Survival of *Rhizoctonia solani* and Other Soilborne Basidiomycetes in Fallow Soil

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ABSTRACT

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Soil in 6,581-cm^3 clay pots was infested with isolates of *Rhizoctonia solani* in AG-1–AG-4, binucleate *Rhizoctonia*-like fungi in CAG-2–CAG-5, *R. zeae*, and *Laetisaria arvalis* and buried to within 5 cm of the pot rim for almost 40 wk of incubation in the field. Soil was sampled at infestation and 86, 211, and 283 days after infestation and plated on a medium selective for soilborne basidiomycetes. One of four isolates of AG-1, one of six AG-2-2, one of three AG-2-1, and eight of 14 isolates of AG-4 were recovered from fallow soil after 283 days. Isolates of AG-3 were not recovered after 86 days. One of two isolates of CAG-3 and one of two CAG-4 and a single isolate of CAG-5 were reisolated from soil after 283 days. Isolates of CAG-2 were not reisolated after 211 days. All isolates of *R. zeae* and a single isolate of *L. arvalis* were recovered from soil after 283 days. Snap bean and corn were planted in each pot of fallow infested soil and a pot of fallow control soil after 283 days. Isolates of AG-2-2 caused crown and brace root rot in corn, and isolates of AG-4 and AG-2-2 and one isolate of CAG-3 caused root and hypocotyl rot in snap bean. Surviving isolates of other fungi were not pathogenic on snap bean or corn, except for one isolate of AG-2-1 that was weakly virulent on snap bean.

Anastomosis groups (AG) 1, 2, and 4 of multinucleate *Rhizoctonia solani* Kühn and binucleate *Rhizoctonia*-like fungi (CAG) 2, 3, 4, and 5 occur naturally in soils and plants of Georgia's coastal plain (3,4,13,17,19,20). The pathogenesis and virulence of isolates of AG-1, AG-2 type 2 (AG-2-2), AG-2-1, AG-4, and CAG-3 and CAG-5 to crops economically important in the Georgia's coastal plain has been established (1,13,17,18–20). In addition, *Rhizoctonia zeae* Voorhees is pathogenic to corn (*Zea mays* L.) roots (19).

Isolates of *Laetisaria arvalis* Burdsall, CAG-2 and CAG-4, from local soil or plant tissues (19,20) in previous studies were antagonistic to *R. solani* AG-4 and AG-2-2 in soil planted to snap bean (*Phaseolus vulgaris* L.) and corn (5).

An important factor common to all the fungi mentioned above is the necessity for survival as temporary or prolonged inhabitors of soil. In soil, plant debris is saprophytically colonized by *R. solani* (6,14) and the fungus may survive several months in plant tissues depending on environmental conditions (3,10). Soil solarization is an effective method of disinfesting soil in warm, dry climates (11), and soil temperatures 10 cm deep in Georgia's coastal plain frequently exceed 35 C from June to September in fallow soil (2). Our objective in this study was to determine the survival of these fungi in a fallow soil of Georgia's coastal plain during about 40 wk of incubation in the field.

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MATERIALS AND METHODS

Two experiments were conducted, the first beginning in March 1978 and the second in December 1980. In the first experiment, two isolates of *R. solani* in AG-4, one each from a corn root and a southern pea (*Vigna unguiculata* (L.) Walp.) hypocotyl, two isolates of CAG-2, one each from a southern pea hypocotyl and an onion (*Allium cepa* L.) bulb, and nine isolates not identified to AG or CAG were used. The identification and source of fungal isolates used in experiment 2 are listed in Table 1. Similar methods were used in both experiments, so only the second experiment is described in detail.

