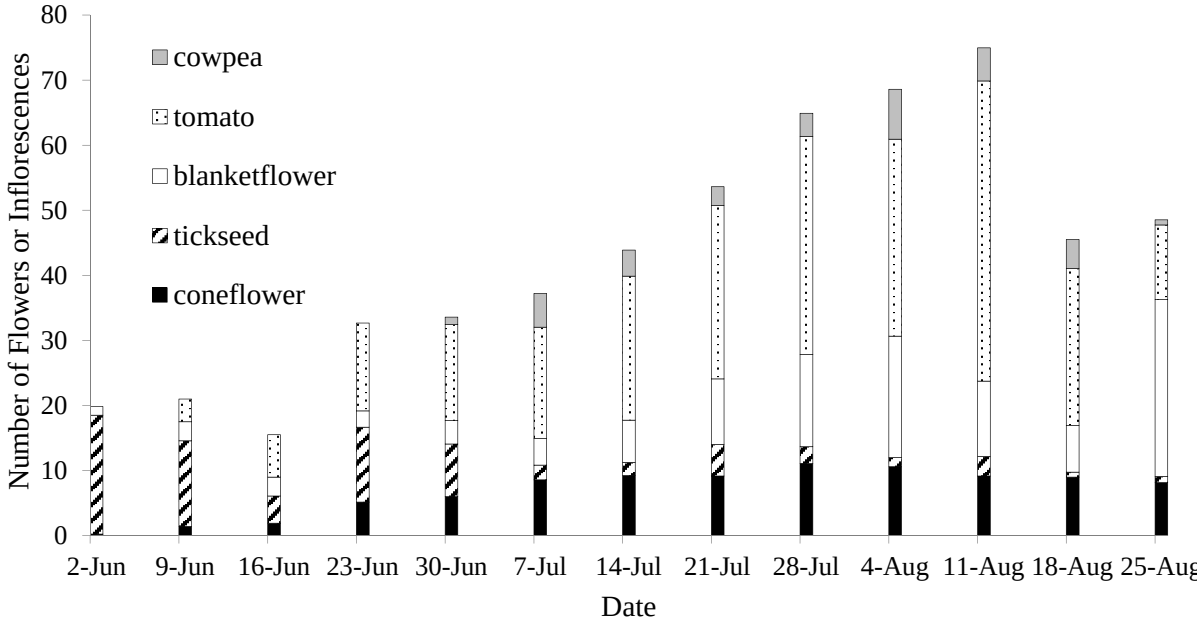

Figure 7 Number of open flowers or inflorescences per plant for purple coneflower, largeflower tickseed, blanketflower, tomato, and cowpea, June 2–August 25, 2009, in Stillwater, Oklahoma, pooled across monocultures and polycultures.

Flowering Phenology and Natural Enemy Abundance

Flowering phenology was similar between monoculture and polyculture plantings for all species; therefore, all treatments were pooled for the illustration of peak flowering times (Fig. 7). Most species (except those species belonging to Asteraceae) exhibited a bimodal peak in flowering; the first peak in flowering occurred from late June to early July and the second from early to mid-August. Both peaks coincided with rainfall events. Two of the ornamental species (purple coneflower and blanketflower) exhibited peak flowering in mid to late August. The other ornamental species, largeflower tickseed, exhibited peak flowering in early June.

The abundance of blanketflower inflorescences was negatively correlated with natural enemy abundance ($r_{Spearman}$ = -0.79, N = 7, P = 0.03). Natural enemy abundance was not correlated with the abundance of flowers or inflorescences of any other plant species (Table 1).

Table 1 Spearman-rank correlation between total natural enemy abundance and flower or inflorescence abundance of five plant species grown in monoculture and polyculture, June 2-August 25, 2009, in Stillwater, Oklahoma; * $P < 0.05$.

Species	Correlation Coefficient
Cowpea	-0.55858
Tomato	-0.42857
Blanketflower	-0.78571*
Coneflower	-0.39286
Tickseed	0.32143

DISCUSSION

Our study demonstrated that natural enemy abundance is higher in polycultures than in monocultures. This finding, in conjunction with similar findings by other researchers (Kloen and Altieri 1990; Andow 1991; Rebek et al. 2005; Ponti et al. 2007), suggests that growing vegetable plants in polyculture with flowering ornamental species is an effective habitat management strategy to increase abundance of natural enemies at small spatial scales, such as those found in home gardens.

There are a number of mechanisms by which polycultures may promote natural enemy abundance. First, the addition of flowering ornamental plants may provide additional pollen and nectar resources and/or may attract alternative prey species, all of which serve as food sources for natural enemies (Landis et al. 2000; Rebek et al. 2005; Isaacs et al. 2009). Second, the ornamental plants may provide a hospitable microclimate and shelter (i.e., refugia) to natural enemies. Refugia are essentially sites where temperature, humidity, light intensity, and other abiotic conditions are at optimal levels for survival of natural enemies. Refuge plants may also harbor alternative prey species for both immature and adult natural enemies (Frank 2010), leading to

increased abundance of natural enemies. Given that we did not detect a significant positive correlation between flowering phenology and natural enemy abundance (see Table 1), the availability of floral resources to natural enemies is likely to be less important than the availability of refugia in determining habitat quality for these arthropods at small spatial scales, such as those found in home gardens. However, future work is needed to determine the mechanism(s) by which polycultures of vegetable and ornamental plants enhance natural enemy abundance.

In addition to supporting higher natural enemy abundance, polycultures have been shown to reduce pest arthropod abundance and improve control of key plant pests compared with monocultures (Kloen and Altieri 1990; Rebek et al. 2006; Ponti et al. 2007). Thus, we expected higher rates of herbivory in monoculture compared to polyculture. However, we failed to detect significant differences between monoculture and polyculture for any plant species. Differences may have been more apparent with other vegetable crops. For example, Ponti et al. (2007) found that broccoli (*Brassica oleracea* L. Italica group) polycultures benefited from reduced herbivore abundance compared to broccoli grown in monocultures.

We predicted that reduced herbivory experienced by plants grown in polyculture would lead to improved health in polyculture plants compared to those in monocultures; however, we found no significant differences in height or photosynthetic activity between plants grown in monoculture and plants grown in polyculture. The lack of differences suggests that no physiological cost exists to vegetables grown in polycultures with ornamental plants at the planting densities and arrangements chosen for this study compared to vegetables grown in monocultures.

Previous work has demonstrated that greater diversity of flowering plants leads to greater diversity of pollinating insects (Potts et al. 2003, 2004). In line with this past work, we found that cowpea grown in polyculture had an additional order of insect floral visitors (Lepidoptera) compared with cowpea grown in monoculture (see Fig. 3A). Higher diversity of pollinators is linked to improved pollinator services and increased plant reproductive success (Albrecht et al. 2012). Future research should investigate whether greater diversity of pollinating insects to cowpeas grown in polyculture results in increased crop yields relative to monoculture plantings.

Our results did not support the hypothesis of higher pollinator visitation rates to plants grown in polyculture vs. monoculture. In fact, we observed the opposite trend for cowpeas. Because large-bodied bees are the primary pollinators of cowpea (Free 1993), this tendency toward an increased visitation rate, mostly from true flies (Diptera), does not necessarily translate to an increase in the number of successful pollinations compared to plants in polyculture, as the pollination efficiency of true flies to cowpea is not known.

Conclusions

Our findings provide evidence that a habitat manipulation strategy in which vegetable and ornamental plants are grown in polyculture has beneficial effects for some crops, including species typically grown in home gardens. Polycultures support a greater abundance of natural enemies. Thus, diversifying plantings to include both vegetable and ornamental species may provide an alternative means to control pest populations, which has important implications for home gardeners. Furthermore, the home gardener may see additional benefits of a diversified garden, including a more diverse pollinator assemblage.

ACKNOWLEDGEMENTS

This project was funded by the Institute for Sustainable Environments at Oklahoma State University (OSU). We thank The Botanic Garden at OSU for providing facilities, as well as Daniel Valdez, Senior Agriculturist at The Botanic Garden at OSU, for assistance in maintaining irrigation to garden plots. We thank Derek Barchenger, Cristina Carballo, and Cameron Goforth for assistance with fieldwork. We also thank Matthew Allen, Lou Anella, Lynn Brandenberger, and Kristen Baum for insightful comments that improved this manuscript.

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CONTRIBUTIONS TO THE FLORA OF CIMARRON COUNTY AND THE BLACK MESA AREA

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Keywords: flora, Cimarron County, Black Mesa, vascular plants, rare plants

ABSTRACT

This paper reports the results of recent collection activities in Cimarron County, including the Black Mesa area, in the state of Oklahoma. A total of 331 taxa in 60 families were collected. Two-hundred and six genera, 279 species and 52 infraspecific taxa were identified. The largest families were the Poaceae with 72 taxa and the Asteraceae with 63. Thirty-six exotic taxa were collected (10.9 % of the flora), including two species new to Oklahoma: *Scorzonera laciniata* and *Ranunculus testiculatus*. Forty-six taxa tracked by the Oklahoma Natural Heritage Inventory were found.

INTRODUCTION

Cimarron County has long been recognized as a botanically significant region in Oklahoma. A total of 95 vascular plants tracked by the Oklahoma Natural Heritage Inventory (ONHI) occur in the county (Oklahoma Natural Heritage Inventory 2013). Included among these is *Asclepias uncialis* Greene, which, prior to 1996, was listed as a likely candidate for federal listing as threatened or endangered by the U.S. Fish and Wildlife Service (United States Department of the Interior 1993). Before this survey, nineteen of the tracked taxa had an ONHI ranking of SH, meaning that reports of occurrences are older than twenty years (Oklahoma Natural Heritage Inventory 2013; NatureServe 2015). The number of taxa in Cimarron County that are rare at the state level is due in part to the presence of Black Mesa, an extension of the Mesa de Maya, which extends for 72 km from east of Raton, New Mexico, though Colorado and into northwestern Cimarron County. The eastern-most extension of the Rocky Mountain foothill vegetation is present in the area; Rogers (1953) found it to be "an excellent example of the intergradation of the flora of the great plains with that of the Rocky Mountain foothills". Our intent for this work was to relocate the rare taxa, update their ONHI ranks, and, hopefully, expand our knowledge of the area's current flora.

The earliest botanical collections from the Black Mesa region were made in 1820 by Edwin James, botanist for Major Stephen Long's expedition to the Rocky Mountains. Eighty-four years later, Per Axel Rydberg, author of *Flora of Colorado* (1906) and *Flora of the Rocky Mountains and Adjacent Plains* (1917), botanized in the area. The first thorough botanical inventory of the Mesa de Maya was completed by Rogers (1953). From 1947 and 1949, he collected along the mesa in Colorado, New Mexico and Oklahoma, as well as from some of the secondary mesas in the area (Rogers 1953). According

https://doi.org/10.22488/okstate.17.100114

to a list published in 1953, Rogers collected 267 taxa from 51 families in Oklahoma, but in a later work (1954) he notes that "approximately five-hundred were found, or could be found". U. T. Waterfall collected at Black Mesa and in Cimarron County within the same time period, adding approximately 30 taxa to the state's flora (Waterfall 1949, 1950a, b). James K. McPherson completed an inventory with the sole focus of Black Mesa in the early 1990s, reporting 236 taxa from 58 families (2003a, b). His collecting activities were confined to the areas of the mesa on the property belonging to the state of Oklahoma (Township 6N, Range 1E, Sections 28–33 and Township 5N, Range 1E, Section 6). Patricia Folley (2003) supplemented the McPherson list with collections made from 1994 through 2003. Folley collected over a wider area than McPherson, surveying the state park around Lake Etling, the roadsides leading to the park and mesa, and some private lands, including Tesequite Canyon (Folley 2003). She found an additional 49 taxa from 25 families. Other botanists have contributed to the knowledge of the Black Mesa/Cimarron County flora over the years, including Delzie Demaree, who worked in the area in the 1930s, George Goodman (from the late 1930s through the early 1970s), John and Connie Taylor (1960s and 1970s), and Larry Magrath in the 1980s (Oklahoma Vascular Plants Database 2015).

STUDY SITE

Cimarron County falls within the High Plains and the Cimarron River Valley geomorphic provinces (Curtis et al. 2008). The High Plains province consists of flat uplands over Tertiary-era Dakota sandstones and is found throughout most of the county (Rogers 1953). The Cimarron River Valley is found in the northeastern part of the county and is distinguished by dissected valleys of Mesozoic-era shale and sandstone. The Black Mesa, the flat, eroded remnant of a Tertiary-era lava flow, is located in this area (Curtis et al. 2008). The highest point in Oklahoma, at 1515 m, is on the mesa. Rolling, low hills and canyons surround the mesa.

Four soil associations occur within Cimarron County. Travessilla-Kim soils are only found in the northeastern corner of the county. They consist of "loam, calcareous, and humus-poor soils on steep slopes" (Carter and Gregory 2008). Dalhart-Vona soils are found primarily in the southern half of the county; these are "very deep loamy soils on gentle slopes" (Carter and Gregory 2008). Sherm-Ulysses type soils dominate the eastern half of the county. These soils are "very deep, silty and clayey, humus-rich soils on gentle slopes" (Carter and Gregory 2008). Conlen-Pastura-Plack soils are the least common soil type in the county; they consist of "loamy and calcareous soils on moderately steep slopes" (Carter and Gregory 2008). Potential vegetation types in Cimarron County include shortgrass high plains, sandsage grassland, piñon pine/juniper mesa, and bottomland forest (Duck and Fletcher 1943; Hoagland 2008).

Cimarron County has a dry climate, falling within Trewartha's steppe or semi-arid type (1968). Average annual precipitation ranges from 38–50 cm, with most falling from May through August. Thunderstorms occur in the spring and summer. Average temperature is 13–14°C. The average high (in July) is 34°C, and the average low (in January) is -7°C. South-southwesterly winds are dominant and relative humidity ranges from 29–84%. Over 70% of days are sunny (Oklahoma Climatological Survey 2015).

METHODS

Plants were collected at 100 sites throughout Cimarron County (Fig. 1; Table 1). Collection sites were chosen based on location information from the Oklahoma Natural Heritage Inventory Database and

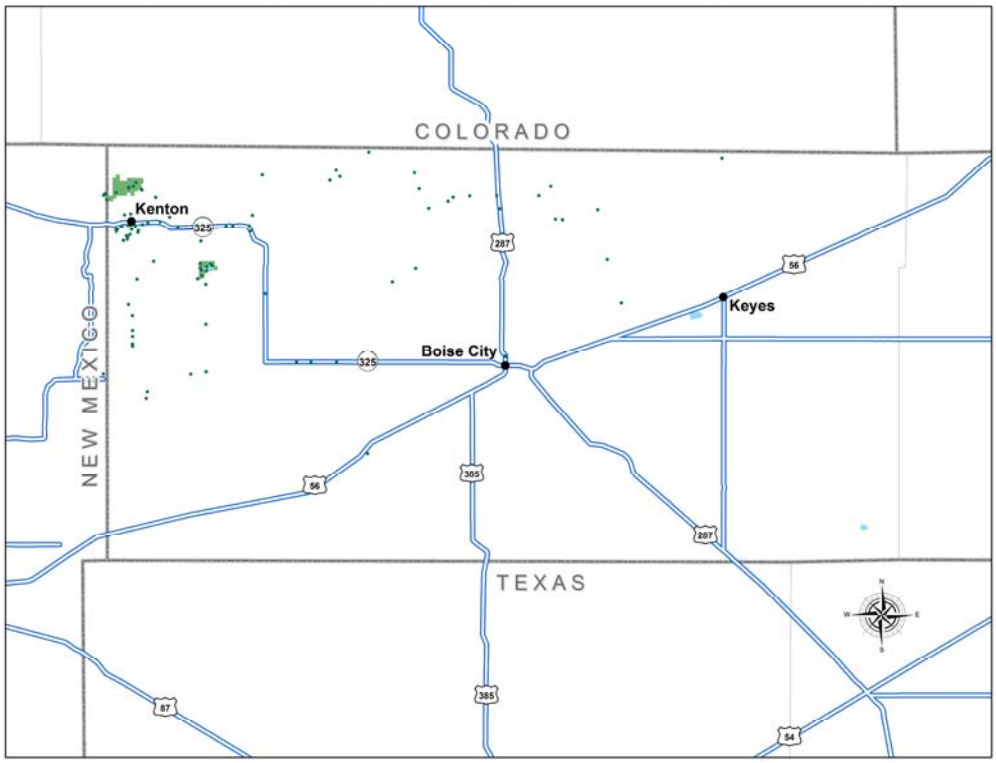

Figure 1 Cimarron County, Oklahoma. Dots indicate collection sites. Shaded areas indicate Black Mesa Nature Preserve lands. Map by Todd Fagin, Oklahoma Biological Survey.

the Oklahoma Vascular Plants Database. Additional collections were also made opportunistically. Coordinates of each site were collected using a Garmin GPSmap 76Cx unit. Sites were located between latitudes N36.98989 and N36.62313 and longitudes W102.67913 and W102.68063. Elevations ranged from 1118 m to 1513 m. Field work began in March of 2013, with subsequent monthly trips until September. An additional trip was made in May of 2014. One example of each taxon encountered was collected and processed according to standard herbarium protocols. Specimens were deposited at the Robert Bebb Herbarium (OKL) at the University of Oklahoma. Manuals used to identify plants included Great Plains Flora Association (1986), Tyrl et al. (2010) and Allred and Ivey (2012); the collections of the Robert Bebb Herbarium were also used to verify identifications. Taxonomy follows the Integrated Taxonomic Information System (2015). Duration and nativity to Oklahoma were determined using the PLANTS Database (USDA-NRSC 2015); if the information from PLANTS was ambiguous, Taylor and Taylor (1991) was consulted. Vegetation classifications were assigned based on Hoagland (2000).

