# 13158 - Rooting Medium

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We use this protocol and it's working

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ABSTRACT

This is part of the Leiboff Lab maize transformation protocol for somatic embryogenesis of B104 immature embryos. This protocol is a combination of Chen et al. 2022 and Kang et al. 2022 with some modifications based on material availability. This protocol is intended for the GRF-GIF/BBM somatic embryogenesis transformation strategy with the LBA4404 Met- auxotrophic *Agrobacterium* strain.

Growing shoots with visible leaves will be transferred from Shoot Maturation Medium 13329A to Rooting Medium 13158 approximately 38-45 days after infection (DAI). 13158 should be used for 7-14 days, before moving rooted, vigorous plantlets to soil in a humid dome. Rooting Medium contains no added plant hormones, relying on endogenous auxin synthesis and transport to encourage root growth. 13158 is high in sucrose, uses Bialaphos as a plant selective agent, and uses carbenicillin to control microbial contamination.

13158 solid media should be prepared in 25x100 (deep) petri plates, planning for <10 calli per plate. Material grown on 13158 will be sealed with micropore tape and be incubated at 28C in the dim to moderate light (80-150 umol/$m^2$/sec). Regenerated T0 plants are ready to move off 13158 into soil when they have roots longer than 3 cm.

## Planning

1 Estimate the volume of 13158 you will need based on the following:

$$Volume = 30mL * NumberPlates$$

Make sure to round up! Check the table below to plan your media

## Mixing Heat-Stable Ingredients

2 Retrieve the following heat-stable ingredients:

1. Murashige & Skoog Basal Salt Mixture, 'Basal MS' - Stored in Main Lab, 4C Refrigerator, Top Shelf
2. Sucrose - Stored in Main Lab, Chemical shelf 'S', use Fowler refillable container
3. Agar, Phyto - Stored in Main Lab, Chemical shelf 'A'

3 Retrieve a graduated cylinder for measuring your final solution
Place a stir bar at the bottom on a beaker that is ~1.5x the volume of your solution
Rinse stir bar+beaker and graduated cylinder with MQ H2O, discard rinse water in sink
NOTE: Any soap or detergent residue will interfere with the tissue culture process; if you see suds, rinse again or find different glassware

4 Add approximately 90% of your final media volume in MQ H2O to your beaker
Place beaker on a magnetic stir plate

Turn stir plate on to generate a vigorous stir

5 Using a fresh weigh paper and dry spatula/scoopula/pipette tip for each ingredient, add the following to your beaker:

| | A | B | C | D | E |
|---|---|---|---|---|---|
| | Ingredient | 100 mL | 200 mL | 300 mL | 600 mL |
| | Basal MS | 0.44 g | 0.88 g | 1.76 g | 3.52 g |
| | Sucrose | 4.0 g | 8.0 g | 12.0 g | 24.0 g |

6 Thoroughly rinse all used tools with running water
Place clean tools in drying rack
Return chemical reagents to their original storage location

## Adjust solution pH to 5.7 with 0.1 M KOH

7 Turn on the Hannah Instruments pH meter
Unscrew and remove the small green pH probe exchange cover and set cap aside
Gently remove the probe from the storage tube and set storage tube aside
Using squeeze bottle, rinse the glass probe with H2O, catch rinse water in a waste beaker
Gently blot probe with laboratory tissue paper to dry

8 Using adjustable arm, lower the pH probe into the beaker with stir plate on
Ensure that the stir bar does not strike the probe
Electrode at the base of the probe must be fully submerged

9 Using a plastic transfer pipette, add 0.1M KOH to your solution until you measure pH 5.7
NOTE: KOH can be added rapidly until pH 5.4, then add one drop at a time to reach pH 5.7
Solution pH between 5.6 - 5.8 is acceptable

10 Using the adjustable arm, remove the pH probe from the beaker
Using squeeze bottle, rinse the glass probe with H2O, catch rinse water in a waste beaker
Gently blot probe with laboratory tissue paper to dry
Return the probe to the storage tube -- Ensure the electrode bulb is fully submerged in storage solution
Return and secure the small probe exchange cover
Turn off the pH meter