Each isolate was grown on 3% (w/w) cornmeal-sand for 12–14 days. Clay pots were used as containers for infested soil. Pots were 25 cm wide top inside diameter (i.d.) × 17 cm wide bottom i.d. ($\bar{x}$ i.d. = 21 cm) × 19 cm inside height (volume = 6,581 cm^3), with 1-cm-thick walls and a drainage hole in the bottom. For each isolate, 100 ml of cornmeal-sand inoculum ($\bar{x}$ = 0.04% (w/w) organic matter) was mixed with 6 L of moist (7.8% w/w) heat-treated (77 C for 30 min with aerated steam) Dothan loamy sand (about 85, 11, and 4% sand, silt, and clay, respectively), with an organic matter content of 0.54% (w/w) and a pH of 5.7, and placed in a single clay pot. A separate pair of plastic gloves was worn to prepare the control and infest soil with each isolate.

Pots of soil were buried on 1.2-m centers in a plowed, harrowed, and smoothed field plot, leaving about 5 cm of the pot rim above ground. Each pot was watered with 140 ml of well water. Thereafter, the infested potted soil and plot area were exposed to natural weathering. The pots were arranged in a completely random design. Because one pot of soil was infested per isolate, statistical analysis of data was not possible.

Small weeds in the plot area were sprayed as needed with glyphosate. Herbicide contamination of artificially infested soil was prevented by covering each clay pot with a larger plastic pot and directing sprays close to the soil surface. Infested soil inside the clay pots was hand-weeded when weeds were tall enough to grasp and remove without contacting the soil. Each rainfall was recorded and minimum-maximum soil temperatures were recorded weekly in the upper 10 cm of soil in a clay pot adjacent to the experiment.

The soil in the second experiment was infested and potted on 1 December and the field plot area was established on 2 December 1980. A 100-ml sample of soil infested with each of the 41 isolates in Table 1 and a noninfested control were collected at infestation to determine initial inoculum concentration. Additional soil samples were collected for assay 86, 211, and 283 days after preparation. Three soil cores 2.5 × 10 cm were collected from each pot on each sampling date. Soil cores from each pot were mixed, moistened to about 10% (w/w), and placed on tannic acid-benomyl agar (TABA) (19) with a multiple-pellet soil sampler (9). Fifteen pellets of 100–120 mg were deposited on agar in each of five 9-cm-diameter petri dishes to determine initial inoculum concentration. At later assays, 10 or 15 petri dishes of TABA were prepared from each sample. Populations were calculated as colony-forming units (cfu) per 100 g of oven-dried soil. Petri dishes of soil were incubated in the dark at 26 ± 1 C for 48 hr and colonies of the various fungi enumerated. Hyphal tips of colonies selected at random were transferred to fresh PDA and potato-dextrose/yeast extract/casein hydrolysate agar (PDYCA)

(19) to confirm identification of the various fungi.

At the last sampling, soil in each pot was mixed with fertilizer (17, 34, 51, and 189 μg/g of N, P, K, and $CaCO_3$, respectively), and 150 ml of soil was collected at random from each pot. Five seeds each of Funk's G-4507 corn and Eagle snap bean were planted in each pot on 10 September 1981 and watered.

In the first test, 10 Eagle snap bean seeds were planted per pot, and all plants were removed from each pot after 2 wk for disease indexing in August 1978, March 1979, and September 1979 (6, 12, and 18 mo, respectively, after the soil was infested). Soils were assayed on TABA just before each crop was planted. Seeded crops and emerging and expanding seedlings were watered as needed. The snap bean and corn plants in each pot of the second experiment were counted and harvested 14 days after planting, when the first trifoliate leaves were expanding in snap bean and corn was in the three-leaf stage. Root-hypocotyl and root-mesocotyl disease indices were determined for snap bean and corn, respectively, using a weighted visual scale where 1 = <2, 2 = 2–10, 3 = 11–50, 4 = >50% discoloration and necrosis, and 5 = dead plant. In this scale, 1 = an avirulent isolate, 2 = a weakly virulent isolate, 3 = a moderately to highly virulent isolate, and 4 or 5 = a highly virulent isolate.

Lesions were removed from diseased plants, surface-disinfested 15–30 sec with 0.5% NaOCl (w/v), and incubated on water agar in petri dishes. Hyphal tips were transferred to PDA and PDYCA and identified.