RESULTS AND DISCUSSION

Three-hundred and thirty-one taxa in 60 families were collected in this study (Appendix A). Two-hundred and six genera, 279 species, and 52 infraspecific taxa were identified. Two-hundred thirty-one taxa were perennials; there were 96 annuals and four biennials. Thirty-six taxa were non-native to Oklahoma, including two species new to the state (*Scorzonera laciniata* in the Asteraceae and *Ranunculus testiculatus* in the

Ranunculaceae); non-native taxa accounted for 10.9% of the total flora. The Poaceae had the greatest number of exotic taxa with 11; the Brassicaceae had five. The largest families were the Poaceae with 72 taxa and the Asteraceae with 63. Forty-six taxa tracked by the Oklahoma Natural Heritage inventory were found (Table 2). *Asclepias uncialis*, the former candidate for federal listing, was not located.

Vegetation classes encountered in this study included the *Artemisia filifolia/Sporobolus cryptandrus-Schizachyrium scoparium* shrubland association. It is found on sandy soils and stabilized dunes in the northwestern and central portions of the study site. Associated taxa included *Andropogon gerardii* ssp. *hallii*, *Abronia fragrans,* and *Eriogonum annuum* (Duck and Fletcher 1943; Hoagland 2000).

Two intergrading variations of shortgrass prairie were noted. The *Bouteloua curtipendula-B. gracilis-B. dactyloides* herbaceous association is found on rocky slopes and well-drained soils in the southern part of the study area (Duck and Fletcher 1943; Hoagland 2000). Plants found here included *Muhlenbergia torreyi*, *Ratibida columnifera,* and *Sphaeraclea coccinea*. The *Bouteloua gracilis-Hilaria jamesii* herbaceous association is found in northwestern Cimarron County on slopes and uplands (Hoagland 2000). Plants found in this type included *Cylindropuntia imbricata*, *Melampodium leucanthum*, and *Zinnia grandiflora*.

The *Bouteloua gracilis-Hilaria jamesii* herbaceous association intergrades with the fourth vegetation type, the *Juniperus monosperma* woodland alliance. This alliance includes *the Juniperus monosperma/Bouteloua curtipendula* woodland association and the *Juniperus monosperma-Pinus edulis/Bouteloua curtipendula* woodland association and is found in northwestern Cimarron County. Plants from this type included *Bouteloua gracilis*, *Cercocarpus montanus,* and *Prunus virginiana* (Hoagland 2000).

Herbaceous wetland vegetation was found at only a few sites, including those with seeps, lakes, and intermittently flowing streams and rivers. Plants found in this vegetation type included *Polypogon monspeliensis*, *Populus deltoides*, *Salix exigua*, and *Tamarix chinensis*. Vegetation of disturbed areas includes taxa found around lawns, stock tanks, campgrounds, parking lots, and gravel pits. Plants in this vegetation type included *Conyza canadensis*, *Descurainia sophia*, *Kochia scoparia*, and *Malva neglecta*.

Table 1 Collection sites in Cimarron County

Latitude	Longitude	Township, Range, and Section
36.623130	-102.68063	Sec. 24-T2N-R3E
36.690420	-102.95001	Sec. 33-T3N-R1E
36.698390	-102.9484	Sec. 28-T3N-R1E
36.719380	-102.89576	Sec. 13-T3N-R1E
36.719660	-103.00208	Sec. 18-T3N-R1E
36.722180	-102.877	Sec. 18-T3N-R2E

36.733790	-102.7183	Sec. 15-T3N-R3E
36.733800	-102.76698	Sec. 17-T3N-R3E
36.733920	-102.74949	Sec. 16-T3N-R3E
36.739900	-102.51231	Sec. 10-T3N-R5E
36.741100	-102.51344	Sec. 10-T3N-R5E
36.753940	-102.96656	Sec. 4-T3N-R1E
36.756350	-102.96655	Sec. 4-T3N-R1E
36.765380	-102.96653	Sec. 32-T4N-R1E
36.772640	-102.96652	Sec. 32-T4N-R1E
36.780300	-102.87736	Sec. 30-T4N-R2E
36.790710	-102.96668	Sec. 29-T4N-R1E
36.804340	-102.97145	Sec. 20-T4N-R1E
36.806220	-102.37202	Sec. 13-T4N-R6E
36.817480	-102.80509	Sec. 13-T4N-R2E
36.829480	-102.87738	Sec. 7-T4N-R2E
36.832480	-102.65052	Sec. 8-T4N-R4E
36.835430	-102.96116	Sec. 9-T4N-R1E
36.836080	-102.88737	Sec. 6-T4N-R2E
36.840080	-102.88219	Sec. 6-T4N-R2E
36.845780	-102.87656	Sec. 5-T4N-R2E
36.846420	-102.88263	Sec. 6-T4N-R2E
36.848380	-102.62216	Sec. 3-T4N-R4E
36.849240	-102.88435	Sec. 6-T4N-R2E
36.850360	-102.87642	Sec. 31-T5N-R2E
36.850360	-102.87642	Sec. 31-T5N-R2E

36.851790	-102.86967	Sec. 32-T5N-R2E
36.853670	-102.87138	Sec. 32-T5N-R2E
36.854090	-102.88454	Sec. 31-T5N-R2E
36.856980	-102.94078	Sec. 34-T5N-R1E
36.859200	-102.38917	Sec. 34-T5N-R6E
36.881280	-102.88344	Sec. 19-T5N-R2E
36.882050	-102.97772	Sec. 20-T5N-R1E
36.883330	-102.97295	Sec. 20-T5N-R1E
36.886460	-102.97238	Sec. 20-T5N-R1E
36.887550	-102.97424	Sec. 20-T5N-R1E
36.889260	-102.96963	Sec. 20-T5N-R1E
36.891690	-102.96015	Sec. 21-T5N-R1E
36.892660	-102.98643	Sec. 19-T5N-R1E
36.893120	-102.82283	Sec. 22-T5N-R2E
36.893790	-102.95947	Sec. 16-T5N-R1E
36.895370	-102.9677	Sec. 17-T5N-R1E
36.895760	-102.98476	Sec. 18-T5N-R1E
36.895960	-102.98691	Sec. 18-T5N-R1E
36.897520	-102.91134	Sec. 13-T5N-R1E
36.897950	-102.96324	Sec. 16-T5N-R1E
36.898540	-102.98034	Sec. 17-T5N-R1E
36.899190	-102.97931	Sec. 17-T5N-R1E
36.899370	-102.8527	Sec. 16-T5N-R2E
36.899560	-102.84465	Sec. 16-T5N-R2E
36.899830	-102.82454	Sec. 15-T5N-R2E

36.900190	-102.96891	Sec. 17-T5N-R1E
36.900640	-102.97209	Sec. 17-T5N-R1E
36.901170	-102.96404	Sec. 16-T5N-R1E
36.901410	-102.95449	Sec. 16-T5N-R1E
36.903700	-102.94789	Sec. 16-T5N-R1E
36.904800	-102.93303	Sec. 15-T5N-R1E
36.907530	-102.44369	Sec. 7-T5N-R6E
36.908160	-102.45214	Sec. 7-T5N-R6E
36.910440	-102.92143	Sec. 11-T5N-R1E
36.912730	-102.82081	Sec. 11-T5N-R2E
36.913450	-102.97624	Sec. 8-T5N-R1E
36.914270	-102.96875	Sec. 8-T5N-R1E
36.919640	-102.4009	Sec. 10-T5N-R6E
36.920710	-102.51988	Sec. 9-T5N-R5E
36.921370	-102.60638	Sec. 10-T5N-R4E
36.921370	-102.60638	Sec. 10-T5N-R4E
36.929980	-102.58279	Sec. 1-T5N-R4E
36.931820	-102.99784	Sec. 6-T5N-R1E
36.934710	-102.9383	Sec. 3-T5N-R1E
36.934710	-102.93839	Sec. 3-T5N-R1E
36.934850	-102.57666	Sec. 1-T5N-R4E
36.936330	-102.55646	Sec. 21-T6N-R5E
36.936870	-102.52358	Sec. 33-T6N-R5E
36.936880	-102.47233	Sec. 36-T6N-R5E
36.937150	-103.0018	Sec. 6-T5N-R1E

36.938120	-103.00098	Sec. 31-T6N-R1E
36.938800	-103.00023	Sec. 31-T6N-R1E
36.939080	-102.99954	Sec. 31-T6N-R1E
36.940380	-102.98649	Sec. 31-T6N-R1E
36.943380	-102.95534	Sec. 33-T6N-R1E
36.944330	-102.95544	Sec. 33-T6N-R1E
36.945420	-102.618	Sec. 34-T6N-R4E
36.945760	-102.97118	Sec.32-T6N-R1E
36.947060	-102.97128	Sec. 32-T6N-R1E
36.947930	-102.96566	Sec. 33-T6N-R1E
36.948080	-102.45784	Sec. 31-T6N-R6E
36.952680	-102.96242	Sec. 28-T6N-R1E
36.955610	-102.72656	Sec. 27-T6N-R3E
36.960120	-102.71428	Sec. 27-T6N-R3E
36.962150	-102.80867	Sec. 26-T6N-R2E
36.964600	-102.62363	Sec. 28-T6N-R4E
36.967830	-102.71885	Sec. 22-T6N-R3E
36.982940	-102.24962	Sec. 13-T6N-R7E
36.989890	-102.67913	Sec. 13-T6N-R3E

Table 2 Taxa located during this study that are tracked by the Oklahoma Natural Heritage Inventory (Oklahoma Natural Heritage Inventory 2013; NatureServe Explorer 2015). Status ranks are on a 1–5 scale, with a 1 indicating the taxa is critically imperiled. G ranks are at the global level and S ranks are at the subnational or state level. Infraspecific taxa are assigned a T rank. A taxon with NR indicates that it has not been ranked at the global level (NatureServe 2015). Highlighted taxa were re-ranked as a result of this survey.

Family	Taxon	Ranking
Amaranthaceae	*Krascheninnikovia lanata* (Pursh) A. Meeuse & A. Smit	S1G5
Apocynaceae	*Asclepias macrotis* Torr.	S1G4
Asteraceae	*Ambrosia confertiflora* DC.	S1G5
Asteraceae	*Artemisia carruthii* Alph. Wood ex Carruth.	S2G4?
Asteraceae	*Brickellia brachyphylla* (A. Gray) A. Gray	S1G5
Asteraceae	*Brickellia californica* (Torr. & A. Gray) A. Gray	S1G5
Asteraceae	*Brickellia eupatorioides* (L.) Shinners var.	S1G5T5
Asteraceae	*Ericameria nauseosa* (Pall. ex Pursh) G.L. Nesom &	S1G5T5
Asteraceae	*Picradeniopsis woodhousei* (A. Gray) Rydb.	S2G4G5
Asteraceae	*Solidago velutina* DC. ssp. *sparsiflora* (A. Gray) Semple	S1G5?TNR
Boraginaceae	*Cryptantha cinerea* (Greene) Cronquist var.	S2G5T5?
Boraginaceae	*Cryptantha thyrsiflora* (Greene) Payson	S2G4
Cactaceae	*Cylindropuntia imbricata* (Haw.) F.M. Knuth	S2G5
Cactaceae	*Echinocereus reichenbachii* (Terscheck ex Walp.) J.N.	S3G5
Cactaceae	*Echinocereus viridiflorus* Engelm.	S1G5
Cactaceae	*Escobaria vivipara* (Nutt.) Buxb.	S1G5
Cactaceae	*Opuntia polyacantha* Haw. var. *polyacantha*	S2G5T5
Convolvulaceae	*Cuscuta umbellata* Kunth	S1G5
Crossomataceae	*Glossopetalon spinescens* A. Gray var.	S1G5TNR
Cupressaceae	*Juniperus monosperma* (Engelm.) Sarg.	S2G4G5
Fabaceae	*Dalea formosa* Torr.	S2G5

Fabaceae	*Dalea jamesii* (Torr.) Torr. & A. Gray	S1G5
Fabaceae	*Desmanthus cooleyi* (Eaton) Trel.	S2G5
Fabaceae	*Hoffmannseggia drepanocarpa* A. Gray	S2G5
Fabaceae	*Lupinus plattensis* S. Watson	S1G4
Grossulariaceae	*Ribes cereum* Douglas	S1G5
Malvaceae	*Sphaeralcea angustifolia* (Cav.) G. Don	S2G5
Nyctaginaceae	*Abronia fragrans* Nutt. ex Hook.	S2G5
Papaveraceae	*Argemone squarrosa* Greene	S1G4
Pinaceae	*Pinus edulis* Engelm.	S1G5
Plantaginaceae	*Penstemon fendleri* Torr. & A. Gray	S1G5T4?
Poaceae	*Aristida arizonica* Vasey	S1G4
Poaceae	*Bouteloua barbata* Lag.	S1G5
Poaceae	*Bouteloua eriopoda* (Torr.) Torr.	S1G5
Poaceae	*Hesperostipa neomexicana* (Thurb.) Barkworth	S1G4G5
Poaceae	*Hilaria jamesii* (Torr.) Benth.	S1G5
Poaceae	*Muhlenbergia phleoides* (Kunth) Columbus	S1G5
Poaceae	*Muhlenbergia porteri* Scribn. ex Beal	S1G5
Poaceae	*Muhlenbergia torreyi* (Kunth) Hitchc. ex Bush	S1G4
Poaceae	*Piptatherum micranthum* (Trin. & Rupr.) Barkworth	S1G5
Polygonaceae	*Eriogonum jamesii* Benth.	S1G5
Polygonaceae	*Eriogonum lachnogynum* Torr. ex Benth.	S1G4?
Polygonaceae	*Eriogonum tenellum* Torr.	S1G5
Rosaceae	*Cercocarpus montanus* Raf.	S1G5
Rosaceae	*Rubus deliciosus* Torr.	S1G4?
Selaginellaceae	*Selaginella underwoodii* Hieron.	S1G5?

DISCUSSION

One-hundred sixty taxa from 46 families reported in the Rogers, McPherson, and Folley studies were not found (Appendix B), and only 46 of the 95 taxa tracked by the Oklahoma Natural Heritage Inventory were located. One explanation for this difference is land access. For instance, we were not able to collect in Tesequite Canyon, which is known to have populations of tracked taxa (Oklahoma Natural Heritage Inventory 2015), as was done in the Folley study. We were uncomfortable botanizing along some of the public roads, as well. Another explanation could be that vegetation changes have occurred in the area. Vegetation analysis by Graham et al. (unpubl. data) indicates a decrease in the amount of grassland/herbaceous vegetation and an increase in forest/shrubland since 1992. This is most probably due to the increased amount of cholla (*Cylindropuntia imbricata*) in the area.

The most likely explanation for our results, however, is drought. Cimarron County is considered to be the epicenter of the exceptional drought experienced by the High Plains regions of northern Texas, southwestern Kansas, northeastern New Mexico, southeastern Colorado, and the northwestern Oklahoma panhandle (Lindsey 2008; South Central Climate Science Center 2013). Throughout the survey period, western Cimarron County experienced exceptional, extreme, or extreme/severe drought (National Oceanic and Atmospheric Administration et al. 2015). Rogers (1953) stated that the "severe drouth of the 1930s had a disturbing effect on the vegetation", but noted a "great recovery" in the following decade. Although the National Weather Service predicts that the drought status for the area will likely be removed, another "great recovery" is unlikely (U. S. Geological Survey 2014). The area could be as much as 5°C hotter by the end of the century, and decreases in precipitation, runoff, and amounts of soil water storage are also likely (U. S. Geological Survey 2014).

ACKNOWLEDGEMENTS

This work was supported by a grant from the Department of Interior, U. S. Fish and Wildlife Service. The authors wish to thank Todd Fagin (Oklahoma Biological Survey/Department of Geography and Environmental Sustainability, University of Oklahoma) for assistance with map preparation.

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Waterfall, U.T. 1950a. Some additions to the Oklahoma flora. *Rhodora* 52:19–24, 35–41.

Waterfall, U.T. 1950b. Some results of a third summer's botanizing in Oklahoma. *Rhodora* 52:165–175.

APPENDIX A

List of Plant Taxa in Cimarron County and Black Mesa, Oklahoma

Taxa list with duration, vegetation type, and nativity. A=annual, B=biennial, P=perennial; AFSA=*Artemisia filifolia* shrubland association, BCBGBD=*Bouteloua curtipendula-Bouteloua gracilis-Bouteloua dactyloides* herbaceous association, BGHJ=*Bouteloua gracilis-Hilaria jamesii* herbaceous association, DAOF=Disturbed area/old field vegetation, HWV=herbaceous wetland vegetation, JMWA=*Juniperus monosperma* woodland alliance. An asterisk (*) indicates a taxon that is non-native to the United States. A dagger (†) indicates a tracked taxon. Taxonomy follows the Integrated Taxonomic Information System (2015). Duration and nativity to Oklahoma were determined using the PLANTS Database (USDA-NRSC 2015); if the information from PLANTS was ambiguous, Taylor and Taylor (1991) was consulted. Vegetation classifications were based on Hoagland (2000).