## Bring solution to target volume, add phytoagar, and autoclave

11 Turn off the stir plate and remove your beaker
Hold a large stir bar in your hand to stabilize the one in your beaker
Pour your solution into the graduated cylinder -- Do not transfer the stir bar
Add a small amount (50-100 mL) of water to your beaker
Carefully add water from the beaker to the graduated cylinder until your solution reaches the target volume -- Do not transfer the stir bar

12 Retrieve a clean dry bottle and matching cap
Using a fresh weigh paper and dry spatula/scoopula:

| | A | B | C | D | E |
|---|---|---|---|---|---|
| | Ingredient | 100 mL | 200 mL | 300 mL | 600 mL |
| | Phytoagar | 0.6 g | 1.2 g | 1.8 g | 3.6 g |

Add phytoagar to dry bottle
NOTE: Adding phytoagar to dry bottle avoids clumping which is undesirable for final media

13 Loosely place the cap over the bottle
Add a small piece of autoclave tape on the cap and bottle
Place the bottle in an autoclave-safe bin
Autoclave 20-25 min using the 'Liquid' setting
NOTE: Recommended autoclaves are in Cord 3112 and 4112. Complete cycle will take ~1 hr.

14 Rinse all used tools and glassware in running water
Place clean items on drying rack
Return chemical reagents to their original storage location

## Adding Heat-sensitive Ingredients

15 Return to the autoclave to pick up your solution -- Be prompt, sucrose can degrade if left too long
Using autoclave gauntlets, gently seal the cap of the bottle
Swirl the autoclaved solution to evenly mix phytoagar

16 Carefully return to the lab with autoclave bin and sealed bottle
Place your sealed solution into the large 55C water bath in the main lab
Discard any liquid remaining in the autoclave bin and return to bin storage
NOTE: Your solution needs to reach ~55C before adding the heat-sensitive ingredients

17 Retrieve the following heat-sensitive ingredients:
1. Carbenicillin (100 mg/mL), 'Carb' - Stored in Main Lab, -20C Freezer, Antibiotics #2
2. Bialaphos (1 mg/mL ) - Stored in Main Lab, -20C Freezer, Tissue Culture Box #2

Place reagents in a tube rack and move to laminar flow hood to thaw

18 Turn on the laminar flow hood, airflow and lamp

Using 70% EtOH spray bottle and paper towels, sterilize the working area inside the laminar flow hood
Retrieve sterile **deep** petri plates (25x150 mm plates) - Stored in Main Lab, Tissue Culture Room, 'Nonstandard Petri Plates' Cabinet
Using a fine-tipped sharpie, write '13158' and the date along the bottom rim of the plate

19 When your solution reads 55C with a digital thermometer gun,
transfer your sealed bottle to the laminar flow hood.
The bottle should be warm, but safe to handle.
Sterilize the outside of the bottle and your gloved hands with 70% ethanol spray.

20 Using a fresh filter tip for each ingredient, add the following to your bottle:

| | A | B | C | D | E |
|---|---|---|---|---|---|
| | Ingredient | 100 mL | 200 mL | 300 mL | 600 mL |
| | Carb | 100 uL | 200 uL | 300 uL | 600 uL |
| | Bialaphos | 500 uL | 1000 uL | 1500 uL | 3000 uL |

Used tips may be disposed of in regular lab waste -- no contact with rDNA or modified cells is anticipated.

21 Gently swirl media bottle to mix thoroughly, but avoid introducing bubbles.
Pour media into plates, ~30 mL per plate.
NOTE: Each plate should be more than 25% full with media.
Close plates to solidify in laminar flow hood.

22 Using paper towels, clean any spilled media and discard in regular lab waste.
When plates are poured, rinse media bottle in lab sink and hang on bottle rack to dry.
Return reagents to their original storage location.
Using 70% EtOH spray bottle and paper towels, sterilize the working area inside the laminar flow hood for the next worker.

23 Leave closed plates to solidify in the laminar flow hood with the fan on, 3 hrs - overnight.
NOTE: Keep plates ~10 cm (4 in) away from the back of the flow hood to avoid drying out.
When plates are solid, wrap in a clean plate bag or individually seal with parafilm and store upside-down at 4C, up to 1 week.