All cultures of *R. solani* and *Rhizoctonia*-like binucleate fungi were identified to anastomosis group with known tester isolates (15). *L. arvalis* and *R. zeae* were identified by morphological characteristics in culture (7,19).

Table 1. Survival of soilborne basidiomycetes in fallow soil near Tifton, GA

Anastomosis groups (AG) and *Rhizoctonia solani* isolates	Source	Population[a] (cfu/100 g soil) At infestation	After 86 days	After 211 days	After 283 days	After 297 days RHDI[b] (snap bean)	RMDI[b] (corn)
AG-1							
Rhs-8	Grain sorghum foliage	810	65	0	12	2.0	1.0
Rhs-10	Peanut foliage	282	0	0	0	1.0	1.0
Rhs-12	Rice sheath (Texas)	528	275	0	0	4.0	1.0
Rhs-13	Lima bean foliage	528	664	59	0	2.0	1.0
AG-2-2							
Rhs-35	Peanut seed	258	16	0	0	4.7	2.8
Rhs-36	Corn brace root	434	8	23	0	4.0	2.6
Rhs-37	Corn brace root	410	49	0	0	2.3	1.4
Rhs-38	Corn brace root	270	0	0	0	3.0	2.0
Rhs-39	Corn brace root	0	8	0	9.4	1.0	1.0
Rhs-44	Corn brace root	305	40	8	0	4.3	3.3
AG-2-1							
Rhs-33	Peanut seed	287	24	0	0	1.0	1.0
Rhs-34	Peanut seed	575	113	0	0	1.0	1.0
770	Corn brace root	822	550	174	94	2.0	1.0
AG-3							
Rhs-63	Potato tuber (Idaho)	868	348	0	0	1.0	1.0
Rhs-64	Potato tuber (Maryland)	822	97	0	0	1.3	1.0
AG-4							
Rhs-93	Cucumber root	845	437	101	70	4.3	1.0
Rhs-97	Wheat root	481	219	151	5	4.3	1.0
Rhs-98	Soybean root	12	0	0	0	1.2	1.0
Rhs-100	Tomato fruit	458	81	40	9	1.2	1.0
Rhs-103	Peanut lateral branch	798	470	13	0	2.8	1.0
Rhs-107	Peanut seed	880	389	30	0	1.0	1.0
Rhs-109	Peanut seed	35	8	0	19	5.0	1.6
Rhs-110	Corn brace root	270	0	0	0	2.3	1.0
Rhs-111	Corn brace root	657	470	47	0	1.3	1.0
Rhs-112	Corn fibrous root	470	381	40	14	1.4	1.0
Rhs-113	Corn fibrous root	622	421	0	0	1.0	1.8
SRB-2	Peanut seed	880	405	83	14	3.3	1.0
114	Pole bean hypocotyl	680	405	20	5	1.3	1.0
2634	Soil under corn	798	459	94	42	3.5	1.0
CAG-2							
BN-2	Arrow leaf clover root	540	599	303	0	1.0	1.0
BN-5	Onion root	822	607	47	0	1.0	1.0
CAG-3							
BN-6a	Peanut shell	857	235	168	192	3.7	1.0
BN-8a	Cucumber root	376	178	0	0	1.3	1.0
CAG-4							
BN-9	Corn fibrous root	305	0	0	0	1.4	1.0
BN-12	Soybean root	481	348	40	5	1.5	1.0
CAG-5							
BN-14a	Cucumber root	880	445	121	33	1.5	1.0
R. zeae							
899-II-4H	Soil under corn	845	526	525	148	2.3	1.0
899-IV-2H-1	Soil under corn	493	429	357	253	1.8	1.0
899-IV-12N	Soil under corn	481	405	263	113	1.3	1.0
899-IV-3H	Soil under corn	646	470	276	148	1.0	1.2
Laetisaria arvalis							
899-IV-8H	Soil under corn	880	348	694	626	1.0	1.0
Control		0	0	0	0	1.0	1.0

[a] Colony-forming units per 100 g of oven-dried soil.
[b] Root-hypocotyl disease index and root-mesocotyl disease index derived from a visual weighted scale: 1 = <2, 2 = 2–10, 3 = 11–50, and 4 = >50% discoloration or necrosis and 5 = dead plant.