Alismataceae
Alisma subcordatum Raf., P, HWV

Amaranthaceae
Amaranthus palmeri S. Watson, A, AFSA
Amaranthus tuberculatus (Moq.) J.D. Sauer, A, AFSA
Atriplex canescens (Pursh) Nutt., P, BGHJ
**Chenopodium album* L., A, BGHJ
Chenopodium berlandieri Moq., A, DAOF
Chenopodium incanum (S. Watson) A. Heller, A, BGHJ
Chenopodium leptophyllum (Moq.) Nutt. ex S. Watson, A , DAOF
Chenopodium pratericola Rydb., A, BGHJ
Chenopodium simplex (Torr.) Raf., A, BCBGBD
Chenopodium standleyanum Aellen, A, JMWA
Froelichia floridana (Nutt.) Moq., A, JMWA
**Kochia scoparia* ssp. *scoparia* (L.) Schrad., A, DAOF
†*Krascheninnikovia lanata* (Pursh) A. Meeuse & A. Smit, P, JMWA
Monolepis nuttalliana (Schult.) Green, A, DAOF
**Salsola tragus* L., A, BCBGBD
Tidestromia lanuginosa (Nutt.) Standl., A, AFSA

Amaryllidaceae
Allium drummondii Regel, P, BGHJ

Anacardiaceae
Rhus aromatica Aiton var. *pilosissima* (Engl.) Shinners, P, BGHJ
Toxicodendron rydbergii (Small ex Rydb.) Greene, P, JMWA

Apiaceae
Cymopterus montanus Nutt. ex Torr. & A. Gray, P, JMWA

Apocynaceae
Apocynum androsaemifolium L., P, JMWA
Asclepias asperula (Decne.) Woodson ssp. *capricornu* (Woodson) Woodson, P, JMWA
Asclepias engelmanniana Woodson, P, AFSA
Asclepias latifolia (Torr.) Raf., P, AFSA
†*Asclepias macrotis* Torr., P, JMWA
Asclepias subverticillata (A. Gray) Vail, P, AFSA
Asclepias viridiflora Raf., P, BCBGBD

Asparagaceae
**Asparagus officinalis* L., P, BCBGBD
Yucca glauca Nutt., P, AFSA

Asteraceae
†*Ambrosia confertiflora* DC., P, AFSA
Ambrosia grayi (A. Nelson) Shinners, P, DAOF
Ambrosia psilostachya DC., P, DAOF
Ambrosia trifida L., A, BGHJ
Amphiachyris dracunculoides (DC.) Nutt., A, AFSA
†*Artemisia carruthii* Alph. Wood ex Carruth., P, BCBGBD
Artemisia filifolia Torr., P, AFSA
Artemisia ludoviciana Nutt., P, BGHJ
Baccharis salicina Torr. & A. Gray, P, HWV
Berlandiera lyrata Benth., P, AFSA
†*Brickellia brachyphylla* (A. Gray) A. Gray, P, BGHJ
†*Brickellia californica* (Torr. & A. Gray) A. Gray, P, BGHJ
†*Brickellia eupatorioides* (L.) Shinners var. *chlorolepis* (Woot. & Standl.) B.L. Turner, P, BGHJ
Cirsium ochrocentrum A. Gray ssp. *ochrocentrum*, P, BGHJ
Cirsium undulatum (Nutt.) Spreng., P, BGHJ
Conyza canadensis (L.) Cronquist, A, DAOF
Diaperia prolifera (Nutt. ex DC.) Nutt., A, BGHJ
Dyssodia papposa (Vent.) Hitchc., A, JMWA
Engelmannia peristenia (Raf.) Goodman & C.A. Lawson, P, BGHJ
†*Ericameria nauseosa* (Pall. ex Pursh) G.L. Nesom & Baird var. *graveolens* (Nutt.) Reveal & Schuyler, P, JMWA
Erigeron bellidiastrum Nutt., AFSA, A
Erigeron flagellaris A. Gray, B, AFSA
Gaillardia pinnatifida Torr., P, BGHJ
Gaillardia pulchella Foug., A, BGHJ
Grindelia squarrosa (Pursh) Dunal, P, BGHJ
Gutierrezia sarothrae (Pursh) Britton & Rusby, P, BGHJ
Helianthus annuus L., A, BGHJ
Helianthus ciliaris DC., P, BCBGBD
Helianthus petiolaris Nutt., A, DAOF
Heterotheca stenophylla (Gray) Shinners var. *angustifolia* (Rydb.) Semple, P, JMWA
Heterotheca subaxillaris (Lam.) Britton & Rusby spp. *latifolia* (Buckley) Semple, A, BGHJ

Heterotheca villosa (Pursh) Shinners var. *villosa*, P, JMWA
Hymenopappus flavescens A. Gray, B, AFSA
Hymenopappus tenuifolius Pursh, B, BGHJ
**Lactuca serriola* L., A, DAOF
Liatris punctata Hook. var. *punctata*, P, AFSA
Lygodesmia juncea (Pursh) D. Don ex Hook., P, JMWA
Machaeranthera tanacetifolia (Kunth) Nees, A, JMWA
Melampodium leucanthum Torr. & A. Gray, P, BGHJ
Packera plattensis (Nutt.) W.A. Weber & A. Löve, P, BGHJ
Palafoxia sphacelata (Nutt. ex Torr.) Cory, A, BCBGBD
†*Picradeniopsis woodhousei* (A. Gray) Rydb., P, BGHJ
Pseudognaphalium canescens (DC.) W.A. Weber ssp. *canescens*, B, BGHJ
Ratibida columnifera (Nutt.) Woot. & Standl., P, BCBGBD
Ratibida tagetes (James) Barnhart, P, DAOF
**Scorzonera laciniata* L., P, DAOF
Senecio flaccidus Less. var. *flaccidus*, P, BGHJ
Senecio riddellii Torr. & A. Gray, P, JMWA
Solidago gigantea Aiton, P, DAOF
†*Solidago velutina* DC. ssp. *sparsiflora* (A. Gray) Semple, P, BGHJ
Symphyotrichum subulatum (Michx.) G.L. Nesom, A, HWV
**Taraxacum officinale* F.H. Wigg., P, DAOF
Tetraneuris acaulis (Pursh) Greene var. *acaulis*, P, JMWA
Tetraneuris scaposa (DC.) Greene var. *scaposa*, P, BGHJ
Thelesperma ambiguum A. Gray, P, AFSA
Thelesperma filifolium (Hook.) A. Gray, P, BGHJ
Thelesperma megapotamicum (Spreng.) Kuntze, P, BGHJ
Townsendia exscapa (Richardson) Porter, P, BGHJ
**Tragopogon dubius* Scop., A, JMWA
Vernonia marginata (Torr.) Raf., P, JMWA
Xanthisma spinulosum (Pursh) D.R. Morgan & R.L. Hartm. var. *spinulosum*, P, BGHJ
Xanthium strumarium L., A, HWV
Zinnia grandiflora Nutt., P, BGHJ

Boraginaceae

†*Cryptantha cinerea* (Greene) Cronquist var. *jamesii* (Torr.) Cronquist, P, AFSA,
Cryptantha minima Rydb., A, AFSA
Cryptantha thyrsiflora (Greene) Payson, P, BGHJ
Lappula occidentalis (S. Watson) Greene var. *cupulata* (A. Gray) Higgins, A, DAOF
Lappula occidentalis (S. Watson) Greene var. *occidentalis*, A, DAOF
Lithospermum incisum Lehm., P, BGHJ
Onosmodium bejariense DC. ex A. DC. var. *occidentale* (Mack.) B.L. Turner, P, JMWA

Brassicaceae

**Camelina microcarpa* DC., A, BCBGBD
Descurainia pinnata (Walter) Britton ssp. *brachycarpa* (Richardson) Detling, A, JMWA
**Descurainia sophia* (L.) Webb ex Prantl, A, DAOF
Erysimum asperum (Nutt.) DC., P, BGHJ

Erysimum capitatum (Douglas ex Hook.) Greene, P, BGHJ
**Erysimum repandum* L., A, BGHJ
**Lepidium densiflorum* Schrad., A, DAOF
Physaria ovalifolia (Rydb.) O'Kane & Al-Shehbaz ssp. *ovalifolia*, P, JMWA
Rorippa sinuata (Nutt.) Hitchc., P, HWV
**Sisymbrium altissimum* L., A, BGHJ

Cactaceae
†*Cylindropuntia imbricata* (Haw.) F.M. Knuth, P, BGHJ
†*Echinocereus reichenbachii* (Terscheck ex Walp.) J.N. Haage, P, AFSA
†*Echinocereus viridiflorus* Engelm., P, JMWA
†*Escobaria vivipara* (Nutt.) Buxb., P, JMWA
Opuntia humifusa (Raf.) Raf. var. *humifusa*, P, BGHJ
Opuntia macrorhiza Engelm., P, JMWA
Opuntia phaeacantha Engelm., P, BGHJ,
†*Opuntia polyacantha* Haw.var. *polyacantha*, P, JMWA

Cannabaceae
Celtis reticulata Torr., P, BGHJ

Caryophyllaceae
Paronychia jamesii Torr. & A. Gray, P, BGHJ
Paronychia sessiliflora Nutt., P, BGHJ

Cleomaceae
Polanisia dodecandra (L.) DC., A, BGHJ

Commelinaceae
Commelina erecta L., P, JMWA
Tradescantia occidentalis (Britton) Smyth var. *occidentalis*, P, BGHJ

Convolvulaceae
**Convolvulus arvensis* L., BGHJ, P
Convolvulus equitans Benth., BGHJ, P
†*Cuscuta umbellata* Kunth, A, DAOF
Evolvulus nuttallianus Schult., P, BGHJ
Ipomoea leptophylla Torr., P, BGHJ

Crossomataceae
†*Glossopetalon spinescens* A. Gray var. *planitierum* (Ensign) Yatsk., P, JMWA,

Cucurbitaceae
Cucurbita foetidissima Kunth, P, BGHJ
Cyclanthera dissecta (Torr. & A. Gray) Arn., A, JMWA

Cupressaceae
†*Juniperus monosperma* (Engelm.) Sarg., P, JMWA

Cyperaceae
Carex gravida L.H. Bailey, P, HWV
Carex muehlenbergii Schkuhr ex Willd., P, HWV
Schoenoplectus acutus (Muhl. ex Bigelow) Á. Löve & D. Löve var. *acutus*, P, HWV
Schoenoplectus pungens (Vahl) Palla var. *pungens*, P, HWV

Euphorbiaceae
Croton texensis (Klotzsch) Müll. Arg., A, BGHJ
Ditaxis mercurialina (Nutt.) J.M. Coult., P, JMWA
Euphorbia dentata Michx., A, AFSA
Euphorbia exstipulata Engelm., A, BGHJ
Euphorbia fendleri Torr. & A. Gray, P, JMWA
Euphorbia glyptosperma Engelm., A, AFSA
Euphorbia lata Engelm., P, BGHJ
Euphorbia marginata Pursh, A, BCBGBD
Euphorbia missurica Raf., A, BCBGBD
Euphorbia serpyllifolia Pers. var. *serpyllifolia*, A, BCBGBD
Tragia ramosa Torr., P, JMWA

Fabaceae
Amorpha canescens Pursh, P, JMWA
Astragalus missouriensis Nutt., P, BGHJ
Astragalus mollissimus Torr., P, BGHJ
Dalea aurea Nutt. ex Fraser, P, BGHJ
Dalea candida Michx. ex. Willd var. *oligophylla* (Torr.) Shinners, P, JMWA
Dalea enneandra Nutt. ex Fraser, P, AFSA
†*Dalea formosa* Torr., P, JMWA
†*Dalea jamesii* (Torr.) Torr. & A. Gray, P, BGHJ
Dalea lanata Spreng., P, BGHJ
Dalea tenuifolia (A. Gray) Shinners, P, BGHJ
Dalea villosa (Nutt.) Spreng., P, DAOF
†*Desmanthus cooleyi* (Eaton) Trel., P, BGHJ
Glycyrrhiza lepidota Pursh, P, BGHJ
†*Hoffmannseggia drepanocarpa* A. Gray, P, BGHJ
Hoffmannseggia glauca (Ortega) Eifert, P, BCBGBD
†*Lupinus plattensis* S. Watson, P, AFSA
**Medicago sativa* L., P, BGHJ
**Melilotus officinalis* (L.) Lam., A, DAOF
Mimosa borealis A. Gray, P, BGHJ
Oxytropis lambertii Pursh, P, AFSA
Pediomelum cuspidatum (Pursh) Rydb., P, BGHJ
Prosopis glandulosa Torr. var. *glandulosa*, P, BGHJ
Psoralidium tenuiflorum (Pursh) Rydb., P, BGHJ
Robinia pseudoacacia L., P, DAOF
Sophora nuttalliana B.L. Turner, P, BGHJ

Fagaceae
Quercus mohriana Buckley ex Rydb., P, JMWA

Geraniaceae
**Erodium cicutarium* (L.) L'Hér. ex Aiton, A, DAOF

Grossulariaceae
Ribes aureum Pursh var. *villosum* DC., P, BCBGBD
†*Ribes cereum* Douglas, P, JMWA

Juncaceae
Juncus interior Wiegand, P, HWV
Juncus torreyi Coville, P, HWV

Krameriaceae
Krameria lanceolata Torr., P, BGHJ

Lamiaceae
Hedeoma drummondii Benth., P, BGHJ
**Marrubium vulgare* L., P, BGHJ
Monarda pectinata Nutt., A, AFSA
Salvia reflexa Hornem., A, JMWA
Teucrium laciniatum Torr., P, JMWA

Linaceae
Linum pratense (Norton) Small, A, BGHJ
Linum rigidum Pursh var. *rigidum*, A, BCBGBD

Loasaceae
Mentzelia multiflora (Nutt.) A. Gray, A, AFSA
Mentzelia nuda (Pursh) Torr. & A. Gray, P, AFSA
Mentzelia oligosperma Nutt. ex Sims, P, BGHJ

Malvaceae
Callirhoe involucrata (Torr. & A. Gray) A. Gray, P, BCBGBD
**Malva neglecta* Wallr., A, DAOF
†*Sphaeralcea angustifolia* (Cav.) G. Don, P, AFSA
Sphaeralcea coccinea (Nutt.) Rydb., P, BCBGBD

Martyniaceae
Proboscidea louisianica (Mill.) Thell., ssp. *louisianica*, A, AFSA

Moraceae
**Morus alba* L., P, JMWA

Nyctaginaceae
†*Abronia fragrans* Nutt. ex Hook., P, AFSA

Mirabilis albida (Walter) Heimerl, P, JMWA
Mirabilis linearis (Pursh) Heimerl var. *subhispida* (Heimerl) Spellenb., P, JMWA
Mirabilis nyctaginea (Michx.) MacMill., P, JMWA

Oleaceae
Forestiera pubescens Nutt., P, BGHJ

Onagraceae
Oenothera cespitosa Nutt., P, JMWA
Oenothera cinerea (Wooton & Standl.) W.L. Wagner & Hoch ssp. *cinerea*, P, BCBGBD
Oenothera curtiflora W.L. Wagner & Hoch, A, DAOF
Oenothera hartwegii Benth. ssp. *pubescens* (A. Gray) W.L. Wagner & Hoch, P, BGHJ
Oenothera serrulata Nutt., P, BCBGBD
Oenothera suffrutescens (Ser.) W.L. Wagner & Hoch, P, BGHJ
Oenothera triloba Nutt., P, BGHJ

Orobanchaceae
Orobanche ludoviciana Nutt. ssp. *multiflora* (Nutt.) T.S. Collins ex H.L. White & W.C. Holmes, A, BGHJ

Papaveraceae
†*Argemone squarrosa* Greene, P, BGHJ
Corydalis aurea Willd. ssp. *occidentalis* (Engelm. ex A. Gray) G.B. Ownbey, A, BCBGBD

Pinaceae
†*Pinus edulis* Engelm., P, JMWA

Plantaginaceae
Penstemon albidus Nutt., P, BGHJ
Penstemon ambiguus Torr., P, BGHJ
†*Penstemon fendleri* Torr. & A. Gray, P, AFSA
Plantago patagonica Jacq., A, BGHJ
Veronica anagallis-aquatica L., P, HWV

Poaceae
**Aegilops cylindrica* Host, A, DAOF
Andropogon gerardii Vitman ssp. *hallii* (Hack.) Wipff, P, AFSA
Andropogon gerardii Vitman ssp. *gerardii*, P, BCBGBD
Aristida adscensionis L., A, BGHJ
†*Aristida arizonica* Vasey, P, BGHJ
Aristida havardii Vasey, P, BCBGBD
Aristida oligantha Michx., A, AFSA
Aristida purpurascens Poir., P, BCBGBD
Aristida purpurea Nutt. var. *purpurea*, P, BGHJ
Bothriochloa barbinodis (Lag.) Herter, P, BGHJ
**Bothriochloa ischaemum* (L.) Keng, P, AFSA
Bothriochloa laguroides (DC.) Herter, P, BGHJ
†*Bouteloua barbata* Lag., A, JMWA