RESULTS

Experiment 1. Populations of one of the two isolates of AG-4 averaged 429, 437, and 157 cfu/100 g of soil 0, 43, and 183 days after preparation, respectively. The isolate caused moderate to severe reddish brown, sunken cankers on roots and hypocotyls of Eagle snap bean planted 31 August, and cultures of *R. solani* in AG-4 were reisolated from snap bean. The second isolate of AG-4 averaged 692 cfu/100 g of soil at infestation but was not detected after that. In snap bean, only light root and hypocotyl discoloration was observed, and the fungus could not be isolated from these tissues. Four of nine unidentified isolates caused moderate to severe root and hypocotyl rot in snap bean, and populations in soil averaged 429, 255, and 76 cfu/100 g of soil 0, 43, and 183 days after preparation, respectively. Populations of the two isolates of CAG-2 averaged 61 cfu/100 g of soil at planting. One isolate averaged 16 cfu/100 g of soil 183 days after preparation, but the other isolate was not detected. The surviving isolate of CAG-2 caused a light superficial tan to reddish brown discoloration in cortical tissue on snap bean but did not cause cankers.

After snap bean was removed, the soil in each pot was mixed on 28 September and left until the following March. Soils were reassayed and snap bean was replanted. Populations of the AG-4 isolates and the four unidentified isolates that caused disease the previous August

averaged 13 cfu/100 g of soil. The isolate of AG-4 and three unidentified isolates caused slight to moderate root and hypocotyl disease with reddish brown, sunken cankers and were reisolated from snap bean. In the final soil assays 18 mo after preparation, AG-4 was not detected and only one of the unidentified isolates was recovered at 5 cfu/100 g of soil. Root and hypocotyl disease severity was slight to moderate in all treatments. *Fusarium solani* (Mart.) Appel. & Wr. f. sp. *phaseoli* (Burk) Snyd. & Hans. and *Pythium myriotylum* Drechs. were isolated more frequently than *R. solani* from diseased plants.

Experiment 2. Three of four isolates of AG-1 were recovered from fallow soil 86 days after infestation, and different single isolates were recovered after 211 and 283 days (Table 1). Populations decreased rapidly from infestation to 283 days. Isolate Rhs-12 (AG-1), incitant of rice (*Oryza sativa* L.) sheath blight in Texas, caused a high root-hypocotyl disease severity on snap bean. The other isolates of AG-1 were avirulent or weakly virulent to snap bean, and none were pathogenic to corn (Table 1).

One isolate of *R. solani* in AG-2-2 and one isolate of AG-2-1 were recovered from fallow soil 283 days after infestation. Isolate Rhs-39 (AG-2-2) was not recovered at preparation, but a culture of AG-2-2 closely resembling Rhs-39 was isolated from fallow soil after 86 and 283 days. The root-hypocotyl disease index of snap bean and root-mesocotyl disease index of corn were slight to severe with isolates Rhs-35-38 and Rhs-44 of AG-2-2. Isolate Rhs-39 (AG-2-2) and isolates Rhs-33 and Rhs-34 (AG-2-1) were avirulent to both hosts, and isolate 770 (AG-2-1) was weakly virulent to snap bean (Table 1).

Two isolates of AG-3 from black scurf on potato (*Solanum tuberosum* L.) tubers, one isolate each from Idaho and Maryland, were recovered from fallow soil at 86 days but not at 211 or 283 days after infestation, and neither isolate was pathogenic to snap bean or corn (Table 1).

Isolates of AG-4 had a wide range of survival in fallow soil. The decrease in populations through sampling dates at 86, 211, and 283 days after infestation was more closely related to the initial inoculum concentration than many isolates in other AGs (Table 1). From 14 isolates of AG-4, 12, 10, and 8 were recovered 86, 211, and 283 days, respectively, after infestation. Isolates Rhs-98 with 12 and Rhs-110, with 270 cfu/100 g of soil at infestation, were not recovered later, and these isolates, respectively, were avirulent and weakly virulent to snap bean and both isolates were avirulent to corn. However, isolates Rhs-97, Rhs-109, SRB-2, and 2634 with 5, 19, 14, and 42 cfu/100 g of soil, respectively, were highly virulent to snap bean. The other isolates of AG-4 were weakly virulent to snap bean, and all isolates of AG-4 were avirulent to corn (Table 1).

The inoculum density of isolate BN-2 (CAG-2) was higher after 86 days than at infestation, and a high population was recovered after 211 days (Table 1). Isolate BN-5 (CAG-2) also maintained a high population after 86 days, decreased sharply after 211 days, and the isolates of CAG-2 were not recovered after 283 days. The isolates of CAG-2 were nonpathogenic to snap bean or corn. Isolate BN-6a (CAG-3) maintained a high population throughout the test and was highly virulent to snap bean but avirulent to corn. Isolate BN-8a (CAG-3) was recovered only at infestation and after 86 days, and this isolate was avirulent to snap bean and corn. Isolate BN-9 (CAG-4) was not recovered after infestation and was nonpathogenic to both hosts. A high population of isolate BN-12 (CAG-4) was recovered at infestation and after 86 days, then decreased sharply through 211 days after infestation. Isolate BN-12 was avirulent to snap bean and corn. Isolate BN-14a (CAG-5) had a high population (cfu/100 g of soil) at infestation that decreased about 50% after 86 days and maintained high to moderate populations after 211 and 283 days. Isolate BN-14a was avirulent to snap bean and corn (Table 1).

All isolates of *R. zeae* maintained high populations throughout the test. Isolates 899-IV-2H-1, 899-IV-12N, and 899-IV-3H were avirulent and isolate 899-II-4H was weakly virulent to snap bean. Isolates of *R. zeae* were avirulent to corn (Table 1). Populations of *L. arvalis* were high throughout the test, with recovery 211 and 283 days after infestation being greater than recovery after 86 days. *L. arvalis* was nonpathogenic to both hosts (Table 1).

Based on morphological characteristics, isolations from discolored or necrotic tissue yielded cultures considered to be the original isolates. One isolate of *R. solani* in AG-1 (one culture) was recovered from snap bean and none from corn. Four isolates of AG-2 (11 cultures of AG-2-2 and five of AG-2-1) were reisolated from snap bean and four (15 cultures of AG-2-2 and three of AG-2-1) from corn. Isolates of AG-3 were not recovered from either host. Two isolates of AG-4 (five cultures) were reisolated from snap bean and one (three cultures) from corn. Isolates of CAG-2, CAG-3, CAG-4, and CAG-5 were not recovered from either host. Two isolates of *R. zeae* (two cultures) were reisolated from corn and none from snap bean. *L. arvalis* was not recovered from either host. No discolored or diseased plants occurred in the control. One culture of *R. zeae* was isolated from corn roots in the control, but nothing was isolated from snap bean.

There were large variations in inoculum concentrations among samples collected at infestation. This was more prevalent in isolates of *R. solani* AG-1, AG-2, AG-4, and *R. zeae*, where there were more isolates per group. However, substantial initial variations in populations also occurred between two isolates in each of CAG-2, CAG-3, and CAG-4.

The minimum temperature range in the upper 10 cm of soil during experiment 2 was −4–22.3 C, and the maximum range was 23.6–40.4 C, and 74.1 cm of rain fell on the infested potted soil. During the test period, total moisture deficit (rainfall − net evaporation) exceeded rainfall by 40.0 cm. In 1981, large moisture deficits of −13.5, −14.7, −16.3, and −10.9 cm occurred during April, May, June, and July, respectively; however, there was an 8.9-cm surplus in August.

DISCUSSION

The combined effects of temperature extremes and prolonged drought created a harsh environment for survival of fungi in soil in the absence of either host or adequate substrate. The survival of numerous isolates of *R. solani* and other basidiomycetes in soil indicates that fallowing fields for one summer would reduce inoculum densities but would not eliminate pathogens or saprophytes in undisturbed soil. Soil solarization with a plastic cover would have increased soil temperatures (11) and eliminated more isolates. Also, keeping the soil continuously wet by irrigation has hastened propagule decline (8).