Bouteloua curtipendula (Michx.) Torr., P, AFSA
Bouteloua dactyloides (Nutt.) Columbus, P, BGHJ
†*Bouteloua eriopoda* (Torr.) Torr., P, BGHJ
Bouteloua gracilis (Kunth) Lag. ex Griffiths, P, BCBGBD
Bouteloua hirsuta Lag. , P, BGHJ
**Bromus arvensis* L., A, DAOF
**Bromus catharticus* Vahl, A, DAOF
**Bromus racemosus* L., A, BGHJ
**Bromus tectorum* L., A, DAOF
Calamovilfa gigantea (Nutt.) Scribn. & Merr., P, BCBGBD
Cenchrus spinifex Cav., P, BGHJ
Chloris verticillata Nutt., P, AFSA
Chloris virgata Sw., A, BGHJ
**Cynodon dactylon* (L.) Pers., P, DAOF
Distichlis spicata (L.) Greene var. *stricta* (Torr.) Thorne, P, BGHJ
Echinochloa muricata (P. Beauv.) Fernald, A, DAOF
Elymus canadensis L., P, BGHJ
Elymus elymoides (Raf.) Swezey, P, JMWA
Elymus virginicus L., P, AFSA
**Eragrostis cilianensis* (Bellardi) Vignolo ex Janch., A, AFSA
Erioneuron pilosum (Buckley) Nash, P, JMWA
†*Hesperostipa neomexicana* (Thurb.) Barkworth, P, BGHJ
†*Hilaria jamesii* (Torr.) Benth., P, BGHJ
Hopia obtusa (Kunth) Zuloaga & Morrone, P, AFSA
Hordeum jubatum L., P, DAOF
Hordeum pusillum Nutt., A, DAOF
Leptochloa fusca (L.) Kunth spp. *fasicularis* N.W. Snow, A, HWV
Muhlenbergia asperifolia (Nees & Meyen ex Trin.) Parodi, P, AFSA
Muhlenbergia paniculata (Nutt.) Columbus, P, DAOF
†*Muhlenbergia phleoides* (Kunth) Columbus, P, BGHJ
†*Muhlenbergia porteri* Scribn. ex Beal, P, JMWA
†*Muhlenbergia torreyi* (Kunth) Hitchc. ex Bush, P, BCBGBD
Munroa squarrosa (Nutt.) Torr., A, BGHJ
Panicum capillare L., A, DAOF
Panicum hallii Vasey, P, BGHJ
Panicum virgatum L., P, JMWA
Pascopyrum smithii (Rydb.) Barkworth & D.R. Dewey, P, AFSA
Paspalum setaceum Michx. var. *stramineum* (Nash) D.J. Banks, P, DAOF
†*Piptatherum micranthum* (Trin. & Rupr.) Barkworth, P, JMWA
Poa fendleriana (Steud.) Vasey, P, JMWA
**Polypogon monspeliensis* (L.) Desf., A, HWV
Schizachyrium scoparium (Michx.) Nash, P, AFSA
Setaria macrostachya Kunth, P, DAOF
**Setaria viridis* (L.) P. Beauv., A, DAOF
Sorghastrum nutans (L.) Nash, P, BGHJ
**Sorghum halepense* (L.) Pers., P, BGHJ
Sporobolus airoides (Torr.) Torr. , P, BGHJ

Sporobolus cryptandrus (Torr.) A. Gray, P, AFSA
Sporobolus pyramidatus (Lam.) Hitchc., P, AFSA

Polemoniaceae
Ipomopsis laxiflora (J.M. Coult.) V.E. Grant, A, JMWA

Polygalaceae
Polygala alba Nutt., P, BGHJ

Polygonaceae
Eriogonum annuum Nutt., A, AFSA
†*Eriogonum jamesii* Benth., P, BCBGBD
†*Eriogonum lachnogynum* Torr. ex Benth., P, BGHJ
†*Eriogonum tenellum* Torr., P, JMWA
Persicaria amphibia (L.) Delarbre, P, HWV
Persicaria lapathifolia (L.) Gray, A, HWV
**Polygonum aviculare* L., A, DAOF
Rumex altissimus Alph. Wood, P, HWV
**Rumex crispus* L., P, HWV
Rumex venosus Pursh, P, DAOF

Portulacaceae
Phemeranthus parviflorus (Nutt.) Kiger, P, AFSA
Portulaca oleracea L., A, JWMA
Portulaca pilosa L., A, DAOF

Potamogetonaceae
Zannichellia palustris L., P, HWV

Pteridaceae
Cheilanthes eatonii Baker, P, JMWA
Notholaena standleyi, P, JMWA

Ranunculaceae
Delphinium carolinianum Walter ssp. *virescens* (Nutt.) R.E. Brooks, P, JMWA
Ranunculus abortivus L., P, HWV
Ranunculus sceleratus L., A, HWV
**Ranunculus testiculatus* Crantz, A, DAOF

Rosaceae
†*Cercocarpus montanus* Raf., P, JMWA
Prunus virginiana L. var. *demissa* (Nutt.) Torr., P, JMWA
†*Rubus deliciosus* Torr., P, JMWA

Rutaceae
Ptelea trifoliata L., P, JMWA

Salicaceae
Populus deltoides W. Bartram ex Marshall, P, HWV
Salix amygdaloides Andersson, P, HWV
Salix exigua Nutt., P, HWV
Salix nigra Marshall, P, HWV

Santalaceae
Comandra umbellata (L.) Nutt. ssp. *pallida* (A. DC.) Piehl, P, JMWA

Sapindaceae
Sapindus saponaria L. var. *drummondii* (Hook. & Arn.) L.D. Benson, P, DAOF

Selaginellaceae
†*Selaginella underwoodii* Hieron., P, JMWA

Solanaceae
Chamaesaracha coniodes (Moric. ex Dunal) Britton, P, JMWA
Datura quercifolia Kunth, A, DAOF
Physalis hederifolia A. Gray var. *fendleri* (A. Gray) Cronquist, P, JMWA
Physalis longifolia Nutt. var. *longifolia*, P, AFSA
Quincula lobata (Torr.) Raf., P, JMWA
Solanum elaeagnifolium Cav., P, DAOF
Solanum ptychanthum Dunal, A, BCBGBD
Solanum rostratum Dunal, A, AFSA
Solanum triflorum Nutt., A, DAOF

Tamaricaceae
**Tamarix chinensis* Lour., P, HWV

Verbenaceae
Glandularia bipinnatifida (Nutt.) Nutt. var. *ciliata* (Benth.) B.L. Turner, A, BGHJ
Glandularia canadensis (L.) Nutt., P, JMWA
Glandularia pumila (Rydb.) Umber, A, BGHJ
Phyla cuneifolia (Torr.) Greene, P, HWV
Verbena bracteata Cav. ex Lag. & Rodr., A, AFSA

Violaceae
Hybanthus verticillatus (Ortega) Baill., P, BGHJ

Vitaceae
Vitis vulpina L., P, JMWA

Zygophyllaceae
Kallstroemia parviflora Norton, A, AFSA
**Tribulus terrestris* L., A, AFSA

APPENDIX B

List of Plant Taxa in Cimarron County and Black Mesa, Oklahoma Not Found by Buthod and Hoagland

Taxa from the published lists of Rogers (1953), McPherson (2003a, b), and Folley (2003) that were not found by Buthod and Hoagland. R=Rogers collection, M=Mcpherson collection, F=Folley collection. Taxonomy has been updated and follows the Integrated Taxonomic Information System (2015).

Amaranthaceae
Amaranthus retroflexus L., M
Chenopodium albescens Small, R
Cycloloma atriplicifolium (Spreng.) J.M. Coult., R
Froelichia gracilis (Hook.) Moq., R
Guilleminea densa (Humb. & Bonpl. ex Schult.) Moq. var. *densa*, R
Salsola kali L. ssp. *tenuifolia* Moq., M
Suckleya suckleyana (Torr.) Rydb., M

Amaryllidaceae
Allium canadense L. var. *fraseri* Ownbey, M

Anacardiaceae
Rhus aromatica Aiton var. *simplicifolia* (Greene) Cronquist, R
Toxicodendron radicans (L.) Kuntze, M

Apiaceae
Cymopterus glomeratus (Nutt.) DC., M

Apocynaceae
Asclepias arenaria Torr. , M
Asclepias involucrata Engelm. ex Torr. , R
Asclepias pumila (A. Gray) Vail, R, M
Asclepias uncialis Greene, M
Funastrum crispum (Benth.) Schltr., R, M

Araceae
Lemna minor L., M

Asparagaceae
Nolina texana S. Watson, F (collections are actually *Nolina greenei* S. Watson ex Trel.; Hess 2002))
Yucca harrimaniae Trel., F

Aspleniaceae
Asplenium septentrionale (L.) Hoffm., M

Asteraceae
Antennaria parvifolia Nutt., R

Artemisia dracunculus L., R, M
Baccharis wrightii A. Gray, R
Bidens cernua L., F
Brickellia eupatorioides (L.) Shinners var. *corymbulosa* (Torr. & A. Gray) Shinners, R
Chaetopappa ericoides (Torr.) G.L. Nesom, R, M
Ericameria nauseosa (Pall. ex Pursh) G.L. Nesom & Baird var. *nauseosa*, R, M
Erigeron nudiflorus Buckley, R
Erigeron tracyi Greene, M
Nothocalais cuspidata (Pursh) Greene, M
Oonopsis foliosa (A. Gray) Greene var. *foliosa*, R
Packera tridenticulata (Rydb.) W.A. Weber & A. Löve, R, M
Pericome caudata A. Gray, R, M, F
Picradeniopsis oppositifolia (Nutt.) Rydb. ex Britton, R
Psilostrophe villosa Rydb., F
Solidago mollis Bartlett , M
Solidago petiolaris Aiton, M
Stephanomeria pauciflora (Torr.) A. Nelson, R, M
Symphyotrichum ericoides (L.) G.L. Nesom, R, M
Symphyotrichum fendleri (A. Gray) G.L. Nesom, M
Symphyotrichum oblongifolium (Nutt.) G.L. Nesom, M
Verbesina encelioides (Cav.) Benth. & Hook. f. ex A. Gray, M
Vernonia fasciculata Michx., F
Xanthisma spinulosum (Pursh) D.R. Morgon & R.L. Hartm. var. *glaberrimum* (Rydberg) D.R. Morgan & R.L. Hartm., R

Boraginaceae
Cryptantha cinerea (Greene) Cronquist var. *cinerea*, R
Cryptantha crassisepala (Torr. & A. Gray) Greene, R
Euploca convolvulacea Nutt., F
Lithospermum multiflorum Torr. ex A. Gray, F

Brassicaceae
Boechera fendleri (S. Watson) W.A. Weber, M

Cactaceae
Opuntia fragilis (Nutt.) Haw., F

Campanulaceae
Lobelia cardinalis L., F

Cleomaceae
Peritoma serrulata (Pursh) DC., R, F
Polanisia jamesii (Torr. & A. Gray) Iltis, F

Cupressaceae
Juniperus scopulorum Sarg., M

Cyperaceae
Carex brevior (Dewey) Mack. , F
Cyperus croceus Vahl, F
Cyperus schweinitzii Torr., R, M
Schoenoplectus tabernaemontani (C.C. Gmel.) Palla, M
Scirpus atrovirens Willd., F
Scirpus pallidus (Britton) Fernald, R

Cystopteridaceae
Cystopteris fragilis (L.) Bernh., F

Equisetaceae
Equisetum laevigatum A. Br., R

Euphorbiaceae
Ditaxis humilis (Engelm. & A. Gray) Pax, R, M
Euphorbia geyeri Engelm., R
Euphorbia spathulata Lam., R

Fabaceae
Astragalus ceramicus E. Sheld., F
Astragalus crassicarpus Nutt., R
Astragalus crassicarpus Nutt. var. *paysonii* (E.H. Kelso) Barneby, M
Astragalus gracilis Nutt., R
Astragalus hallii A. Gray, R
Astragalus lotiflorus Hook. , R, M
Astragalus puniceus Osterh., M
Colutea arborescens L., F
Dalea candida Michx. ex Willd var. *candida*, R
Dalea compacta Spreng. var. *compacta*, R
Dalea nana Torr. ex A. Gray, R
Dalea purpurea Vent. var. *purpurea*, R
Hedysarum boreale Nutt., R
Melilotus albus Medik., R
Pediomelum argophyllum (Pursh) J.W. Grimes, M
Pediomelum hypogaeum (Nutt.) Rydb. var. *hypogaeum*, R
Pomaria jamesii (Torr. & A. Gray) Walp., R, M
Vicia americana Muhl. ex Willd. , M
Vicia ludoviciana Nutt. ex Torr. & A. Gray var. *leavenworthii* (Nutt. ex Torr. & A. Gray) Broich, R

Fagaceae
Quercus gambelii Nutt., R
Quercus grisea Liebm., R
Quercus X *undulata* Torr., R

Lamiaceae
Salvia azurea Michx. ex Lam. var. *grandiflora* Benth., M

Linaceae
Linum lewisii Pursh , R, M

Loasaceae
Mentzelia decapetala (Pursh ex Sims) Urb. & Gilg, R, M

Lythraceae
Lythrum alatum Pursh, R

Nyctaginaceae
Mirabilis glabra (S. Watson) Standl., R, M
Mirabilis linearis (Pursh) Heimerl var. *linearis*, R

Onagraceae
Oenothera albicaulis Pursh, R
Oenothera engelmannii (Small) Munz, R, F
Oenothera lavandulifolia Torr. & A. Gray, M
Oenothera pallida Lindl. ssp. *latifolia* (Rydb.) Munz, F

Orobanchaceae
Castilleja sessiliflora Pursh, R, M

Papaveraceae
Argemone polyanthemos (Fedde) G.B. Ownbey, R

Plantaginaceae
Penstemon angustifolius Nutt. ex Pursh var. *caudatus* (A. Heller) Rydb., R

Poaceae
Achnatherum hymenoides (Roem. & Schult.) Barkworth, R, M
Achnatherum scribneri (Vasey) Barkworth, R, M
Andropogon virginicus L., F
Aristida barbata E. Fourn., R
Aristida divaricata Humb. & Bonpl. Ex Willd., R
Aristida purpurea Nutt. var. *fendleriana* (Steud.) Vasey, R
Aristida purpurea Nutt. var. *longiseta* (Steud.) Vasey, R
Aristida purpurea Nutt. var. *wrightii* (Nash) Allred, R, M
Bothriochloa saccharoides (Sw.) Rydb., M
Bouteloua hirsuta Lag. var. *hirsuta*, M
Bromus japonicus Thunb. ex Murray, R
Bromus lanatipes (Shear) Rydb., R, M
Cenchrus incertus M.A. Curtis, R
Cenchrus longispinus (Hack.) Fernald, M
Dichanthelium oligosanthes (Schult.) Gould, R
Digitaria californica (Benth.) Henrard, R
Digitaria cognata (Schult.) Pilg., R
Echinochloa crus-galli (L.) P. Beauv., M

Enneapogon desvauxii P. Beauv., R
Eragrostis curtipedicellata Buckley, R
Eragrostis intermedia Hitchc., R
Eragrostis secundiflora J. Presl, R
Eragrostis sessilispica Buckley, R
Eragrostis trichodes (Nutt.) Alph. Wood, M
Hesperostipa comata (Trin. & Rupr.) Barkworth, R, M
Leptochloa dubia (Kunth) Nees, R
Muhlenbergia arenicola Buckley, R
Muhlenbergia racemosa (Michx.) Britton, Sterns & Poggenb., R, F
Phalaris caroliniana Walter, R
Phragmites australis (Cav.) Trin. ex Steud., R
Poa nemoralis L., R
Poa pratensis L., R
Setaria leucopila (Scribn. & Merr.) K. Schum., M
Sphenopholis obtusata (Michx.) Scribn., R
Tridens muticus (Torr.) Nash var. *elongatus* (Buckley) Shinners, R
Triplasis purpurea (Walter) Chapm., R
Vulpia octoflora (Walter) Rydb., R, M

Polemoniaceae
Giliastrum rigidulum (Benth.) Rydb., F

Polygonaceae
Polygonum ramosissimum Michx. , M

Pteridaceae
Astrolepis sinuata (Lag. ex Sw.) D.M. Benham & Windham ssp. *sinuata*, R
Cheilanthes feei T. Moore, R, M
Cheilanthes lanosa (Michx.) D.C. Eaton, M
Pellaea atropurpurea (L.) Link, R, M

Ranunculaceae
Clematis hirsutissima Pursh var. *scottii* (Porter) R.O. Erickson, M
Ranunculus cymbalaria Pursh, R

Rhamnaceae
Ceanothus herbaceus Raf., R

Rosaceae
Fallugia paradoxa (D. Don) Endl. ex Torr., R
Physocarpus monogynus (Torr.) J.M. Coult., R, M
Prunus americana Marshall , M
Rosa woodsii Lindl., F

Rubiaceae
Galium texense A. Gray, M

Salicaceae
Salix interior Rowlee, M

Selaginellaceae
Selaginella densa Rydb., R

Solanaceae
Solanum nigrum L., R

Tamaricaceae
Tamarix gallica L., R, M

Urticaceae
Parietaria pensylvanica Muhl. ex Willd., M

Verbenaceae
Verbena plicata Greene, R

Vitaceae
Parthenocissus quinquefolia (L.) Planch., M
Vitis acerifolia Raf., R, F,

Woodsiaceae
Woodsia oregana D.C. Eaton, R, M

ANTIFUNGAL ACTIVITY IN EXTRACTS OF PLANTS FROM SOUTHWESTERN OKLAHOMA AGAINST *ASPERGILLUS FLAVUS*

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Key words: antifungal,* Aspergillus flavus, *medicinal plant, undergraduate research

ABSTRACT

The use of medicinal plants has been an integral part of human civilization since antiquity. Naturally occurring pesticidal compounds are synthesized by the plant defense system, which includes antimicrobial proteins and lower molecular weight natural products. In this study, plants were collected from southwestern Oklahoma, and plant tissues were extracted and assayed for antifungal activity against *Aspergillus flavus*, a mycotoxin producing fungus. Out of the 84 plant tissue extracts tested, 40 extracts exhibited complete to very strong inhibition of fungal growth. Extracts were dialyzed in Tris buffer using 3,500 molecular weight cut-off dialysis membrane to remove low molecular weight compounds. After dialysis, the majority of the plant extracts lost antifungal activity against *A. flavus*. Four plant extracts, however, retained complete activity. The source plants of these four extracts were identified as belonging to Asparagaceae. Three of the extracts came from three different plants of the genus *Allium*. The fourth extract was from *Camassia scilloides*.