All of our isolates of *R. solani* in AG-1 corresponded morphologically with at least one of Sherwood's (16) three sclerotial types of AG-1. Isolates of AG-1 collected in Georgia's coastal plain, and one isolate from Texas, were from necrotic foliar tissue. This agrees with the reported infection and disease loci of Parmeter et al (15) for AG-1. In our study, isolates of AG-1 were reisolated from heat-treated soil artificially infested with these fungi and from one snap bean plant growing in this soil (Table 1).

On host plant foliage (1,13) and in culture (16), isolates of AG-1 produced copious quantities of sclerotia varying in size from 0.4 to 9.0 mm in diameter, depending on the sclerotial type of the isolate (16). Our isolates of AG-1 produced numerous sclerotia in cornmeal-sand, but the isolates most likely survived in the fallow soil as free sclerotia because of the low organic matter content (<1%) in the soil.

We did not isolate a culture of AG-1 from soil before this study. Although isolates of AG-1 may vary from temporary to prolonged inhabitors of local soils, this group of fungi requires special conditions of temperature, moisture, and humidity (1,16) to become serious foliar pathogens. Therefore,

compared with AG-4 and AG-2-2, AG-1 is of minor importance to crops grown in open fields in Georgia's coastal plain.

One of the two isolates of *R. solani* in AG-3 used in our study was obtained from Idaho and the other from Maryland. These isolates came from cool climates and may have succumbed to heat and drought stresses present in the fallow soil. Only one of nine of Sherwood's isolates of AG-3 grew at 28 C in culture on PDA (16).

Isolates of AG-3 in the United States have been primarily associated with potato plants and tubers (15,16), and this crop is rarely grown commercially in Georgia's coastal plain. We have not isolated a culture of AG-3 from field soils in this area. Five cultures of AG-3 were isolated (21 May 1981) from black scurf sclerotia on five potato tubers from an irrigated home garden near Tifton, GA. These cultures readily anastomosed with tester isolates of AG-3. After the preceding incident, cultures of AG-3 were isolated from black scurf sclerotia on locally purchased tubers grown in two other states (D. K. Bell, *unpublished*). To our knowledge, black scurf has not recurred in Georgia's coastal plain since 1981. The present unimportance of AG-3 in this area, especially in home gardens, may result from the absence of a susceptible host, adverse conditions of moisture or temperature, or misidentification or no identification of disease caused by this AG of fungi.

Severe crown and brace root rot of corn was observed in irrigated fields in Georgia's coastal plain. Cultures of AG-2-2, the primary pathogen, and AG-2-1 were isolated from severely diseased lateral roots on lodged plants 2–6 wk after tasseling. Cultures of AG-2-2 or AG-2-1 were rarely isolated from diseased corn roots from nonirrigated fields (19). Growth of isolates of AG-2-2 and AG-2-1 may become quiescent during prolonged moisture deficits. All of our isolates of AG-2-2 survived large moisture deficits in fallow soil from April through July, and most isolates caused disease in snap bean and corn planted 9.4 mo after infestation of soil.

In pathogenicity tests conducted in growth chambers, isolates of AG-2-2 and AG-2-1 were equally highly and weakly virulent, respectively, on 2- to 5-wk-old corn plants at temperature ranges of 8–21, 16–18, and 20–34 C (19). These temperatures were within the minimum and maximum soil temperature ranges of our study, and they also encompass soil temperatures often encountered during production of snap bean and corn in Georgia's coastal plain.