INTRODUCTION

The history of plant use for medicinal purpose is as old as human civilization. Six thousand year old excavated clay slabs from early Sumerian civilizations revealed recipes for drug preparation using over 250 different plants (Petrovska 2012). Additionally, an Egyptian scroll dated about 1500 BP mentioned more than 850 plant based medicines (Petrovska 2012). Hippocrates (460–377 BP) also believed in the power of plants to cure ailments and used 300 different plant species to heal his patients. Pedanious Dioscorides (50–70 AD) assembled *De Materia Medica* where he described comprehensive use, preparation, side effects, and cultivation of 600 plants (Sumner 2000). There are examples from every culture about healing abilities of plants. Modern research indicates that the majority of ethnobotanical claims are valid and correspond with our current knowledge of plant-derived compounds. Benefits of

https://doi.org/10.22488/okstate.17.100115

traditional plants have been explored via scientific research, which led to the discovery of many valuable drugs for the modern world. Examples include reserpine from *Rauwolfia serpentina* (Indian snake root), vincristine from *Catharanthus roseus* (Madagascar periwinkle), artimisinin from *Artemisia annua,* (sweet sagewort), capsaicin from *Capsicum annuum* (chili pepper), morphine from *Papaver somniferum* (poppy), atropine from *Atropa belladonna* (deadly night shade), silymarin from *Silybum marianum* (milk thistle), and ephedrine from *Ephedra sinica* (Chinese ephedra) (Farnsworth et al. 1985; Houghton 1995; Sumner 2000; Gupta, et al. 2005; Goutam 2015).

Plants provide a rich source for the discovery of new drugs. A number of bioactive compounds can be isolated from different parts of a single plant. *Azadirachta indica* (neem) is one such plant. Neem plant is known as the 'village dispensary' in India because of its wide spectrum of biological activities and no-cost availability due to the widespread growth of this plant in the region (Arora et al. 2008; Asif 2013). Over 135 bioactive compounds have been isolated from different parts of this plant. Some are well known to exhibit antiviral, antifungal, antibacterial, anti-insect, and antitumor activity. There are over 250,000 plant species in the world, but only 6% have been screened for biological activity. According to Farnsworth et al. (1985), there are 119 plant-derived drugs used today all over the world, and all of those came from less than 90 plant species. The possibility of finding novel compounds from plants that can be exploited for medicinal use is enormous.

Although the origin of many life-saving modern medicines came from natural sources, tremendous achievements in synthetic chemistry have made it possible for pharmaceutical companies to design and introduce drugs at a faster pace. To find medicinal compounds from a plant source, a large number of plants need to be screened. Once identified, the active compounds have to be purified and characterized. Many of these natural compounds are structurally complex. Therefore, to go from discovery to high throughput commercial production can be technically challenging and time consuming. Some of these issues have contributed to the decline of plant-based drug discovery (Gupta et al. 2005). However, over the last few decades, the world has watched the reemergence of infectious diseases once thought to be eradicated (Gupta et al. 2005; Lam 2007; Petrovska 2012). At the same time, incidences of pest and pathogen resistance against antimicrobial products have increased in alarming numbers. There is also an increase in the prevalence of multidrug resistant bacterial pathogens. In light of these facts, there is renewed interest in looking into nature's wealth for newer and better medicines.

Plants are a storehouse of naturally occurring pesticidal compounds that are molecularly diverse. Plants have developed an arsenal of defense mechanisms from protection against pests and pathogens. As a result, different and unique chemicals compounds are synthesized by plants. These diverse molecules in plants are under constant evolutionary selection. This makes the plant kingdom a continuously wealthy source for finding new antimicrobial compounds (Hossain 1999).

The compounds that are synthesized by plant defense systems, either to prevent pathogen attack or to destroy invading pathogens, include proteins and lower molecular weight natural products. Defense-related proteins produced by plants include hydroxyproline-rich glycoproteins; glycine-rich proteins; amylase inhibitors; proteinase inhibitors; toxic proteins such as lectins and thionins; hydrolases such as chitinases and β-1,3-glucanases; anti-microbial peptides such as defensins; and other cysteine-rich proteins (Hossain 1999). Lower molecular weight natural products include various

alkaloids, tannins, flavonoids, terpenes, etc. (Goutam 2015).

Southwestern Oklahoma has a rich history in the use of medicinal plants. Jordan et al. (2006) documented the use of over 100 species of vascular plants by the Plains Apache tribe. Thirty-nine of those species were used in rituals and for medicinal purposes. According to the study, out of the 105 documented species used by the tribe, 98 are native to southwestern Oklahoma and occur throughout the western U. S. and Great Plains (Jordan et al. 2006). Students from the Cameron University Biology Department collected plants from four different locations in southwestern Oklahoma. Aqueous crude and dialyzed extracts from collected plants were screened for antifungal activity against *Aspergillus flavus*, which was chosen because this fungus produces carcinogenic mycotoxins known as aflotoxins. This study was part of an assignment for a medicinal plants class. In this report we present the result of the study.

MATERIALS AND METHODS

Collection of Plant Materials

Forty-seven species of plants were collected from four different locations in southwestern Oklahoma (Table 1). These locations include Medicine Park, East Lawton, Stephens County, and Anadarko. Plants were collected based on ethnobotanical information (Jordan et al. 2006) as well as field observation. Therefore, not all of the collected plants have known medicinal use. The field observations of healthy plants growing in the midst of plants infested by pests and pathogens could indicate defense related compounds protecting these plants. Such field observations were a part of the collection process. On location, the collected materials were photographed, bagged, and kept in ice. Once transported to the laboratory, the plant materials were placed in -80^{o}C for long term storage.

Table1 List of plants screened for antifungal activity against *Aspergillus flavus*
PCN, Plant Collection Number. AF, Stephens County; MP, Medicine Park; CSL, East Lawton; AN, Anadarko. R, root; L, leaf; B, bulb; Fl, flower; Yl, young seedling. NI, not identified. Approximate latitudes and longitudes of the locations are: Lawton (N34.069424, W98.417781); Medicine Park (N 34.733270, W 98.483923); Anadarko (N35.069203, W96.265657); Stephens County (N34.36, W98.23).

Extract. #	PCN #	Scientific Name	Common Name	Family
1	AF#19 L	NI		
7	AF#19R			
2	AF#10R	*Yucca glauca* Nutt.	Soapweed	Agavaceae
4	AF#10L			
37	AF#10F			
3	AF#13L	*Artemisia* sp.		Asteraceae

5	AF#18L	NI	NI	NI
25	AF#18L			
6	AF#1L	*Ulmus* sp.	Elm seedling	Ulmaceae
8	AF#17L	*Callirhoe involucrata* (Torr. & A. Gray) A. Gray	Purple Poppy	Malvaceae
18	AF#17R			
9	AF#17Fl			
10	AF#15F	*Castilleja indivisa* Engelm	Indian Paint Brush	Orobanchaceae
15	AF#15R			
11	AF#9L	Rumex crispus L.	Curly Dock	Polygonaceae
29	AF#9R			
16	AF#	NI		
12	AF#5L	*Achillea* sp.	Yarrow	Asteraceae
14	AF#5R			
17	CSL#7L	*Nothoscordum bivalve* (L.) Britton	False garlic	Asparagaceae
38	CSL#7R			
16	AF#FR	NI		
19	AF#4L	*Daucus carota* L.	Wild carrot	Apiaceae
20	MPGA#1S	*Echinocereus reichenbachii* (Terscheck ex Walp.) J.N. Haage	Lace Echinocereus	Cactaceae
23	MPGA#1R			
21	AF#21S	*Opuntia* sp.	Prickly pear	Cactaceae
33	AF#21R			
24	AF #8 R	*Callirhoe involucrata* (Torr. & A. Gray) A. Gray	Purple Poppy seedling	Malvaceae
22	AF#14L	*Cirsium undulatum* (Nutt.) Spreng	Wavy-leaf Thistle	Asteraceae
27	AF#14R			
26	AN#1L	NI		
28	AF#6L	*Medicago lupulina* L.	Legume	Fabaceae
30	AF#11L	*Polygala senega* L.	Senega snake root	Polygalaceae
31	AF#2L	*Capsella bursa-pastoris* (L.) Medik.	Shepherd's Purse	Brassicaceae
39	AF#2Fl			
32	MPGP#1FL	NI		
35	MPGP#1L			

34	AF# 20L	NI		
36	CSL#10 F	NI		
40	AN#42	*Equiseteum* sp.	Rough horsetail	Equisetaceae
41	MP#23R	NI		
42	MP#24		Moss	
43	MP#9L	*Camassia scilloides* (Raf.) Cory	Wild hyacinth	Asparagaceae
76	MP#9F			
78	MP#9B			
44	MP#8L	NI		
46	MP#4L	*Glandularia bipinnatifida* (Nutt.) Nutt.	Verbena	Verbenaceae
50	MP#4F			
47	MP#5L	*Callirhoe leiocarpa* R.F. Martin	Tall poppy mallow	Malvaceae
60	MP#5F			
48	MP#1F	*Sapindus* sp.	Soap berry	Sapindaceae
49	MP#3F	*Tradescantia tharpii* E.S. Anderson &Woodson	Spiderwort	Commelinaceae
66	MP#3R			
45	MP#3L			
51	MP#6L	NI		
75	MP#6R			
52	MP#22Fl	*Allium canadense* L.	Wild onion	Asparagaceae
57	MP#22R			
70	MP#22B			
58	MP#19L	*Ambrosia* sp.	Ragweed	Asteraceae
85	MP#19R			
59	MP#26R	*Amsonia ciliata* Walter	Blue Star	Apocynaceae
61	MP#26Fl			
65	MP#26L			
62	MP#14L	NI		
79	MP#14R			
63	MP#15L	*Physaria gracilis* (Hook) O'Kane & Al-Shehbaz	Yellow-flowered bladderpod	Brassicaceae
80	MP#15R			
81	MP#15Fl			

64	CSL #B	*Allium drummondii* Regel	Drummond's onion	Asparagaceae
72	CSL#1L			
73	CSL#1Fl			
67	MP#11F	*Yucca glauca* Nutt.	Soapweed	Asparagaceae
68	CSL#3L	*Vicia sativa* L.	Common vetch	Fabaceae
69	MP#10 L	*Rosa* sp.	Wild rose	Rosaceae
71	MP#7R	*Allium canadense* L.	Wild onion	Asparagaceae
54	MP#7FL			
74	CSL#11L			
77	MP#13L	*Oenothera* sp.	Gaura	Onagraceae
82	MP#16L	NI		
83	MP#18L	*Erodium cicutarium* L.	Stork's bill	Geraniaceae
84	MP#18R			

Plant Tissue Extraction

Plant materials (seeds, fruits, leaves, roots, stems) were extracted in 10 mM Tris-HCl (pH 8.0) containing 0.2 g of insoluble PVP (polyvinylpolypyrrolidone; Sigma Chem. Co, Cat. # P6755) for each g of frozen plant tissue (Hossain 1999). Mostly, five volumes of buffer were used for each gram of fresh weight of tissue. The buffer volume was adjusted for mucilaginous and starchy tissue. All extractions were carried out at room temperature. Plant tissues were homogenized in liquid nitrogen using a mortar and pestle, and the homogenate was filtered through a double layer of Miracloth. The filtrate was centrifuged at 12,000 x g for 15 min in a Sorvall SS34 rotor. The supernatant fluid was collected, and the pellet containing debris and insoluble PVP was discarded. The clarified supernatant fluid, referred to as the crude extract was tested for antifungal activity.

Dialysis

To remove soluble, low-molecular-weight materials from the crude extract, 2 ml of each crude extract was dialyzed extensively against 10 mM Tris-HCl (pH 8.0) using a 3,500 molecular weight cut-off dialysis membrane (Spectra/Por). Dialysis was routinely carried out in 4 L beakers, and the dialysis buffer was changed at least three times over a 24–48 h period. Total volume of extracts dialyzed in each 4 L beaker was 20–30 ml. After dialysis, the crude extract (referred to as dialyzed extract) was tested for antifungal activity.

Source of Fungal Pathogen

The antifungal activity of all extracts was evaluated on the basis of activity against an *Aspergillus flavus* (ATCC # 22548) culture obtained from the American Type Culture Collection, Waldorf, MD. Working cultures of *A. flavus* (Fig. 1) were grown at room temperature on half-strength potato dextrose agar (PDA; Difco # 0013-17-6). Inoculated fungal plates were kept at room temperature for 10 days until the mycelial growth covered 75% of the plate. At that point, the plates were stored at 4ºC for future use.

Figure 1 *Aspergillus flavus* (ATCC 22548) on half strength Potato Dextrose Agar. Inoculated fungal plates were kept at room temperature for two to three weeks until the hyphal growth covered three fourths of the plate. For fungal assay, conidia were scraped from 2–3 weeks old plates.

Antifungal Bioassay

The assay used to detect antifungal activity in plant extracts was originally developed by Duvick et al. (Duvick et al. 1992). Conidia of *A. flavus* were collected by scraping the colony with a sterile loop and suspending the conidia in sterile water containing 0.01% Tween 20. Conidia from this stock solution were diluted with synthetic culture medium to a final concentration of ~290 conidia/90µl of growth medium. The latter contained 0.037 g NaCl, 0.0625 g $MgSO_4 \cdot 7H_2O$, 0.25 g $CaNO_3$, 2.5 g glucose, 0.25 g yeast extract, and 0.125 g casein enzyme hydrolysate in 1 L of 7.5 mM sodium phosphate buffer, pH 7.0. Ninety µl of the culture medium containing conidia were added to each well of a 96-well, U-bottom microtiter plate. Ten µl of crude extract or crude dialyzed extracts were added to each well. Four replicates (individual wells) were used for each sample. All of the assays were conducted in 96-well microtiter plates with four control wells in each plate. In these control wells, extraction buffer (10 mM Tris pH 8.0) was added to *A. flavus* conidia instead of plant extracts. The microtiter plate was covered with parafilm and incubated in the dark at 25ºC for 48 h. Conidial germination and fungal growth were observed after 48 h using a Nikon SMZ 1500 stereomicroscope equipped with digital CCD camera and NIS software. A rating scale of 0 to 4 was used to evaluate the inhibition of fungal growth (Fig. 2).

The ratings were based on the relative growth of fungi in comparison to the buffer control. A rating of zero indicated no inhibition of fungal growth, and a rating of four was given in the case of complete inhibition of fungal growth. Rating of 1–2 was based on approximately 50% or more hyphal growth compared to control. Rating of 3 was based on approximately 10–20% hyphal growth compared to control. Intermediate values (such as 1.5, 2.5, and 3.5) were assigned to distinguish between ratings when possible. Values from the four replicates were averaged.

Plant extract numbers were assigned by the students in the class, and the assay was performed by Dr. Tahzeeba Frisby. The plant collection numbers (PCN) matching the extracts were not given to Dr. Frisby, for an unbiased bioassay. Extracts were matched with their respective plant collection number after the bioassay data was collected.

Plant Identification

Plants collected for the study are common to southwestern Oklahoma. All plants were collected in the second week of April 2015. All of the plants were carefully identified using published field guides and keys (McCoy 1987; Freeman and Schofield 1991; Kindscher 1992; Ladd 1995; Loughmiller and Loughmiller 1996; Foster and Hobbs 2002; Barker 2006; Tyrl et al. 2008; Foley 2011). The Oklahoma Vascular Plants Database and the U. S. Wildflower Database of Wildflowers of Oklahoma were also consulted. Source for the authorities of the scientific names was the Integrated Taxonomic Information Service (www.itis.gov).

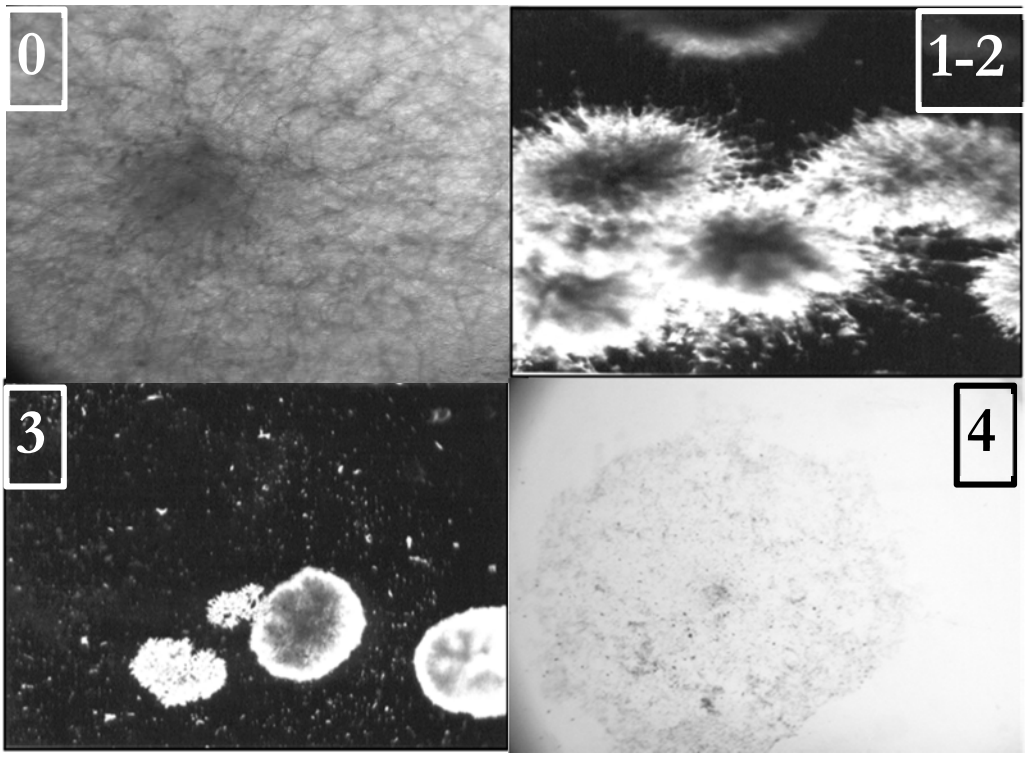


Figure 2 Antifungal rating based on the relative growth of fungi in the buffer control. Ratings: 0, no inhibition of fungal growth; 1, slight inhibition; 2, moderate inhibition; 3, strong inhibition; 4, no fungal growth.

RESULTS

Fungal Growth Inhibition by Crude Extracts

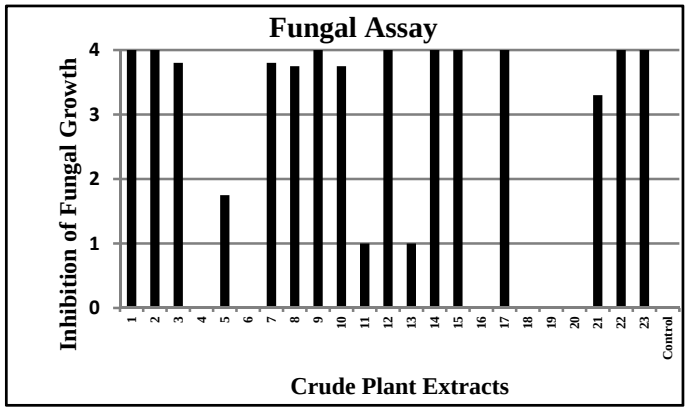
Eighty-four crude extracts obtained from 47 plant species (see Table 1) were screened for antifungal activity against *A. flavus*. Out of those, 29 exhibited complete inhibition of fungal growth (rating of 4; Fig. 3). An additional 18 extracts exhibited strong inhibition with a rating of 3.0 or above but less than 4. Nine more extracts exhibited a rating of 1 to 2.5. Twenty-seven extracts showed no inhibition (see Fig. 3). Growth was comparable to that observed in the control. Mycelial growth in the control buffer was extensive after 48 h incubation and covered the total surface of each well of the microtiter plate.

Fungal Growth Inhibition by Dialyzed Extracts

Forty-one crude extracts exhibiting very strong to complete inhibition of fungal growth (rating 3.5 to 4) were selected for further analysis. These extracts were exhaustively dialyzed (3,500 MWCO) in extraction buffer (10mM Tris pH 8.0) and assayed for antifungal activity against *A. flavus* as described above. Out of the 41 extracts, 29 exhibited a complete loss of antifungal activity after dialysis (Fig. 4). A loss of substantial activity was observed in extract 3, which was obtained from the leaves of an *Artemisia* species (Fig. 4A; Table 2). After dialysis, this extract showed slight inhibition of fungal growth after 48 hours. A partial loss of antifungal activity also was observed in dialyzed extracts from the root of *Yucca glauca* (extract 2) and from

the leaves of *Tradescantia tharpii* (extract 45), *Camassia scilloides* (extract 43), and *Oenothera* sp. (extract 77), but unlike extract 3, these retained moderate inhibition of *A. flavus* growth (see Fig. 4; see Table 2). However, dialyzed extracts 70 and 71 indicated strong activity with a rating of 3.4 and 3.1 respectively (Fig. 4B). Antifungal activity was also retained by dialyzed extracts 52, 54, and 73 and *Camassia* bulb extract 78 (see Fig. 4; Fig. 5, see Table 2). According to our bioassay results, these four dialyzed extracts completely inhibited conidial germination of *A. flavus* (see Fig. 5). Thus, among the 84 extracts screened for antifungal activity against *A. flavus,* these were the only four extracts that exhibited complete inhibition of fungal growth both before and after dialysis. Even after one week, no fungal growth was observed in these extracts.

3A



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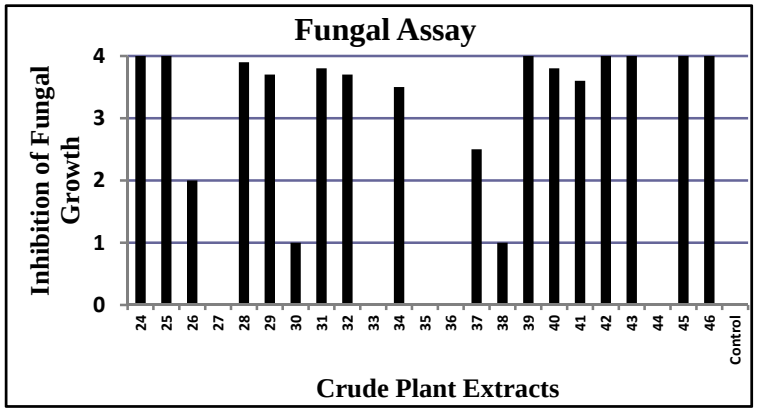
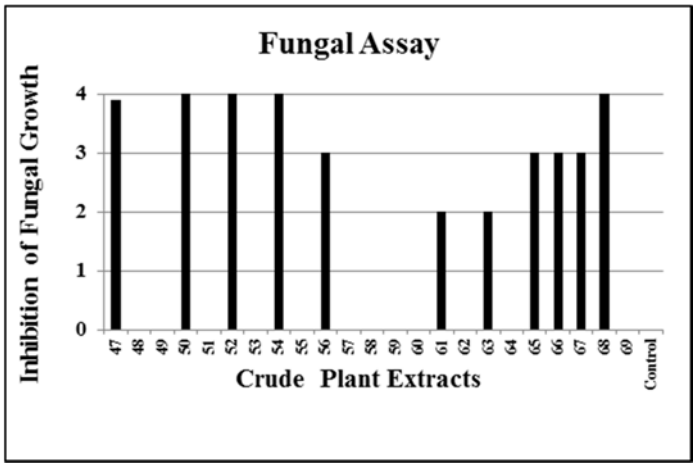


3C



3D

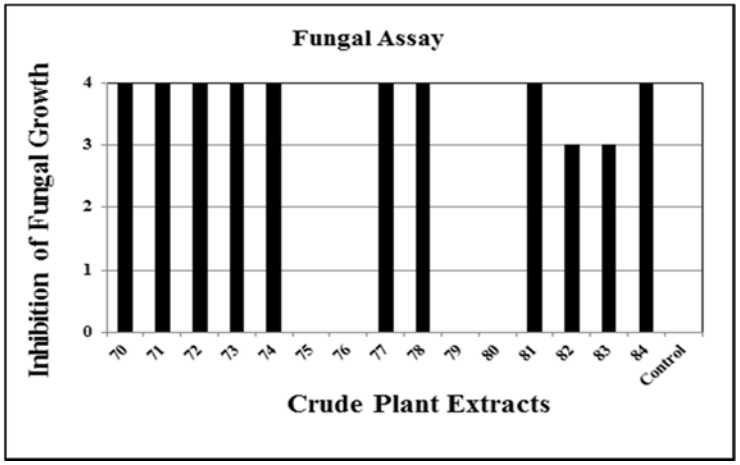
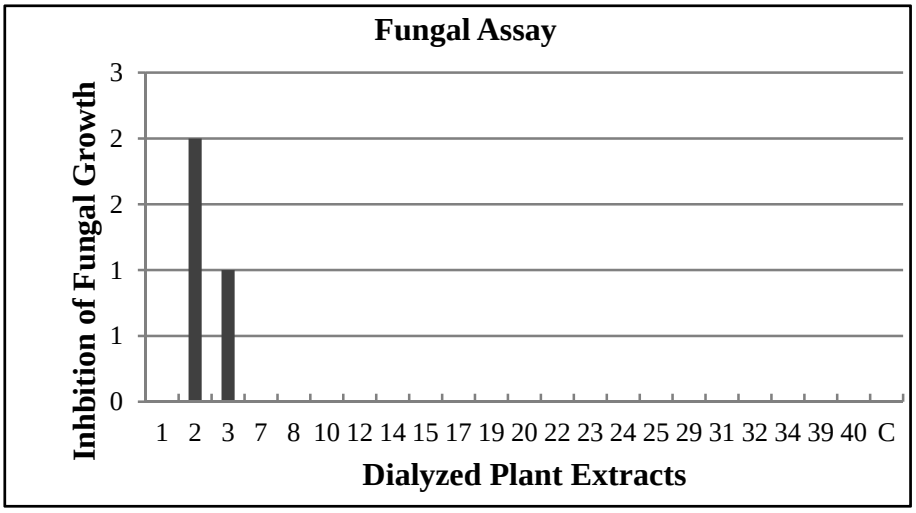


Figure 3 Evaluation of antifungal activity of crude extracts from Plant Collection Number (PCN) 1-84. (3A) Crude extracts from PCN 1-23. (3B) Crude extracts from PCN 24-46. (3C) Crude extracts from PCN 47-69. (3D) Crude extracts from PCN 71-84. Antifungal activity was measured using the standard assay with *Aspergillus flavus*. Rating of 0 = no inhibition of fungal growth and rating of 4 = complete inhibition of conidial germination and hyphal growth. PCN, plant collection number.

4A



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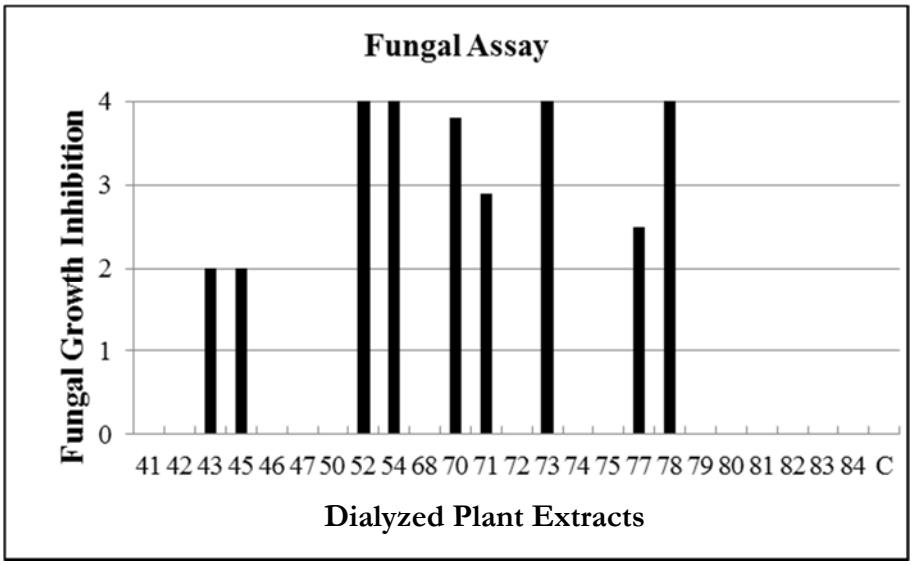
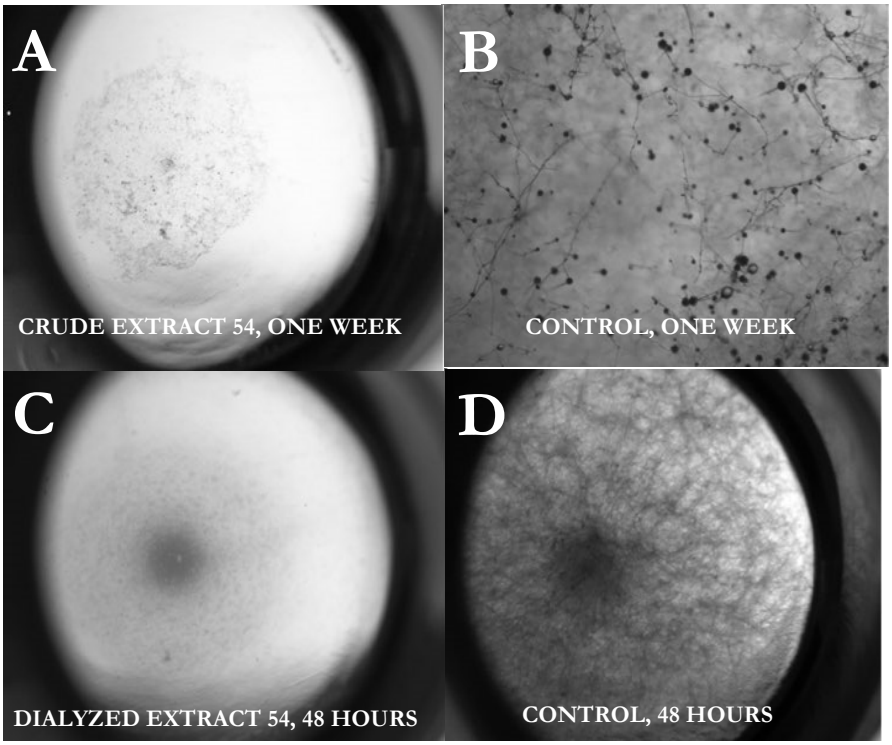
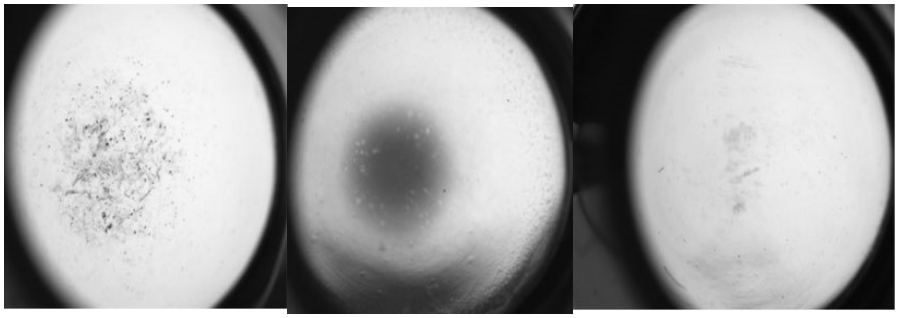


Figure 4 Evaluation of antifungal activity of dialyzed extracts from PCN 1-84 and Controls. (4A) Dialyzed extracts from PCN 1-40. (4B) Dialyzed extracts from PCN 41-84. Antifungal activity was measured using the standard assay with *Aspergillus flavus*. Rating of 0 = no inhibition of fungal growth and rating of 4 = complete inhibition of conidial germination and hyphal growth. PCN, plant collection number.



Panel 1 (A) Crude extract 54 retained complete inhibition of conidial germination and hyphal growth after one week. (B) *Aspergillus flavus* growth in Tris buffer control after one week. After one week of incubation conidia were visible in the control well. (C) Dialyzed extract 54 retained complete inhibition of conidial germination after 48 hours. (D) Control growth after 48 hours.



Panel 2 Antifungal activity of dialyzed extracts 52, 73 and 78. All three dialyzed extracts exhibited complete inhibition of conidial germination after 48 hours. Dark areas were due to the extract settling in the center of the well. Conidial germination and fungal growth were observed using a Nikon SMZ 1500 stereomicroscope equipped with digital CCD camera and NIS software.

Figure 5 Antifungal activity of crude and dialyzed extracts 54, 52, 73 and 78

Table 2 List of plants retaining antifungal activity after dialysis
PCN, Plant Collection Number. AF, Stephens County; MP, Medicine Park; CSL, East Lawton. R, root; L, leaf; B, bulb; Fl, flower.