In a field of Bonifay sand (about 95–98% sand) naturally infested with *R. solani*, cultures of AG-2-2 and AG-2-1 were isolated from 0.1% and 0.03%, respectively, of Florunner peanut (*Arachis hypogaea* L.) seed from pods left on or in the soil after harvest (3). Similar seed from field microplots of Dothan loamy sand infested 15 cm deep (about 1:500 cornmeal-sand, inoculum-soil) yielded 2.3% cultures of AG-2-2 and 3.0% cultures of AG-2-1 (D. K. Bell and D. R. Sumner, *unpublished*). These fungi also may survive in infested roots and crowns of corn left in the soil after harvest. However, this has not been proven. With our methodology, cultures of AG-2-2 and AG-2-1 were not consistently isolated from soil, unless the populations were $\geq$ 25 cfu/100 g of soil. Only 6–11 cfu/100 g of soil were recovered from soil collected by pushing a 2.5-cm-diameter probe through soil and decaying roots of 8- to 12-wk-old field-grown corn plants and plating the soil on TABA with a multiple-pellet soil sampler (9,19). In our study, the discrepancy between recovery of isolates of AG-2-2 from soil 7.0 and 9.4 mo after infestation vs. isolations from plants and the disease indices highlight the inconsistency in recovery of the fungus from soil. A similar situation exists with AG-2-1, except that the isolates of AG-2-1 were less virulent than those of AG-2-2 on snap bean and corn.

Our isolates of AG-2-2 and AG-2-1 produced a partial to continuous layer of "dark brown, velutinous to lanose, plectenchymatous, crustaceous" sclerotia within 2–3 wk on PDA or PDYCA. Single sclerotia measured up to 5 mm in diameter (19). Numerous single sclerotia and sclerotial aggregates (both <5 mm diameter) were produced by isolates of AG-2-2 and AG-2-1 in cornmeal-sand. However, we have not observed sclerotia associated with diseased roots and crowns of corn or hypocotyls of snap bean or southern pea. In our study, the isolates of AG-2-2 and AG-2-1 most likely survived in fallow soil as free sclerotia, because there was <1% organic matter in the infested soil and it was evenly distributed.

Survival of isolates of *R. solani* in AG-2-2 and AG-2-1 in fallow soil exposed to natural weathering for 9.4 mo is of considerable importance in soils of Georgia's coastal plain. Crops of snap bean, narrow-leafed (blue) lupine (*Lupinus angustifolius* L.), or southern pea may be planted after an early crop of corn. Snap bean, narrow-leafed lupine, and southern pea may be severely damaged by isolates of AG-2-2, and southern pea is susceptible to isolates of AG-2-1 (18,19).

R. solani AG-4 is the most widespread, destructive *Rhizoctonia* root pathogen in Georgia's coastal plain (3,4,17,18,21), and several isolates of AG-4 survived 9.4 mo in fallow soil. However, isolates of CAG-2 and one isolate of CAG-4 survived, and both CAG-2 and CAG-4 have shown potential as biological control agents for AG-4 (5). In mixed populations in fallow fields, the inoculum potential of AG-4 may be reduced to low levels. *L. arvalis* survived in high populations. It is common in soils of Georgia's coastal plain and is a known antagonist of *R. solani* (7). We made no attempt to investigate the influence of fallow on soilborne bacterial or fungal antagonists of *R. solani*.

If fallow soil were disturbed to control weeds (i.e., tillage in field plots), inoculum densities of many basidiomycetes would decrease more rapidly. Propagules would be more exposed to weathering and antagonism, competition, and parasitism by other microorganisms. Deep-turning (20–30 cm) of soil with a moldboard plow reduced populations of *R. solani* in AG-4 but did not eliminate propagules of the pathogen (12,21).

Sclerotia were abundant in the cornmeal-sand inoculum of most basidiomycetes used in our test, but the fungi were more exposed to natural degradation in this study than they would have been in naturally colonized plant debris such as peanut shells (3). Allowing fields to lie fallow would be less effective in reducing populations of pathogenic basidiomycetes in colonized plant debris than in our study, where total organic matter was only 0.58%. The practice of fallowing land would not be feasible in intensive cropping systems, but fallow coupled with polyethylene mulches or turning with a moldboard plow during the summer (11) may be an effective method of reducing populations of soilborne pathogens for homeowners and growers who plant fall vegetables in Georgia's coastal plain.

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