Extract Number	PCN Number	Plant Tissue	Plant Name	Inhibition of Fungal Growth after Dialysis
2	AF #10R	Root	*Yucca glauca*	Moderate (rating 2)
3	AF #13L	Leaf	*Artemisia* sp.	Slight (rating 1)
43 78	MP#9L MP#9B	Leaf Bulb	*Camassia scilloides*	Moderate (rating 2) Complete (rating 4)*
45	MP#3L	Leaf	*Tradescantia tharpii*	Moderate (rating 2)
52 70	MP#22Fl MP22#B	Flower Bulb	*Allium canadense* (white flower)	Complete (rating 4)* Strong (rating 3.4)
54 71	MP#7Fl MP#7B	Flower Bulb	*Allium canadense* (light pink flower)	Complete (rating 4)* Strong (rating 3.1)
73	CSL#1B	Bulb	*Allium drummondii* (deep pink flower)	Complete (rating 4)*
77	MP#13L	Leaf	*Oenothera* sp.	Moderate (rating 2.5)

All six of the dialyzed extracts exhibiting strong to complete inhibition of *A. flavus* belong to two genera of Asparagaceae (see Table 2). Both the bulb and leaf of *Camassia scilloides* (PCN 9; Fig. 6C) possess antifungal activity against *A. flavus*, but the activity of the extract from the bulb of the plant is more potent. The other five extracts belong to the genus *Allium* (see Table 2). These plants were collected from East Lawton (CSL) and Medicine Park (MP). Extracts 52 and 70 were obtained from PCN MP#22 which had white flowers (Fig. 6A). Extracts 54 and 71 were from PCN MP#7 that had slightly pink flowers (Fig 6B). Extract 73 was obtained from CSL#1 which had a very distinct bulb and deep magenta-pink flowers (Fig. 7). According to the Oklahoma Vascular Plant Database, seven different *Allium* species are found in Comanche County, and *Allium* species exhibiting antifungal activity were tentatively identified based on the external morphology of the plants (see Table 2).

A. PCN MP#22 B. PCN MP#7 C. PCN MP#9

Figure 6 Plants exhibiting complete inhibition of fungal growth in crude and dialyzed extracts. (A and B), *Allium canadense*. (C) *Camassia scilloides*. Extracts from flowers of *A. canadense (ex #52 and 54)* and the bulb of *C. scilloides* (ex #78) exhibited complete inhibition of *A. flavus* growth. PCN, plant collection number.

A. PCN CSL#1 B. PCN CSL#1FL C. PCN CSL#1B

Figure 7 *Allium drumandii* collected from East Lawton. Both crude and dialyzed extracts (#73) from the flower of *A. drumandii* completely inhibited *Aspergillus flavus* growth in fungal bioassay. (A and B) Entire plant and flowers of *A. drumandii* respectively. (C) Characteristics fibrous structure around the bulbs of *A. drumandii*. PCN, plant collection number; Fl, flower; B, bulb.

DISCUSSION

In this study, most of the plant extracts that exhibited antifungal properties lost the activity after dialysis. Thus, it appears that most of the antifungal activity in crude extracts was due to soluble metabolites with molecular weights less than 3,500 Da. However, our results indicate that six extracts from two genera of Asparagaceae contained macromolecular compounds with molecular weights greater than 3,500 Da that were capable of strong to complete inhibition of fungal growth. To our knowledge, this is one of the first reports of antifungal activities from *Camassia scilloides*, *Allium canadense,* and *A. drummmondii* against *Aspergillus flavus*.

It is possible that the activity exhibited by some of the extracts may be due to the combinatorial effect of more than one type of compound. For example, crude extracts 2, 3, 43, 45, and 77, with ratings of 4, exhibited complete inhibition of conidial germination. After dialysis, however, these extracts lost approximately half or more of the inhibitory activity indicating a possible combined action of both low molecular weight compounds as well as molecules larger than 3,500 Da.

It is important to remember that the activities detected in the 84 aqueous extracts used in our study do not reflect the total antifungal activity present in the plant tissue or the potential to produce defense-related proteins and metabolites in response to fungal invasion. Furthermore, the loss of activity in many extracts may be due to protein denaturation and/or precipitation which may occur during storage and dialysis of the extracts. Also, many antifungal metabolites may not be soluble or may be only sparingly soluble in aqueous extracts. Therefore, organic solvent extraction would be required to isolate those compounds. Frequently, plant defenses are not expressed constitutively but are often produced in response to pathogen attack. These defense-related compounds include different types of proteins, which are induced upon infection by various pathogens, including fungi (Heisey and Gorgam 1992; Hu and Reddy 1997; Hu and Zhu 1997; Mohr et al. 1998; Cardoza et al. 2002). These defense-related proteins are comprised of enzymes responsible for the production of phytoalexins and other defensive metabolites, as well as pathogenesis-related proteins such as chitinase, glucanase and protease inhibitors. It is possible that extracts without antifungal activity are from plants that do not produce defense-related molecules constitutively and have not been induced. Even though antifungal compounds are not produced constitutively by these plants, they may very well possess the ability to activate defense genes in response to various elicitors of defensive compounds present in the tissue.

In this study, all 84 extracts were tested against *A. flavus* which is a filamentous ascomycete. Antifungal compounds present in the extracts may or may not have a broad range of antifungal activity. The activity may vary for different fungi due to different modes of action, or different fungi may be more or less sensitive to certain defense compounds. None of these extracts were tested against any representatives from oomycetes, such as *Phytophthora* sp. or *Pythium* sp. Many known antifungal proteins such as PR-1 and PR-5 specifically affect oomycetes, such as *Phytophthora infestans* (Woloshuk et al. 1991). Consequently, if these 84 extracts were tested for inhibitory activity against oomycetes, completely different results may have been obtained in this study.

The bioassays used in this study were rated after 48 hours of incubation, but the fungal growth in the microtiter plates was monitored and recorded for up to one week. Close observation of conidial germination and hyphal growth in the bioassay revealed that there may be different classes of mechanisms of antifungal activity

present in the extracts. In one class, conidial germination was completely inhibited after 48 h of incubation. Some examples from this category include crude extracts 1, 2, 14, 17, 25, 37, 43, 46, 50, 52, 54, 68, 73, and 78. In these extracts, conidia did not germinate even after 72 h of incubation. In the second class, conidial germination did occur, but the reduction in germination was coupled with an alteration in hyphal growth. For example, crude extract 56 strongly inhibited conidial germination and retarded hyphal growth, forming extremely branched and distorted hyphae (Fig. 8). Thus, our study indicates that extracts may exert their effect in at least two different ways. The first is to inhibit conidial germination and the second to distort hyphal growth.

The activities observed in our antifungal screens may not reflect all of the activities initially present in the extracts. The inhibitor may break down naturally or may be inactivated or detoxified through the action of endogenous activities in the extract (such as hydrolases) or reactive components in the extract (such as phenolic compounds or oxygen). The extraction conditions used in these studies were not designed to protect sensitive or unstable activities. Antioxidants, metal chelators, protease inhibitors, and/or reductants were not included in the buffer. The only protectant used was PVP, which was added to reduce the concentration of potentially reactive phenolics.

CONCLUSIONS

Our results showed that most of the extracts that exhibited antifungal activity before dialysis lost their activity after dialysis. This indicates that most of the antifungal activity was due to the presence of soluble metabolites with molecular weights less than 3,500 Da. Four extracts retained complete antifungal activity after dialysis. Three of the extracts were obtained from *Allium* and the fourth from *Camassia* sp. Our study also indicates that antifungal activity retained in these dialyzed extracts is due to macromolecular compounds with molecular weights greater than 3,500 Da.

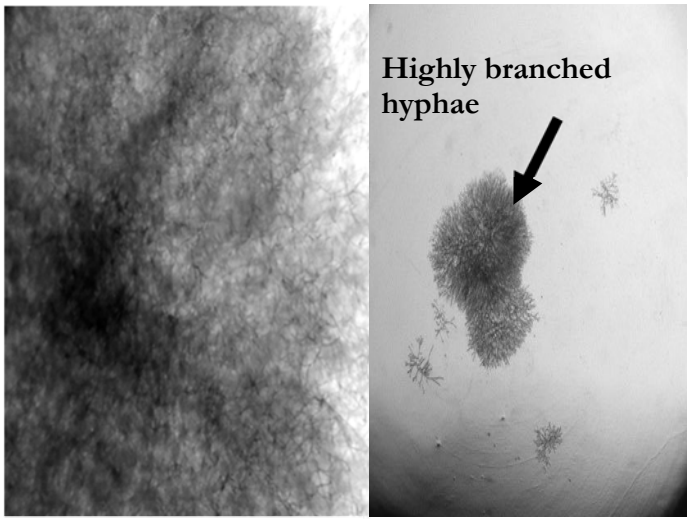


Figure 8 Antifungal activity of crude extract 56 after 48 hours. (A) Hyphal growth in control. (B) Crude extract 56 strongly inhibited conidial germination and retarded hyphal growth, forming extremely branched and distorted hyphae. Antifungal activity was measured using the standard assay with *Aspergillus flavus*. Rating of 0 = no inhibition of fungal growth and rating of 4 = complete inhibition of conidial germination and hyphal growth. Conidial germination and fungal growth were observed using a Nikon SMZ 1500 stereomicroscope equipped with digital CCD camera and NIS software.

ACKNOWLEDGEMENTS

We want to thank Cameron University and the Department of Biological Sciences for their support. We are grateful to Dr. Dennis Frisby for his reading of the manuscript, insightful discussion, and encouragement.

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Oklahoma Native Plant Record
Volume 15, December 2015

KUDZU, *PUERARIA MONTANA* (LOUR.) MERR. ABUNDANCE AND DISTRIBUTION IN OKLAHOMA

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Key words: invasion, invasive species, mapping, federal noxious weed

ABSTRACT

Invasive species are a growing problem in the United States, and kudzu (*Pueraria montana*) (Lour.) Merr. is one of the most well documented invaders of southeastern states. Documenting the invasion of kudzu in Oklahoma, however, has not been a targeted focus in previous studies; thus, maps of its occurrence differ among sources. Our primary objective was to locate and confirm the presence of kudzu throughout Oklahoma. Specifically, we attempted to confirm previously recorded populations of kudzu and estimate the extent of the invasion at those sites. In addition, we wanted to locate stands of kudzu within Oklahoma that had not been recorded and to assess the extent of invasion. A survey was sent to state and county officials to acquire information on locations and general knowledge of kudzu. Points of occurrence and estimated extent of invasion in hectares were then placed in ArcMap programming to create a consolidated map of kudzu. Samples were collected, pressed, and placed in the University of Oklahoma's Bebb Herbarium (OKL). We determined the majority of kudzu locations are in the southeastern portion of the state and total a minimum of 32.4 hectares. Results of the survey indicated half of the respondents polled were unaware of kudzu's presence in the state.

INTRODUCTION

Invasive species are a growing concern in the United States, as well as across the globe. There are approximately 17,000 native species of vascular plants in the U. S., compared to a continually increasing estimate of 6,000 nonnative species (Forseth and Innis 2004). Invasive species can be detrimental to the environments they occupy and cause major ecosystem changes (Mitich 2000). Kudzu, *Pueraria montana* (Lour.) Merr. (Fabaceae) is an introduced, leguminous vine which causes major changes in areas in which it invades. Kudzu is listed as one of the world's 100 worst invasive species of all time (Sage et al. 2009). First introduced at the 1876 Centennial Exposition in Pennsylvania, kudzu has since made a lasting impact on the southeastern U. S. (Brown 2010). Upon introduction, the vine was sold to the public to aid with soil erosion control and as forage for livestock; additionally, the Soil Conservation Service (currently Natural Resources Conservation Service) and other national agencies encouraged the planting of kudzu (Forseth and Innis 2004). Eventually, evidence indicated that the vine overtopped mature trees, took over native plant dominated areas, buildings, and disturbed areas, and became a financial burden to those who

https://doi.org/10.22488/okstate.17.100116

tried to control and eradicate the invader. Kudzu has been found to alter a landscape abruptly as it can grow up to 30 cm a day and between 10 to 30 m in one growing season (Mitich 2000). Additionally, kudzu fixes nitrogen and releases isoprene into the environment, which can create pollution in the atmosphere, further reducing environmental value (Hickman et al. 2010).

Kudzu is one of the worst invasive species in the U. S. and is continuing its spread across the country (Fig. 1). It has been estimated that the vine covers 2.83 million hectares in the Southeast, in 1955 was declared a weed by the U. S. Department of Agriculture (Alderman 1998) and declared a federal noxious weed in 1999 (Mitich 2000). Kudzu has a wide climatic range which facilitates its ability to continue spreading northward (Mitich 2000). It has been suggested that kudzu is limited in its range by annual rainfall, which needs to be a minimum of 100 cm a year (Mitich 2000). The vine is also considered to be limited in its distribution by lack of hardiness; however, it has exceeded many expert predictions in range expansion (Mitich 2000). This area includes Oklahoma, which was once believed to be unsuitable habitat for kudzu (Mitich 2000).

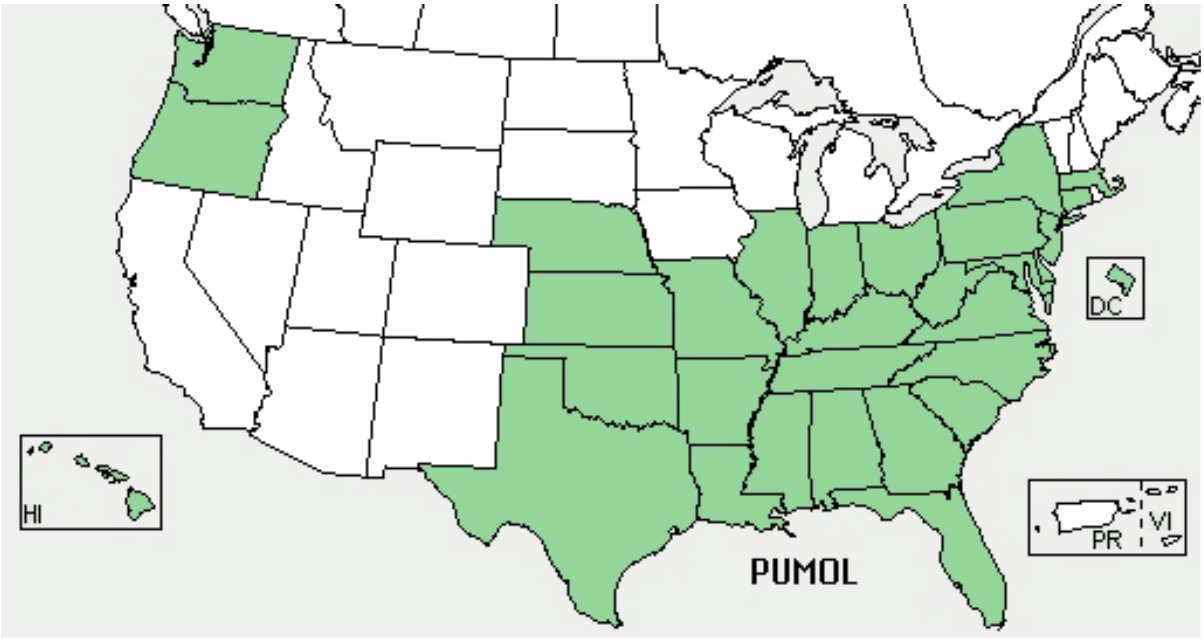

Figure 1 Distribution map of kudzu across the United States, in the USDA PLANTS database http://plants.usda.gov/core/profile?symbol=PUMO

Kudzu is present in Oklahoma but has not been the focus of a targeted survey in the state. Because of the variability in data, maps illustrating the distribution of kudzu are inconsistent among sources (e.g., state and national agencies). Thus, a need for an updated map has arisen for future management of the species. For instance, the Oklahoma Vascular Plant Database map, whose data are based on herbarium records, indicates 22 counties with kudzu (Fig. 2), while a map from Early Detection and Distribution Mapping Systems (EDDMapS) includes 12 counties

(Oklahoma Vascular Plant Database 2014; EDDMaps 2014). While some of the occurrences overlap, there are some inconsistencies. Importantly, none of these maps are based on a compilation of reliable field observations and specimens that have been critically examined by experts. Thus, we attempted to confirm previously known locations, obtain information about new sightings, and collect specimens for confirmation. A survey was utilized to cover Oklahoma as a whole and to gather as much information as possible about the plant from knowledgeable persons primarily within the Oklahoma State University Extension Service. Surveys have been found to be a useful tool when other forms of data sources or collection methods are not adequate, and in this case it was not practical to reach as many people through other methods (Innovation Insights 2006). Survey reports were then confirmed by groundtruthing and utilized to create a detailed map of kudzu locations and the extent of invasion at each site.

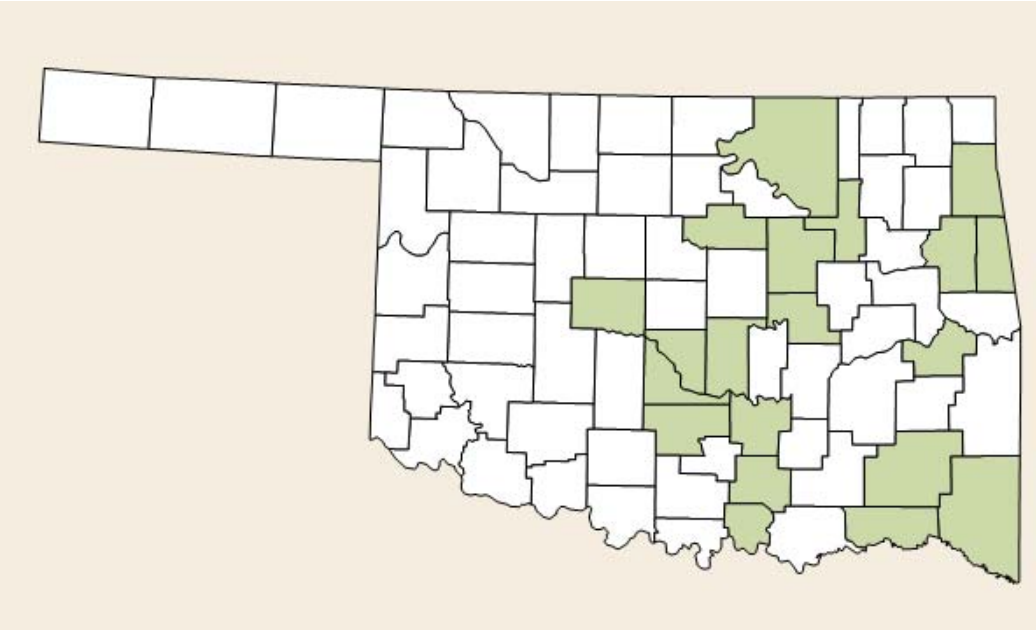

Figure 2 Oklahoma Vascular Plant Database map of kudzu occurrence by county http://www.oklahomaplantdatabase.org

METHODS

Kudzu location, extent of invasion, and date of record were obtained from available records, which included the OVPD records of herbarium specimens, information collected by the Oklahoma Invasive Plant Council (OkIPC; K. Hickman, unpublished), directed contact with botanists in the state, and through a survey sent to OSU Extension personnel, land managers known to have experience with kudzu, Oklahoma Department of Wildlife Conservation employees, and the OkIPC. The survey provided the majority of data collected.

A link to the kudzu survey, which was created through Survey Monkey (www.surveymonkey.com), was sent out through email. Five questions were asked regarding the respondent's knowledge of kudzu and its presence in Oklahoma.

Questions asked in the survey included: 1) What county of Oklahoma are you currently working or residing in? 2) Have you seen or heard of Kudzu inhabiting land in Oklahoma? 3) If so, please provide the locations of the kudzu sightings. 4) In acres, how large of an area would you estimate that the infestation is at each site? 5) Please provide contact information for verification and/or additional inquiries. Approximately two hundred invitations were emailed to OSU County Extension offices, Oklahoma Department of Wildlife Conservation, Oklahoma Department of Transportation, and the Oklahoma Invasive Plant Council members. These agencies and organizations were chosen based on previous experience we have had with them concerning invasive species and the ability to send mass emails to the group. Also, individuals were included who had knowledge of Oklahoma vegetation and ecosystems, or who dealt with invasive species frequently.

We used ArcGIS ArcMap v. 10.1 (Esri, Redlands, CA) software to create distribution maps. A state overview illustrating counties with kudzu present was created, along with more detailed maps of the counties displaying extent of the invasion of kudzu. Estimates of the extent invaded were made on sites (19), approximated from GoogleEarth imagery (4), or reported in the surveys (5). Points were added to the map for individual stands of kudzu across Oklahoma, illustrating area invaded for each location within the county. For our map, we included sites that were confirmed to have kudzu; we did not include locations of kudzu that we visited and confirmed kudzu was not present. Mapped points (Table 1) only include confirmed locations of kudzu, but not sites in OVPD that were not confirmed via a visit or sites visited where no plants were found.

Samples of kudzu were collected from all confirmed sites visited (16) to create herbarium voucher specimens. We traveled to some, but not all of the locations, due to time constraints of the project (see Table 1). Sites chosen to visit were those with larger infestations reported or those reported in the survey. Samples of individual plants were cut in sections including leaves, flowers, and pods (if available, as samples were taken throughout the project year). Specimens were deposited at the University of Oklahoma's Bebb Herbarium (OKL).

RESULTS

The survey received 52 responses from the approximately 200 emails sent, which indicates a return rate of close to 25%. Of those, over 50% (28) respondents had knowledge of kudzu in Oklahoma, while 46% (24) reported having not seen or heard of the vine's encroachment within the state. Of those surveyed, 17 provided locations, and 10 estimated dimensions of the area invaded of kudzu. Of those reported, 9 locations were new, previously unrecorded sites of kudzu.

Maps (Figs. 3, 4) were created using data from the survey and previously known locations (confirmed by groundtruthing) of kudzu (see Table 1). If kudzu was confirmed as absent from a site, then it was removed from the map. A gray scale was utilized to illustrate the extent of invasion of kudzu in each county. Figure 3 presents specific locations of kudzu in the state along with their corresponding extent of invasion, while Figure 4 illustrates presence and extent by county. Based on our results, at least 32.4 hectares of land are invaded by kudzu in Oklahoma across 28 sites.

Table 1 Locations of kudzu identified from previous documentation (Oklahoma Vascular Plants Database), survey results, or on-site discoveries. Kudzu was confirmed present or absent via site visits or previous documentation. Estimates of the extent of kudzu invasion were obtained during on-site visits using GPS or GoogleEarth imagery.

Site Name	Longitude	Latitude	Source of Location Data	Source of Extent of Invasion	Status of Kudzu on Site
Idabel	-94.709	33.896	Discovered by Marli Claytor	Google Earth to estimate coverage	Confirmed present
Claremore	-95.599	36.299	From survey	Site Visit	Confirmed present
Antlers	-95.637	34.233	Discovered by Marli Claytor	Google Earth to estimate coverage	Confirmed present
P St. & Springdale Rd. , Ardmore	-97.108	34.159	Previous documentation	Site visit	Confirmed present
Marsden Rd. Love Co.	-97.195	34.070	Site visit	Site visit	Confirmed present
Tater Hill Rd. Ardmore	-97.008	34.144	Previous documentation	Site visit	Confirmed present
Shawnee	-96.962	35.333	Previous documentation /survey	Google Earth to estimate coverage	Confirmed present
Haskell	-95.611	35.754	From survey	Google Earth to estimate coverage	Confirmed present
Eufaula	-95.339	35.281	Previous documentation	Site visit	Confirmed present
Cleveland County	-97.164	35.233	Previous documentation /survey	Site visit	Confirmed present
Dickson	-96.928	34.188	From survey	From survey	Inconclusive
Red River	-95.500	33.877	Previous documentation	Unavailable	Inconclusive

Untitled Placemark-Hulbert	-95.226	35.869	Previous documentation	Site visit	Confirmed present
Shoals	-95.398	33.968	Previous documentation	Site visit	Inconclusive
North Eufuala	-95.387	35.391	Previous documentation	Site visit	Confirmed present
Norman	-97.156	35.232	Previous documentation / from survey	Site visit	Confirmed present
Okemah	-97.399	35.430	Previous documentation	Site visit	Confirmed present
Washita River Tributary	-97.510	34.779	Previous documentation	Site visit	Confirmed present
Fittstown	-96.635	34.618	Previous documentation	Site visit	Confirmed present
Durant	-96.410	34.056	Previous documentation	Site visit	Confirmed present
Duncan	-97.986	34.594	Previous documentation	Site visit	Confirmed present
Stillwater	-97.063	36.113	Previous documentation	Site visit	Confirmed present
Osage	-96.304	36.242	From survey	From survey	Confirmed present
Osage	-96.282	36.246	From survey	From survey	Confirmed present
Adair			From survey	Unavailable	Inconclusive
Caddo	-98.324	35.464	Previous documentation		Confirmed absent
Marshall	-96.685	34.148	Previous documentation		Confirmed absent
Pontotoc	-96.634	34.579	Previous documentation		Confirmed absent

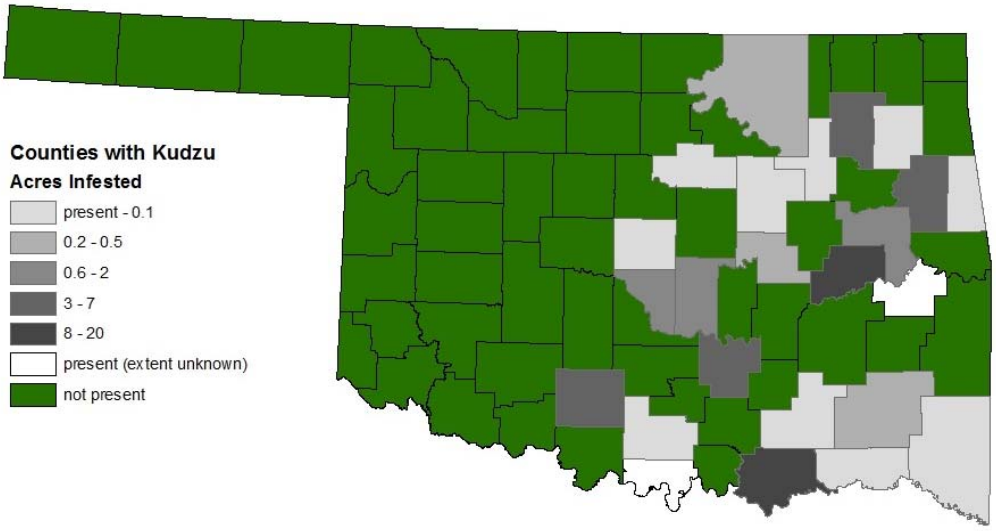

Figure 3 Distribution map of counties with confirmed kudzu invasion, showing acres invaded. Acres represent total acres for all sites within each county. Map created using ArcMap.

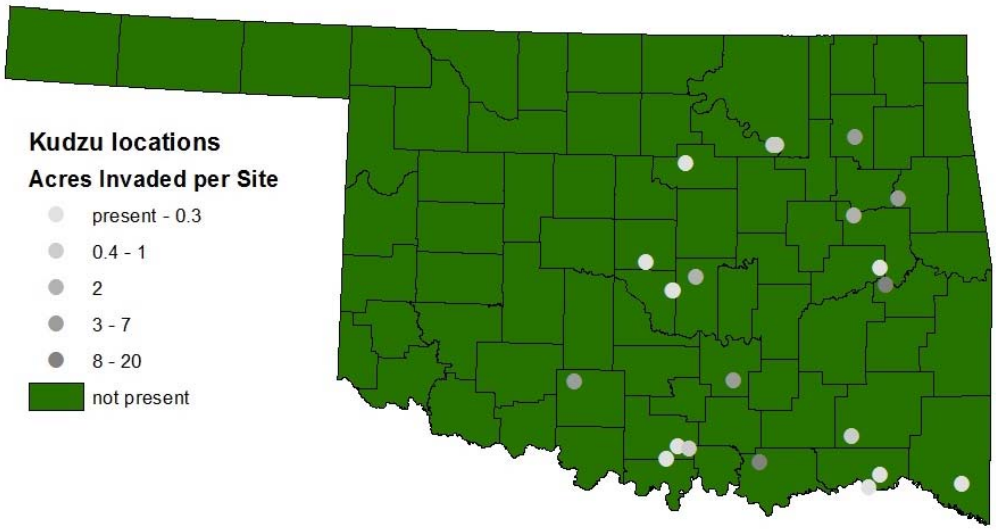

Figure 4 Locations of kudzu across the state of Oklahoma, featuring area invaded for each site. Map was created using ArcMap programming with data from the survey.

DISCUSSION

The survey was successful in acquiring important information on kudzu throughout the state. Nearly half of the respondents had no knowledge of kudzu being present in the state, which indicates very little familiarity with the vine even from knowledgeable professionals. Close to 30% of all sightings reported were new locations in the state. This prompts the question: if we had sent out more surveys, how many more new locations would have been documented?

The new distribution map aids in assessing current and future invasion of kudzu. In comparison to the OVPD, our map includes 22 counties reported while the other has only 20; additionally, not all OVPD counties are included in the new map as some reports could not be confirmed or old populations were found to no longer exist as determined through our site visits (see Figs. 2, 3). It can be observed that kudzu currently exists primarily in the eastern portion of the state. Climatic restrictions are most likely limiting the range of kudzu (Jarnevich and Stohlgren 2009. Once kudzu has invaded an ecosystem it is very difficult to eradicate, further facilitating its spread across Oklahoma. It is likely that kudzu will continue not only its coverage north, but also invade more hectares where stands currently persist (Jarnevich and Stohlgren 2009).

Currently there are at least 32.4 hectares invaded with kudzu in Oklahoma, which is extremely small in comparison to the total seven million hectares invaded in the United States (Eskridge and Alderman 2010). This does not mean we can ignore the problem, but presents our state with an opportunity to stop a problem while we can. If our state began an Early Detection and Rapid Response (EDRR) program for kudzu, it would be possible to limit the future spread of the vine and keep our state and economy safe from the detriment of invasion. EDRR programs work to develop a system of effectively addressing issues of invasive species through the steps of: early detection and reporting of new plants, identification and collection of specimens, verification of new plant records, archival of new records where appropriate, rapid assessment of new records, and rapid response to new records determined to be invasive (Westbrooks 2004). To stop this problem now would save the state financially in the long run. More studies need to be conducted on kudzu, and there is a current study on viability of kudzu seeds in Oklahoma (Zoeller and Hickman, unpublished). This study will be crucial in estimating to what extremes kudzu can further invade Oklahoma.

Education for the state needs to occur to stop the further expansion of kudzu. The creation of our updated map will aid in educating citizens on where the vine resides and if they should be on alert for presence in their area. To inform the public, the first step will be to train county and state officials to properly identify kudzu and instruct citizens on how to handle the issue. Kudzu has caused major damage in the southeastern United States, but this destruction can be reduced through proper education and effectively implementing an EDRR program.

ACKNOWLEDGEMENTS

We would like to thank those who participated in the survey or contributed a location. Thanks to Oklahoma State University Natural Resource Ecology and Management Department and also the Oklahoma State University Honor's College. Finally, we would like to thank the others who helped complete this project: Aaron Cromer, Dwayne Elmore, Gail Wilson, and Mark Gregory.

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Critic's Choice Essay

MISTLETOE, *PHORADENDRON SEROTINUM* (RAF.) JOHNSTON

Reprinted from *Gaillardia*, Spring 1993

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Every Oklahoma child quickly becomes familiar with the common mistletoe, the green leaved growth on naked branches of large trees in mid-winter. This native plant occurs over most of the state and is particularly popular as one of the year-end holiday decorations. We all know it is permissible to steal a kiss from someone standing "under the mistletoe."

Although considered by many to be a parasite, in reality the plant is only semi-parasitic. It does obtain water, minerals, and perhaps some proteins from the host, but it is able to carry out photosynthesis and therefore produce most of its own food. In spite of the plant invading its tissue, the host is seldom harmed, unless of course there is a very heavy infestation.

Just 100 years ago in February 1893, mistletoe became the floral emblem of the Territory of Oklahoma. In 1909, the Second State Legislature conferred the same designation for the State of Oklahoma. The following explanation for its selection appeared in the *Chronicles of Oklahoma*, the publication of the Oklahoma Historical Society.

Tradition has it that the first grave made in Oklahoma country in the winter after the Opening of 1889 was covered with mistletoe since there were no other floral offerings in the new country except the green of the mistletoe with its white berries growing in great clusters on the elms along the dry creek beds and branches. All through the winter, the green bank of the lonely grave could be seen far across the prairie against the sere brown grass or the melting snow of early spring. Thus, the mistletoe became associated with sacred thoughts among the pioneer settlers.

In Oklahoma, mistletoe is most commonly associated with *Ulmus americana* (American elm), a species which has been badly ravaged by Dutch elm disease, a fungus with tissue choking the water translocating tissues. Mistletoe may also be found on hackberries, oaks, maples, ash, sycamore, and other native deciduous trees. This is fortunate; otherwise, the species might well become a candidate for rare or endangered status.

The plants are dioecious (unisexual: staminate and pistillate flowers on different individuals). Flowers are about 2 mm across, without petals, and borne on spike-like stalks from the bases of the leaves. The fruit, which are readily consumed by birds, are whitish, mucilaginous, one-seeded drupes, appearing during the winter. It has been suggested that dispersal takes place when the sticky seeds are "glued" to a twig as a bird wipes its bill, or the ingested, but unharmed, seeds are deposited on a limb with fecal material.

https://doi.org/10.22488/okstate.17.100117

Used as a medicinal plant by Indians and pioneers, a tea was prepared to relax nervous tension and muscle irritability and to increase blood pressure. Other uses were to lessen bleeding, promote clotting, stimulate uterine contraction, and arrest postpartum hemorrhage. However, caution is advisable. Like virtually all medications, mistletoe can be poisonous under certain conditions such as improper dosage levels, sensitive individuals, or with the very young, elderly, or feeble. There is no reliable information on safe dosages. Although consumption of the fruit is harmless to pigs, 13 Hereford cattle, forced to consume the plant when their pasture was reduced, died within 10 hours after the onset of symptoms. Death was due to collapse of the cardiovascular system. Several deaths among children, having consumed the fruit, have been documented.

Such is the state's floral emblem, the Oklahoma mistletoe, *Phoraendron serotinum* — an interesting, beneficial, and potentially dangerous member of our native flora.

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Common names should be referenced to a scientific name using nomenclature that has been revised according to the Integrated Taxonomic Information Service (ITIS) database (http://www.itis.gov). Abbreviations of authorities for scientific names should follow *Authors of Plant Names* (Brummitt and Powell 1992). Titles of periodicals should be abbreviated following Botanico-Peridoicum-Huntianum and its supplement, except in historic publications when original format may be used.

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In this issue of Oklahoma Native Plant Record Volume 15, December 2015: