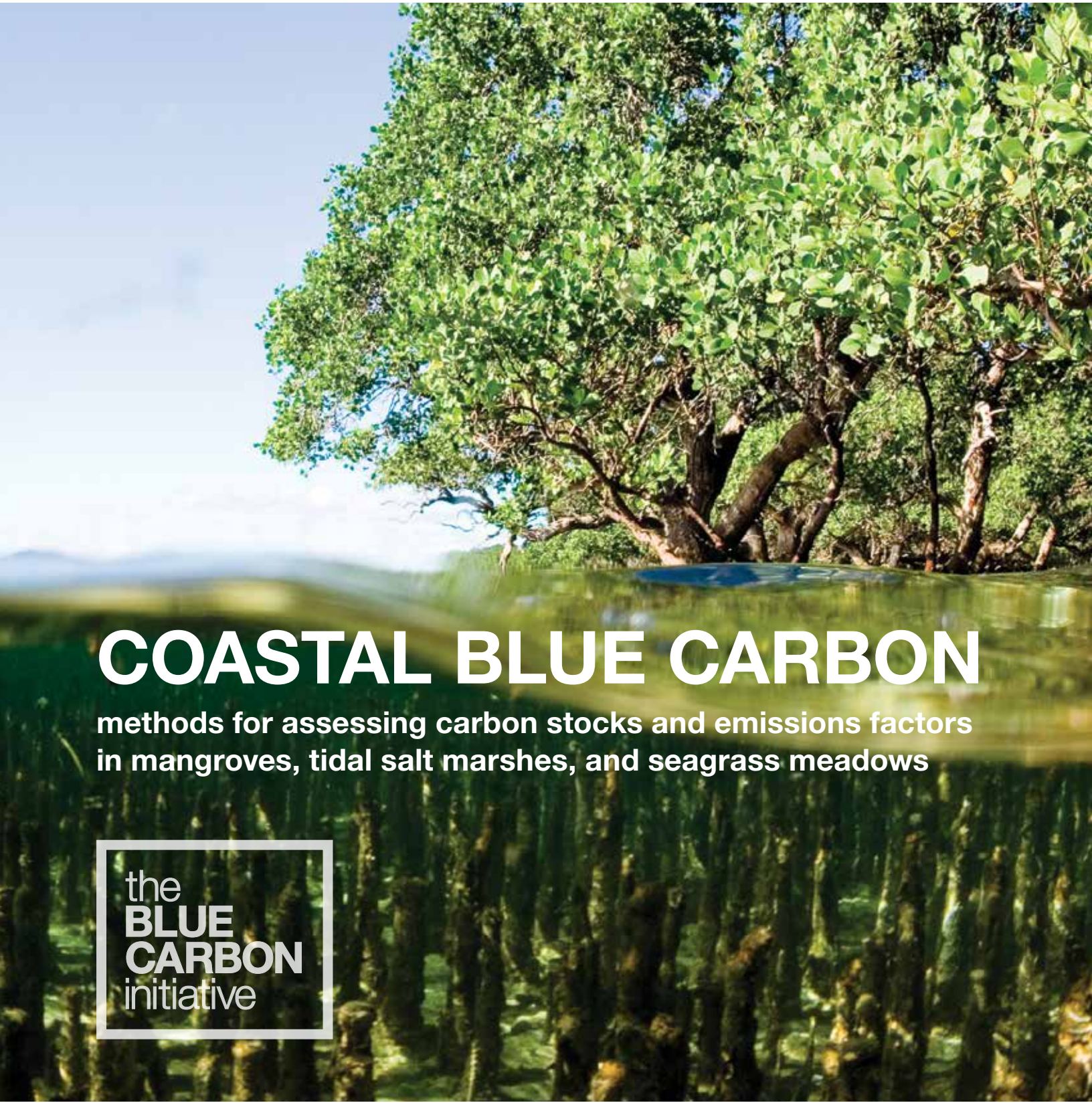

COASTAL BLUE CARBON

methods for assessing carbon stocks and emissions factors in mangroves, tidal salt marshes, and seagrass meadows

the BLUE CARBON initiative

CONSERVATION INTERNATIONAL

IUCN

United Nations Educational, Scientific and Cultural Organization

Intergovernmental Oceanographic Commission

the BLUE CARBON initiative

Coordinators of the International Blue Carbon Initiative

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ACRONYMS

A

AGEDI	Abu Dhabi Global Environmental Data Initiative
ArcView	ArcGIS for Desktop pages
Arc	Closed segment of a differentiable curve in the two dimensional plane
ASF	Alaska Satellite Facility

B

B	Biomass

C

C	Carbon
$CaCO_3$	Calcium carbonate
CDM	Clean Development Mechanism
CGIAR	Consultative Group on International Agricultural Research
CH_4	Methane
CHN analyzer	Elemental analyzer of mainly carbon, hydrogen and nitrogen
CI	Confidence Interval
CI	Conservation International
CIFOR	Center for International Forestry Research
CO_2	Carbon Dioxide
C_{org}	Organic Carbon

D

D_{30}	Mainstem diameter at 30 cm height
DAAC	NASA's Distributed Active Archive Center
D_{base}	Mainstem basal diameter
DBD	Dry Bulk Density
dbh	Diameter at Breast Height
DEM	Digital Elevation Maps
DIC	Dissolved Inorganic Carbon
D_{max}	Mainstem maximum diameter of sampled trees
D_{top}	Mainstem top diameter

E

ECU	Edith Cowan University Western Australia
eqiv	Equivalent
Eqn	Equation

Equiv	Equivalent
ESRI	Environmental Systems Research Institute
EVI	Enhanced Vegetation Index

F

FIU	Florida International University
FURG	Fundação Universidade Federal do Rio Grande, Federal University Foundation of Rio Grande

G

GHG	Greenhouse Gas
GIS	Geographic Information System
GLAS	Geoscience Laser Altimeter System
GPS	Global Positioning System

H

H	Horizontal transverse waves
H	Height
H	Hydrogen
H_2O_2	Hydrogen peroxide
HCl	Hydrogen chloride
HH	Horizontal transmitting, Horizontal receiving waves
H_{max}	Maximum Height
HV	Horizontal transmitting, vertical receiving waves

I

ICESat	Ice, Cloud, and land Elevation Satellite
ID	Identity Document, Identifier
IITA	International Institute of Tropical Agriculture
IOC	Intergovernmental Oceanographic Commission
IPCC	Intergovernmental Panel on Climate Change
IUCN	International Union for Conservation of Nature

J

JAXA	Japan Aerospace Exploration Agency's

K

KIOST	Korea Institute of Ocean Science and Technology
KKP	Agency for Research and Development of Marine and Fisheries (Baltitbang KP), Indonesia

L

LCCS	United Nation's Land Cover Classificaiton System
LPDAAC	Land Processes Distributed Active Archive Center
ln	Natural Logarithm
LOI	Loss on Ignition

M

MH	SET marker horizon
MODIS	Moderate Resolution Imaging Spectroradiometer

N

n	Number of subsamples
N	Nitrogen
N_2	Dinitrogen
N_2O	Nitrous oxide
NASA	National Aeronautics and Space Administration
ND	No Data
NDVI	Normalized Difference Vegetation Index
NSIDC	National Snow and Ice Data Center

O

O	Oxygen
OEH NSW	Office of Environment and Heritage North South Wales
ORNL	Oak Ridge National Laboratory
OSU	Oregon State Universitypage 20

P

ρ	Wood density
PALSAR	Phased Array L band Synthetic Aperture Radar
PCA	Principal Component Analysis
PVC	Polyvinyl chloride

Q

QMD	Quadratic Mean Diameter

R

R^2	Coefficient of determination
RAE	Restore American's Estuaries

REDD	Reducing Emissions from Deforestation and Forest Degradation
RFDI	Radar Forest Degradation Index

S

SAR	Synthetic aperture radar
SCUBA	Self Contained Underwater Breathing Apparatus
SD	Standard Deviation
SERC	Smithsonian Environmental Research Center
SET	Surface Elevation Table
SRTM	Shuttle Radar Topography Mission
STS	NASA's Space Transportation System
SWBD	SRTM Water Body Data

T

T	Temperture
T_1	Initial Assessment
T_2	Subsequent Assessments
TanDEM X	TerraSAR X add on for Digital Elevation Measurement
th	Tree Height

U

UNEP	United Nations Environment Programme
UNESCO	United Nations Educational Scientific and Cultural Organization
USD	United States dollar
USGS	US Geological Survey
UTS	University of Technology Sydney
UW	University of Wisconsin, USA
UWB	Bangor University, Wales, UK

V

V	Vertical transverse waves
VCS	Verified Carbon Standard
VH	Vertical transmitting, Horizontal receiving waves
VI	Vegetation Index
VV	Vertical transmitting, Vertical receiving waves

W

WCMC	World Conservation Monitoring Centre
W	Length of the plant canopy

UNITS LIST

atmos	Atmosphere
ºC	Degree Celsius
cm	Centimeter
cm^3	Cubic centimeter
e	Euler's number, approx. 2.71828
h	Hour
ha	Hectare
g	Gram
K	Kelvin
kg	Kilogram
L	Liter
m	Meter
m^2	Square meter
Mg	Megagram
mg	Milligram
min	Minute
mL	Milliliter
mol	Mole
%	Percent
π	Mathematical constant, the ratio of a circle's circumference to its diameter, approximately equal to 3.14159
$	Dollar
σ	Standard deviation
t	Metric ton

KEY WORDS

Active Remote Sensing – A remote-sensing system, such as radar, that produces electromagnetic radiation and measures its reflection back from a surface.

Activity Data – Geographical data showing the types of land coverage and use in a given area.

Allochthonous Carbon – Carbon produced in one location and deposited in another. In the context of blue carbon systems, this type of carbon results from the hydrodynamic environment in which they are found where sediments and associated carbon is transported from neighboring ecosystems (offshore and terrestrial).

Allometric Equations – Allometric equations establish quantitative relationships between key characteristics that are easy to measure (i.e., stem height/diameter) and other properties that are often more difficult to assess (i.e., biomass).

Autochthonous Carbon – Carbon produced and deposited in the same location. In the context of blue carbon systems, this type of carbon results from vegetation uptake of CO_2 from the ocean and/or atmosphere that gets converted for use by plant tissue and decomposes into the surrounding soil.

Blue Carbon – The carbon stored in mangroves, tidal salt marshes, and seagrass meadows within the soil, the living biomass above ground (leaves, branches, stems), the living biomass below ground (roots), an the non-living biomass (litter and dead wood).

Carbon Inventory – A carbon inventory is an accounting of carbon gains and losses emitted to or removed from the atmosphere/ocean over a period of time. Policy makers use inventories to establish a baseline for tracking emission trends, developing mitigation strategies and policies, and assessing progress

Carbon Pool – Carbon pools refer to carbon reservoirs such as soil, vegetation, water, and the atmosphere that absorb and release carbon. Together carbon pools make up a carbon stock.

Carbon Stock – A carbon stock is the total amount of organic carbon stored in a blue carbon ecosystem of a known size. A carbon stock is the sum of one or more carbon pools.

Emission Factors – A term used to describe changes in the carbon content of a pre-defined area due to change in land coverage and use (i.e., conversion from mangroves to shrimp ponds) or changes within a land use type (i.e., nutrient enrichment of seagrass).

Flux Method – This method estimates the GHG flux between the soil and vegetation and the atmosphere/water column through direct measurements or by modeling and results in Tier 2 and 3 estimates.

Gain-loss Method – This method estimates the difference in carbon stocks based on emissions factors for specific activities (e.g., plantings, drainage, rewetting, deforestation) derived from the scientific literature and country activity data and results in Tier 1 and 2 estimates.

Inorganic Soil Carbon – The term soil inorganic carbon refers to the carbon component of carbonates (i.e., calcium carbonate) and can be found in coastal soils in the form of shells and/or pieces of coral.

IPCC Tiers – The IPCC has identified three tiers of detail in carbon inventories that reflect the degrees of certainty or accuracy of a carbon stock inventory (assessment).

Tier 1 – Tier 1 assessments have the least accuracy and certainty and are based on simplified assumptions and published IPCC default values for activity data and emissions factors. Tier 1 assessments may have a large error range of +/- 50% for aboveground pools and +/- 90% for the variable soil carbon pools.

Tier 2 – Tier 2 assessments include some country or site-specific data and hence have increased accuracy and resolution. For example, a country may know the mean carbon stock for different ecosystem types within the country.

Tier 3 – Tier 3 assessments require highly specific data of the carbon stocks in each component ecosystem or land use area, and repeated measurements of key carbon stocks through time to provide estimates of change or flux of carbon into or out of the area. Estimates of carbon flux can be provided through direct field measurements or by modeling.

Mangrove – A mangrove is a tree, shrub, palm or ground fern, generally exceeding one half meter in height that normally grows above mean sea level in the intertidal zone of marine coastal environments and estuarine margins. A mangrove is also the tidal habitat comprising such trees and shrubs.

Passive Remote Sensing – A remote-sensing system, such as an aerial photography imaging system, that only detects energy naturally reflected or emitted by an object.

Resolution – In remote sensing resolution of an image is an indication of its potential detail, where the smaller the pixel the higher the detail. In other words, 250 meters resolution data could identify any earthly feature that is 250 meters by 250 meters (useful for mapping ecosystem extent). Higher resolution data, such as 30 meters can be used to monitor in more detail (useful for identifying encroachment by aquaculture).

Seagrass Meadows – Seagrasses are flowering plants belonging to four plant families, all in the order Alismatales, which grow in marine, fully saline environments. There are 12 genera with some 58 species known.

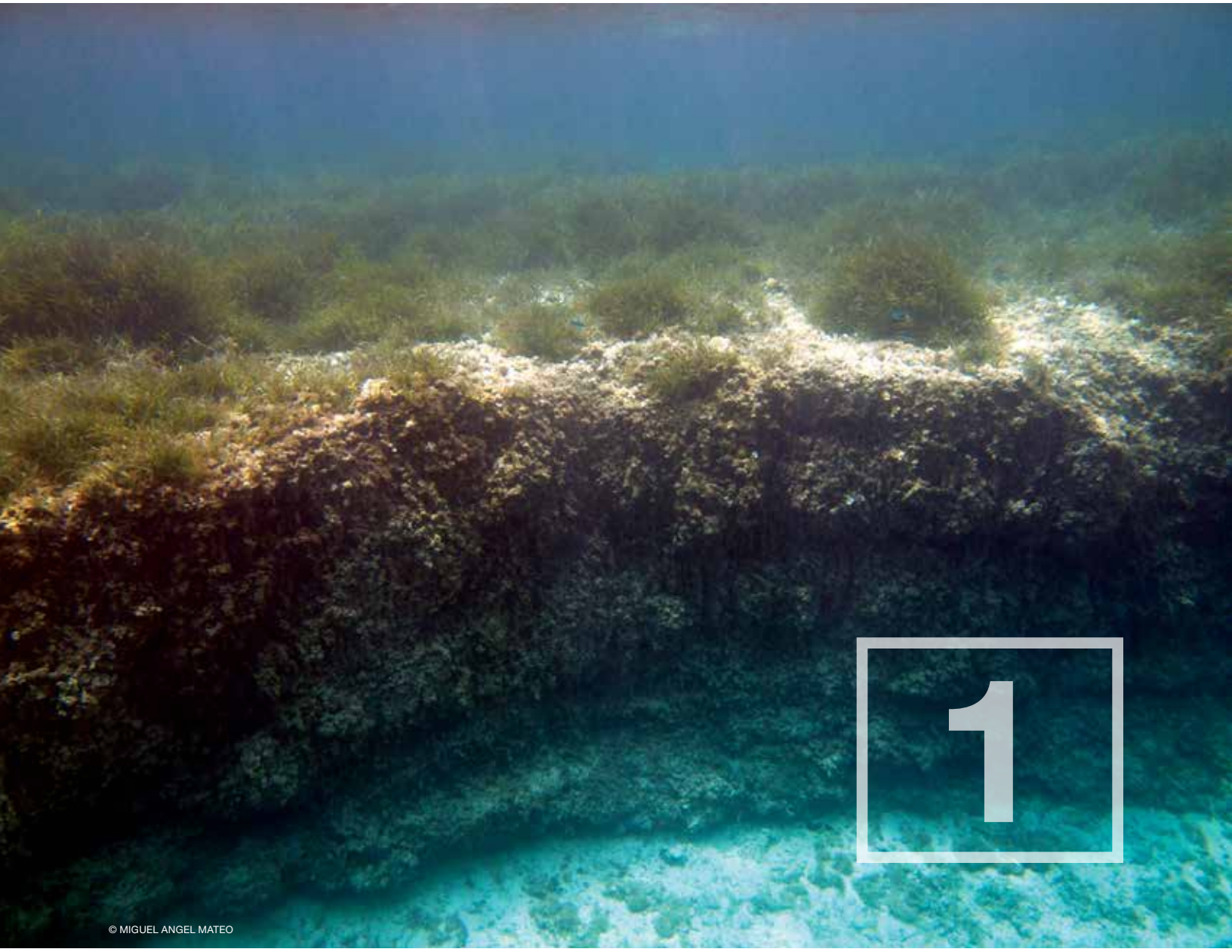
Soil Organic Carbon – The term soil organic carbon refers to the carbon component of the soil organic matter. The amount of soil organic carbon depends on soil texture, climate, vegetation and historical and current land use/management.

Soil Organic Matter – The term soil organic matter is used to describe the organic constituents in the soil (undecyed tissues from dead plants and animals, products produced as these decompose and the soil microbial biomass).

Stock Difference Method – This method estimates the difference in carbon stocks measured at two points in time and results in Tier 3 estimates.

Stratification – A technique used to divide large heterogeneous sites (which require many samples to account for variation) into smaller more homogeneous areas (where fewer samples are needed) and is also useful when field conditions, logistical issues, and resource limitations prevent dense sampling regimes

Tidal Salt Marsh – A tidal salt marsh is a coastal ecosystem in the upper intertidal zone between land and open salt water or brackish water that is regularly flooded by the tides. It is dominated by dense stands of salt tolerant plants such as herbs, grasses, or low shrubs.



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Why Measure Carbon Stocks

1

BACKGROUND AND CONTEXT

Coastal ecosystems are critical to maintaining human well-being and global biodiversity. In particular, mangroves, tidal salt marshes, and seagrasses provide numerous benefits and services that contribute to people's ability to mitigate and adapt to the impacts of climate change (**Fig. 1.1**). Many of these services are essential for climate adaptation and resilience along coasts, including protection from storm surge and sea level rise, erosion prevention along shorelines, coastal water quality regulation, nutrient recycling, sediment trapping, habitat provision for numerous commercially important and endangered marine species, and food security for many coastal communities around the world (Kennedy 1984; Robertson & Alongi 1992; King & Lester 1995; Hogarth 1999; Beck *et al*. 2001; Kathiresan & Bingham 2001; Saenger 2002; Mumby 2006; Gedan *et al*. 2009; Barbier *et al*. 2011; Sousa *et al*. 2012; Cullen-Unsworth & Unsworth 2013). In addition, these ecosystems help mitigate climate change by sequestering and storing significant amounts carbon, known as coastal blue carbon, from the atmosphere and oceans (Duarte *et al*. 2005; Bouillon *et al*. 2008; Lo Iacono *et al*. 2008; Duarte *et al*. 2010; Kennedy *et al*. 2010; Donato *et al*. 2011; Mcleod *et al*. 2011; Fourqurean et al. 2012a; Pendleton *et al*. 2012; Chmura 2013; Lavery *et al*. 2013).

Figure 1.1 Blue carbon ecosystems: mangroves (top left, © Sterling Zumbrunn, CI), seagrasses (bottom left, © Miguel Angel Mateo), and tidal salt marshes (right, © Sarah Hoyt, CI)

Despite their benefits and services, coastal blue carbon ecosystems are some of the most threatened ecosystems on earth, with an estimated 340 000 to 980 000 hectares being destroyed each year (Murray *et al*. 2011). Although their historical extent is difficult to determine due to dramatic losses which occurred before accurate mapping was possible, it is estimated that up to 67% of the historical global mangrove range, 35% of tidal salt marshes, and 29% of seagrasses have been lost. If these trends continue at current rates, a further 30–40% of tidal marshes and seagrasses and nearly all unprotected mangroves could be lost in the next 100 years (Pendleton *et al*. 2012).

Increasingly, coastal ecosystems are being recognized for their important role in carbon sequestration and, when degraded, their potential to become sources of carbon emissions. Progress has been made to include these systems in international and national policy and finance mechanisms, but full integration of coastal management activities as part of countries' portfolio of solutions to mitigate climate change has not yet been realized. This opportunity to incorporate coastal blue carbon into policies and management could lead to additional coastal ecosystem conservation (restoration and protection) worldwide, which would preserve and enhance the multiple benefits these ecosystems provide to humans.

NEED FOR THE MANUAL

There is a rapidly growing body of scientific knowledge on the direct and indirect effects of climate change and human development on coastal ecosystems. Increased attention is being paid to mangroves, tidal salt marshes, and seagrasses for their carbon sequestration capabilities, as well as other important ecosystem services. If properly planned and managed, coastal blue carbon could function as a potential funding mechanism for coastal ecosystem conservation and restoration. To achieve this goal, managers need to be able to assess carbon stocks (total amount of carbon stored within a distinct area) and monitor changes in carbon stocks and greenhouse gas (GHG) emissions over time. Until recently, coastal ecosystem managers and other stakeholders interested in quantifying blue carbon have lacked practical tools and guidance to allow for proper carbon analyses. This is particularly true in developing countries where there may be large data gaps and a lack of technical and financial resources to carry out complex analyses. New guidelines and methodologies have begun to emerge in the last few years, all of which refer to the need for internationally accepted measuring and monitoring procedures for carbon accounting (Appendix A: Additional Guidance Documents). This guide will provide managers, scientists, and field practitioners with standardized recommendations and techniques for carbon measurement and analysis in blue carbon systems and directly support the assessment and accounting of blue carbon globally.

OBJECTIVE OF THE MANUAL

The objective of this manual is to provide standardized methods for field measurements and analysis of blue carbon stocks and flux in coastal ecosystems. The manual is designed to provide users with relevant background information on key concepts, and guide them in a step-by-step process, pointing out stages where expert advice or additional technical data may be required. The goal is to utilize these assessments to support improved conservation and restoration of coastal ecosystems through various management and policy approaches, regulatory frameworks, and participation in voluntary carbon markets.

WHO IS THE MANUAL FOR?

The manual has been designed to be used by a wide range of stakeholders, including natural resource managers, scientists, community groups, and local and national government agencies interested in assessing blue carbon stocks. It can be implemented in a range of situations, with a focus on developing country contexts, and can be tailored to the needs of specific areas according to resource availability. The manual has been designed for users with local knowledge about the system being assessed, but without necessarily a detailed technical knowledge of how to conduct blue carbon measurements.

MANUAL STRUCTURE

The manual outlines the rationale and project design for measuring blue carbon in the field and approaches for data analysis and reporting. Effort was made to ensure consistency with international standards, the Intergovernmental Panel on Climate Change (IPCC) guidelines, and other relevant sourcebooks.

The manual is structured as follows:

Chapter 1: Introduces the role of blue carbon in climate change mitigation and outlines the manual's purpose and objectives;

Chapter 2: Outlines the main steps to prepare a robust field measurement plan;

Chapter 3: Provides protocols and guidance for measuring organic carbon stocks found in the soils of all three ecosystems;

Chapter 4: Provides protocols and guidance for measuring organic carbon stocks, found in above- and belowground biomass, with specific protocols designed for each ecosystem;

Chapter 5: Highlights methods for determining the changes in carbon stocks over time and monitoring greenhouse gas emissions;

Chapter 6: Gives an overview of remote sensing options and applications;

Chapter 7: Provides guidance on managing large data sets and data sharing; and

Appendices: There are several appendices; they contain supplementary information, worked through examples, lists of equations, and more.

WHAT IS BLUE CARBON

Blue carbon is the carbon stored in mangroves, salt tidal marshes, and seagrass meadows within the soil, the living biomass aboveground (leaves, branches, stems), the living biomass belowground (roots), and the non-living biomass (e.g., litter and dead wood) (Mcleod *et al*. 2011). Similar to the carbon stored in terrestrial ecosystems, blue carbon is sequestered in living plant biomass for relatively short time scales (years to decades). Unlike terrestrial ecosystems, carbon sequestered in coastal soils can be extensive and remain trapped for very long periods of time (centuries to millennia) resulting in very large carbon stocks (Duarte *et al*. 2005; Lo Iacono *et al*. 2008). The difference in soil carbon accumulation in terrestrial versus coastal systems is that potential carbon storage in upland soils is limited by high availability of oxygen, allowing for aerobic microbial carbon oxidation and release back into the atmosphere (Schlesinger & Lichter 2001). In blue carbon systems, however, the soil is saturated with water keeping it in an anaerobic state (low to no oxygen), and it continually accretes vertically at high rates resulting in continuous build-up of carbon over time (Chmura *et al*. 2003). Some of the largest examples of carbon stocks in coastal sediments include the *Posidonia oceanica* seagrass meadows in Portlligat Bay, Spain, and mangroves in Belize which have accreted carbon-rich soils more than 10 meters thick and are more than 6000 years old (McKee *et al*. 2007; Lo Iacono *et al*. 2008; Serrano et al. 2014). Similarly, tidal salt marsh sediments in northern New England are 3–5 meters thick, 3000–4000 years old, and are composed of up to 40% organic carbon (Johnson *et al*. 2007).

Figure 1.2 Tidal salt marsh soil sample, Beaufort, NC (© Jennifer Howard, CI)

The carbon found in blue carbon ecosystems can be classified as either autochthonous or allochthonous and depending on the project, may need to be assessed separately (Middelburg *et al*. 1997; Kennedy *et al*. 2010).

- **Autochthonous Carbon**: This type of carbon is produced and deposited in the same location. Plants remove carbon dioxide (CO_2) from the atmosphere/ocean through photosynthesis (primary production) and convert it for use by plant tissue (such as leaves, stems, roots/rhizomes) to increase plant biomass. A large portion of plant biomass is allocated to the roots where it decomposes very slowly in anaerobic conditions, thus storing the carbon within the sediments (**Fig. 1.2**) (Middelburg *et al*. 1997; Kennedy *et al*. 2010).
- **Allochthonous Carbon**: This type of carbon is produced in one location and deposited in another. Blue carbon ecosystems exist in very hydrodynamically active settings; they are constantly battered by waves, tides, and coastal currents that transport sediments and associated organic carbon from adjacent ecosystems (offshore or terrestrial). The plants found in these systems have complex root structures and canopies that are efficient at trapping sediment as it moves through the system, adding to the local carbon stock as a result (**Fig. 1.2**).

The ratio of carbon originating within the ecosystem to that trapped from external sources varies between blue carbon systems. In seagrass meadows, an estimated 50% of carbon stored in soils can be of external origin (allochthonous) (Kennedy *et al*. 2010), while most of the sequestered carbon in mangrove and tidal salt marsh systems is directly produced by the plants within the system (autochthonous) (Middleton & McKee 2001). However, in some settings there are significant allochthonous contributions found in mangroves and marshes, derived from adjacent terrestrial or marine ecosystems (Middelburg *et al*. 1997; Bouillon *et al*. 2003; Adame *et al*. 2012).

WHY MANAGE FOR BLUE CARBON

Globally, numerous policies, coastal management strategies, and tools designed for conserving and restoring coastal ecosystems have been developed and implemented. Policies and finance mechanisms being developed for climate change mitigation may offer an additional route for effective coastal management. Blue carbon now offers the possibility to mobilize additional funds and revenue by combining best-practices in coastal management with climate change mitigation goals and needs.

Mangroves, tidal salt marshes, and seagrasses are under high levels of pressure from coastal development and land-use change (Alongi 2002; Gedan *et al*. 2009; Saintilan *et al*. 2009; Waycott *et al*. 2009). When vegetation is removed and the land is either drained or dredged for economic development, (e.g., mangrove forest clearing for shrimp ponds, draining of tidal marshes for agriculture, and dredging in seagrass beds—all common activities in the coastal zones of the world), the sediments become exposed to the atmosphere or water column resulting in the carbon stored in the sediment bonding with the oxygen in the air to form CO_2 and other GHG that get released into the atmosphere and ocean (Yu & Chmura 2009; Loomis & Craft 2010; Donato *et al*. 2011; Kauffman *et al*. 2011; Lovelock *et al*. 2011; Ray *et al*. 2011; Callaway *et al*. 2012; Fourqurean *et al*. 2012a) (**Fig. 1.3**). Not only do these activities result in CO_2 emissions but they also result in losses of biodiversity and critical ecosystem services.

Coastal blue carbon ecosystems offer coastal protection through wave attenuation and erosion prevention (King & Lester 1995; Gedan *et al*. 2011). These services are already recognized as a vital function of mangroves (Mazda *et al*. 1997; Massel *et al*. 1999; McIvor *et al*. 2012a; McIvor *et al*. 2012b), but they gained more prominence in the aftermath of the December 2004 Indian Ocean tsunami (Danielsen *et al*. 2005; Kathiresan & Rajendran 2005; Alongi 2008), the November 2013 typhoon Haiyan that hit the Philippines (Gross 2014), and other recent destructive cyclones and hurricanes (Tibbetts 2006; Williams *et al*. 2007; Das & Vincent 2009). These systems also regulate water quality, serve as critical habitats for many fish and shellfish species, provide wood and other products to local populations, and host

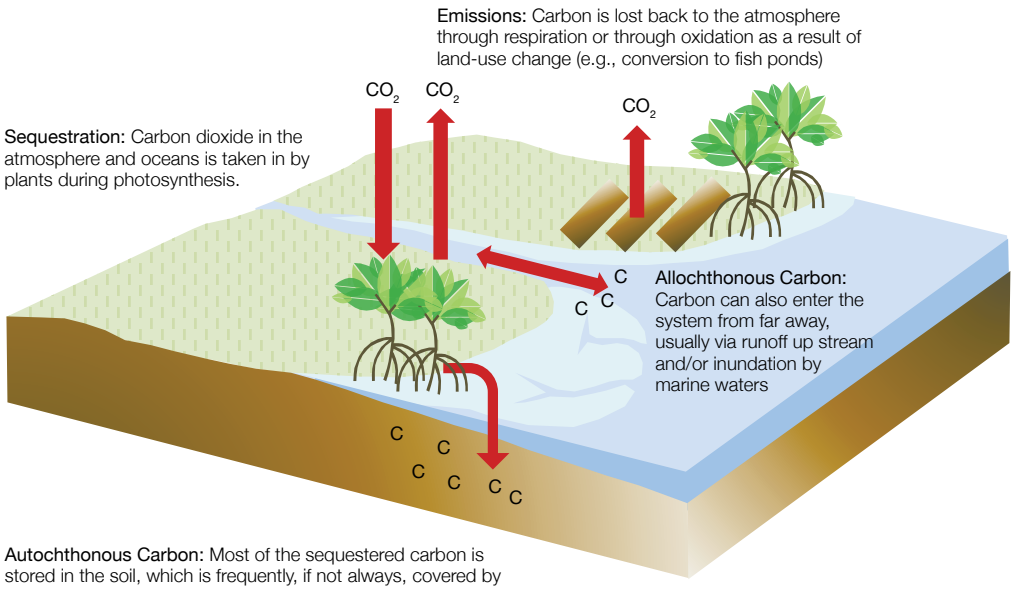

Figure 1.3 Mechanisms by which carbon moves into and out of tidal wetlands

a diverse array of rare and endangered species (Valiela & Teal 1979; Beck *et al*. 2001; Duke *et al*. 2007; FAO 2007; Barbier *et al*. 2011; Cullen-Unsworth & Unsworth 2013). They are a source of nutrients to adjacent ecosystems, provide sheltered living space for economically important species, and are valued for aesthetics and ecotourism (Barbier *et al*. 2011).

While this manual is focused on the evaluation of carbon stocks as a tool for conservation of coastal vegetated ecosystems, it is important to remember that actions to conserve the carbon stocks also ensure the preservation of these and other critical ecosystem services.

KNOWLEDGE GAPS

Despite the wealth of research that has been conducted, knowledge gaps still exist. Continued work in the areas outlined below will help to further refine current estimates and evaluations.

- **Geographical extent**: While mangroves are fairly well mapped, large areas containing seagrass meadows remain largely unsurveyed, (e.g., Southeast Asia, eastern and western South America and the west coast of Africa). Similarly, the global extent of tidal salt marsh and rates of marsh and seagrass meadow loss are currently undocumented.
- **Sequestration and storage**: Limited data are available in the scientific literature on the carbon sequestration and storage rates of blue carbon ecosystems in Africa, South America, and Southeast Asia.
- **Emissions and removals:** Additional mapping of converted, and degraded and revegetated blue carbon ecosystems and the quantification of emissions from exposed organic soils, and from disturbed or degraded seagrass meadows as well as quantification of removals to restored coastal ecosystems, is needed to enable inclusion in relevant databases (e.g., the IPCC Emission Factor Database).
- **Human drivers**: Emission rates associated with specific human activities over time for various drivers of ecosystem degradation, or loss (e.g., drainage, burning, harvesting, or clearing of vegetation at different intensity levels) are limited at the moment, especially for seagrasses. Removal rates to restored coastal ecosystems are also currently lacking.
- **Coastal Erosion**: A significant amount of eroded coastal carbon is thought to be dissolved in the ocean water where it enters the ocean-atmosphere system. The remaining eroded carbon is deposited in offshore sediments and sequestered. The fate of carbon eroded from blue carbon ecosystems is an ongoing topic of scientific research.

BLUE CARBON INVENTORIES

To explicitly address the role of blue carbon ecosystems in climate change mitigation and human wellbeing through policy, regulatory, finance, or other mechanisms, the carbon stock in these ecosystems and the existing or potential carbon emissions resulting from changes to those ecosystems must be quantified. This process is referred to as creating a carbon inventory. Carbon inventories can be undertaken at site-level, regional, national, and global scales. Specific examples of activities that require a blue carbon inventory include quantifying the total GHG emissions that result from land use changes, and estimating the avoided carbon emissions and the resulting climate change mitigation potential of a given coastal conservation project or activity.

Creating a carbon inventory for a given area requires understanding 1) the past and present distribution of coastal vegetated ecosystems linked to the human uses of the area, 2) the current carbon stock within the project area and rate of carbon accrual, and 3) the potential carbon emissions that will result from expected or potential changes to the landscape. Carbon emissions are normally expressed in megagrams (Mg) or metric tons[1] of carbon (C) per hectare (ha), for a given change in land use in a given time frame. Results can also be reported in tons of CO_2 per ha. To convert Mg C/ha to Mg CO_2/ha, multiply the Mg C/ha by 3.67 (the molar ratio of CO_2 to C). CO_2 equivalents (equiv) per hectare is a metric used to express carbon emissions produced by non-CO_2 emissions (e.g., methane) and allows for comparability between GHG.

The IPCC guidelines identified "activity data" and "emission factors" as being required to calculate the carbon emissions or removals for a given area. Those two distinct types of data are described here:

- **Activity data**: This term refers to geographical data showing the types of land coverage and use in a given area such as pristine mangrove forest, degraded tidal marsh, agricultural land, grassland, or aquaculture ponds. These data also include the expected rates of change in land uses over time—for example the rate of conversion of mangrove areas to shrimp ponds. Remote sensing is commonly used to classify land-use types and to track changes between different land uses over time. However, additional field assessments and mapping are often necessary, especially in coastal environments where accurate remote imaging may be challenging (Chapter 6: Remote sensing methods).
- **Emission factors**: Emission factors: This term refers to changes (loss or gain of carbon) the investigated area that has resulted from changes in land coverage and use (e.g., loss of carbon due to conversion of mangrove to aquaculture, and tidal marsh to agricultural land or gain of carbon through revegetation or restoration of coastal ecosystems. Positive values for emission factors indicate loss of carbon from biomass and soil, to the atmosphere and negative values indicate removal of carbon from the atmosphere to the biomass and soil (sequestration). Accurate quantification of emission factors requires ground-based measurements of ecosystem carbon stocks and their change over time, (Chapters 3: General principles of field sampling soil carbon pools and Chapter 4: General Principles of field sampling vegetative carbon pools for relevant methods for measuring carbon stocks in mangroves, tidal marshes, and seagrasses meadows) or direct measurement of how much carbon is emitted or sequestered over time (Chapter 5).

TIERS OF DETAIL IN CARBON INVENTORIES

There is a clear need to align methods with international standards such as those described by the IPCC's 2013 Supplement to the 2006 IPCC Guidelines for National Greenhouse Gas Inventories: Wetlands (IPCC 2013) and other relevant sourcebooks. According to the IPCC, carbon inventories can be achieved at various levels of detail or certainty, often determined by the purpose of the inventory and the resources available. The IPCC has identified three tiers of detail in carbon inventories that reflect the degrees of certainty or accuracy of a carbon stock inventory (or assessment) (**Table 1.1**).

1 A metric ton is the same as a mega gram (Mg) or 1 000 000 grams.

The methods described in this manual are relevant to achieving the highest level of assessment of carbon stocks in ecosystems—Tier 3. The IPCC recommends that countries aspire for Tier 3 for the measurement of key carbon stocks/sources/sinks. However, Tier 3 assessments are more costly to implement, require higher levels of technical resources and capacity, and are not always possible.

Table 1.1 Tiers that may be used to assess carbon emission factors (GOFC-GOLD 2009).

TIER	REQUIREMENTS	COMMENTS
1	IPCC default factors	Tier 1 assessments have the least accuracy and certainty and are based on simplified assumptions and published IPCC default values for activity data and emissions factors. Tier 1 assessments may have a large error range of +/- 50% for aboveground pools and +/- 90% for the variable soil carbon pools.
2	Country-specific data for key factors	Tier 2 assessments include some country or site-specific data and hence have increased accuracy and resolution. For example, a country may know the mean carbon stock for different ecosystem types within the country.
3	Detailed inventory of key carbon stocks, repeated measurements of key stocks through time or modelling	Tier 3 assessments require highly specific data of the carbon stocks in each component ecosystem or land use area, and repeated measurements of key carbon stocks through time to provide estimates of change or flux of carbon into or out of the area. Estimates of carbon flux can be provided through direct field measurements or by modelling.

GLOBAL BLUE CARBON STOCKS

When Tier 2 or 3 estimates are not possible, Tier 1 estimates can be performed. The globally averaged estimates, shown in **Table 1.2**, can be used to give a Tier 1 estimate of carbon stocks within any given area if site-specific data does not exist. They are based on globally averaged carbon stock estimates for mangroves, tidal marshes, and seagrass meadows according to current literature. However, these estimates have a high degree of uncertainty.

Table 1.2 Mean and range values of soil organic carbon stocks (to 1 m depth) for mangrove, tidal marsh, and seagrass ecosystems and CO_2 equivalents. Examples of how carbon is distributed amongst the different ecosystems and the variation within each ecosystem (IPCC 2013)

ECOSYSTEM	CARBON STOCK Mg/ha	RANGE Mg/ha	CO_2Mequiv/ha
Mangrove	386	55 – 1376	1415
Tidal salt marsh	255	16 – 623	935
Seagrass	108	10 – 829	396

A Tier 1 assessment of a carbon stock within a project area can be achieved by multiplying the area of an ecosystem by the mean carbon stock for that ecosystem type.

FOR EXAMPLE

Questions being asked:

- How much carbon is stored in the biomass and top 1 m of soil in 564 hectares of mangrove forests on your project site?
- And how does that relate to CO_2 emissions if all the organic carbon in the upper 1 m of sediment is oxidized to carbon dioxide?

Total Carbon (MgC/ha) * Area (ha) = Tier 1 total carbon stock for the project site (Mg)

- Where Total Carbon = the mean carbon stock for a given ecosystem (from **Table 1.2**)
- Area = the area of the ecosystem being investigated

Answer to the first question

- *386 MgC/ha * 564 ha = 217 704 Mg of Blue Carbon in the study area*

Total potential CO_2 emissions per hectare (Mg CO_2/ha) = Conversion factor for the CO_2 that can be produced from the carbon present in the system * carbon in the system

- Conversion factor = 3.67, the ratio of the molecular weights of CO_2 (44) and carbon (12)
- Carbon in the system = the mean carbon stock for a given ecosystem

Answer to the second question

- *217 704 Mg of Blue Carbon * 3.67 = 798 974 Mg CO_2 in the study area*

CONCLUSION

This manual provides specific instructions for the field collection and laboratory analysis of carbon pools in mangrove, tidal salt marsh, and seagrass systems and some additional guidance for measuring GHG emissions, such as CO_2 and methane, which may be appropriate for some projects. Depending on the level of detail and the accuracy of the measurements used, this manual should be able to provide estimates that meet the IPCC standards for Tiers 2 and 3. It should be noted that the technical aspects of quantifying coastal ecosystem carbon and removals described in this guide are only one of several elements of complete carbon accounting. Other important elements including social, political, and economic factors—for example, addressing permanence, leakage, and governance—are not covered here. Definitions and information on those topics can be found in the IPCC guidelines (IPCC 2007) and associated sourcebooks (GOFC-GOLD 2009).

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Conceptualizing the Project and Developing a Field Measurement Plan

MEASURING CARBON STOCKS VS. CARBON POOLS

A *carbon stock* is the amount of organic carbon (C_{org}) stored in a blue carbon ecosystem, typically reported as megagrams of organic carbon per hectare (Mg C_{org}/ha) over a specified soil depth. These stocks are determined by adding all relevant carbon pools within the investigated area. Relatedly, *carbon pools* are reservoirs such as soil, vegetation, the ocean, and atmosphere that store and release carbon. Relevant blue carbon pools include:

- The living aboveground biomass, primarily herbaceous (for seagrass and tidal salt marsh) and woody (for mangroves) plant mass. This biomass also includes epiphytic organisms (e.g., algae and microbes living on the main plants).
- The dead aboveground biomass, primarily leaf detritus (in all three ecosystems) or wood (in mangroves), and other organic debris such as macro-algae.
- The living belowground biomass dominated by roots and rhizomes.
- The belowground carbon comprised of dead plant tissues and soil organic matter ('autochthonous' and 'allochthonous' carbon).

Carbon pools in blue carbon ecosystems are further differentiated into short-term pools (e.g., prevailing less than 50 years, e.g., living biomass) and long-term pools (e.g., prevailing for centuries or millennia, e.g., soil organic carbon). For blue carbon purposes, long-term carbon pools are the most important for determining carbon mitigation potential (IPCC 2007; Kyoto Protocol 1998).

CARBON STOCK ASSESSMENTS

During planning, a project's end goal must be clearly defined as this will influence both the design and execution of the assessment process. A clear end goal dictates geographic areas to include, carbon pools to measure, the level of specificity required, and the need/time scale for future reassessment. Moreover, the available resources must be considered to maximize the project's cost effectiveness.

The planning process has four essential elements:

1) Conception;
2) Carbon pool field sampling;
3) Sample preparation and laboratory analysis; and
4) Calculations for scaling up carbon stocks to the project area.

Conceptualizing the project is discussed in detail in this chapter. Detailed descriptions of the approaches and techniques required for field sampling of carbon pools and the specific laboratory techniques for analyzing each pool are presented in Chapter 3 (General Principles of Field Sampling of Soil Carbon Pools in Coastal Ecosystems) and Chapter 4 (General Principles of Field Sampling of Vegetative Carbon Pools in Coastal Ecosystems). Remote sensing options that may inform field site location and scaling up of results are presented in Chapter 6 (Remote Sensing and Mapping).

The approaches outlined here represent best practices based on the latest peer-reviewed research. However, project managers are encouraged to explore the literature and choose the best method for their project or they may choose to adapt these methods according to their local knowledge, training, resource constraints or other data collection needs, or the evolving nature of IPCC and related sourcebook guidelines.

CONCEPTUALIZING THE PROJECT

The main steps needed to prepare a robust field measurement plan are summarized in **Fig. 2.1**. Each step should be taken in a consistent, well-justified and well-documented manner. While Pearson *et al*. (2007) provides guidance on project design for terrestrial forests, much of the discussion is applicable to blue carbon ecosystems and, consequently, highly influenced the recommendations below.

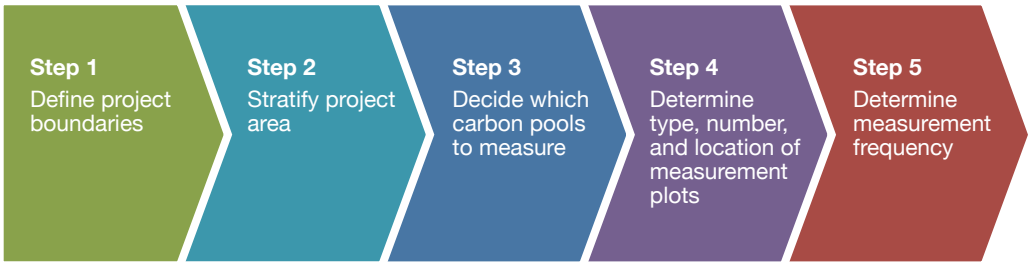

Figure 2.1 Steps to preparing a measurement plan

Define the Project Boundaries

Determining the project's spatial boundaries will depend on its scope and objectives. Project areas may range from a single site (tens of hectares) to national-scale assessments (hundreds of thousands of hectares). They may contain ecosystems that have been degraded or converted. The project area may be one contiguous block of land, or consist of many small patches of land spread over a wide area. Once the boundaries have been determined, every effort should be made not to change them; however, if changes are unavoidable, they must be well-documented, and any estimates of total carbon stock or carbon stock change must be adjusted to reflect the change in area.

Once the project location and scale have been decided, the next step is to map the area. Maps allow field teams to optimize their campaign by selecting locations for sampling that maximize geophysical range as well as environmental and biophysical variables within the area. Maps can also be used to verify accessibility through roads, tidal channels, and rivers. Starting with thorough and accurate mapping is valuable not only for determining where to sample, but also for extrapolating carbon measurements from individual samples to large scale project sites. If future reassessments are anticipated, an accurate initial map of the area will be vital for determining changes to the carbon stock and ecosystem services. Topographic, land use, soil, and vegetation maps, as well as aerial photographs may be acquired from local government agencies and used to discern project boundaries with varying accuracy. Satellite images and remote sensing techniques are very useful in mapping blue carbon ecosystems and for gathering information about the ecosystem more generally. Detailed information on remote sensing can be found in Chapter 6 (Remote Sensing and Mapping).

Stratifying the Project Area

Stratification is used to divide large heterogeneous sites (which require many samples to account for variation) into smaller more homogeneous areas (where fewer samples are needed) and is also useful when field conditions, logistical issues, and resource limitations prevent dense sampling regimes (**Fig. 2.2**). Stratification divides the project area into sub-areas or "strata" that are relatively ecologically homogenous (e.g., species diversity and geomorphology). Many tools exist to assist in stratification including local knowledge, satellite imagery, and geographic information systems.

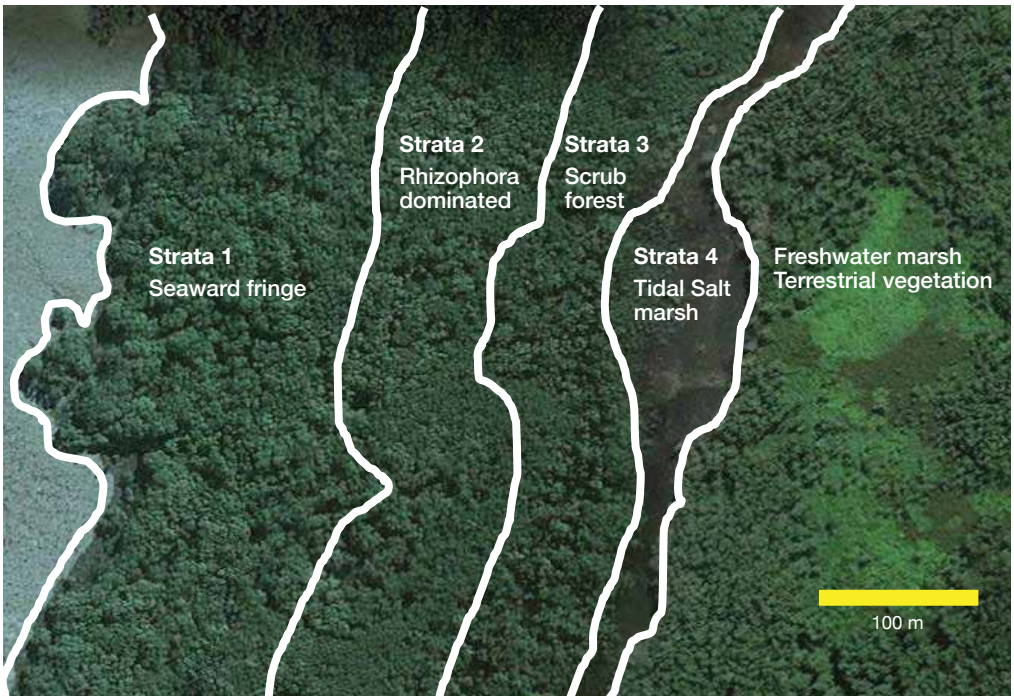

Figure 2.2 Example of mangrove stratification (© Boone Kauffman, OSU)

Stratification should be carried out such that the criteria used to define the strata are related to the variable being measured. For our purposes, the main variable being measured is carbon; thus, the features that influence the blue carbon content are used to determine the strata for a project area. Blue carbon stocks are largely influenced by variation in vegetative species and vegetative density. For example, mangroves contain tall forest, dwarf mangrove, shrub forests, and nypa palm (also sometimes known as Nipa, *Nypa fruticans*); tidal salt marshes consist of grass, shrub, and reed growth forms; and seagrass species vary according to water depths. These variations can guide strata delineation. Other factors that may be used to define strata include:

1) Existing land use (tidal salt marsh areas being used for agricultural purposes);
2) Potential land use (areas vulnerable to conversion for aquaculture or development);
3) Variations in soil characteristics (soil depth or type, or sediment grain-size);
4) Geomorphological features (proximity to geologic features, drainage features); and
5) Proximity to the ocean (areas tidally flooded daily or areas only flooded at the highest high tides).

While stratification is used to reduce the number of samples that are needed and increase carbon stock estimate accuracy, it is important to note that stratifying using strict criteria that create many small strata (that all need to be sampled) or conversely using loose criteria that create only a couple large strata (where some variation will be missed) negates this advantage. The strata size and number should be a balance between accuracy desired, time required, and resources available.

Decide Which Carbon Pools to Measure

Each stratum established in the project area will usually contain more than one carbon pool. The project purpose and objective will determine which carbon pools within each stratum to measure. Not all pools will be significant or require quantification for all projects. Projects may choose not to account for one or more carbon pools if they can prove that they will not significantly change the assessment results, but it is always better to measure all carbon pools in at least a few representative sampling sites.

In general, a carbon pool should be measured:

- If it is a significant portion (e.g., > 5%) of the total carbon in the stratum;
- If it is likely to or has changed significantly (either naturally due to climate change and extreme weather or due to human impacts such as land-use change or dredging); or
- If the carbon pool is unknown

Small carbon pools or those unlikely to be affected by change may be excluded or sampled less frequently depending on the project budget and other constraints. In most blue carbon systems, **soil carbon is by far the dominant carbon pool**. However, it is usually necessary to measure additional carbon pools in order to conform to requirements for carbon project certification. For example, both national carbon accounting and carbon market projects require four basic carbon pool measurements: aboveground living biomass (e.g., trees, grass, shrubs), aboveground dead biomass (e.g., leaf litter, downed wood), belowground living biomass (e.g., roots and rhizomes), and soil carbon.

In all three habitats, it is common to have vegetation and sediment that has traveled from surrounding habitats to the project site. For example, seagrass beds will often have a few mangrove propagules and leaves, and seagrass leaves are common in mangroves and tidal salt marsh sediments. Organic matter derived from terrestrial uplands can also be transported to and incorporated within blue carbon ecosystems. In most cases, this allocthonous organic material does not make up a significant proportion of the total ecosystem biomass and can be ignored. However, if the allocthonous material present is significant (> 5%), it can be classified as its own pool and quantified directly. In some cases allocthonous organic carbon present in the soil pool can be quantified using stable isotopes, but this may not be practical in all areas or needed for all projects (Johnson *et al.* 2007).

MANGROVE CARBON POOLS

Similar to most terrestrial forest ecosystems, mangroves can be roughly divided into four carbon pools (**Fig. 2.3**):

- Aboveground living biomass (trees, scrub trees, lianas, palms, pneumatophores);
- Aboveground dead biomass (litter, downed wood, dead trees);
- Belowground living biomass (roots and rhizomes); and
- Soil carbon which includes the dead below-ground biomass.

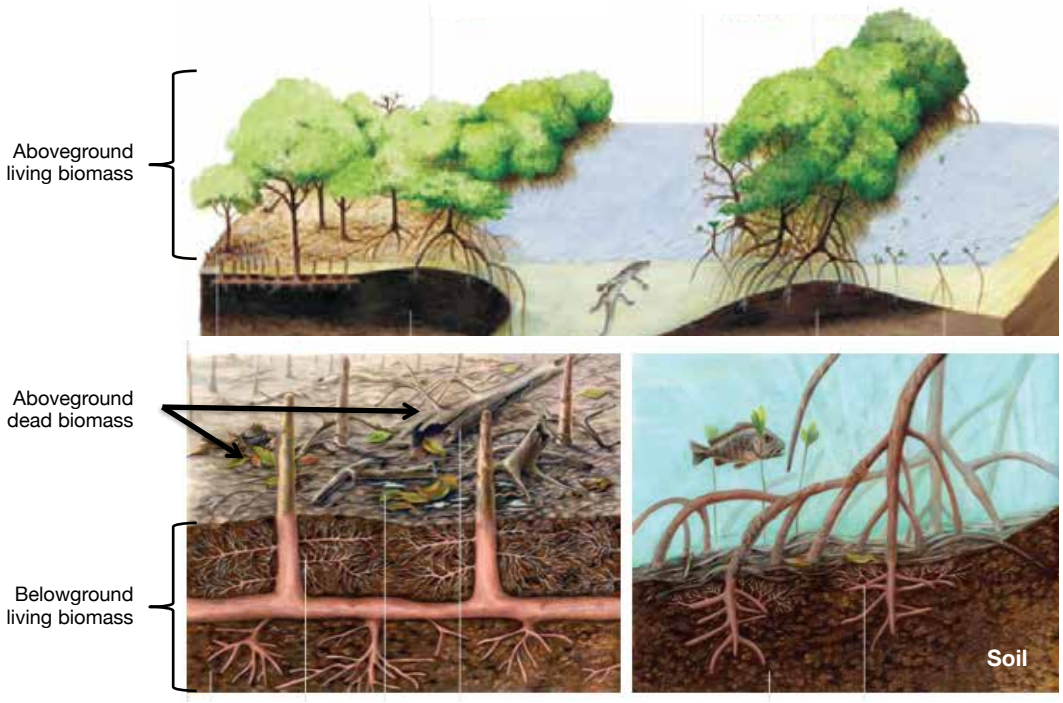

Figure 2.3 Carbon pools in mangrove ecosystems

In mangroves, all trees are included in the assessment because they are a large carbon pool (up to 21% of the carbon stock), are relatively easy to measure (due to well documented allometric equations that convert plant biomass to carbon content), and are heavily affected by land use. Dead wood can be an important pool (2.5–5.0% of the carbon stock), but may be especially important following disturbances such as land-use activities or tropical storms (Kauffman & Cole 2010). The live root component (5–15% of the total belowground carbon stock) is difficult to measure, but some allometric equations do exist (Chapter 4). Non-tree vegetation and leaf litter are usually minor ecosystem components and often can be excluded from measurements without compromising the accuracy.

Blue carbon is mostly stored belowground in organic-rich soils many meters deep where it can remain for very long times (up to millennia). The large size of these belowground pools and their poorly understood vulnerability to land-use change makes their measurement extremely important.

TIDAL SALT MARSH CARBON POOLS

Three major carbon pools can be considered in tidal salt marsh ecosystems. (**Fig. 2.4**):

- Aboveground living biomass (shrubs, grasses, herbs, etc.);
- Belowground living biomass (roots and rhizomes); and
- Soil carbon.

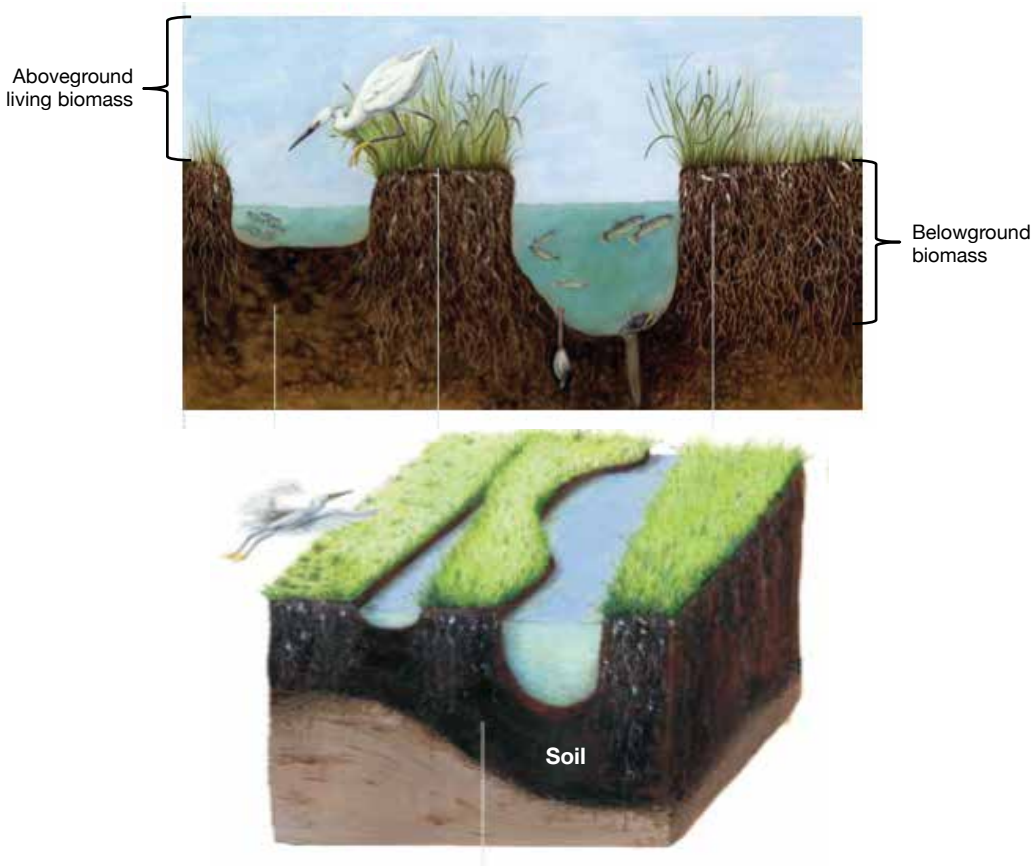

Figure 2.4 Carbon pools in tidal salt marsh ecosystems

In tidal salt marshes, most annual primary production occurs in the belowground biomass (roots and rhizomes) (Valiela *et al*. 1976) with root-to-shoot ratios (e.g., belowground to aboveground biomass ratio) ranging from 1.4 to 50 in salt marsh plants (Smith *et al*. 1979; Darby & Turner 2008a). Thus, most carbon in tidal salt marshes is stored in the living belowground biomass and the non-living soil carbon pool. These two pools are often difficult to separate and therefore treated as a single carbon pool (Chmura *et al*. 2003). Dead aboveground biomass is usually carried away with the regular tides and can usually be excluded from measurements without compromising the accuracy.

SEAGRASS CARBON POOLS

Three major carbon pools can be considered in seagrass meadows. (**Fig. 2.5**):

- Aboveground living biomass (seagrass leaves and epiphytes);
- Belowground living biomass (roots and rhizomes); and
- Soil carbon.

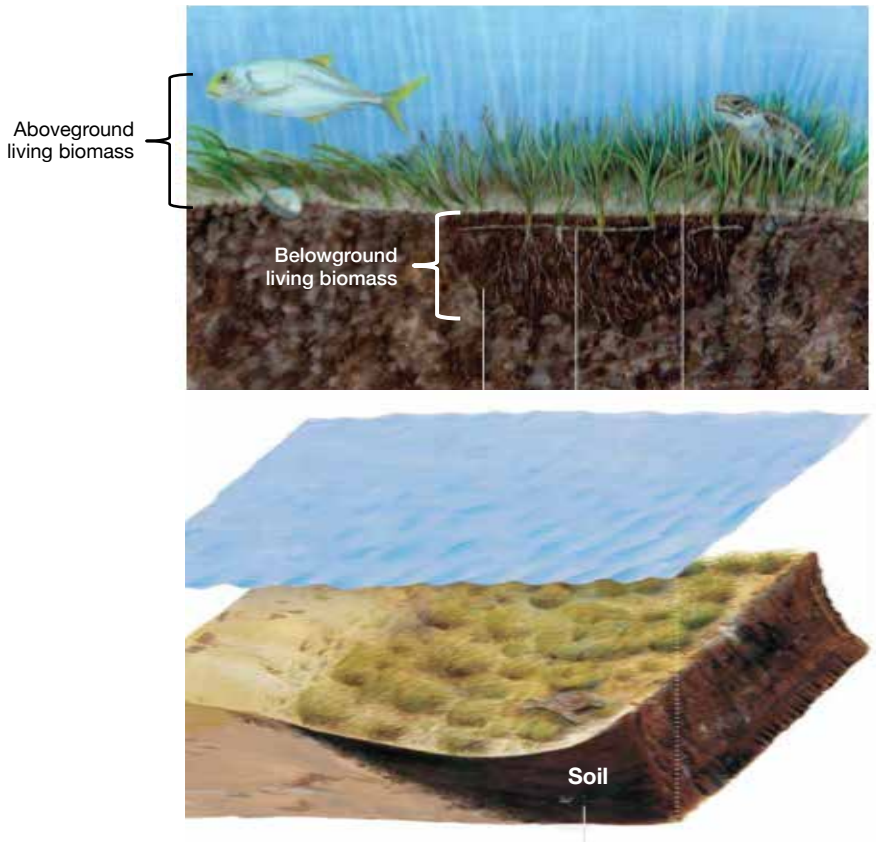

Figure 2.5 Carbon pools in seagrass ecosystems

The largest carbon pool in seagrass ecosystems is the soil carbon. Dead aboveground biomass is usually negligible since seagrass leaves are rapidly decomposed and/or rapidly exported from seagrass meadows through the movement of tidal waters. Epiphytes are noted as a carbon pool; however, their size varies by species and location.

On a global scale, belowground living seagrass biomass only represents 0.3% of the total organic carbon pool found below the surface. Therefore, it can usually be combined with the soil carbon pool estimate without significantly over-estimating the soil organic carbon pool (Fourqurean *et al*. 2012a).

Determine Plot Type, Number, and Location

Determining the minimum number of plots needed to ensure accuracy will aid in keeping initial field measurement and long-term monitoring program costs as low as possible. However, sampling density will ultimately be determined by the project goal and desired accuracy.

A Tier 2 National assessment requires data sufficient for estimating national or regional carbon stocks and may be achieved with a relatively low sampling density that covers a relatively large area. In contrast, a carbon market project requires a higher level of accuracy achieved through increased sampling over a smaller area over time.

Based on the carbon pools present in the ecosystem and the project area stratification, there is a need to determine the optimal shape, size, and sampling intensity necessary to accurately describe the ecosystem properties without needless redundancy. As such, plot design should be done with the project objectives, accuracy, sampling efficiency, and safety in mind.

PLOT TYPE

For assessing blue carbon stocks, two sampling plot types can be used: permanent and temporary. Permanent plots have greater long-term value and credibility in determining carbon stock changes through time, but temporary plots may be more practical.

Permanent plots are persistent, well-demarcated areas that allow for directly comparable measurements to be taken over time.

- **Advantages**: Stratification and plot design are only done once; they are statistically more accurate for determining carbon stock changes over time because the same plot and vegetation is measured at both points in time; they provide low cost verification as an independent verification organization can measure permanent plots and make a direct comparisons (Pearson *et al*. 2007).
- **Disadvantages**: Sites can be manipulated and enhanced (improved management, increased plantings, etc.) to make it appear that more carbon has been sequestered than is true for the rest of the strata; plots can be lost due to natural disasters or anthropogenic intervention thereby requiring enough plots to provide an accurate measure should some be destroyed. (Pearson *et al*. 2007).

Temporary plots are used to generate a single blue carbon measurement. They can be used to determine carbon stock change over time; however, measurements are not directly comparable thereby reducing accuracy.

- **Advantages**: They are cheaper to set up as they do not require permanent demarcations; a new location can be chosen relatively easily if the area where the original samples were taken is lost.
- **Disadvantages**: More plots may be needed to achieve the required precision level.

NUMBER OF PLOTS

The optimum number of plots depends on the accuracy level required, the inherent biomass variability between plots within the same strata, and the cost associated with sampling. Ideally there would be an existing carbon estimate for the study site, and the variance associated with those measurements would be known. Under those circumstances Pearson *et al*. (2007) provides comprehensive methods for determining plot number based on known intra-strata variation (an online tool for calculating the number of plots is also available at: http://www.winrock.org/resources/winrock-sample-plot-calculator). However, the more likely scenario

is that no information is available. In this situation, the first time the area is sampled, it is recommended to examine as many plots as the resources (budget and staff time) allow. Subsequent measurements can then use these initial data to determine if more or fewer plots are needed to achieve the desired accuracy level.

It is important to note that some areas may naturally be highly variable; therefore, the minimum number of plots needed may not be known or practical. High variability areas (largely due to soil carbon variations at various depths) will have higher uncertainties (the uncertainty level simply needs to be reported along with any results). The project manager will need to determine how much effort is feasible.

PLOT LOCATION

Plot location should be arranged to minimize disturbance to the ecosystem, while accounting for the variation within strata. There are several methods by which plot placement can be determined. The most common are:

1) **Linear:** This method can be used when either the stratification procedure shows that strata are most logically based on the distances from a location (river, shore, tidal channel) or when traversing the distance between randomly placed plots is prohibitive; however, the actual variability may not be represented (**Fig. 2.6a**).
2) **Random:** Within each strata, plots are picked at random to increase the likelihood of capturing the true variation within and across strata (**Fig. 2.6b**).
3) **Probability-based grid:** This method uses a square or hexagonal cell overlay placed within the defined strata where one random point is sampled from each cell. This method allows for sampling to be evenly spread throughout the strata while still maintaining the assumptions required for a random sample (**Fig. 2.6c**).

The method chosen will depend on study site accessibility, but when possible random or probability-based grid plot design is recommended. If the strata were defined properly, no significant differences will exist between plots. If significant differences ($p > 0.05$) do exist, sampling additional plots for robust estimates may be needed, or simply report the variability found within the stratum. The latter, however, reduces accuracy.

PLOT SHAPE AND SIZE

There are many acceptable plot sizes and shapes that can adequately describe ecosystem composition, biomass, and carbon content. Sample plot shape and size is a trade-off between accuracy, time, and cost for measurement. For example, large square plots (e.g., 100 m^2) are relatively simple to define, require only basic equipment to mark boundaries (measuring tapes and stakes), and sampling in these areas is relatively time- and resource-efficient. However, sampling a small number of larger plots limits the total area that can be surveyed. As a result, the entire project area may not be represented and, as a consequence, carbon assessments extrapolated from these plots will not be as accurate. In contrast, sampling many small, circular plots (e.g., 10 plots, 14 meters diameter) will be more time- and resource-intensive, but will encompass more variation in the project area and support more accurate carbon accounting. Many smaller plots are, therefore, more suitable where variation within the project area is high and resources are available.

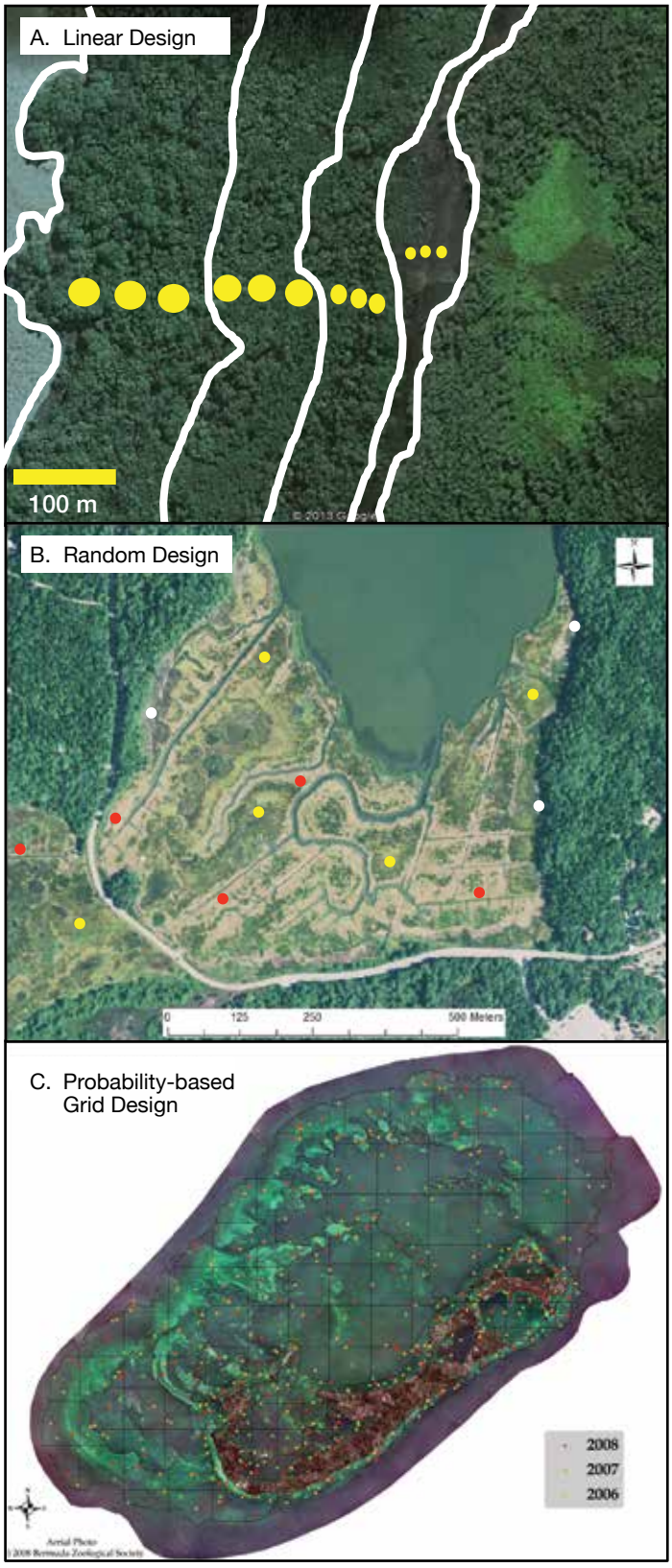

Figure 2.6 Plot location strategies. A) Linear plot design is common in densely vegetated areas where traversing between sites is prohibitive and when there is an inherent gradient (© Boone Kauffman, OSU). B) Random plot design places plots at random location within each strata, this assures that all strata are equally represented (red, yellow, and white dots represent potential sites for plot locations in the low, high and higher marsh, respectively) (© Beverly Johnson, Bates College). C) Probability-based grid design utilizes software (e.g., ArcView) to create grids that fit over, and are proportional to each stratum. Software is used to generate random points in each grid to be surveyed (© Sarah Manuel, Department of Conservation Services, Bermuda).

Plot size is predominantly determined by map resolution or dominant vegetation size. Plot size based on map resolution is useful when trying to validate remote sensing techniques for creating carbon maps. Plot size based on vegetation size may vary within a single assessment to accommodate different pools or strata within a system. For example, in a mangrove system, the largest trees and plants are sampled in the largest plots (e.g., 100 m^2). The high density of smaller trees, lianas, and palms make it practical to sample them in smaller plots or a series of plots (e.g., 10 m^2). Litter, seedlings, and grasses can be adequately sampled in even smaller plots (e.g., 1 m^2). Similarly, plots for a large seagrass species such as *Posidonia* spp could be bigger (e.g., 1 m^2) than the plots for grass-dominated salt marshes (e.g., 0.50 m^2), or smaller seagrass species, such as *Halophila* spp or *Zostera* spp (e.g., 0.25 m^2).

SUBPLOTS AND CLUSTERING

If several plot sizes are required for sampling, it is often most efficient to determine the largest size needed and nest the smaller plots into the larger one. Nested plots are designed such that the largest vegetation components (e.g., trees) are measured over the entire plot, but smaller components (e.g., shrubs, grasses) are measured over a smaller area within the larger plot (**Fig. 2.7a**). Clustering is when the largest plot area is divided into sub-plots such that the total area being sampled remains the same but the cluster design is able to capture more variation found within a plot location increasing overall precision.

For example, it might be determined that a mangrove area requires a circular plot about 1520 m^2 (22 m radius) in area for the larger components but nested plots about 250 m^2 (9 m radius) in area is fine for the smaller components. The total area to be measured can be divided into five

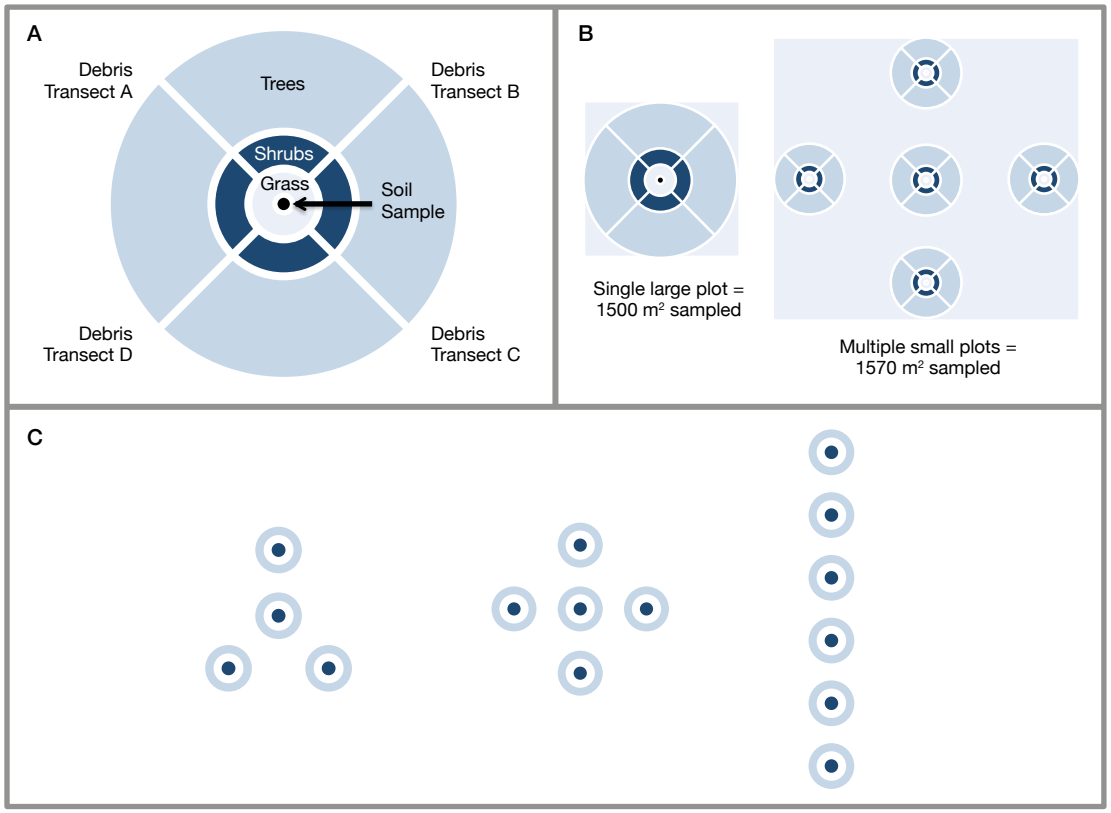

Figure 2.7 Plot nesting and clustering designs. A) Nested plot design where smaller plots are centered. B) Example showing the spatial coverage using single large plots vs. multiple small plots, the area sampled is similar but the total area represented is larger with multiple plots. C) Examples of radial and linear clustered plot designs. (USDA 2008; Kauffman & Cole 2010; Donato *et al*. 2011; Kauffman & Donato 2011)

subplots consisting of a larger sub-plot that has a 10 m radius (~ 314 m^2 * 5 subplots = 1570 m^2) and nested 4 m radius sub-plot (~ 50 m^2 * 5 subplots = 250 m^2). The resulting total area sampled is roughly the same, but the clustered sub-plots reduce between-plot variance and, therefore, the total number of plots necessary to achieve a desired accuracy (USDA 2008; Kauffman & Donato 2011) (**Fig. 2.7b**)

It is important to make some practical considerations when planning sample subplot design and layout. There are many benefits to linear subplot layouts (**Fig. 2.7c**). Linear subplots ease maneuvering in dense mangroves or muddy intertidal marshes, minimize trampling damage, and encompass the variation along an inherent gradient. A radial plot design may be more appropriate for smaller sites and produce more representative data.

Determine Measurement Frequency

The frequency required to conduct (and repeat) carbon stock assessments depends on the assessment objectives and the rate of expected change in the ecosystem being studied. Carbon stocks change in mangroves, tidal salt marshes and seagrass meadows for numerous reasons and vary in impact and time-response. Such changes can include natural disturbances (e.g., typhoons), variations in plant productivity and natural carbon sequestration rates, changes in land cover due to land-use activities (e.g., aquaculture or upland agriculture), and alterations due to climate change (e.g., sea level rise). Sampling frequency also involves establishing requirements for regulation, management or financing, and resource availability. It also depends on the pool being measured. For example, the aboveground biomass pool in seagrass beds will change more rapidly than the carbon stock in underlying soils. Also, seasonal growth/die off patterns in aboveground living biomass oscillates throughout the year and will most likely cancel each other out. For standing stock measurements, we recommend sampling be conducted at peak aboveground biomass (typically late summer). repeated sampling should occur at the same time of year (Fourqurean *et al*. 2001).

Given blue carbon ecosystem dynamics, approximately five-year intervals are sufficient to monitor aboveground pools (Pearson *et al*. 2005; Pearson *et al*. 2007). For carbon pools that respond more slowly (e.g., soils associated with mangroves, tidal salt marshes, and seagrass meadows), longer periods can be used—perhaps 10 or even 20 years between sampling events, if no sudden perturbations affecting soil integrity occurred. However, long intervals risk that natural or anthropogenic disturbances will be missed (Pearson *et al*. 2007). Therefore, irregular or unexpected events, such as strong tropical storms, rapid sea level rise, or land-use change, may justify sampling at more frequent intervals than originally planned.

CONCLUSION

A project designed with the end goal in mind is mandatory to obtain reliable and robust carbon stock estimates. Project designs will vary depending on local requirements and the ecosystem type. Once the project details have been decided, measurements can commence. Field techniques for measuring the aboveground and belowground living biomass in the different ecosystems vary between mangroves, tidal salt marshes, and seagrass meadows and are described in the ecosystem specific sections of Chapter 4. However, the techniques for sampling carbon contained in the sediments and soils are generally applicable to all three ecosystems and discussed in Chapter 3.

QUICK GUIDE

Step 1: Define the project boundaries

- Depends on the scope and objective of the project (single area up to national scale assessments)
- Ensure that the area to be assessed adequately represents the range of species and growth forms found in that ecosystem

Step 2: Stratify the project area

- If the project area is composed of various distinct biological structures (e.g., tall mangrove tree areas, dwarf mangrove areas, palm areas), it may be desirable to stratify the project area into sub sections of relatively homogeneous units/strata
- Remote sensing and satellite images are useful for this purpose, but local expertise is also needed
- Care should be taken to not include adjacent marine, upland, or freshwater ecosystems

Step 3: Decide which carbon pools to measure

- Common carbon pools to measure are:
  - Aboveground living biomass (trees, shrubs, grass, etc.)
  - Aboveground dead biomass (downed wood, leaf litter, etc.)
    - May not be relevant to all ecosystems. Tidal marshes and seagrass meadowstend to not have large enough pools of downed wood and leaf litter due to the composition of the local vegetation and removal of debris by tidal waters and currents.
  - Belowground biomass of live vegetation (roots and rhizomes)
    - Sampling will depend on feasibility
  - Soil
    - The most carbon-rich pool in these ecosystems
- A pool should be measured if
  - It is large
  - It is likely to be affected by land use
  - Future land use is uncertain
  - The pool size is uncertain
- Determine type, shape and size, number, and location of measurement plots
  - Type
    - Decide if plots are going to be temporary (single measurement) or permanent (continued monitoring)
  - Shape and size
    - Reliable data can be obtained from circular or rectangular plots
    - Plot shape and size is determined by the level of accuracy needed, time, risk, and cost
    - A nested plot design is recommended with sizes corresponding to the spatial scale of the component of interest
  - Number
    - Should have enough plots to reach a high level of statistical certainty ($p < 0.05$)
    - An online tool for calculating the number of plots needed is available at www.winrock.org/Ecosystems/tools.asp
    - If the project area has been stratified plot number must be determined for each strata
  - Location
- To avoid bias, plot selection should be random (e.g., along a transect) and selected without any prior knowledge of composition or structure within strata

Step 4: Determine measurement frequency

- Depends on the rate of expected change (natural disasters, land use change, climate change, etc.), requirements for participation in carbon markets, and the cost involved in sampling and laboratory analysis linked to resource availability
- Annually may yield best estimates but is costly and often more than is needed to monitor changes
- 5-year intervals are common and coincide with recommendations for participating in carbon markets
- 10–20-year intervals are also common but run the risk of missing natural or anthropogenic disturbances

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Field Sampling of Soil Carbon Pools in Coastal Ecosystems

GENERAL CONSIDERATIONS

Once the details of the project and sampling requirements have been determined, field sampling of the ecosystem carbon pools can begin. Field techniques for measuring the aboveground and belowground living biomass in different ecosystems vary between mangroves, tidal salt marshes, and seagrass meadows and are described in the ecosystem specific sections of Chapter 4. However, the techniques for sampling carbon contained in the soils, discussed in this chapter, are generally applicable to all three ecosystems. It is important to note that belowground carbon is sometimes referred to as sediment carbon or as soil carbon. For the purposes of this document, we use these terms interchangeably.

Belowground carbon pools usually termed soil carbon—dominated largely by the living and decomposing roots, rhizomes, and leaf litter—are usually the largest pool in vegetated coastal ecosystems and their measurement is critical for determining long-term changes in carbon stocks associated with disturbance, climate change, and land management changes. Belowground Soil carbon pools usually constitute 50% to over 90% of the total ecosystem carbon stock of mangroves (Donato *et al*. 2011; Kauffman *et al*. 2011) (**Fig. 3.1**). The proportional contribution of soil carbon is often higher (> 98%) for tidal salt marshes (Johnson *et al*., in prep) and seagrasses (Fourqurean *et al*. 2012a). Despite the importance of belowground soil carbon pools, they are the least studied. This is likely due to the novelty and recent recognition of the significance of belowground soil carbon in these systems as an important source of carbon globally (Smith 1981; Chmura *et al*. 2003; Laffoley & Grimsditch 2009; Donato *et al*. 2011; Fourqurean *et al*. 2012a). It is important to note that soil carbon takes a long time to accumulate and recently established or restored blue carbon ecosystems may not have a significant soil carbon pool for several years.

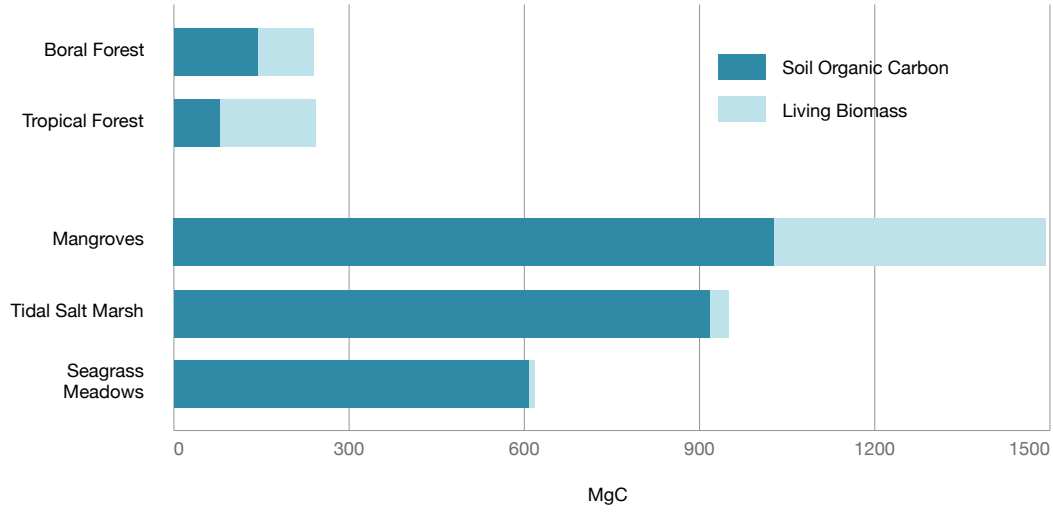

Figure 3.1 Mean carbon storage in the above- and belowground biomass in coastal vegetative ecosystems vs. terrestrial forest (Pan *et al*. 2011; Fourqurean *et al*. 2012a; Pendleton *et al*. 2012)

All soils contain both organic and mineral components; the percentage of each is what classifies a soil type as either an organic or mineral soil. For the purposes of this work, organic soils are defined as those comprised of more than 20% organic matter, whereas mineral soils are those comprised of less than 20% organic matter (USDA 1999). However, the criteria soil scientists use to define organic and mineral soils are much more specific than those presented here and are not defined consistently across the globe. Organic rich soils develop

where there are high rates of organic matter burial and preservation and low rates of mineral/soil deposition. Mineral rich soils develop when there is a large flux of sediment derived from terrestrial sources (e.g., from river input), estuarine sources (e.g., tidal movement of sediment), or from calcium carbonate produced by calcifying organisms (e.g., shell material). In general, organic soils appear dark and have a high concentration of decomposing plant fragments. Mineral soils are sandier and contain more shell fragments (**Fig. 3.2**).

Figure 3.2 Examples of organic and mineral soil. (A) Organic soil; Terraba Sierpe National Park, Costa Rica (© Sarah Hoyt, CI), (B) Sand & clay (mineral) soils; Patos Lagoon, southern Brazil (Bruno Lainetti Gianasi, © Margareth Copertino, FURG)

Soil carbon accumulation in upland forest usually does not exceed 30 cm and corresponds to the depth of common anthropogenic activities that may affect the soil pool (e.g., tilling). Hence, many carbon assessments of upland forests have limited their field sampling of soils to 30 cm depth. Mangroves, tidal salt marshes, and seagrass meadows often have organic-rich soils that range from 10 cm to over 3 m in depth and the disturbance of the organic-rich soils due to land-use and climate change in coastal ecosystems will likely affect deeper layers through drainage, oxidation, collapse, sea level rise, etc. (Hoojoer *et al*. 2006; Pendleton *et al*. 2012). Therefore, it is important to sample to greater depths in coastal ecosystems than in their terrestrial counterparts (a minimum depth of 1 m is standard but depths 3–5 m are common).

To accurately quantify the soil carbon pool, soil cores must be collected, subsampled, and analyzed for a specific depth (usually 1 m). Three parameters must be quantified for each field plot, sub-plot, and/or coring site to estimate the soil carbon pool:

1) Soil depth;

2) Dry bulk density; and

3) Soil organic carbon content ($\%C_{org}$)

Soil depth is determined with a soil depth probe or during the coring and sampling process. The dry bulk density and $\%C_{org}$ of soil are used to calculate carbon density. Because soil bulk density and $\%C_{org}$ vary with depth and location, there is not always a consistent pattern of carbon density with depth. Consequently, it is essential that an adequate number of soil cores (1 per plot, at least 3 plots per stratum) are collected and studied for a three-dimensional assessment of the carbon stock in each stratum.

Table 3.1 Equipment typically needed for field collections of soil carbon (**Fig. 3.3** images of equipment).

TOOL	PURPOSE
Soil depth probe (optional)	For measuring soil depth
Measuring tape	For measuring thickness of soil sampled and depth along the soil core
Sharp knife or 25 ml syringe	To subsample core
Soil coring device	To sample the soil core; (can also be used to determine soil depth)
GPS	To record the coring position
Plastic sample bags	To store samples
Waterproof writing utensils and tape	To label samples
Camera	To archive sample appearance and sample number

Figure 3.3 Equipment typically needed for field collections of soil carbon. (A) Measuring tape for measuring depth along the soil core (© Sarah Hoyt, CI), (B) Syringe used to subsample core (© Sarah Hoyt, CI), (C) Knife used for subsampling (© Boone Kauffman, OSU), (D) Core & sample bag (© Kcrishna Barros, UFC), and (E) GPS to mark coring position (© Boone Kauffman, OSU)

SOIL DEPTH

The ability to measure the depth of soil depends on two factors: 1) the nature of the soil (mineral vs. organic soil) and 2) the depth of soil relative to the equipment being used. Measuring the depth of soil is most difficult in deep (> 5 m) mineral rich soils and the easiest in shallow (< 5 m) organic rich soils (**Fig. 3.4**). In organic soils, the soil depth is usually defined as the depth to parent materials such as bedrock, or some other hard substrate (coral/minerogenic sands). It is sometimes feasible to accurately measure organic soil depth with a soil depth probe such as a bamboo pole or avalanche probe. When possible, extensions for the poles should be available to ensure complete penetration. In many cases "depth to refusal" (e.g., the depth at which the pole can no longer be inserted) is considered a reasonable estimate of organic soil thickness. "Depth to refusal" assumes that the organic soil is generally easier to penetrate with a rod than underlying sands and/or bedrock. Because the "depth to refusal" depends on the diameter of the pole and strength of those who are using it, as well as changes in the underlying soils, it is important to validate that "depth of refusal" represents organic soil thickness, at least initially, by also taking soil cores. If "depth to refusal" corresponds to the lower limit of the soil as seen in a soil core, then the pole method is sound. It is important to note that the presence of roots and fibers may stop penetration of the soil depth probe; thus, it is mandatory to measure soil depth in different random locations.

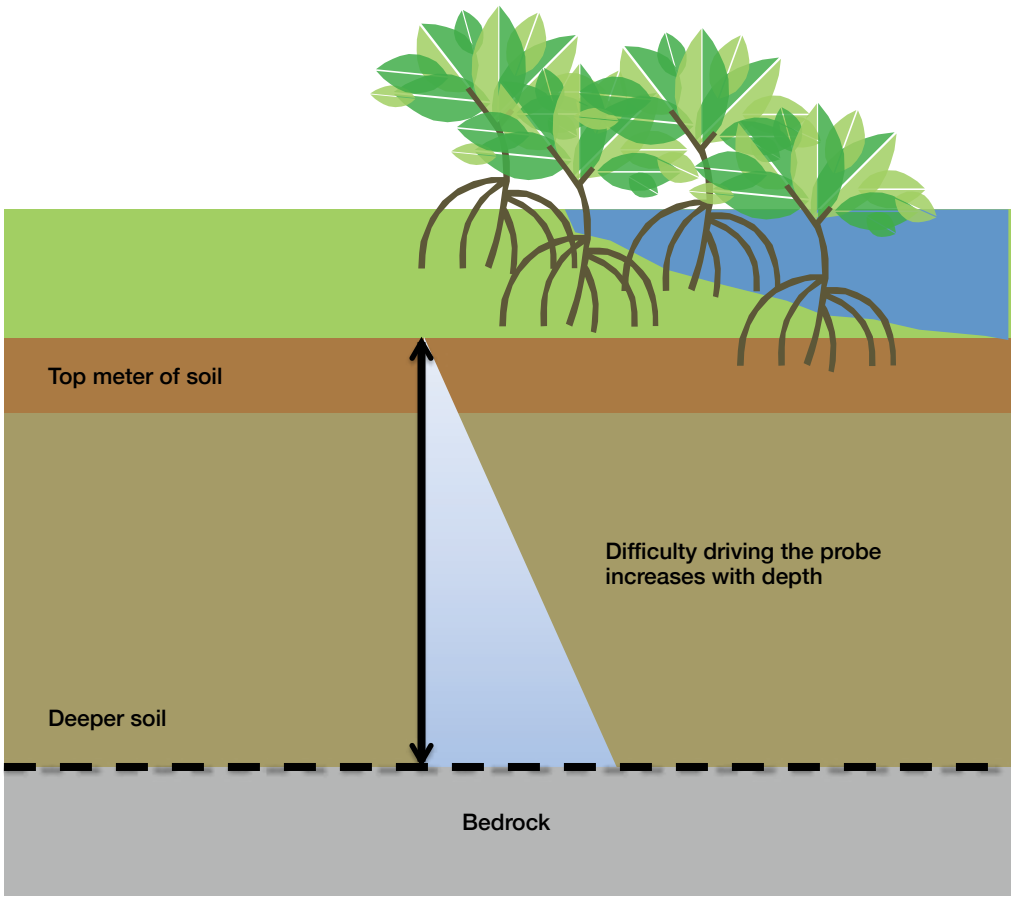

Figure 3.4 Measuring soil depth with a soil depth probe

In some instances (such as the organic-rich seagrass soils) the soil depth can only be accurately determined through the use of sophisticated sampling equipment or geophysical techniques, due to the presence of fibrous material and the large depth over which the organic soil has accumulated. Sampling can be achieved using a coring device, but even this may be very difficult for very deep soils as it requires heavy coring equipment. In addition, the depth of soil may be harder to define unless there is a clear change in soil type or there is an impenetrable boundary. All of these issues are compounded in mineral rich soils as they are often deep and harder to penetrate. In cases where the probe cannot be pushed or even hammered to the depths that the actual soil layer reaches, the only way to accurately establish organic layer depth is to take deep cores and use inspections of core samples to identify organic layers.

SOIL CORING

Obtaining soil samples for bulk density measurements and carbon content analysis requires soil-sampling equipment that allows for extraction of a relatively undisturbed soil sample that has undergone minimal compaction (**Table 3.2** summary of common soil coring devices). Specialized gouge augers for organic or peat soils are recommended such as the Russian peat corer, or Eijkelkamp gouge auger. Both are long (up to ~ 2m) semi-cylindrical chambers that are pushed into the soil, twisted, and then pulled out. The samples recovered should have undergone minimal compaction and extensions can be added so that long cores (3–5m) can be recovered. The Russian peat corer has a fin that closes prior to extraction thereby preventing soils from sloughing back out of the bottom of the corer. The Eijkelkamp auger has an open bottom, and soils can be lost out the bottom if they are wet or lack cohesiveness, such as unconsolidated sands.

In some mangrove, tidal salt marsh, and seagrass locations, simple piston coring devices are often effective. Such a device uses the suction created by a fixed piston at the top of the soil surface to pull the core sample into the core barrel as the core barrel is pressed/hammered into the soil.

In areas of high sand content where piston corers and gouge augers cannot easily penetrate the ground, the only options left are to either manually hammer a tube into the ground or use a vibracorer. A vibracorer entails attaching a heavy vibrating power head to an aluminum or plastic pipe and vibrating it into the underlying soils.

Because bulk density measurements may be altered by any coring technique (particularly hammering) if the soil is compressible, experimentation with different soil sampling equipment in representative sites is recommended to ensure the sampling of relatively undisturbed cores. The type of coring gear needed will vary according to the vegetation and soil type. For example gouge augers may be sufficient for organic rich marsh soils, but vibracores may be the best option for mangroves and seagrass rooted in sandy/muddy soils.

The presence of coarse plant fibers embedded in the soil may either prevent core penetration ("core refusal") or cause a "nail effect" (penetration of the corer without soil entering the tube). To sample cores in fiber-rich soils, it is desirable to ensure a sharp cutting edge on the bottom of the core tube. In practice this can be accomplished by sharpening and serrating the end of the core tube or by attaching a removable coring head. This coring method combines manual and mechanical percussion with rotation to cut through the fibers (Serrano *et al*. 2012).

Table 3.2 Soil coring devices.

CORING DEVICES	Russian Peat Corer	Eijkelkamp Gouge Auger
	Semicylindrical chamber with rotating fin designed to fill chamber from side; extensions available up to several meters.	Semicylindrical chamber with an open end; extensions available up to several meters.
	Advantages	Advantages
	Extensions allow coring up to 5m deep; undisturbed, uncompacted, soils; minimal sloughing out the bottom.	Extensions allow coring up to several meters; undisturbed, uncompacted soils recovered; simple construction, portable.
	Disadvantages	Disadvantages
	Depth recovered depends on strength of people coring. Fin may get jammed during the coring process.	Depth recovered depends on strength of people coring. Soils can slough out the bottom if they are wet or sloppy.
Piston Corer	**Bucket Soil Auger**	**Vibracorer**
Semicylindrical chamber with an open end; extensions available up to several meters.	Cylinder or barrel to hold the soil, which is forced into the barrel by cutting lips.	Large pipe is vibrated in the soil using a motor to force the core into the bottom.
Advantages	Advantages	Advantages
Can be used in saturated soils. No hassle with casings and coring tubes.	Universal approach to looking at soils in diverse settings.	Long cores recovered in one simple step.
Disadvantages	Disadvantages	Disadvantages
Rinse before and after use to avoid wear of piston, small diameter.	Provides a semi-undisturbed soil sample.	Compaction possible. Tripod or lifting equipment needed to extract cores. Not particularly portable.

Steps for Taking a Soil Core (Mangroves and Tidal Salt Marshes)

1) At the sampling location, the organic litter and living leaves, if present, should be removed from the surface before inserting the corer.

2) Steadily insert the coring device vertically into the soil until the top of the corer is level with the soil surface. The descent rate of the core has to be kept low (e.g., gentle hammering) to minimize core compaction. If the coring device will not penetrate to full depth, do not force it, there may be a large root or coral fragment in the way; instead try another location or use a coring system that is capable of cutting fibers (**Fig. 3.7**).

3) Once at depth, twist the coring device to cut through any remaining fine roots, and seal the top end (the vacuum will prevent the loss of the sample). Gently pull the coring device out of the soil while continuing to twist as it is being extracted. This twisting assists in retrieving a complete soil sample (**Fig. 3.7**).

Steps for Taking a Soil Core (Seagrass Meadows)

Figure 3.5 Sampling a soil core using a soil auger (© Boone Kauffman, OSU)

Steps for taking soil samples in seagrass systems are a bit unique because the soils are saturated with water, do not hold their shape as well, and are more susceptible to compaction. Further they can be underwater, requiring the operator to either hold their breath or use SCUBA. For fine-grained soils, thin-walled PVC pipe can be used as a core tube, and a piston can be constructed from a rubber bung, an eye bolt, washers and nuts (**Fig. 3.6**). For coarse-grained soils, which are harder to core, a thick-walled PVC pipe fitted with a piston is recommended.

Figure 3.6 Seagrass coring devices. PVC tubes, rubber stopper, and syringe (© CI/Sarah Hoyt)

The PVC pipe or core tube or barrel can then be driven using a sledgehammer or a post-pounder (**Fig. 3.7**). After the core barrel is driven to the desired depth cap the top with a stopper or duct tape and remove the core. The core barrel may be very difficult to remove and the use of a chain (or other non-stretching line) along with a hand-held winch is recommended. A portable tripod can be constructed from iron pipe (or a ladder) and a chain-block is used to keep the core barrel straight as it is being removed (**Fig. 3.8**). Another option is to excavate the core barrel out of the surrounding soil.

Once the corer is removed, cap the bottom with duct tape and keep upright while it is being transferred to the lab for subsampling. Note that it is very important to keep soils upright during transportation so that the core layers do not mix within the tube. If it is logistically difficult to transport the entire core vertically to the lab, subsamples should be taken at the site (see below).

Core Compression

Figure 3.7 A demonstration of method used to drive corer into soil in seagrass meadows. (A) Shallow water with a sledgehammer (© Sarah Hoyt, CI), (B) shallow water with a post-pounder (© Sarah Hoyt, CI), (C and D) deep water with a sledgehammer (© James Fourqurean, FIU)

Figure 3.8 Set up for core sampling in seagrass ecosystems. Instruments for seagrass soil coring: (A) ladder with crank for removing coring device from soft soil in shallow water (© Sarah Hoyt, CI), (B) sturdy tripod with weighted pulley system for removing coring device from soft soil in either shallow or deep water (© Oscar Serrano, ECU)

Compression of sediment layers (also known as core compaction or core shortening) comes from three sources; 1) weight from the sediments layers as they build over time, 2) decomposition of organic matter with aging, and 3) shifting of sediments during the coring process. Sediment layers settle one on top of another with the top layers creating pressure on the lower layers. As a result, sediment layers are tightly pressed together, and the top organic-rich and low-density layers may become denser with aging. These forms of compaction occur naturally and are difficult to determine, and therefore, are not considered. However, driving the coring tube into sediments will often compress the sediment, causing depth-variable changes in the bulk density of the sample (this is particularly true for seagrass soils) and this may skew the estimate of carbon stocks (**Fig. 3.9**). Cores that are much shorter than the depth to which the core tube was inserted in the soil may also result from the above-described "nail effect," (page 44) in which the core tube becomes plugged and consequently penetrates the soil as a solid rod or nail. Ideally, compressed samples would not be used in the soil carbon analysis, but it is sometimes unavoidable. Efforts should be made to limit compression as much as possible and record each sample where it occurs to allow corrections.

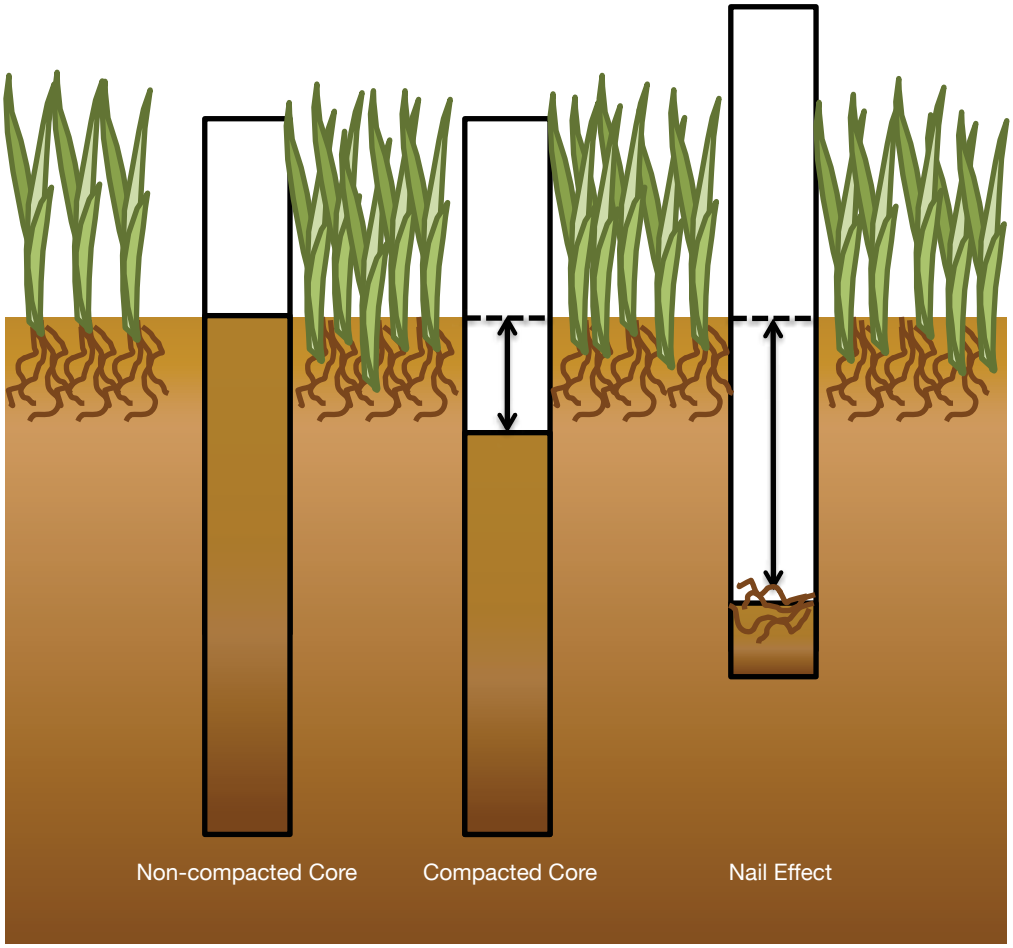

Figure 3.9 Diagram of soil core compaction that can occur while sampling. The top of a non-compacted core will be level with the surrounding ground (left). Cores can be compacted due to the force applied to the corer as it is driving into the soil (middle). The nail effect occurs when something (roots, rocks, shells, etc.) gets caught in the corer and compacts the soil underneath it (right).

If significant compaction has occurred, take another core nearby. Repeat until there is minimal compaction. However, even the most efficient practices for minimizing core compression (e.g., specially designed augers, coring at a low descent rate, and use of rotation and cutting head), can result in core shortening of up to 30% (Morton & White 1997). In these cases, a compression correction factor should be used to compensate for the "artificial" compression in the core sample recovered.

The compaction correction factor is calculated by dividing the length of sample recovery by the length of core penetration. During sample processing the corrected sample length is determined by multiplying the desired depth interval by the compaction correction factor.

FOR EXAMPLE

- A sample is recovered that is 150 cm long
- But the depth reached by the corer was 175 cm
- This will give you soil compaction of 25 cm, a compaction correction factor can be found by dividing the length of the sample by the corer depth (150 cm / 175 cm = 0.86).
- If we then wanted to obtain a sample that represents the top 10 cm of the soil we would need to multiply the depth interval (10 cm) by the compaction correction factor (10 x 0.86) giving a new sample recovery measurement of 8.57 cm.

For simplicity, a uniform compaction correction factor may be used for the entire length of the core. However, this technique assumes that all parts of the core are compacted equally, which may not be the case since bulk density and compactibility are likely to vary over the depth of the core. Thus, a more complex, but more accurate, method is to determine the degree of compression several times at different intervals during the coring process.

Dense Soils

If using an open-faced auger (e.g., Russian peat auger) the soil sample will be readily accessible and ready for archiving and subsampling. If using a close-faced auger coring system (e.g., piston auger), the core liner must first be cut open. To do this, remove the core liner and soil sample from the coring apparatus and cut the plastic or metal core line. lengthwise along opposite sides with a hacksaw, electric rotary tool, knife, or vibrating saw. It's important to control the cut depth to cut through the liner wall without cutting significantly into the soil and to avoid getting chips of plastic/liner into the sample. Once the liner wall is cut through along

Figure 3.10 Soil core liner that has been cut lengthwise to expose the soil for archiving and subsampling. This core is in the process of being split. A clean face is exposed in the lower part of the image. (© Boone Kauffman, OSU)

opposite sides, use a knife to cut the soil core lengthwise into two half-cylinders (also known as splits) using vertical cuts in discrete steps. Between each vertical cut, clean the blade properly, and slowly open the core. Take care to remove any plastic/metal chips resulting from cutting the liner using a brush and forceps. Clean up the face of one of the splits with a knife by gently scraping off a very thin layer from the surface of the split (upper 1 mm) by dragging the knife across the core barrel (**Fig. 3.10**). This will provide a fresh exposure of the soil for photographic archiving and description.

Loose Soils

For looser soils, there is a risk of mixing the soil layers if the core is laid on its side (for transport or subsampling, **Fig. 3.11**). A syringe can be used as a mini-corer to accurately subsample loose/saturated soils (**Fig. 3.12**). In these cases, soil cores are collected using a corer with predrilled sampling ports. For example, Fourqurean *et al*. (2012b) use a 5.2 cm diameter, diver-operated piston corer that is driven into the soils until refusal using a sledgehammer to when taking cores in seagrass meadows (Fourqurean *et al*. 2012b). The core tube is pre-drilled with 2.5 cm diameter sampling ports at 3 cm intervals. Before inserting the pre-drilled corer into the soil, the sampling ports are covered with duct tape. After the corer is extracted from the soil, it is kept upright to ensure no mixing occurs and returned to shore for sub-sampling. The tape is then slowly peeled downward, starting from the upper port and finishing at the lowest port, then a piston sub-corer made of a 25 mL cut-off polyethylene syringe (2.0 cm diameter) is inserted into each port, starting at the top, to extract a soil sample of known volume. It is important to always collect the same volume of soil in the syringe or note the volume sampled each time.

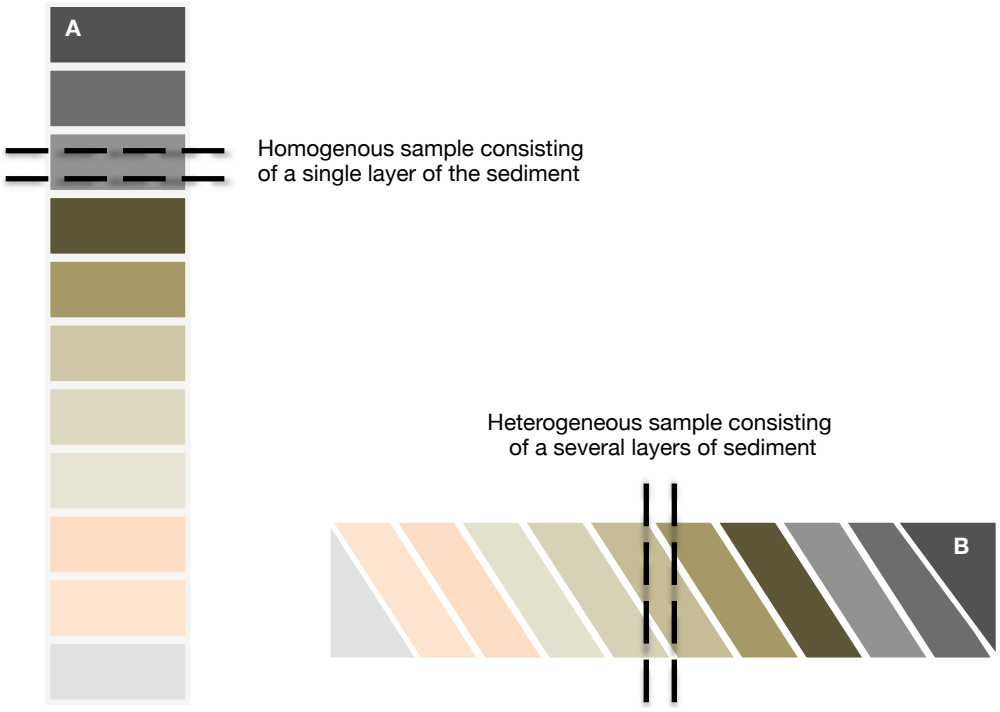

Figure 3.11 Examples of cores from water saturated/loose soil types. (A) Cores should be kept upright to prevent soil layers from mixing and allow for a consistent subsample. (B) Soft, unconsolidated soils when placed on their side allow for mixing of the layers, making the subsample inaccurate.

Figure 3.12 Cores are collected using a corer with predrilled sampling ports and sub-cores removed using cut-off polyethylene syringes (© Sarah Hoyt, CI).

Archiving the Core Prior to Sampling

A photographic archive of the appearance of the soil core is useful for planning the subsample technique and laboratory analyses. For example, if the soils are dark and have many plant fragments, they will be rich in carbon and less material will be needed for organic carbon analysis. If the soils are dominated by light colored sand, then more material will need to be analyzed to determine the organic carbon content.

To archive the core, take a GPS recording of your coring site and assign the site a unique label then photograph the entire core from top to bottom and record changes that occur with depth. For mangroves and many tidal salt marsh samples, photos can be taken in the field once the core has been recovered and one of the splits has been cleaned. Extend a tape measure along the core starting at the top end and document the split from top to bottom (surface to depth) using detailed photographs of core sections in overlapping frames so that the images can be lined up for a complete core image. Be sure to include the tape measure in these images of the core. Place a label with the core ID so that is appears in all photographs and identifies which direction is the top and bottom of the core and use a polarizing filter to limit the light reflected off the wet surface of the core.

Seagrass soils are more difficult because they must be kept upright. In this case, record a general description of the core subsamples as they change with depth observing zones of different color, texture, presence of plant debris and shells, sediment type (mud, sand, gravel), etc. Take photographs to complement the written descriptions, again making sure to have an ID visible in all photographs.

SAMPLING A SOIL CORE

Ideally, once the core is removed it is transported in its entirety to the laboratory for analysis. However, this is often not possible, and samples must be taken from the core in the field. The depths at which samples are taken from a soil core are an important decision. Preferably, it is best to sample the entire depth of the soil core, although this may not always be possible or practical. When soils are several meters deep the standard practice is to sample the top meter extensively and fewer samples of the deeper material (**Fig. 3.13**).

- **Mangroves**: Kauffman *et al*. (2011) and Donato *et al*. (2011) use a highly depth-aggregated sampling technique with samples taken from mangrove soils at depth ranges of 0–15 cm, 15–30 cm, 30–50 cm, 50–100 cm, and > 100 cm. At depths > 100 cm, soil samples are recommended to be collected at a maximum of 2 m intervals. These sampling intervals are

deemed adequate for mangroves because carbon content generally changes slowly with depth (Donato *et al*. 2011; Kauffman *et al*. 2011).

- **Tidal salt marshes and seagrass meadows**: Variations in carbon content are most significant in the upper 20 to 50 cm of soil (Choi *et al*. 2001; Connor *et al*. 2001; Choi & Wang 2004; Johnson *et al*. 2007; Fourqurean *et al*. 2012b); therefore, we recommend taking more detailed depth profiles. For example, 5 cm-thick samples can be collected continuously throughout the soil (or upper 50 cm). As organic content of these soil cores changes more slowly with depth below 50 cm, it may be practical to take fewer subsamples separated by larger intervals.

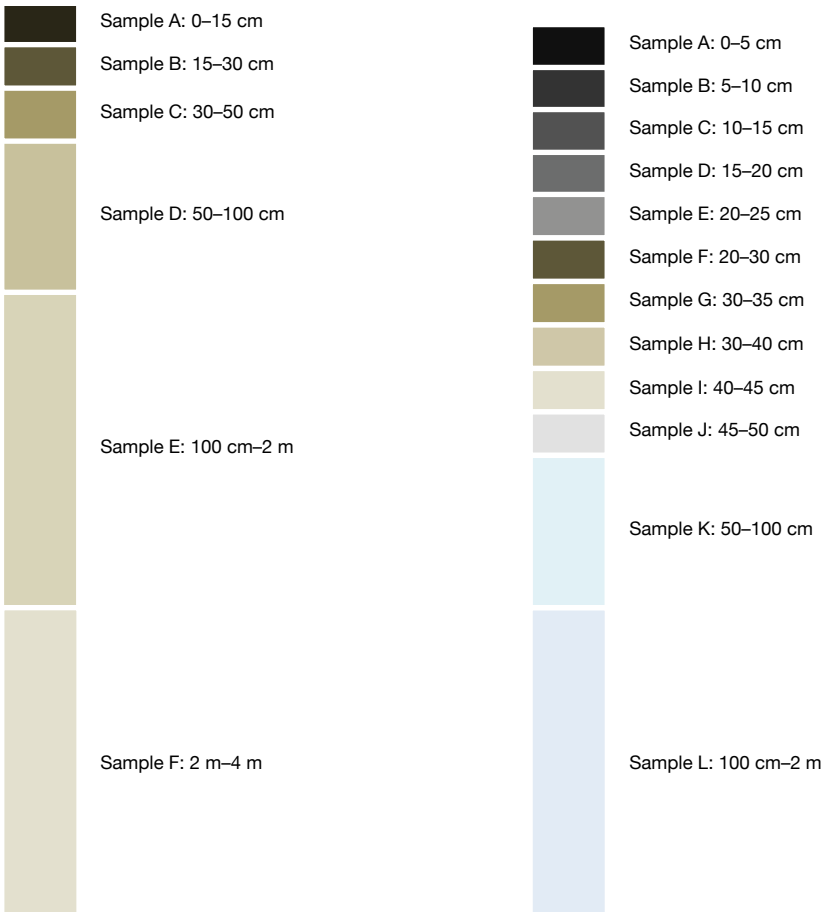

Figure 3.13 Alternative core sampling strategies

It is imperative that the samples be collected in such a way that its original volume can be determined. For example, if whole core sections are removed, the volume can be calculated using the depth interval of the section and the diameter of the core barrel. If using a syringe, the volume can be determined directly where 1 cc is equal to 1 cm^3.

SUBSAMPLING A SOIL CORE

The most accurate, and sometimes most practical, technique for subsampling is to determine the bulk density for each depth interval and then homogenize the subsample and determine the organic carbon content. Alternatively, subsamples can be taken directly from each depth

interval. To do this use a ruler or tape measure to determine the depths from which the subsamples will be collected. Subsample sizes are usually about 5 cm deep and will contain between 5 and 50 g of sample, depending on core barrel size and sediment composition. If not sampling the entire core, samples should be collected at the approximate mid-point of each desired depth range. For example, if sampling the 0–15 cm depth interval, the sample would ideally come from the 5–10 cm depth; for the 50–75 cm depth the sample would be collected at the 60–65 cm depth, and so on (**Fig. 3.14**). For dense soils, a knife can be used to remove subsamples (**Fig. 3.15**). The blade of the knife should be cleaned between each subsample. Upon collection, samples are each placed in individual, numbered plastic containers/bags with the site, plot number, core identification, soil depth, date, coring device used, diameter of core barrel for calculating volume, and any other relevant information (**Fig. 3.16**).

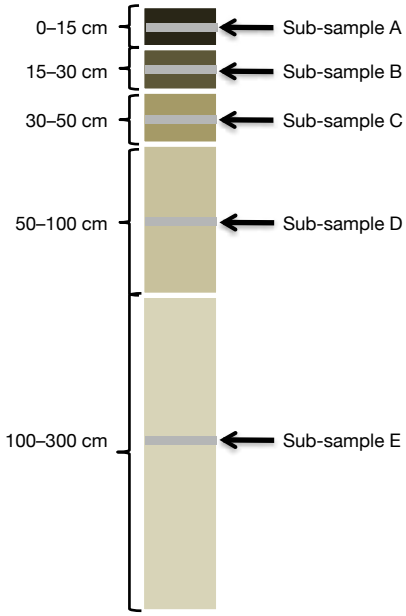

Figure 3.14 Core sub-sampling strategy

Figure 3.15 Collection of soil samples from open-face auger. (A) Cutting soil away from auger face, (B) Measuring and marking the depth intervals, (C) Cutting a sample, (D) Removal of sample from auger in numbered container (© Boone Kauffman, OSU)

Figure 3.16 Samples are each placed in individual, numbered containers. The number corresponds with sample identification information recorded in the field notes. (© Boone Kauffman, OSU)

ARCHIVING SAMPLES

The proper labelling of the cores and samples in the field is essential to avoid confusion and common mistakes in sample identification. Each sample/subsample should be labelled with a core ID, sample depth, and depth interval. A general recommendation is to print several copies of template labels on waterproof paper to bring with you in the field. Write on the label using a permanent marker, and attach the labels using duct tape or another water-resistant tape.

To minimize decomposition of organic matter and microbial growth, samples should be kept cold (4 ºC) and if possible, either frozen or dried (see section on sample preparation) within 24 hours of collection. Prior to analysis, frozen samples should be thawed and dried. Once dried, samples can be stored for years with minimal decomposition. Quarantine treatment (e.g., irradiation) does not affect the organic carbon concentration of dried samples.

LABORATORY ANALYSIS

To accurately determine the soil carbon density, two parameters must be quantified: soil dry bulk density and organic carbon content (C_{org}). Once dry bulk density (mass of dried soil/original volume) is determined, it can be used with C_{org} to determine the carbon density of the soil at specific depth intervals. The procedures for this analysis are as follows.

Determining Soil Dry Bulk Density

Dry bulk density (DBD) is determined from the mass of a fully dried sample and its original volume.

- Dry bulk density (g/cm^3) = Mass of dry soil (g) / Original volume sampled (cm^3)

DETERMINING ORIGINAL VOLUME SAMPLED

To determine the original volume you will need to know the type and internal diameter of the coring device used (e.g., closed tube coring device or syringe) and the thickness of the sample (if cut from a larger core) or the length of the sample (if taken with a syringe). The volume of the soil can be calculated using the mathematical formula for determining the volume of a cylinder, as follows:

- If the sample came from an intact core, use the following equation:
  - Original pre-dried volume of soil sample = [π * (radius of core barrel)^2] * (depth of the sample, h)
- If the sample came from a core split, the same equation can be used to determine the volume from an intact core, but volume calculated must be halved.
- If the sample was taken using a syringe, volume can be measured directly from the syringe where 1 cc = 1 cm^3.

DETERMINING THE DRY MASS

Dispense the soil sample onto a pre-weighed container, such as a petri-dish or a beaker and place in a 60 ºC oven to dry. The sample can be spread or carefully broken up into smaller pieces to improve the speed at which the soil will dry (**Fig. 3.17**).

Figure 3.17 Removal of sample from syringe and preparing it for oven drying. (A) Sample in syringe, (B) Depositing sample on pre-weighed petri dish, (C) Sample when first removed from the syringe, and (D) Spreading the sample with a spatula. (© Hilary Kennedy, UWB)

The soil sample should be dried until it reaches a constant weight. To determine when your soil has reached a constant weight, dry it at 60 ºC for at least 24 hours, and then cool it to room temperature in a desiccator for at least 1 hour before weighing (**Fig. 3.18**).

Weigh your sample in the petri dish before returning to the oven, dry it for another 24 hours, and re-weigh. This cycle is repeated until successive weight differences are less than 4% (always use the same balance). Typically, this process requires at least 48–72 hours.

Figure 3.18 Soil sample cooling to room temperature in desiccator (© Hilary Kennedy, UWB)

While we recommend drying the samples at 60 °C, other protocols recommend drying at 105 ºC for bulk density determination. This higher temperature is not advised because, some part of the soil organic matter may begin to be lost (oxidized) at temperatures greater than 60 ºC. Thus, the weight loss recorded at 105 ºC would potentially represent both water loss and loss of organic matter, resulting in an underestimation of organic carbon.

Once the sample has reached a stable weight, the mass of the sample, along with the volume calculated above, is used to determine DBD. Note that the inorganic carbon (e.g., carbonate shells) should NOT be removed prior to bulk density analysis. Some representative distributions of bulk density at various depths in blue carbon soils are shown in **Fig. 3.19**.

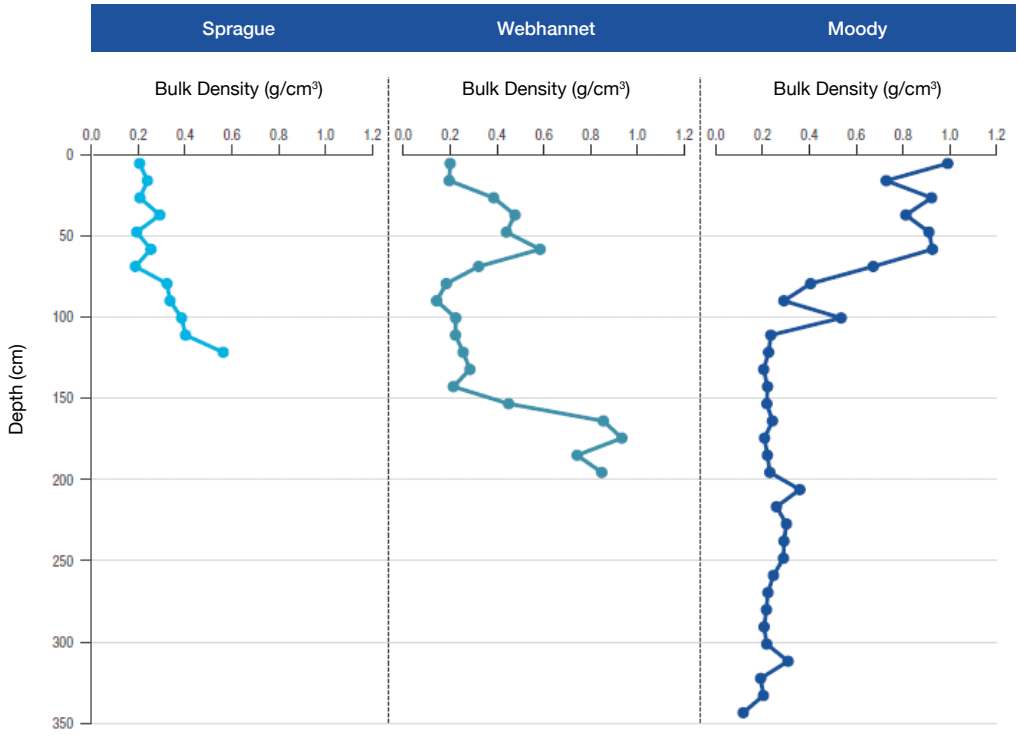

Figure 3.19 Bulk density of cores from Sprague Marsh, Phippsburg, Maine (N 44º 44' 21.64" / W 69º 49'48.90"), Webhannet Marsh, Wells, Maine (N 43º 18' 14.82" / W 70º 34' 16.61"), and Moody Marsh, Wells, Maine (N 43° 16' 26.19" / W 70º 35' 12.21"). The lowest core depth represents the depth to refusal at each site (Johnson *et al*. in prep).

Determining Organic Carbon Content (% C_{org})

The organic carbon content of a soil sample can be measured using a variety of methods; the method chosen will depend largely on accessibility to necessary equipment. Options for

measuring the organic carbon content include: 1) using an automated elemental analyzer (sometimes called a CHN analyzer since many elemental analyzers are configured to simultaneously measure carbon (C), hydrogen (H), and nitrogen (N) content); 2) using combustion and empirical relationships between organic carbon and organic matter (known as Loss on Ignition, LOI); or 3) using wet chemistry techniques such as the Walkley-Black method, which is simple and requires minimal equipment. The pros and cons of each method are presented in **Table 3.3**. The results obtained using the wet method cannot be considered quantitative, and the process produces toxic wastes and so is only appropriate for labs equipped for safe use and disposal of chemical oxidants and low-resolution studies (Nelson et al. 1996; Sollins *et al*. 1999). Because of the limitations of the wet chemistry technique, we only describe the elemental analyzer and LOI methods here. The use of any of these techniques depends on availability as well as budgetary and capacity constraints (**Fig. 3.20**).

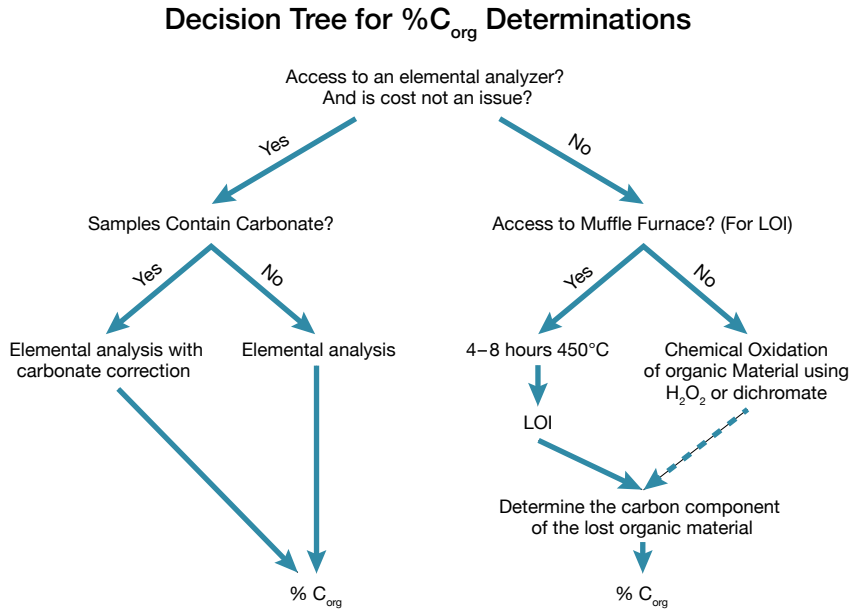

Figure 3.20 Decision tree to determining which method is best for calculating the organic carbon component of soil

Table 3.3 Comparison of laboratory techniques to determine percent organic carbon.

DRY COMBUSTION METHOD		WET COMBUSTION METHOD
Elemental Analyzer	LOI	H_2O_2 and Dichromate Digestion (Walkley-Black method)
Pros	Pros	Pros
Quantitative measure of carbon content.	Semi-quantitative measure of organic carbon content; low cost and simple technology.	Semi-quantitative measure of organic carbon content; low cost and simple chemistry.
Cons	Cons	Cons
Requires special instrumentation; can be costly.	Percent organic carbon determined from empirically derived relationships between carbon and organic matter.	H_2O_2 does not always digest carbon equally; produces hazardous waste.

HOMOGENIZATION

Before the carbon content can be determined, each individual sample/subsample should be homogenized. Dried samples are assessed, and any large items, such as stones and twigs are removed, and large clumps are broken up with a spatula. Homogenization can be done by manually grinding the dried soils to a powder of consistent particle size using a mortar and pestle or it can be done automatically using a pulverizer or mill (**Fig. 3.21**). Whichever method is used, it is important to clean the grinding device (e.g., with ethanol) between each soil sample to ensure minimal cross-contamination. The homogenized samples (hereafter called raw soil sample) can then be used for determining the organic and inorganic carbon content.

Figure 3.21 Grinding and homogenization of a soil sample. (A) Mortar and pestle, (B) Agate pot in commercially available mill, (C) Agate pot with beads to help pulverize and homogenize the soil sample. (© Hilary Kennedy, UWB)

ESTIMATING % ORGANIC CARBON USING AN ELEMENTAL ANALYZER

For this method dry combustion is used to determine the total carbon (organic and inorganic) for each sample. It is the most suitable method for routine analysis of total carbon, and we recommend use of an elemental analyzer if possible (Sollins *et al*. 1999). An elemental analyzer is a laboratory instrument used to determine the elemental composition of a sample. The analyzer uses a high temperature induction furnace and either infrared spectroscopy or gas chromatographic separation of gases and thermal conductivity detection to measure the carbon, hydrogen, and nitrogen (as well as other elements) content of the sample.

When using an elemental analyzer, samples are automatically dropped onto the top of a quartz tube maintained at around 1000 ºC, packed with oxidation reagents and catalysts, and there is a constant flow of helium through the column. When the sample drops onto the top of the column, the helium stream is temporarily enriched with pure oxygen. Flash combustion takes place, producing carbon dioxide, water, and nitrogen. The water is removed using a desiccant, and the CO_2 is separated from N_2 by gas chromatography. The output of this process is a graph where the amount of carbon is proportional to the area under the CO_2 peak (**Fig. 3.22**), and is reported in units of percent carbon (% C). The instrument is calibrated

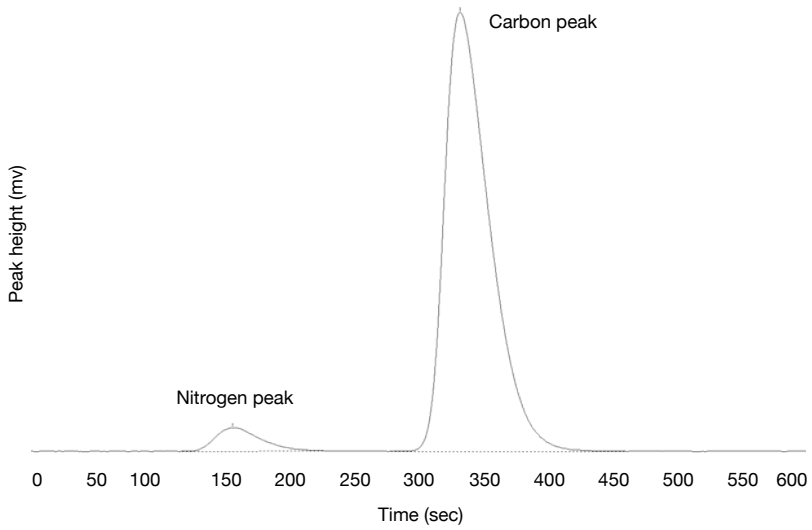

Figure 3.22 Chromatogram results from an elemental analyser, showing nitrogen and carbon peaks from combusted sample (© Hilary Kennedy, UWB)

using an organic compound as a standard, such as acetanilide. The precision of the analysis must be determined using international standards and monitored using an internal standard with a composition close to that of the sample.

If an elemental analyzer is not available, samples can be sent to a commercial laboratory where costs for elemental analysis typically range between $10 and $20 USD per sample. Provided the proper tools are available (microbalance and tin boats), it is possible to save money by weighing out samples and shipping them to a qualified laboratory. In this case, add samples into pre-weighed tin capsule using a spatula, then close and compress using forceps. Weigh the tin capsule with the sample inside, and subtract the weight of the empty tin capsule to determine sample weight. Create a record of where each sample is in the tray, including weights, and ship to the laboratory. The laboratory will need the weight of each sample to calculate the % C in your soil. While awaiting analysis, samples can be stored in a tray inside a desiccator (**Fig. 3.23**). Ask the laboratory facilities in which you will run the organic carbon analysis for advice before starting to encapsulate the samples (amount of organic carbon needed for robust analyses in their facilities, size of tin capsules needed, etc.). A good option is to run the organic carbon analysis in a few representative samples first and make adjustments as needed before running all of your samples.

Elemental analyzers determine the total carbon content of a sample, including the organic and inorganic carbon. To correct for this, the inorganic carbon content must be determined.

Correcting for Inorganic Carbon Content

Inorganic carbon in the form of carbonates (i.e., calcium carbonate, $CaCO_3$) can be found in coastal soils in the form of shells and/or pieces of coral and is most often associated with seagrass beds. Calcium carbonate may also be present in some mineral-rich soils found beneath layers of peat. (Carbon-neutral sands, silts, and clays will also likely be present in the sediment in varying proportions, but their presence will not affect the analysis of organic carbon.) Calcium carbonate (hereafter referred to as carbonate) contains carbon, but is not included when determining blue carbon stocks, but it will be converted to CO_2 in an elemental analyzer, biasing the results.

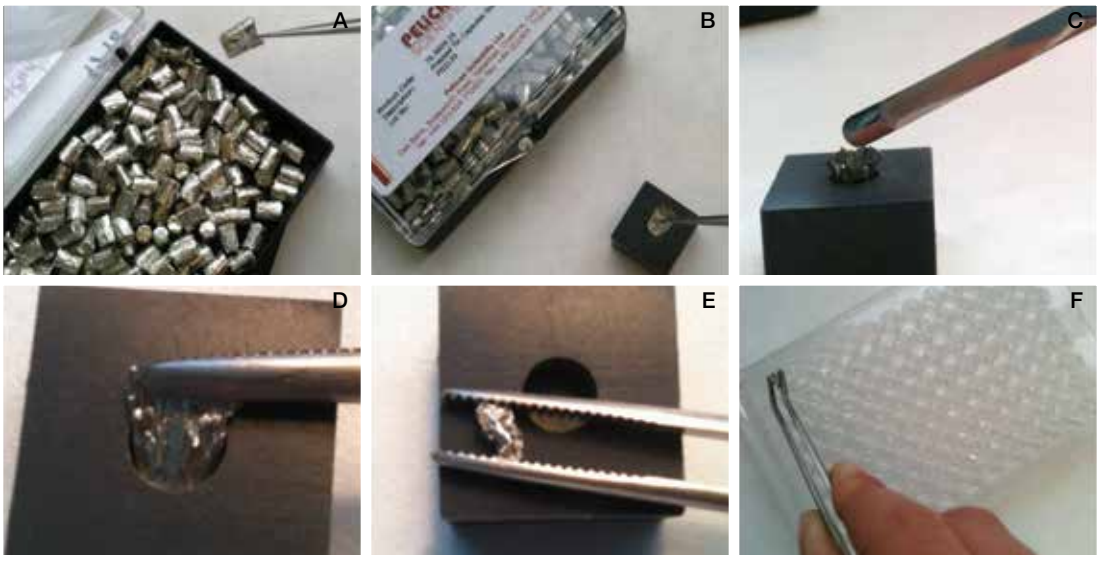

Figure 3.23 Preparing a dried sample for CHN analysis. (A) Extracting a tin capsule to be weighed, (B) After weighing, tin capsule can be placed in clean receptacle, (C) The sample added using a spatula, (D) Forceps are used to close, (E) Compress the tin capsule, and (F) Place the sample in a 96-well plate and store prior to analysis. (© Hilary Kennedy, UWB)

There are two basic methods that correct for the carbonate content of soils.

1) **Acidification**: This approach is easy, cheap, and requires less sophisticated laboratory equipment. Inorganic carbon is volatilized to CO_2 by treating the soil subsample with a strong acid. Inorganic carbon content is estimated from the difference in weight of the subsample before and after treatment. There is a risk that some organic carbon will also be removed using this method leading to a possible under-estimation of organic carbon. Reactions with more dilute acids over longer times periods minimizes the loss of organic carbon due to decomposition.

2) **Elemental Analyzer**: A soil subsample is heated to 500 ºC. At this temperature organic carbon is removed leaving the inorganic carbon in the ash. The inorganic carbon remaining in the ashed subsample is determined using an elemental analyzer.

In both techniques the inorganic carbon content is subtracted from the total carbon (see previous section), what remains it the estimate of organic carbon content.

ACIDIFICATION

Some protocols for removal of carbonate (e.g., decalcification) use a relatively concentrated acid for short periods of time (Mortlock & Froelich 1989), and others use a more gentle method (Weliky *et al*. 1983; Pilskaln & Paduan 1992). We recommend the slower and gentler approach to decalcification described here. First, test to see if the sample contains significant quantities of carbonate by taking a subsample (corresponding to the samples used for total carbon analysis), placing it on a glass surface, and adding a few drops of 1N hydrochloric acid (HCl). If carbonate is present, bubbles of CO_2 will be generated and the sample will effervesce (**Fig. 3.24**).

If carbonate is present, weigh out ~ 1 g of your original homogenized soil sample into a 125 mL beaker or a 50 mL glass conical centrifuge tube (the latter is preferred if samples need to be centrifuged to separate solution from soil; see below). Dilute HCL to 1N and add enough to the beaker to cover your sample and agitate for 15 minutes by manually

shaking the sample or by using an ultrasonic bath or probe. Both techniques help break up any clumps of soil so that the acid can remove all the inorganic carbon present. The acid is gentle enough to leave the organic matter intact. Allow any effervescence to die down and let samples sit overnight (18–24 hours). Add additional HCl, agitate or sonicate for 15 minutes and check for further effervescence.

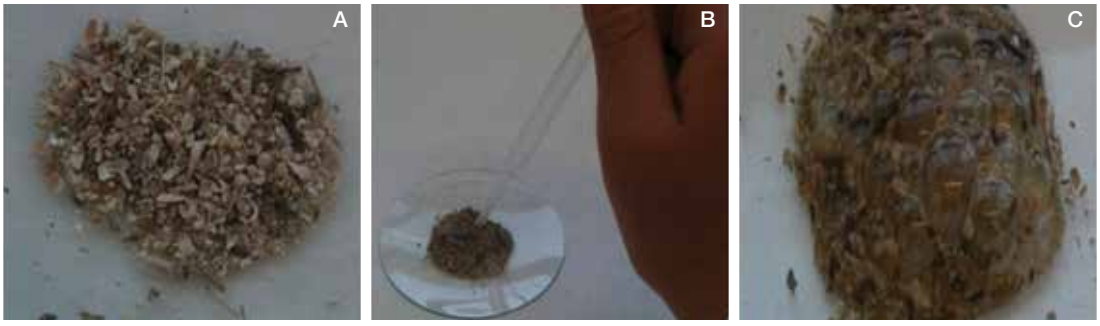

Figure 3.24 Testing for carbonate. (A) Subsample in a watch glass prior to acidification, (B) pipetting a few drops of weak HCl, (C) subsample effervescing. (© Hilary Kennedy, UWB)

If CO_2 is no longer being produced (no new outgassing is observed), the carbonate has been removed. Once the soil has settled to the bottom of the beaker/tube the overlying acid can be decanted. If there is a lot of fine-grained soil suspended in solution, centrifuge the samples to separate the solution from the soil and then decant or remove the liquid with a pipette. After the acid has been removed, add distilled water to the sample, swirl, allow the material to settle (or centrifuge), and decant off the water. Repeat this washing step two more times. Dry at 60 ºC overnight, and weigh the sample.

The mass difference of the sample pre and post acidification is an estimate of calcium carbonate in the sample. However, only 12% of the weight difference can be attributed to carbon (carbon makes up 12% of the molecular weight of calcium carbonate ($CaCO_3$)). Thus in order to estimate the amount of inorganic carbon present, the mass of calcium carbonate is multiplied by 0.12. Finally, subtract the inorganic carbon content from the total carbon content of the subsample (from the elemental analysis described in the previous section) to get the organic carbon content of the sample (**Table 3.4**).

Table 3.4 Determining % Inorganic carbon by acidification method

SAMPLE ID	TOTAL CARBON CONTENT (ELEMENTAL ANALYZER)	DRY MASS BEFORE ACID (mg)	DRY MASS AFTER ACID (mg)	MASS OF CARBONATE (mg)	INORGANIC CARBON (mg)	INORGANIC CONTENT OF SAMPLE	ORGANIC CARBON CONTENT OF SAMPLE
	A	B	C	D = B – C	E = C*0.12	F = (E/B)*100	G = A – F
Example	25%	100	90	10	1.2	1.20%	23.8%

ELEMENTAL ANALYZER

Take a separate subsample (corresponding to the samples used for organic carbon analysis, ~ 0.5 g) of the dried raw soil, weigh it to the nearest milligram, and place it in a temperature-proof vessel (i.e., ceramic crucible). These samples are then put into a furnace heated to 500 ºC for a minimum of three hours (until a constant weight is reached) to volatilize the organic

compounds. The weight of the ash remaining is then determined to the nearest milligram. An elemental analyzer is used, following the procedures outlined above, to determine the carbon content of the ash which is assumed to be all inorganic carbon.

Scale the elemental analyzer results by the ratio of ash weight to sample dry weight to get the inorganic carbon content of the original dry sample. Then, subtract the inorganic carbon content from the total carbon content of the subsample to get the organic carbon content of the sample (**Table 3.5**).

Table 3.5 Determining % Inorganic carbon by elemental analysis

SAMPLE ID	TOTAL CARBON CONTENT (ELEMENTAL ANALYZER)	DRY MASS BEFORE ASHING (mg)	DRY MASS AFTER ASHING (mg)	INORGANIC CARBON CONTENT OF ASHED SAMPLE (mg)	INORGANIC CONTENT OF SAMPLE	ORGANIC CARBON CONTENT OF SAMPLE
	A	B	C	D	E = D*(C/B)	F = A – E
Example	25%	500	250	10%	5%	20%

MEASURING % CARBON VIA LOI ANALYZER

If the cost of using an elemental analyzer is prohibitive, we recommend using the percent loss on ignition technique (often referred to as % LOI). The initial cost of the equipment needed for % LOI analysis (including a muffle furnace and ceramic crucibles) ranges between $5000 USD and $10 000 USD. This relatively simple set-up is very durable and can be used for many years, significantly decreasing cost per sample analysis over the long-term.

LOI is a measure of the mass of sample lost (e.g., oxidised and lost as gas, or volatilised) when heated to high temperatures. Typically the sample is heated to combustion at 450 ºC for 4–8 hours (Heiri *et al*. 2001). This temperature is used to ensure that only organic (not inorganic) carbon is oxidized.

The % LOI is calculated as follows:

- % Loss on Ignition = [(dry mass before combustion (mg) – dry mass after combustion (mg)) / dry mass before combustion (mg)] * 100

Table 3.6 Determining % LOI

SAMPLE ID	INITIAL MASS BEFORE COMBUSTION (mg)	FINAL MASS AFTER COMBUSTION (mg)	DIFFERENCE PRE- AND POST-COMBUSTION	% LOSS ON IGNITION
Example	50	40	10	(10/50)*100 = 20

It is important to note that LOI represents the loss of organic matter, which is composed of carbon, hydrogen, nitrogen, oxygen, sulfur, etc. and not solely the loss of organic carbon. Thus, a relationship needs to be determined to relate % LOI to % C_{org}.

Relationship between organic matter and organic carbon: An equation must be constructed that relates organic matter content (% LOI) to the organic carbon content (% C_{org}) of the same sample. This can be achieved by sending a limited number of samples for organic

carbon analysis using an elemental analyser (see above) and comparing the organic carbon content resulting from that technique to the % LOI results.

If this is not possible, use a value from the literature for a study location/type that most closely resembles your own. The following table (**Table 3.7**) summarizes examples of the relationships between % LOI and % C_{org} in mangrove, tidal salt marsh, and seagrass soils. Additional information on the relationships between % LOI and organic carbon in mangrove, tidal salt marsh, and seagrasses can be found in **Appendix E**. However, there is a large range in the ratios of carbon content (% C_{org}) to organic matter (% LOI) reported in the scientific literature, making standard ratio values possible sources of error in estimating organic carbon content. Therefore, it is good practice to determine the ratio for your particular soils by sending a few samples to a laboratory for elemental analysis. Sending a small number of samples should not be too cost prohibitive and will greatly increase the accuracy of your results.

Table 3.7 Relationship between % LOI and % C_{org} for the different ecosystems. Variability within ecosystems may be due to slight differences in methods used and/or characteristics of the soils.

ECOSYSTEM	RELATIONSHIP STRENGTH (r^2)	RELATIONSHIP BETWEEN % LOI AND % C_{org}	LOCATION (SOURCE)
Mangroves	0.59	% C_{org} = 0.415 * % LOI + 2.89	Palau (Kaufmann *et al*. 2011)
Tidal Salt Marshes	0.98	% C_{org} = 0.47 * % LOI + 0.0008 (% LOI)^2	Maine (Johnson *et al*. in prep)
Tidal Salt Marsh	0.99	% C_{org} = 0.40 * % LOI + 0.0025 (% LOI)^2	North Carolina (Craft *et al*. 1991)
Seagrasses (% LOI > 0.2)	0.87	% C_{org} = 0.40 * % LOI – 0.21	Global data set (Fourqurean *et al*. 2012a)
Seagrasses (% LOI > 0.2)	0.96	% C_{org} = 0.43 * % LOI – 0.33	Global data set (Fourqurean *et al*. 2012a)

While % LOI can be an adequate indicator of organic matter content in many sample types (often defined operationally as % organic matter), it is important to understand the possible limitations of this technique. LOI has been reported to lead to overestimation of organic carbon content in two ways:

1) If a sample containing carbonate (e.g., those underlying seagrass meadows with shoots covered by abundant epiphytes or soils in the region of coral reefs) is heated above 500 ºC, loss of water and CO_2 derived from $CaCO_3$ may also be driven off (Hirota & Szyper 1975; Leong & Tanner 1999).

2) In soils containing > 11% clay minerals, a significant amount of structural water (that is not lost by heating at 60 ºC) may be driven off during heating at this higher temperature (Barillé-Boyer *et al*. 2003).

In both cases, the organic carbon content could be overestimated due to the fact that the % LOI could reflect a loss of organic matter, inorganic carbon, and structural water contained within the sample. A reduction in the error arising from % LOI may be achieved by determining and correcting for the inorganic content (see section below on correcting for inorganic carbon).

CALCULATING TOTAL SOIL CARBON STOCK

The total soil carbon stock within a project area is determined by the amount of carbon within a defined area and soil depth. To calculate the total soil carbon for your project area you will need the following information:

- Soil depth,
- Subsample depth and interval,
- Dry bulk density, and
- % Organic carbon.

The total carbon stock in a project area can be determined as follows:

Step 1: For each interval of the core sampled/analyzed, calculate the soil organic carbon density as follows:

Soil carbon density (g/cm^3) = dry bulk density (g/cm^3) * (% C_{org}/100).

Step 2: Calculate the amount of carbon in the various sections of core sampled by multiplying each soil carbon density value obtained in step 1 by the thickness of the sample interval (cm):

Amount carbon in core section (g/cm^2) = Soil carbon density (g/cm^3) * thickness interval (cm).

Step 3: Sum the amount of carbon in core sections over the recommended total sampling depth (1 m at a minimum). *It is critical that the total sampling depth be included in your report.*

Core #1 summed = Amount carbon in core section A (g/cm^2) + Amount carbon in core section B (g/cm^2) + Amount carbon in core section C (g/cm^2) + …. all the samples from a single core.

*The entire core needs to be included in this calculation. If subsamples were taken along the core (**Fig 3.11**), sum the amount of carbon in each of the sections and then sum over the total depth sampled to get the total carbon stock.

Step 4: Convert the total core carbon from step 3 into the units commonly used in carbon stock assessment (MgC/hectare) using the following unit conversion factors (There are 1 000 000 g per Mg (megagram), and 100 000 000 cm^2 per hectare):

Total core carbon (MgC/hectare) = Summed core carbon (g/cm^2) * (1 Mg/1 000 000 g) * (100 000 000 cm^2/1 hectare).

The unit here is Mg C/hectare (for the top 1 m soil), and is a typical unit used in carbon stock assessment.

REPEAT FOR EACH CORE

Step 5: Determine the average amount of carbon in a stratum for given depth and calculate the associated standard deviation to determine variability/error.

Average carbon in a core = Carbon content for core #1 (determined in step 4) + Carbon content for core #2 + Carbon content for core #3+…. n) / n.

Standard Deviation (σ) determines how closely the data are clustered about the mean, and is calculated as follows:

Core Standard Deviation $$(\sigma) = \left[\frac{(X_1 - \overline{X})^2 + (X_2 - \overline{X})^2 + \ldots (X_n - \overline{X})^2}{(N-1)} \right]^{1/2}$$

- $\overline{X}$ = average carbon in a core
- X_1 = individual result for core #1, in MgC/hectare; X_2 = individual result for core #2, in MgC/hectare, etc.,
- N = total number of results.

Step 6: To obtain the total amount of carbon in the ecosystem, multiply the average carbon value (MgC/hectare) for each core obtained in step 5 above by the area of each stratum (in hectares) to determine MgC for each stratum, and then sum the MgC values for each stratum to determine the total soil carbon stock.

Again, it is critical to note the total depth of the soil cores. Thus, the final unit for soil carbon stock in each project strata will be MgC over a specific depth interval (usually, but not always 1m).

Total organic carbon in a project area (MgC) = (average core carbon from Statum A (MgC/hectare) * area Statum A (hectares)) + (average core carbon from Statum B (MgC/hectare) * area Statum B (hectares) + …

Step 7: To report a value for the variability/error associated with these measurements, calculate the total uncertainty in the data. First, calculate the standard deviation of the average Mg C for each stratum. [Multiply the standard deviation carbon value (MgC/hectare) for each core determined in step 5 (above) by the area of each stratum (in hectares).] Then propagate the uncertainty through the calculations by combining the standard deviations of the average MgC for each stratum as follows:

$\sigma_T = \sigma A2 + \sigma B2 + \ldots \sigma N2$

- Where σ_T = the total variability associated with the measurements,
- σ_A = standard deviation of the core average MgC for stratum A * area of stratum,
- σ_B = standard deviation of the core average MgC for stratum B * area of stratum, and
- σ_N = standard deviation of the core average MgC for remaining stratum * area of each individual stratum.

This approach can be used when adding average values, as is done when combining the data from the individual strata.

Step 8: The final soil carbon stock will be presented in an average value ± the total uncertainty. Alternatively, a minimum and maximum carbon stock can be presented by multiplying by the project area by the minimum and maximum carbon densities.

Total organic carbon in a project area (calculated in Step 6) ± the standard deviation (calculated in Step 7)

Equations and examples are provided in Appendices B and C.

QUICK GUIDE

Step 1: Determine Soil Depth

- Measure depth to parent materials, bedrock, or coral sands.

Step 2: Soil Coring

- Choose soil coring device based on type of soil and degree to which the soil is saturated with water.
- Steadily insert the coring device until the top of the sampler is level with the soil surface.
- Once at depth, twist the coring device to cut through any remaining fine roots, measure the length of pipe outside the sediments and the length of empty pipe, and seal the top end (the vacuum will prevent the loss of the sample).
- Gently pull the coring device out of the soil while continuing to twist as it is being extracted. This twisting assists in retrieving a complete soil sample.
- In the case of seagrasses, the coring device must often be removed from the soil using a winch.
- It is imperative to note the total depth and any compression.

Step 3a: Sampling an Entire Soil Core (if this is not feasible subsampling can be done, next section)

- It is best to sample the entire depth of the soil core; however, this may not always be possible or practical.
- It is imperative to record subsample depth, depth interval and volume.

Step 3b: Subsampling a Soil Core

- Samples should be collected from homogenized sample intervals or from the approximate mid-point of each desired depth range.

Step 4: Archiving samples

- The proper labelling of the cores and samples in the field is essential to avoid confusion and common mistakes in sample identification.
- Each sample/subsample should be labelled with at least a core ID, sample depth, and depth interval.

Step 5: Storing samples

- To minimize decomposition of organic matter, samples should be kept cold (at 4 °C) and, if possible, frozen within 24 hours of collection.

Step 6: Determining Dry Bulk Density (g/cm^3)

- Calculate the volume of soil sampled using the equation (cm^3)
- Determine the dry weight.
- Calculate dry bulk density by dividing the mass of dried soil by the volume of soil sampled (g/cm^3).

Step 7: Determine Organic Carbon Content

- Decide which techniques to use based on desired result, capacity, and budgetary constraints.
- Determine inorganic carbon content.
- Determine organic carbon content.

Step 8: Calculate Total Soil Carbon Stock

- If you subsampled the core, you will need to determine the amount of carbon per cm^3 of the core and then multiply that by the length of the sample interval, then add all the intervals together to determine the total carbon /area represented in the core.
- You must include the variability associated with the measurements and the total soil depth assessed when reporting results.

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Field Sampling of Vegetative Carbon Pools in Coastal Ecosystems

GENERAL CONSIDERATIONS

The vegetative blue carbon pool consists of three different components:

- The living, aboveground biomass dominated by herbaceous (for seagrass and tidal salt marsh) and woody (for mangroves) plant mass. This can also include epiphytic organisms (e.g., algae and microbes living on the main plants) and areal roots (pneumatophores).
- The living, belowground biomass dominated by underground roots and rhizomes.
- The non-living, aboveground biomass, including detritus consisting mainly of leaf litter (found in all three ecosystems), algae, or in mangroves dead and downed wood.

The protocols used to determine the amount of carbon in each pool—in each ecosystem—will vary depending on vegetation type and density. Allometric equations are used to describe the relationship between "measurable parameters" (height, width, circumference, etc.) and total biomass. These equations are commonly used to determine the biomass of materials where it is impractical, overly destructive, or unwise to take an entire sample back to the laboratory (e.g., trees and large shrubs). Many well-established allometric equations exist in scientific literature (many are used in this chapter), and it is recommended to use equations that have been established for similar vegetative species and locations as those found in the study site being investigated.

In all cases, the carbon pool for each vegetation type is determined by multiplying the biomass of each type of plant material (e.g., wood, leaf litter, roots, etc.) by the corresponding carbon conversion factor. The carbon conversion factor represents the fraction of vegetation that is carbon. For example, if the living aboveground wood has been determined to be 45% carbon, then the carbon conversion factor is 0.45. The carbon conversion factor is then multiplied by the total biomass of the above ground wood pool for that plot to determine the amount of carbon found in the aboveground wood pool for a certain area.

The goal of Chapter 4 is to utilize ecosystem-specific techniques to determine the biomass and the organic carbon content (% C_{org}) for each of the relevant blue carbon pools. Once the carbon content for all carbon pools has been established, they are then added together to determine the carbon content of the biomass per unit area for a specific system (MgC/hectare). This value is then added to the soil carbon pool specified for a certain soil carbon depth (Chapter 3) to determine the total carbon stock (MgC/hectare-depth) for a blue carbon ecosystem.

MANGROVES

Mangrove ecosystems are populated by halophytic trees, shrubs, and other plants that grow in brackish to saline tidal waters of tropical and subtropical coastlines (Mitsch & Gosselink 2007). In general, mangroves are restricted to the intertidal zone from approximately mean sea level to the highest high tide water level.

Mangroves are classified into four major associations based on differing vegetative structures that correspond to physical, climatic, and hydrologic features of the environment in which they exist: (1) oceanic fringing mangroves, (2) riverine or estuarine mangroves, (3) basin mangroves, and (4) dwarf or scrub (or chaparro) mangroves (Cintrón *et al*. 1978; Mitsch & Gosselink 2007) (**Fig. 4.1**). These classifications correspond to aboveground biomass ranging from more than

500 MgC/ha in riverine and fringe mangroves (such as in Asia-Pacific regions) to about 8 MgC/ha for dwarf mangroves (Kauffman & Cole 2010; Kauffman *et al*. 2011).

Figure 4.1 Classification of mangroves. (A) Oceanic fringing mangroves (© Enrico Marone, CI), (B) Riverine or estuarine mangroves (© Ginny Farmer, CI), (C) Basin mangroves (© Colin Foster, CI), and (D) Dwarf or scrub mangroves (© Catherine Lovelock, UQ)

Flooding regimes throughout the mangrove habitat cause saline or brackish coastal wetland environments that often consist of anoxic (low oxygen levels) sediments. As such, mangroves possess a number of adaptations to facilitate survival in these unique environments. Most notably, this includes aboveground roots (e.g., their characteristic stilt roots and pneumatophores), which allow gas exchange for belowground root tissues. The vegetation also varies greatly in structure (tree height, density, and species composition) and function, due to differences in temperature, rainfall, hydrology, and substrate (Saenger & Snedaker 1993). Mature mangroves may range from shrub-like stands less than 1 m in height to large trees with trunk diameters > 1 m.

Figure 4.2 Height differences among mangrove vegetation. (A) Small dwarf mangroves < 2 meter in height (© C.I. Feller, SERC), (B) Larger mangroves several meters tall (© Andreas Hutahaean, KKP)

Despite several similarities, mangroves are quite different from upland forests in both composition and structure. Mangroves have stilt roots and pneumatophores, and they usually do not have significant understory vegetation or a well-developed floor litter layer as crabs are usually extremely efficient consumers of fallen leaves and litter is transported away by tides. Because of these and other differences between the structure and environment of mangroves and upland forests, approaches to quantifying their composition, structure, carbon stocks, and status differ. However, some approaches to upland forest sampling may provide guidance in project implementation and design. Notable examples can be found in the scientific literature (Pearson *et al*. 2005; Pearson *et al*. 2007; GOFC-GOLD 2009).

Field Sampling Considerations

Mangroves ecosystems are often extremely difficult environments to conduct field assessments and sampling. The trees often have extremely high stem densities with abundant stilt roots and/or pneumatophores. Project areas are frequently dissected by tidal channels which are difficult to cross. The entire ecosystem may be flooded, especially during high tides. These and a number of other hazards limit mobility and create safety concerns. Most mangroves are also subject to semi-diurnal tidal cycles and can only be sampled during low tides, limiting both the timing and duration of the sampling, especially for components on the forest floor. In the lowest elevation mangroves, sampling may be limited to low tidal periods of as little as 3 to 4 hours. This narrow time window necessitates an efficient sampling plan.

In Chapter 2, we describe approaches to determining the number and location of sampling plots within a project area or strata. Since the various carbon pools within mangroves have distinctly different scales and levels of effort required for sampling, it is usually necessary to assess different components with different size sampling areas. The biomass of trees, shrubs, herbs, lianas, and palms along with non-living vegetation like leaf litter and downed wood must be determined separately and sampled at appropriate scales (**Fig. 4.3**). For example, to get a representative sampling of trees, the sample area may need to be large (e.g., 50 m x 50 m), but if you then want to sample the leaf litter, it is not practical or necessary to collect all the leaf litter in an area that big. Thus a smaller subplot size (e.g., 2 m x 2 m) is more appropriate.

Once size and location have been determined, it is necessary to decide if the sampling plots will be permanent or temporary. Permanent plots are used if the same location will be assessed in the future to determine change. Temporary plots are used when sampling will only be done once or when permanent plots are not feasible. We give some guidance on establishing permanent sampling plots vs. temporary plots in Chapter 2. For further information on the establishment of permanent plots and sampling methods, we recommend the Amazon Forest Inventory Network (www.rainfor.org/) and the Center for Tropical Forest Science (www.ctfs.si.edu/group/Resources/Methods) websites.

With plot size, scale, and type determined, the next step is to assess the carbon content of each relevant carbon pool.

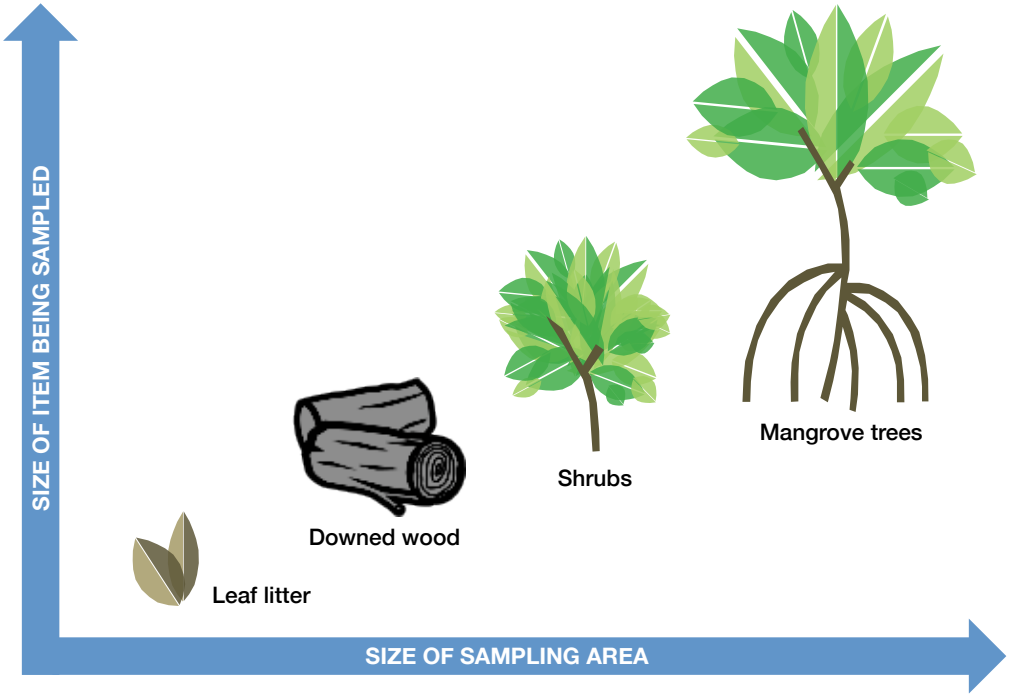

Figure 4.3 Plot scale depends on the component being assessed. Large trees require a large enough area to allow for a representative sampling, smaller trees need less area to get a representative sample, leaf litter, lianas, dead wood, and pneumatophores components are so small and numerous that a relatively small area is adequate for sampling (Kauffman & Donato 2011).

Biomass Estimates

This section provides guidance on how to measure aboveground biomass across a range of vegetation types likely to be encountered in the field. Some differences in sampling procedures are required to accommodate differences in growth forms.

LIVE TREES

Trees dominate the aboveground carbon pool in mangroves, and both their presence and condition are indicators of land-use change and ecological condition. It is essential to measure trees thoroughly and accurately. Basic data that must be recorded for all individual mangrove trees in a plot include:

- Species (there are typically few species present in mangroves, species can usually be identified with on-site training);
- Main stem diameter at breast height (dbh);
- Tree height if feasible; and
- Location and ID.

It is recommended that all live trees be sampled and recorded over the entire plot area, particularly for permanent plots where trends in carbon are being monitored. However, this is different from surveys of upland forests, where only trees greater than 10 cm dbh are measured to standardize methods (GOFC-GOLD 2009). Smaller trees are left out of the carbon calculations because they often constitute a relatively insignificant proportion of the total upland forest carbon stock (Cummings *et al*. 2002). For many mangroves, however,

smaller trees can dominate the stand composition and thus must be included (Lovelock *et al*. 2005; Kauffman & Cole 2010). A tree is included in the survey if at least 50% of the main stem is rooted inside the plot perimeter.

In the interest of efficiency, trees that are smaller in diameter may be measured in subplots to reduce the number of measurements necessary. For example, Kauffman and Cole (2010) measured all trees > 5 cm dbh over the entire plot area, while trees < 5 cm dbh were measured in smaller subareas of known size. The total number of small trees could then be estimated by assuming a constant density over the full plot area.

If numerous seedlings are present, the seedlings can be recorded as a simple count of individuals in a subarea. For our purposes, a seedling is defined as a woody plant with a height of 10–30 cm (Esquivel *et al*. 2008). To determine the carbon content associated with seedlings, a random sample should be collected from outside the plot area for analysis (only necessary to collect from outside the plot if using permanent plots, the idea is to include seedlings in the analysis but leave them in the plot so they can be monitored over time). Carbon content can be determined by drying the seedlings to determine biomass followed by laboratory analysis using an elemental analyzer (Chapter 3), but in many cases it is possible to find a published carbon conversion factor for specific tree species. The average carbon content is then multiplied by the density (seedlings per unit area) to determine their contribution to the plot and strata biomass.

To determine the biomass of mangrove trees, existing allometric equations are applied (Chave *et al*. 2005). Allometric equations give established relationships between the biomass of whole trees (or their components) and readily measured parameters. Accurate species identification is important as it allows the selection of the most appropriate allometric equation for each measured individual mangrove tree. Common parameters include tree diameter, wood density (**Table 4.2**) and tree height.

Diameter at Breast Height (dbh): The diameter of the tree is typically used to calculate the tree volume. The diameter of the tree's main stem is typically measured at 1.3 m above the ground, which is also called the dbh. These measurements are usually made with a diameter tape (if multiple measurements are needed) or tree callipers (for a single measurement for rapid assessment). This is not always a straightforward process due to anomalies in stem structure. **Fig. 4.4** gives an overview of the location for measurement for a variety of different tree configurations.

- If the tree is fairly straight with a tall trunk the dbh can be measured from the ground parallel to the trunk (**Fig 4.4A**)
- If the tree is on a slope, always measure on the uphill side (**Fig 4.4B**)
- If the tree is leaning, dbh is taken according to the trees natural height parallel to the trunk (**Fig 4.4C**)
- If the tree is forked at or below 1.3 m then measure just below the fork (**Fig 4.4D**)
- If the fork is very close to the ground measure as two trees (**Fig 4.4E**)
- For trees with tall buttresses exceeding 1.3 m above ground level, stem diameter is usually measured directly above the buttress (**Fig 4.4F**).

- For stilt rooted species (e.g., *Rhizophora* spp.), stem diameter is often measured starting above the highest stilt (**Fig 4.4G**). For some individual trees with prop roots extending well into the canopy it is not necessary, practical, or accurate to measure above the highest prop root and, typically, tree diameter is measured above the stilt roots where a true main stem exists.

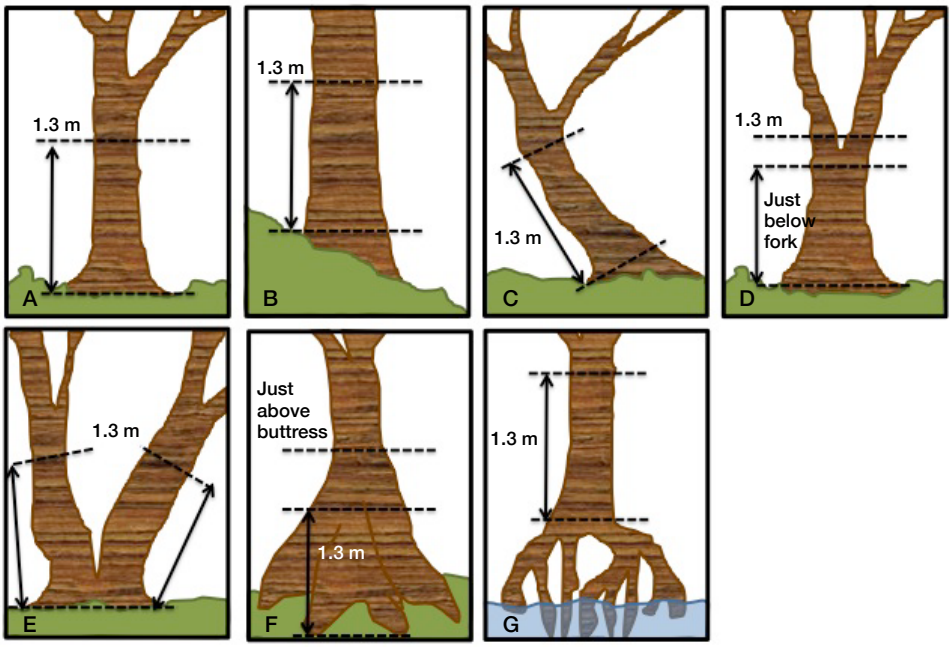

Figure 4.4 Estimating diameter at breast height for irregular mangrove trees (modified from Pearson, *et al*. 2005)

In permanent plots, it is quite important to mark the point of measurement when it is not at 1.3 m above ground level so that repeated measurements can be taken at the same location. This is accomplished by placing tree tags and/or by painting a ring at the exact point of measurement. In some studies, point of measurement is marked with a stainless steel nail. However, when nails are used trees tend to develop wound wood (bulges) where the wood is damaged by the nail. This can cause overestimates of tree growth and therefore is not recommended.

Wood Density: Wood density describes the relationship between the wood's dry weight (g) and the wood's volume (cm^3). The part of the plant (branches, main stem, bark, etc.) harvested for wood density depends on the practicality of obtaining samples and the level of accuracy desired. Wood density requires measuring both the volume of fresh samples and the oven-dry mass of several samples of wood (ideally n > 25 per sample type). Samples are commonly taken by removing small portions of the bark, cutting segments of branches (~2.5 cm), and using an increment borer on the main stem (taken from a consistent height). As a rough guideline, each piece collected for analysis should have a mass of between ~0.5 and 50 g.

Volume is obtained by determining each fresh sample's submerged mass. On a digital balance, place a container of a size sufficient to submerge each sample. Add water to the container, not to the top, but to a height that will allow the displacement of water without it spilling over the sides of the container. Each sample is attached to a needle attached to a ring stand above the scale. The sample is then submerged (without touching the bottom and sides of the container) and the change in mass is recorded. The change in mass (g) divided by

the density of water (g/cm^3) gives the volume of the sample. The density of water is 1 g/cm^3, thus the resulting increase in mass shown on the scale is equivalent to the volume displaced by the sample.

Dry weight is obtained by drying the wood samples in a well-ventilated oven at 100 ºC until constant mass is obtained (typically 24–72 hours, but the time will depend on the size of the sample). We recommend drying at 100 ºC because water within the cell wall can only be completely dried off at these temperatures. For each sample, calculate the wood density using the following equation and determine the mean for each sample type.

- Wood density (g/cm^3) = Dry weight (g) / volume of fresh wood (cm^3)

Wood densities for live trees (which may be different from densities of downed woody debris) are required for certain allometric biomass equations, including the general mangrove equations (see next section). Wood densities of individuals of the same species have been shown to vary greatly between sites. As such, it is desirable to use site-specific wood densities estimated by laboratory analysis of samples taken from the field. If that is not possible local forest agencies might know wood densities of specific species. Other general sources for wood density include the World Agroforestry Database (World Agroforestry Center 2001) and sources books produced by the U.S. Department of Agriculture (Hidayat & Simpson 1994). Examples of wood density for common mangrove species are given in **Table 4.1**.

Allometric equations for mangrove tree biomass: A number of references report allometric equations for mangrove biomass (Saenger 2002; Chave *et al*. 2005; Smith III & Whelan 2006; Komiyama *et al*. 2008; Kauffman & Cole 2010; Kauffman & Donato 2011) and examples complied by Kauffman and Donato (2011) are found in **Table 4.2** (for equations with parameters of dbh and wood density) and **Table 4.3** (for equations with parameters of dbh, wood density, and tree height). Before deciding which allometric equation to use, consider the geographic origin and species that composed the data set from which the equation was derived. Ideally, it is best to use a species-specific equation developed in the region where the sampling is to occur. Given the differences in structure and wood density among species, species-specific equations are likely to yield greater accuracy than general equations. It is also critical to note the maximum diameter from which the equation was derived. Applying the equation to trees that exceed the maximum diameter (D_{max}) can lead to a statistically significant overestimation of the biomass. In very data poor situations, general allometric equations for mangrove trees can be used but the uncertainty is relatively high.

Table 4.1 Wood density of mangrove species (1 ton/m^3 = 1 g/cm^3) (Saenger 2002; Komiyama *et al*. 2005; Donato *et al*. 2012; World Agroforestry Center 2001)

SPECIES	n	AVERAGE WOOD DENSITY (TONNES/m^3)	STANDARD ERROR
Avicennia germinans	5	0.72	0.04
Avicennia marina	6	0.62	0.06
Avicennia officinalis	3	0.63	0.02
Brugueria gymnorrhiza	8	0.81	0.07
Ceriops decandra	2	0.87	0.10
Ceriops tagal	7	0.85	0.04
Excoecaria agallocha	7	0.41	0.02
Heritiera fomes	3	0.86	0.14
Heritiera littoralis	6	0.84	0.05
Laguncularia racemosa	3	0.60	0.01
Rhizophora apiculata	4	0.87	0.06
Rhizophora mangle	7	0.87	0.02
Rhizophora mucronata	9	0.83	0.05
Sonneratia alba	6	0.47	0.12
Sonneratia apetala	2	0.50	0.01
Xylocarpus granatum	7	0.61	0.04
Average		**0.71**	**0.02**

Table 4.2 Allometric equations for computing biomass of mangrove trees where only parameters of diameter (dbh) and wood density are used. The general equations include all aboveground biomass. Individual species equations have been broken down by component. B = Biomass (kg), D = Diameter at breast height (cm), ρ = Wood density (g/cm^3), D_{max} = maximum diameter of sampled trees (cm) (Modified from Kauffman and Donato, 2011).

SPECIES GROUP	N	D_{max}	LOCATION	BIOMASS EQUATION	R^2
General Equation	84	42	Americas	$B = 0.168*\rho*(D)^{2.471}$	0.99
General Equation	104	49	Asia	$B = 0.251*\rho*(D)^{2.46}$	0.98
Avicennia germinans	25	42	French Guinea	$B = 0.14*D^{2.4}$	0.97
Avicennia germinans	8	21.5	Florida, USA	$B = 0.403*D^{1.934}$	0.95
Bruguiera gymnorrhiza (leaf)	17	24	Australia	$B = 0.0679*D^{1.4914}$	0.85
Bruguiera gymnorrhiza (wood)	326	132	Micronesia	$B = 0.0754*\rho*D^{2.505}$	0.91
Laguncularia racemosa	70	10	French Guinea	$B = 103.3*D^{2.5}$	0.97
Laguncularia racemosa	10	18	Florida, USA	$B = 0.362*D^{1.930}$	0.98
Rhizophora apiculata	20	30	Malaysia	$B = 0.1709*D^{2.516}$	0.98
Rhizophora apiculata (wood)	191	60	Micronesia	$B = 0.0695*\rho*D^{2.644}$	0.89
Rhizophora apiculata stylosa (leaf)	23	23	Australia	$B = 0.0139*D^{2.1072}$	0.86
Rhizophora apiculata stylosa (stilt roots)	23	23	Australia	$B = 0.0068*D^{3.1353}$	0.97
Rhizophora mangle	14	20	Florida, USA	$B = 0.722*D^{1.731}$	0.94
Rhizophora spp (racemosa and *mangle)*	9	32	French Guinea	$B = 0.1282*D^{2.6}$	0.92
Sonneratia alba (wood)	346	323	Micronesia	$B = 0.3841*\rho*D^{2.101}$	0.92

Table 4.3 Allometric equations for computing biomass of mangrove trees where parameters of diameter (dbh) and height are used for species specific equations, and diameter and wood density are used for general equation. The general equations include all aboveground biomass. Individual species equations represent wood mass and do not include leaves or roots. B = Biomass (kg), H = Height (m), D = Diameter at breast height (cm), ρ = Wood density (g/cm^3), D_{max} = maximum diameter of sampled trees (cm), H_{max} = maximum height of sampled trees (Modified from Kauffman and Donato, 2011).

SPECIES GROUP	N	D_{max}	H_{max}	BIOMASS EQUATION	R^2
General Equation	84	42		$B = 0.0509*\rho*(D)^2*H$	
Bruguiera gymnorrhiza	325	132	34	$B = 0.0464*(D^2H)^{0.94275}*\rho$	0.96
Lumnitzera littorea	20	70.6	19	$B = 0.0214*(D^2H)^{1.05655}*\rho$	0.93
Rhizophora apiculata	193	60	35	$B = 0.0444*(D^2H)^{0.96842}*\rho$	0.96
Rhizophora mucronata	73	39.5	21	$B = 0.0311*(D^2H)^{1.00741}*\rho$	0.95
Rhizophora spp.	265	60	35	$B = 0.0375*(D^2H)^{0.98626}*\rho$	0.95
Sonneratia alba	345	323	42	$B = 0.0825*(D^2H)^{0.89966}*\rho$	0.95
Xylocarpus granatum	115	128.5	31	$B = 0.0830*(D^2H)^{0.89806}*\rho$	0.95

Variance from Allometric equations: There is a great deal of variation in wood density, morphology, and height-diameter relationships between sites, which can affect the accuracy and utility of any given allometric equation. Different equations can yield very large differences in biomass predictions. In **Fig. 4.5**, predictions generated from different allometric equations using the same dataset from a mangrove stand in Yap, FSM, are shown (Kauffman *et al*. 2011). The biomass prediction of the largest *Bruguiera* tree in this mangrove forest (69 cm dbh) was 2588 kg using the Kauffman and Cole (2010) equation and 7014 kg using the general

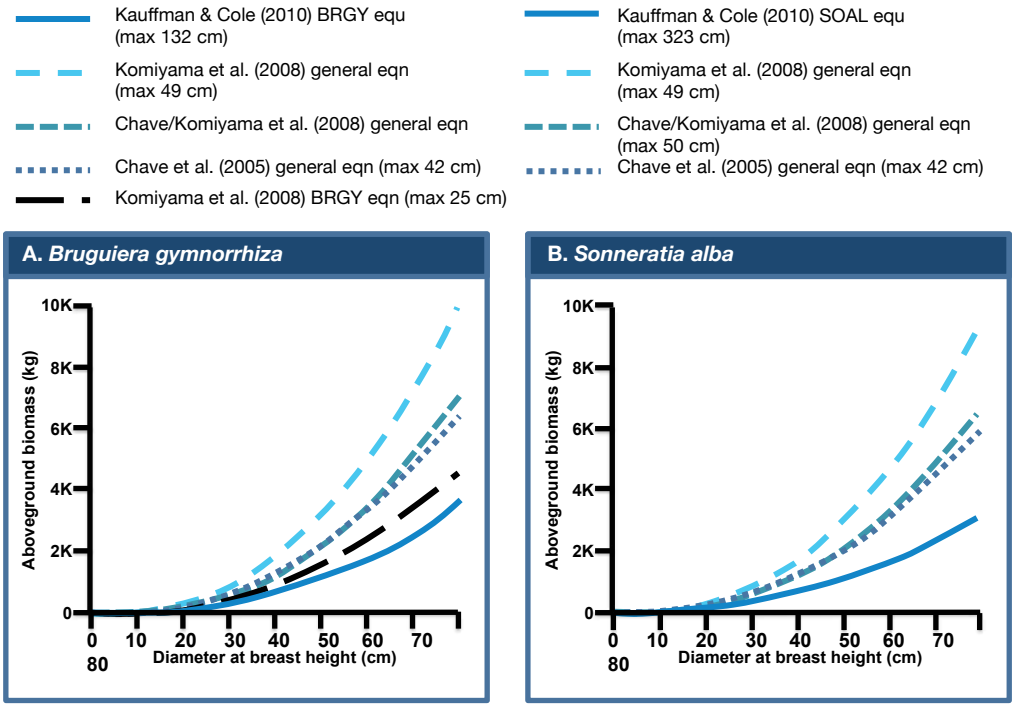

Figure 4.5 Comparison of tree biomass estimates for (A) *Burguiera gymnorrhiza* and (B) *Sonneratia alba*. The numbers in parentheses are the maximum tree diameters used to develop the equations (Chave *et al*. 2005; Komiyama *et al*. 2008; Kauffman & Cole 2010).

equation of Komiyama *et al*. (2008). Similarly, the biomass estimate for a 45 cm dbh *Sonneratia alba* tree was 873 kg using the Kauffman and Cole (2010) formula but > 1500 kg using the other equations. For the largest trees in this stand the differences were even more dramatic. The biomass estimate for an 80 cm dbh tree was 3034 kg using the Kauffman and Cole (2010) equation but over threefold higher (9434 kg) using the Komiyama *et al*. (2008) general mangrove equation. It is important to note that only the equations developed by Kauffman and Cole (2010) encompassed the entire range of diameters encountered at the Yap site, and the allometric equations used only depended on diameter (dbh) and wood density for computing biomass of mangrove trees. These large differences underscore the importance of using the same equations for all trees in a project area, when comparing different mangroves, and especially for permanent mangrove plots through time.

Determining the Carbon within the Live Tree Component (kg C/m^2): The live tree carbon component is determined by multiplying the biomass (kg) of each tree—as determined by an allometric equation or laboratory analysis—by the carbon conversion factor for mangrove species specific to that region. This is done for every tree sampled. Next, all the values for individual tree carbon content are added together to determine the total carbon content from the trees (kg C) for the given plot size (m^2). Carbon conversion factors are based on the percent of the biomass that is made of organic carbon. This can be determined through laboratory analysis using an elemental analyzer (Chapter 3), but in many cases it is possible to find a published carbon conversion factor for specific tree species. Kauffman *et al*. (2011) reported the carbon content of *Bruguiera gymnorrhiza* as 46.3%, *Rhizophora apiculata* as 45.9%, and *Sonneratia alba* as 47.1%. Thus, if local or species-specific values are not available the biomass of these and other trees can be multiplied by 0.46 to 0.5 to obtain carbon content.

EXAMPLE

- Carbon content of each tree (kg C) = tree biomass (kg) * carbon conversion factor (0.46–0.5)
- Carbon in live tree component (kg C/m^2) = (carbon content of tree #1 + carbon content of tree #2 + ……. Tree #n) / area of the plot (m^2)

SCRUB MANGROVES

A great percentage of the world's mangroves have an aboveground structure of small trees less than a few meters in height, often referred to as dwarf mangrove, scrub mangrove, or "manglar chaparro." Currently there are few allometric equations to determine aboveground biomass for these kinds of mangroves; thus, it is an important research need. The few existing equations that are most often used are from measurements of dwarf mangroves in south Florida, USA (Ross *et al*. 2001; Coronado-Molina *et al*. 2004). The equations in Ross *et al*. (2001) utilize the main stem diameter at 30 cm above the ground surface and crown volume to predict aboveground biomass of individual mangroves. Coronado-Molina *et al*. (2004) developed allometric equations utilizing crown area and the number of prop roots. The most accurate method, however, is to develop allometric equations specifically for the plants in your area of interest.

To develop an allometric equation for a new site, at least 15 to 25 trees of each species in question, encompassing the range in size from the smallest seedlings to the largest individuals, should be measured (typically crown diameter, crown volume, crown area, tree height, and/or main stem diameter at 30 cm aboveground, **Fig. 4.6**) and harvested. In the laboratory, individual trees are dried and weighed to obtain biomass. A relationship between

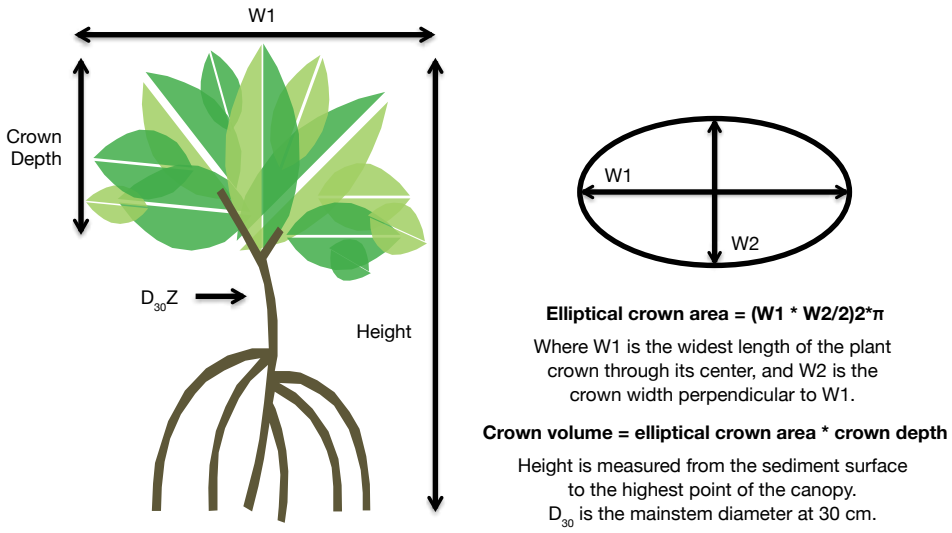

Figure 4.6 Field measurement techniques to calculate the elliptical crown area, crown depth, height, and diameter at 30 cm height (D_{30}) of dwarf mangroves. Aboveground biomass of these small trees is best predicted through allometric equations where aboveground biomass is the dependent variable and crown area, and height and/or crown volume are independent variables (Ross *et al*. 2001).

the tree biomass and the biometric measurements (crown diameter, area and volume, main stem diameter at 30 cm) can then be developed by regression analysis.

Once allometric equations have been established, they can be applied to every scrub mangrove tree within a sample plot. The plot size is typically small; however, since scrub mangroves grow in very dense communities, field measurements can be time consuming (**Fig. 4.2A**). For example, a typical plot size and shape would be a 2 m radius half circle plot (total area 6.3 m^2).

Determining the Carbon within the Scrub Mangrove Component (kgC/m^2): The scrub mangrove carbon component is determined for each tree by multiplying the biomass (kg), as determined by an allometric equation, by a carbon conversion factor for scrub mangroves specific to that region. This is done for every scrub tree in the plot. Next, all the values for individual tree carbon content are added together to determine the total carbon content from the scrub trees (kg C) for the given plot size (m^2).

The carbon conversion factor for scrub mangroves is not well documented in the scientific literature; therefore, it can be determined either through laboratory analysis using an elemental analyzer (Chapter 3), or it is justifiable to use the conversion factor reported for tall mangrove trees (0.46 to 0.5, see section on mangrove trees above).

EXAMPLE

- Carbon content of each tree (kg C) = tree biomass (kg) * carbon conversion factor (0.46–0.5)
- Carbon in live tree component (kg C/m^2) = (carbon content of tree #1 + carbon content of tree #2 + ……. Tree #n) / area of the plot (m^2)

STANDING DEAD TREES

Within each sampling plot, all trees that are dead and standing should be recorded as such and analyzed as a separate pool. The degree to which the tree has decayed will determine how its biomass is calculated. Decay status is broken down as follows (**Fig. 4.7**):

- **Decay status 1:** Small branches and twigs are retained; resembles a live tree except for absence of leaves.
- **Decay status 2:** No twigs/small branches; may have lost a portion of large branches.
- **Decay status 3:** Few or no branches, standing stem only; main stem may be broken-topped.

Figure 4.7 Examples of dead tree decay status. 1) Recently dead trees that maintain many smaller branches and twigs, 2) Trees have lost small branches and twigs, and a portion of large branches, 3) Most branches have been lost and only the main stem remains, and is often broken (Solochin 2009).

The biomass of standing dead trees is then determined according to the decay status of the tree.

Decay status 1: Biomass can be estimated using live tree equations. The only difference is that leaves should be subtracted from the biomass estimate. This can be accomplished either using a leaf biomass equation to determine the quantity of leaves to be subtracted (Clough & Scott 1989; Komiyama *et al*. 2005; Smith III & Whelan 2006), or by subtracting a constant of 2.5% of the live tree biomass estimate.

Decay status 2: Biomass can be calculated by subtracting away a portion of the biomass from the live tree equations. Because they have lost some branches in addition to leaves, both leaf biomass and an estimate of branch loss must be factored in. Commonly, a total of 10–20% of biomass (accounting for both leaves and some branches) is subtracted. This amount can be adjusted and tailored to specific conditions using field observations.

Decay Status 3: Trees have often lost a significant portion of their volume due to advanced breakage; consequently, it is difficult to estimate biomass from the live-tree biomass estimates. Instead, the remaining tree's volume may be calculated using an equation for a frustum (truncated cone). To do this, record the diameter at the base of the tree and the

total tree height using a laser tool or clinometer (a tool used to measure mangrove tree height). The top diameter must be estimated using a taper equation.

Taper equation for estimating the top-diameter of a broken-topped dead tree:

- Estimating the top-diameter of a broken-topped dead tree (cm) = the measured basal diameter (cm) – [100 * tree height (m) * ((the measured basal diameter (cm) – diameter at breast height (cm) / 130)]
  - $d_{top} = d_{base} – [100 * ht * ((d_{base} – dbh) / 130)]$
  - If taper equation results in negative number, use 0 for the next equation.

Then the volume of the dead tree is determined by assuming the tree is a truncated cone:

- Dead tree volume (cm^3) = [π * (100 x tree height (m)) / 12] * [base diameter $(cm)^2$ + top diameter $(cm)^2$ + (base diameter (cm) x top diameter (cm))]
  - Volume (cm^3) = (π * (100*ht) / 12)* ($d2_{base} + d2_{top} + (d_{base} * d_{top})$).

Dead tree biomass (kg) is then determined by multiplying its volume (cm^3) by its wood density (g/cm^3).

- Decay Status 3 dead tree biomass (kg) = Volume of the dead tree (cm^3) * wood density (g/cm^3)) * (1 kg / 1000 g).

Ideally wood density of standing dead trees would be determined in the laboratory; however, if that is not practical, a list of standard densities based on size (**Table 4.4**) can be used for this calculation. It is important to note that the density information in **Table 4.4** was derived from downed wood measured following a typhoon with little or no decay. Studies have shown that there is a broad range of densities for various components (twigs 0.628–0.350, branches 0.60–0.284, prop roots 0.276–0.511, and trunks 0.340–0.234), emphasizing the need for site specific estimations of wood density when possible (Robertson & Daniel 1989).

Table 4.4 The wood density and mean diameter of the standard wood debris size classes of downed mangrove wood (Rhizophora apiculata, Sonneratia alba, and Bruguiera gymnorrhiza) (Kauffman & Cole 2010).

SIZE CLASS (cm DIAMETER)	DENSITY ±SE (g/cm^3)	SAMPLE SIZE (n)
< 0.64	0.48 ± 0.01	117
0.65 – 2.4	0.64 ± 0.02	31
2.54 – 7.6	0.71 ± 0.01	69
> 7.6	0.69 ± 0.02	61

Determining the Carbon within the Standing Dead Tree Component (kg C/m^2): The standing dead tree carbon pool is determined by multiplying the biomass (kg), determined by decay status, of each individual standing dead tree by a carbon conversion factor. Next all the values for individual tree carbon content are added together to determine the total carbon content from the trees (kg C) for the given plot size (m^2).

The carbon conversion factor for standing dead trees can be determined through laboratory

analysis using an elemental analyzer (Chapter 3). If this is not practical, the carbon concentration of dead wood is usually around 50% (Kauffman *et al*. 1995). Thus, the biomass of these trees is typically multiplied by 0.5 to obtain carbon stock value.

EXAMPLE

- Carbon content of each tree (kg C) = tree biomass (kg) * carbon conversion factor (0.46–0.5)
- Carbon in standing dead tree component (kg C/m^2) = (carbon content of tree #1 + carbon content of tree #2 + ……. Tree #n) / area of the plot (m^2)

LIANAS

Lianas are long-stemmed, woody vines that are rooted in the soil of the forest floor and climb up into the forest canopy on trees (**Fig 4.8**). They range from small, indiscrete vines that wind around tree trunks to giant lianas several cm in diameter that seemingly hang in the middle of the forest independent of trees. Large lianas rarely exist in mangroves, but if present they can be measured just like trees.

Figure 4.8 Lianas. (A) Close up view of a typical liana found in mangrove forests (© IITA), (B) image of the extent to which lianas can contribute to the vegetative biomass (© Mark Marathon, Wikimedia Commons)

Every liana in a sample area should be measured if they are being included in calculations. There are a number of equations to determine liana biomass (Schnitzer *et al*. 2006), but for our purposes liana biomass can be estimated using the general allometric equation:

- Biomass for lianas (kg) = (Diameter 130 cm from the soil surface (cm))$^{2.657}$ * $e^{0.968}$ x ln (Diameter 130 cm from the soil surface (cm))
  - $B = D^{2.657} * e^{-0.968} * \ln(D)$

Determining the Carbon within the Liana Component (kg C/m^2): When lianas are present in mangrove forests, their carbon pool is determined by multiplying the biomass (kg) of each individual vine by a carbon conversion factor. Next, all the values for individual liana carbon content are added together to determine the total carbon content from the vines (kg C) for the given plot size (m^2).

The carbon conversion factor for lianas can be determined through laboratory analysis using an elemental analyzer (Chapter 3). If this is not practical, the carbon concentration (based on dry weight) of lianas in the forest of Mexico have been reported to be ~46% (Jaramillo *et al*. 2003); therefore, a default value for the carbon conversion factor of lianas is 0.46.

EXAMPLE

- Carbon content of each liana (kg C) = liana biomass (kg) * carbon conversion factor (0.46)
- Carbon in the liana component (kg C/m^2) = (carbon content of liana #1 + carbon content of liana #2 + ……. liana #n) / area of the plot (m^2)

PALMS AND OTHER NON-TREE VEGETATION

Understory vegetation (e.g., non-mangrove tree seedlings and herbs) is generally negligible in mangroves, and its measurement for ecosystem carbon pools is usually unnecessary (Snedaker & Lahmann 1988).

The most notable exception is the nypa palm (*Nypa fruticans*, **Fig. 4.9**) of the Asia-Pacific region where it may be the dominant species in some locations. Approaches to sampling these vegetation components will depend on their density, structure, and distribution (Snedaker & Lahmann 1988). The biomass of nypa palms and herbaceous vegetation may be determined either through non-destructive sampling or destructive harvests (which is most common for herbaceous vegetation, such as ferns, seagrass, grasses, sedges, rushes, and broad-leaved herbs). Non-destructive approaches are necessary for plots that will be revisited over time, especially for perennial species.

Figure 4.9 Examples of palm plants found in mangroves. (A) *Small Nypa fruticans* (© Andreas Hutahahean, KKP), (B) tall woody stemmed palm plants (© Enrico Marone, CI)

To determine biomass (kg), at least 15 to 25 palm fronds from different individual plants should be collected from outside any permanent sample plots. The samples should cover the observed size distribution of the individual leaves and each sampled frond is cut at ground level. Obtain the dry mass of each frond in the laboratory and calculate the average. Count the total number of nypa leaves within a sample (or subsample) plot and then multiplying that number by the average dry mass.

Palms with woody trunks can be measured in the same manner as broad-leaved trees. Parameters of the palm that need to be measured (dbh, height, etc.) depend on the allometric equation used to determine palm biomass. The most frequently used variable to determine palm biomass is the height of the main stem from the ground to the base of leaves.

Determining the Carbon within the Palm Component (kg C/m²): The palm carbon component is determined by multiplying the biomass (kg), either the average calculated from the collected fronds or from allometric equations for larger woody palms, by a carbon conversion factor for palm species specific to that region.

If the biomass used was determined for the entire plot using the average biomass of the fronds collected multiplied by all the fronds in the plot, then simply multiply that number by the carbon conversion factor to determine the total palm carbon component for your plot.

If the biomass used was determined for larger woody palms, all the values for individual palm carbon content must be added together to determine the total carbon content from the trees (kg C) for the given plot size (m^2).

The carbon conversion factor for palms can be determined either through laboratory analysis using an elemental analyzer (Chapter 3) or a conversion factor of 0.47 can be used (Kauffman *et al*. 1998).

EXAMPLE FOR SMALL PALMS

- Carbon in the small palm component (kg C/m^2) = (Estimated biomass of the palm fronds * carbon conversion factor (0.47)) / area of the plot (m^2).

EXAMPLE FOR LARGE WOODY PALMS

- Carbon content of each large palm (kg C) = palm biomass (kg) * carbon conversion factor (0.47);
- Carbon in large palm component (kg C/m^2) = (carbon content of tree #1 + carbon content of tree #2 + ……. Tree #n) / area of the plot (m^2).

PNEUMATOPHORES

Pneumatophores of mangrove species of the genera *Avicennia*, *Brugueira*, and *Sonneratia* can be of significant structure and biomass, and unlike the stilt roots on *Rhizophora*, these tree parts are not included in the allometric equations of biomass for live trees (**Fig. 4.10**). Pneumatophore density can be determined by counting their numbers in microplots within or immediately adjacent to, the main sampling plot.

A common-sized microplot is 50 x 50 cm^2 but can range from 30 x 30 cm^2 to 1 x 1 m^2. To determine biomass (kg), pneumatophores should be collected from outside any permanent sample plots. All pnematophores within the plot should be counted and 50–100 samples should be collected. The samples should cover the observed size distribution and be cut at ground level. Obtain the dry mass of each pneumatophore in the laboratory and calculate the average.

Figure 4.10 Pneumatophores. (A) Measuring pneumatophore height, (B) Pneumatophores can be measured in or next to microplots. These microplots can be the same plots used to sample litter (described below). (© Boone Kauffman, OSU)

- Biomass for pneumatophores (kg) = Average dry mass of sampled pneumatophores * number of pneumatophores in the microplot.

An allometric equation using pneumatophore height to predict biomass could be developed and would be of value in permanent plots. If such an equation were developed, each pneumatophore in a microplot would need to be measured for height. This would be most relevant for those species containing large pneumatophores such as *Sonneratia alba*.

Determining the Carbon within the Pneumatophore Component (kg C/m^2): The pneumatophore carbon component is determined by multiplying the average biomass (kg)—calculated from the collected pneumatophores—by a carbon conversion factor for species specific to that region.

Since the biomass used was determined for the entire microplot using the average biomass of the pneumatophores collected multiplied by all the pneumatophores in the plot, you simply need to multiply that number by the carbon conversion factor to determine the total pneumatophore carbon component for a microplot.

The carbon conversion factor for pneumatophores can either be determined through laboratory analysis using an elemental analyzer (Chapter 3), or a conversion factor for roots can be used—typically 0.39 (Kauffman & Donato 2011).

EXAMPLE

- Carbon in the pneumatophore component (kg C/m^2) = (Estimated biomass of the pneumatophores * carbon conversion factor (0.39)) / area of the plot (m^2).

LITTER

The litter layer is defined as the recently fallen non-woody dead organic material on the soil surface. Typically, it consists of dead leaves, flowers, fruits, seeds, and bark fragments. In most mangrove settings, the amount of this material (and therefore carbon stock) only represents a very small component of the carbon pool due to the high efficiency of detritus-consuming crabs as well as export through tides and seasonal river flooding.

Figure 4.11 Pre-labelled plastic bags containing litter (© Boone Kauffman, OSU)

If it is measured, litter in most biomass studies is destructively sampled through collection from small microplots, often 0.5 x 0.5 m in size. All organic surface material, excluding woody particles, is collected into a sturdy bag or container. The bags are labelled with the location, date, plot, and sample number (**Fig. 4.11**). Given the wet nature of mangroves, pre-labelled plastic bags with permanent markers may be most efficient for litter samples.

Determining the Carbon within the Leaf Litter Component (kg C/m^2): The samples should be transported to the laboratory, placed in a drying oven, and dried to constant mass. Due to constraints of carrying bulky samples from the field and the limited availability of oven drying space, especially in rural field settings, we suggest determining the wet weight of the entire sample and then extract a well-mixed representative subsample for transportation to a laboratory to be dried to a constant weight. Finally, determine the ratio between wet and dry mass of the subsample by recording the wet mass of the sample and relating it to the dry (constant) mass (Cummings *et al*. 2002).

- Biomass of litter (kg) = (dry mass of subsample (g) / wet mass of the subsample (g)) * wet mass of all the litter in the sample plot (kg)

The litter biomass (kg) can then be estimated for the given plot size (m^2). Mean carbon concentrations of tropical forest leaf litter dry mass have been reported to range from 38–49% (Kauffman *et al*. 1993; Kauffman *et al*. 1995). Therefore, a carbon conversion factor of about 0.45 is recommended.

EXAMPLE

- Carbon in the leaf litter component (kg C/m^2) = (Average biomass of the litter * carbon conversion factor (0.45)) / area of the plot (m^2)

Figure 4.12 Downed wood. (A) Downed wood after a storm in Bangladesh, (B) Uprooted *Rhizophora* and *Sonneratia* following Typhoon Sudal, Yap, FSM (© Boone Kauffman, OSU)

DEAD AND DOWNED WOOD

There are several guides that describe methods for determining downed wood volume and mass (Harmon & Sexton 1996; Waddell 2002). Dead and downed wood material can be a significant component of aboveground biomass, particularly following natural disturbances, such as tropical cyclones (**Fig. 4.12**). Land-use and/or land-cover change may also increase the quantity of downed wood on the mangrove forest floor. To accurately assess ecosystem carbon pools and influences of natural and human disturbances, dead and downed wood is an important variable to measure. Downed wood is usually sampled either by plot-based sampling or by the line-intersect method (Waddell 2002; Baker & Chao 2009). The non-destructive line-intersect technique is recommended.

Line (or planar) intersect technique for sampling downed wood: The line (or planar) intersect technique involves counting intersections of woody pieces along a vertical sampling plane (transect). In each sampling plot, a series of transects should be established to measure this carbon pool. A transect is a straight line across the entire length of the plot. For example, four transects might be established in each of six subplots for a total of 24 transects per plot.

Important rules in measuring downed wood include:

- Dead trees that are standing are not measured in the line intersect technique;
- Downed wood must be broken from the tree where it originated; thus, dead branches and stems still attached to standing trees or shrubs do not count;
- The transect tape must intersect the central axis of a wood piece for it to be counted. This means that if the tape only intersects a corner at the end of a log, it does not count; and
- Any piece can be recorded multiple times if the tape intersects it more than once (e.g., a curved piece, or at both the branch and the stem of a fallen tree).

All downed, dead, woody material (fallen/detached trunks, branches, prop roots, or stems of trees and shrubs) that has fallen and lies on or over the transect line (within 2 m of the ground surface) is counted and classified using this technique. Woody debris can be categorized into four sizes: fine, small, medium, and large wood particles (**Table 4.5**). These size classes are regularly used in forest inventories, and convenient measurement tools exist to streamline field sampling based on these limits (**Fig. 4.13**).

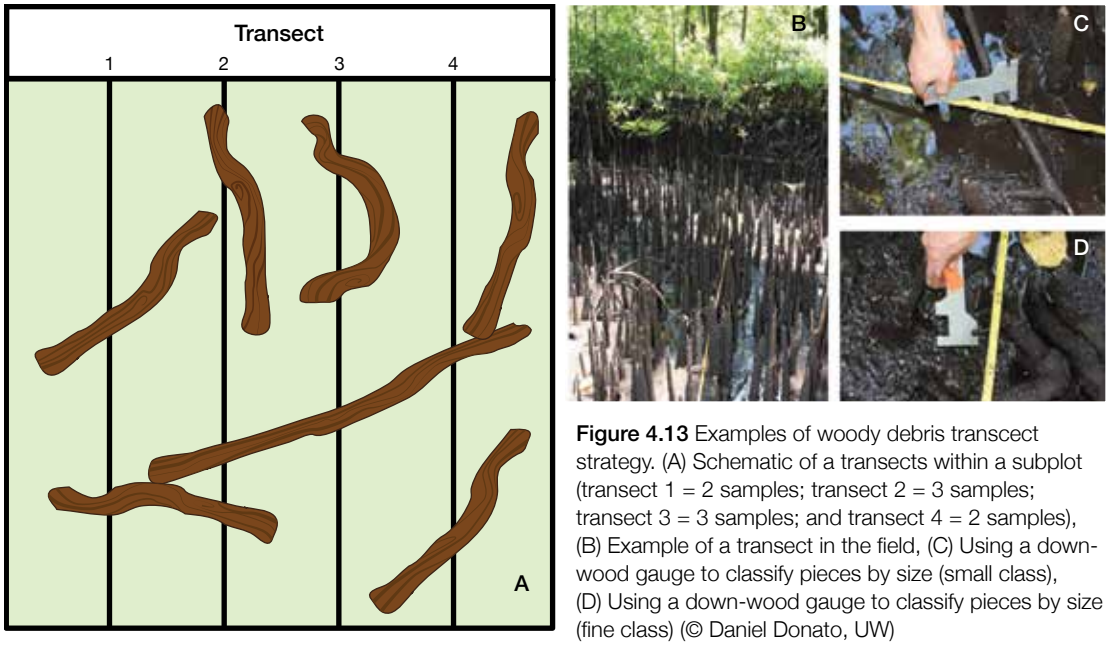

Figure 4.13 Examples of woody debris transcect strategy. (A) Schematic of a transects within a subplot (transect 1 = 2 samples; transect 2 = 3 samples; transect 3 = 3 samples; and transect 4 = 2 samples), (B) Example of a transect in the field, (C) Using a down-wood gauge to classify pieces by size (small class), (D) Using a down-wood gauge to classify pieces by size (fine class) (© Daniel Donato, UW)

Table 4.5 Commonly used size classes of wood.

DESCRIPTION	DIAMETER
Fine	0 – 0.6 cm
Small	0.6 – 2.5 cm
Medium	2.5 – 7.6 cm
Large	≥ 7.6 cm

The number of fine, small, and medium pieces that cross the transect line are counted and tallied separately for each size class. Smaller pieces can be very abundant, and to save time, are only sampled along subsections of each transect (**Fig. 4.14**).

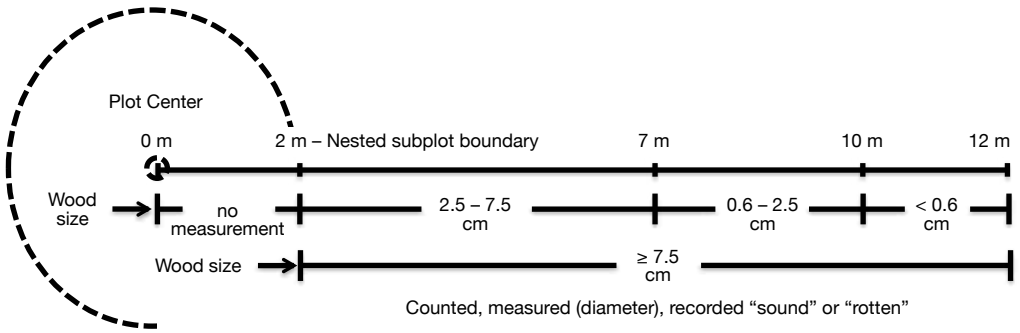

Figure 4.14 Example of a wood debris transect for sampling downed wood in mangroves using the line intersect technique. The diameter of each large wood piece encountered along the entire transect is measured. Smaller diameter pieces are counted but not measured along the designated lengths of the transect.

To convert the count for the smaller class sizes into biomass, an average diameter of wood pieces in each of these classes must be estimated. The average diameter of wood pieces can be derived from measurements of about 50–100 randomly selected pieces of each

size class (fine, small, and medium). A digital calliper is recommended for measuring the diameter of each piece. We recommend using a quadratic mean equation to diameter the average diameter of the wood pieces rather than the mean diameter of wood classes (Brown & Roussopoulos 1974). The quadratic mean diameter (QMD) is calculated as:

- Quadratic mean diameter (cm) = $\sqrt{(\sum \text{diameter of each piece of wood}^2)}$ / number of pieces sampled
  - QMD = $\sqrt{(\sum Di^2)}$ / n

Examples of quadratic mean diameter of down wood from mangrove forests are shown in **Table 4.6**.

In contrast to the smaller wood pieces, the diameter of each large wood piece encountered along the entire transect is recorded (**Fig. 4.14**). For each large piece crossing the transect line, record its diameter at the point where the transect line crosses the midpoint of the wood particle. Also, record whether the decay status is sound (machete bounces off or only sinks slightly when struck) or rotten (machete sinks deeply and wood is crumbly with significant loss).

In a practical sense, usually only the medium and large wood fraction (> 2.5 cm diameter) is a significant carbon pool (Kauffman & Cole 2010; Kauffman *et al*. 2011). If sampling the litter component, it may be simpler to include the smaller wood pieces (< 2.5 cm diameter) in the litter sample and only measure the medium and large wood pieces along the transect line.

Wood density: The density of wood debris is determined from the dry weight of the wood dried at 100 ºC divided by the volume of undried wood. Density must be determined for each wood category sampled (fine, small, medium, large-sound, large-rotten) in order to determine biomass. We recommend collecting at least 20–25 pieces of wood for each species and size class, capturing a representative range of sizes within each class and the full range of species present in the sample. As a rough guideline, each piece collected for wood density determination should have a mass of between ~ 0.5 and 50 g. Randomly collect pieces to determine wood density within the project area; to avoid undue disturbance, do not collect pieces from permanent sample plots. See beginning of this chapter for a discussion on wood density calculations. Examples of wood density for downed wood from mangrove forests are shown in the table below.

Table 4.6 The wood density and mean diameter of the standard wood size classes of downed mangrove forests dominated by *Rhizophora apiculata*, *Sonneratia alba*, and *Bruguiera gymnorrhiza*. Data are from downed wood following a typhoon with no decay. na = not applicable (Modified from Kauffman and Cole, 2010).

SIZE CLASS (cm diam.)	DENSITY ±SE (g/cm^3)	SAMPLE SIZE	DIAMETER (cm)	QUADRATIC MEAN DIAMETER (cm)	SAMPLE SIZE
< 0.64	0.48 ± 0.01	117	0.43 ± 0.15	0.43	50
0.65 – 2.4	0.64 ± 0.02	31	1.33 ± 0.78	1.47	48
2.54 – 7.6	0.71 ± 0.01	69	4.30 ± 0.18	4.52	52
> 7.6	0.69 ± 0.02	61	na	na	na

Downed wood volume is then calculated from line intercept data using scaling equations.

- Wood volume for fine, small, and medium classes per unit of ground area (m^3/ha) = (π^2 x [number of samples x quadratic mean diameter for the size class (cm))^2 / (8 x transect length (m))]
  - Volume (m^3/ha) = π^2 * (Ni * QMDi^2 / (8 * L))
- Wood volume of large (> 7.6 cm diameter) down wood per unit of ground area (m^3/ha) = π^2 * [$\sum$diameter of each piece of wood^2 / (8 x transect length (m))]
  - Volume (m^3/ha) = π^2 * (d1^2 + d2^2 + d3^2 +….dn^2 / (8 * L))
  - Each piece is individually measured

Downed wood biomass (kg/ha) is then calculated as the volume multiplied by its average wood density.

- Downed wood biomass (kg/ha) = volume (m^3/ha) * average wood density (kg/m^3)

Determining the Carbon within the Downed Wood Component (kg C/ha): Finally, convert downed wood biomass to carbon mass. An acceptable default value based upon carbon content of dead wood in tropical forests is 50%, corresponding to a carbon conversion factor of 0.50.

EXAMPLE

- Carbon content of downed dead wood per plot (kgC/ha) = dead wood biomass (kg/ha) * carbon conversion factor (0.5)

BELOWGROUND TREE BIOMASS

Belowground biomass (e.g., roots and the base of stems) is an important component in mangroves. The aboveground-to-belowground biomass ratio for mangroves is generally found to be between 2.0 and 3.0, whereas in upland forests the ratio is higher—between 3.96 and 4.52. Thus, in mangrove forests, a large amount of biomass tends to be allocated below ground (Komiyama *et al*. 2008). Given the extreme difficulty of collecting and measuring root systems, it is beyond the capacity of most mangrove projects to destructively harvest and measure belowground biomass or develop allometric equations. Very few allometric equations exist for belowground biomass of forests, and mangroves are among the least studied forests in this respect. Additional studies would be a valuable contribution to science. Useful belowground equations available for mangroves have been reviewed in Komiyama *et al*. (2008). The general equation reported by Komiyama *et al*. (2008) is:

- Belowground tree biomass (kgC) = 0.199 * ((wood density (g/cm^3)$^{0.899}$) * (tree diameter at breast height (cm))$^{2.22}$

Carbon content of roots are typically lower than that of aboveground tree components. Jaramillo *et al*. (2003) reported that carbon content of roots of tropical forests range from 36–42%. A defensible default value for root carbon content would be 39% (median); making the carbon conversion factor 0.39.

EXAMPLE

- Belowground carbon content of each tree (kg C) = Belowground tree biomass (kg C) * carbon conversion factor (0.39)
- Carbon content of belowground carbon per plot (kg C/m^2) = (belowground carbon content of tree #1 + carbon content of tree #2 + ……. Tree #n) / plot area (m^2)

TIDAL SALT MARSHES

For the purposes of this manual, tidal salt marshes are defined as coastal ecosystems in the upper coastal intertidal zone between land and open salt water that is regularly flooded and covered with vegetation (excluding trees). Tidal salt marshes exclude mangroves, along with tidal freshwater/brackish forests (e.g., Swamp oak, Paperbark, Cyprus swamps, and willows), and un-vegetated salt flats. The halophytic plants that inhabit coastal tidal salt marsh are not exclusively intertidal or marine—a characteristic that sets them apart from mangrove and seagrass vegetation—and are dominated by herbaceous plants including tidal grasses. sedges, and rushes. Tidal saltbushes and shrubs may also be present. The variety of growth forms is important when calculating carbon stocks because each has a different biomass.

Tidal salt marshes establish on soft substrate shores within the tidal reaches of estuaries and embayments and on some open low-wave energy coasts (**Fig 4.15**). At a global scale, tidal salt marshes establish on shorelines unsuitable for mangrove forests or where development is limited (Kangas & Lugo 1990). For this reason, these systems are most common in temperate, subarctic and arctic zones (Long & Mason 1983; Mitsch & Gosselink 2007). In tropical regions, tidal salt marshes may be replaced in the intertidal environment, either by mangroves in humid climates or saltpans in arid climates.

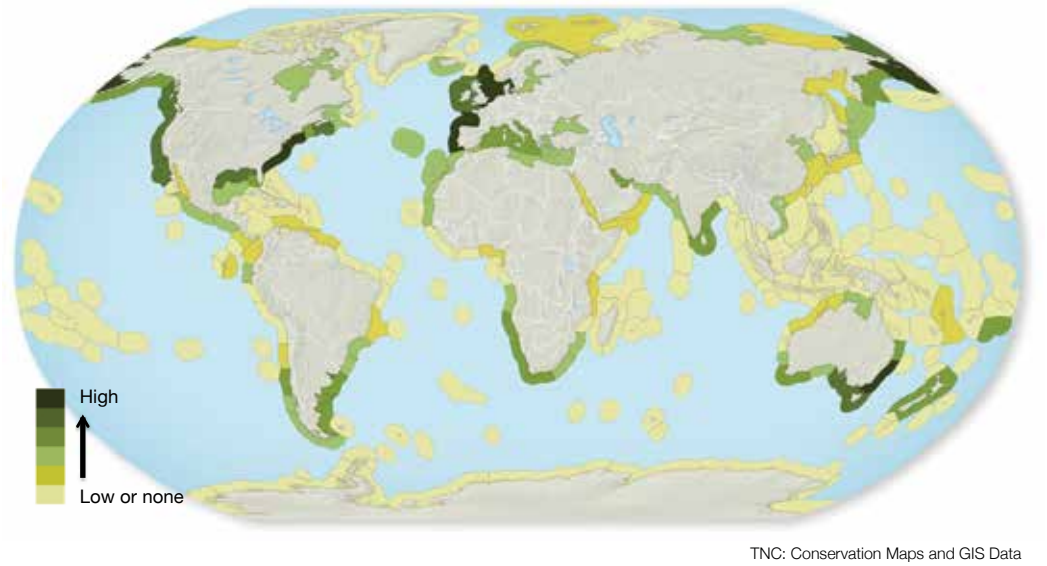

Figure 4.15 Tidal salt marsh abundance by marine ecoregion (Hoekstra *et al*. 2010)

The distribution of tidal salt marsh plants varies across the marsh and with marsh elevation, due to differences in tolerance to soil salinity, oxygen levels, sulphide levels, hydroperiod, etc. (Partridge & Wilson 1987). Along the Atlantic coast of North America, tidal salt marshes are

often divided into different zones, based on variations in plant species. and relative elevation of the ground surface with respect to the tide (Niering & Warren 1980) (**Fig. 4.16**). The low marsh area is located at the lowest elevation and is more frequently inundated by tides. The middle/high marsh is flooded less frequently and the higher high marsh is even less frequently inundated by the tide. Brackish marshes are typically located along the inland edges of the marsh area where freshwater input creates a brackish environment.

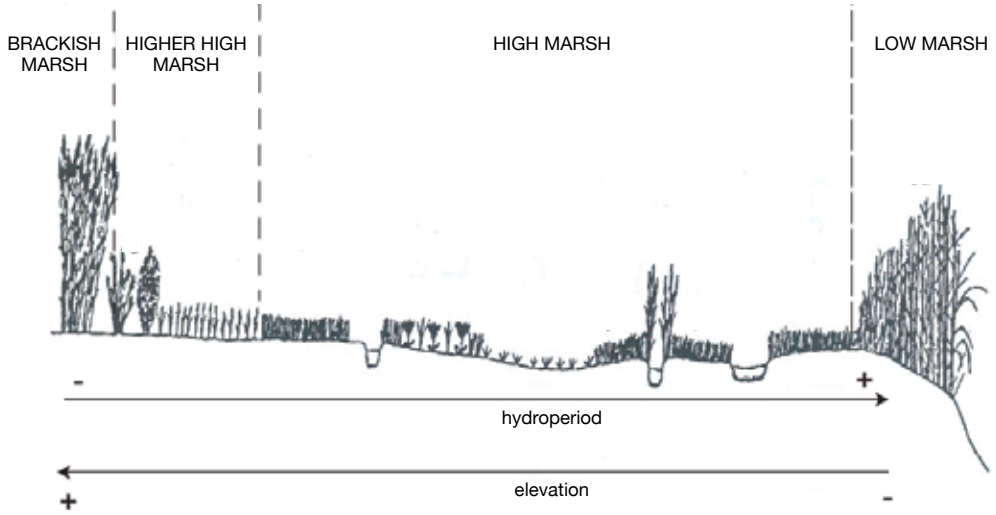

Figure 4.16 Zonation of tidal salt marsh vegetation in herb dominated systems, NE North America (modified from Niering & Warren 1980)

The different zones of the marsh may have floristic and other structural differences that influence above- and below-ground biomass of living plants. Additionally, geochemical properties, including nutrient availability and redox conditions may influence carbon retention in the soil.

Tidal salt marshes provide a host of ecosystem services, including organic matter production and export, nutrient cycling, buffering against storm activity, and carbon sequestration (Nixon 1980; Dame *et al*. 1991; Mitsch & Gosselink 2000; Chmura *et al*. 2003). Globally, tidal salt marshes have suffered from a long history of conversion into non-tidal land (termed "reclamation"), usually through the imposition of embankments or dikes, which prevent flooding of the ecosystems by tides. Other direct, human-induced causes of degradation of coastal tidal salt marshes include their conversion to salt evaporation ponds, aquaculture ponds (i.e., shrimp ponds) (Barg *et al*. 1997), and the alteration of tidal salt marsh drainage for insect control (Daiber 1982). Drainage control has also been implemented to protect low-lying land, leading to changes in the plant composition of the marsh ecosystem.

Although no global assessment of tidal salt marsh decline has been conducted, regional case studies suggest that tidal salt marsh extent has declined significantly over the past century. Wetland loss on the U.S. Mississippi Delta has been estimated to be in the vicinity of 100 km^2/yr (Viles & Spencer 1995) due to the diversion of water and sediment in the Mississippi catchment and delta (Gosselink & Maltby 1990). Overall loss of North American tidal salt marshes is estimated to be 50% since European settlement (Gedan *et al*. 2009). In southeast Australia, tidal salt marsh area has declined by 30–40% since 1950, primarily due to the replacement of tidal salt marshes by mangroves (Saintilan & Williams 1999), and 50% of China's tidal salt marshes were lost due to reclamation since the 1950's (An *et al*. 2007).

These losses have important implications for carbon retention in the system (DeLaune & White 2012).

Tidal salt marshes sequester and store carbon efficiently in the plant biomass and soils. Belowground carbon pools usually constitute between 65 and 95% of the total ecosystem carbon stock of tidal salt marshes (Elsey-Quirk *et al*. 2011), dominating the carbon sequestration, particularly in low and middle marsh areas. Aboveground biomass becomes a more significant component of the total carbon pool in the high marsh. Thus, it is important to be able to differentiate high, middle, and low marsh when creating a sampling strategy.

Field Sampling Considerations

Tidal salt marsh vegetation will vary depending on the season as well as soil moisture, nutrient, and salinity conditions that correspond to variations in tidal flooding. Proper sampling techniques take these factors into account by subdividing areas of interest into strata of (relatively) homogeneous ecological characteristics, and sampling takes place when the biomass is at its peak growth (usually late summer). Sampling may also be limited to low tidal periods of as little as 3 to 4 hours. This narrow window necessitates an efficient sampling plan.

The strata of (relatively) homogeneous ecological characteristics within tidal salt marshes usually occur parallel to the shoreline or the tidal channel. Due to the innate structure of tidal salt marshes, strata will usually form perpendicular to the dominant tidal channel/shoreline. Sufficient sampling plots should be placed in each stratum to characterize biomass within that stratum. Plots within each stratum should be approximately 20 m x 50 m. Within these plots, a minimum of 5–6 smaller subplots (0.25 m x 0.25 m) are randomly located (**Fig. 4.17** and Chapter 2 on project planning and sampling design for more details).

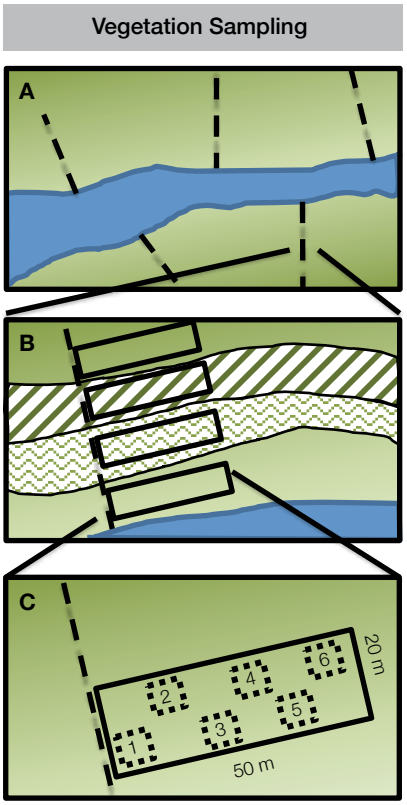

Figure 4.17 Recommended Placement of sampling plots in tidal salt marshes. (A) Transects should occur perpendicular to the dominant channels in the marsh and that cross across vegetation or other structural gradients, (B) Plots along the transects should be contained within strata, and (C) enough quadrats (minimum five) should be sampled in each plot to characterize the biomass.

Tidal salt marsh aboveground biomass can vary substantially between seasons, particularly in cool, temperate, and subarctic climates where the entire aboveground biomass of the tidal salt marsh might be lost during the winter (Darby & Turner 2008a). Other factors include elevated nutrient levels that can decrease the proportion of biomass allocated to the roots (Darby & Turner 2008b), salinity level that can increase root:stem ratio (Parrondo *et al*. 1978), grazing pressure (Giroux & Bédard 1987), and water table position (Saunders *et al*. 2006).We recommend that aboveground and belowground estimates of tidal salt marsh biomass be taken when biomass is likely to be at its greatest—usually mid-late summer. For permanent plots and/or repeated measurements, it is important sample during the same season each time so that results will be comparable.

Biomass Estimates

This section provides guidance on how to measure aboveground biomass across a range of tidal salt marsh types likely to be encountered in the field. Some differences in sampling procedures are required to accommodate differences in growth forms.

GRASSES, SEDGES AND OTHER HERBACEOUS PLANTS

Reeds, sedges and rushes are densely growing plants, with tall, straight stems and are the primary vegetation type in tidal salt marshes. The method commonly used to estimate their aboveground biomass uses a quadrat to define sampling areas within plots. Quadrats for this ecosystem are typically 30 cm x 30 cm, though a larger quadrat should be used if less than 10 individuals are likely to be counted in the 30 cm x 30 cm area. Within each quadrat, count the number of stems per species (tidal salt marshes generally contain large stands of a single dominant species but not always) and measure the total height for each species present within the quadrat. If a stem shows signs of senescence (dead material) at the tip, measure the green height (**Fig 4.18**).

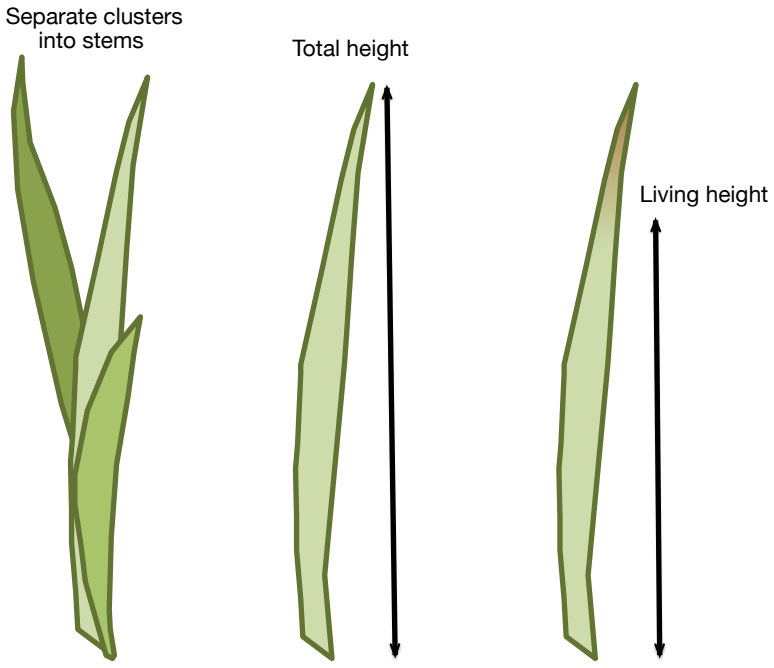

Figure 4.18 Measuring stem height and width

Developing allometric equations specific to the species and location being investigated is the most accurate way to estimate the grass carbon component. To do this, cut at least 50 individual stems over the range of observable heights for each species. Measure each individual stem for height of the green portion.

In the laboratory, determine plant biomass by oven-drying the stems to a constant weight (approximately 72 h at 60 ºC, but the time will vary depending on the size of the stem). For each species, plot the results on a scatter plot representing plant biomass (y-axis) against stem green height (x-axis) (**Fig. 4.19**). Develop an allometric equation (using regression analysis) representing the relationship between plant biomass and stem height. Apply this equation to the average height of the plants sampled in the quadrat for an estimate of total mass per species within the 30 x 30 cm area.

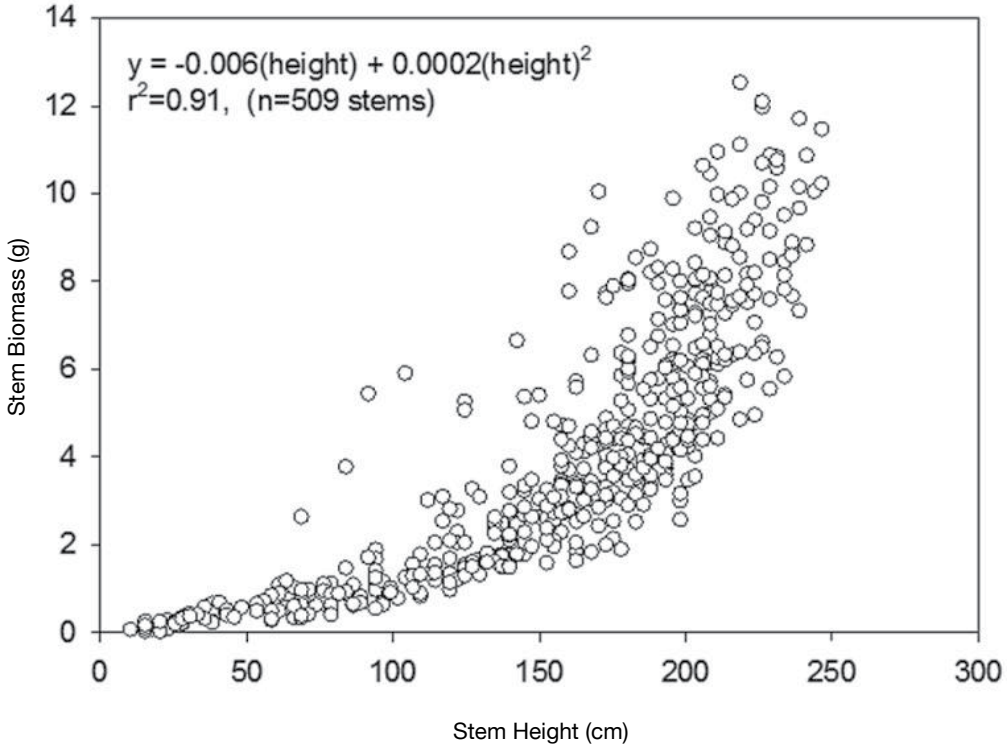

Figure 4.19 Allometric equations between stem biomass and height (Craft 2013)

Determining the carbon pool in tidal salt marsh grasses (kg C/m²): The grass carbon component is determined for each individual species by multiplying the biomass (kg)—as determined by an allometric equation—by a carbon conversion factor for the specific grass species. This is done for every stem in the quadrat. Next, all the values for individual stem carbon content are added together to determine the total carbon content from the grass (kg C) for the given quadrat size (m^2).

The carbon conversion factor for grasses can either be determined through laboratory analysis using an elemental analyzer (Chapter 3) or a conversion factor of 0.45 (Fang *et al*. 1996).

EXAMPLE

- Carbon in the grass component (kg C/m^2) = (Estimated biomass of the grass (kg) * carbon conversion factor (0.45)) / area of the quadrat (m^2)

SHRUBS

The presence of shrubs typically varies depending on the location of the tidal salt marsh. In the northern hemisphere, shrubs make up a minor component of the marshes and are usually of the genus *Atriplex*, *Borricha*, and *Iva* (Pennings & Moore 2001), while in the southern hemisphere—particularly in higher latitudes—shrubs of the genus *Tecticornia* may be the dominant growth form (Saintilan *et al*. 2009). The suggested approach to estimating aboveground biomass for tidal salt marsh shrubs is similar to the protocol for scrub mangroves (see section on scrub mangroves above). Currently, very few allometric equations exist for tidal salt marsh shrubs; therefore, equations should be developed on the basis of predictors including stem diameter at 30 cm above-the-ground surface, and crown volume/area or projected cover.

Crown diameter, width, volume. and area, as well as main stem diameter at 30 cm aboveground should be measured for each shrub in the plot area. To properly measure carbon content, at least 15–25 shrubs of each species, encompassing the range of sizes encountered and located outside any permanent plots that might be established, should be sampled in their entirety. In the laboratory, individual bushes are dried and weighed to obtain biomass. Biometric measurements (crown diameter, area and volume, and main stem diameter at 30 cm) can then be applied as independent variables predicting aboveground plant mass by regression analysis (**Fig. 4.20**). Once allometric equations have been established, they can be applied to every shrub within a sample plot.

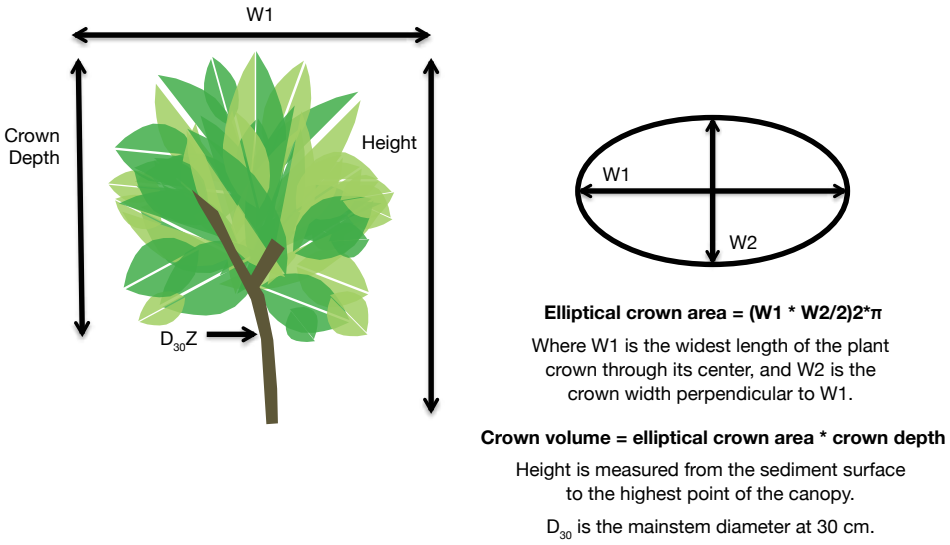

Figure 4.20 Field measurement techniques to calculate the elliptical crown area, crown depth, height, and diameter at 30 cm height (D_{30}) of shrubs (adapted from Kauffman and Donato 2012). Aboveground biomass is best predicted through allometric equations where aboveground biomass is the dependent variable and crown area, and height and/or crown volume are independent variables (Ross *et al*. 2001).

Determining the carbon pool in shrubby tidal salt marsh (kg C/m²): The shrub carbon component is determined by multiplying the biomass (kg), as determined by an allometric equation, by a carbon conversion factor for shrubs specific to that region. This is done for every shrub sampled. Next, all the values for individual shrub carbon content are added together to determine the total carbon content from the shrubs (kg C) for the given plot size (m^2).

The carbon conversion factor for shrubs can either be determined through laboratory analysis using an elemental analyzer (Chapter 3), or it is justifiable to use the conversion factor reported for scrub mangrove trees (0.46 to 0.5).

EXAMPLE

- Carbon content of each shrub (kg C) = shrub biomass (kg) * carbon conversion factor (0.46–0.5);
- Carbon in shrub component (kg C/m^2) = (carbon content of shrub #1 + carbon content of shrub #2 + ……. shrub #n) / area of the plot (m^2).

BELOWGROUND BIOMASS

Belowground root and rhizome biomass can contain 50–95% of the overall vegetative biomass in tidal salt marshes (Valiela *et al*. 1976; Elsey-Quirk *et al*. 2011). Belowground biomass can be estimated using allometric equations that relate belowground biomass to known aboveground biomass, or it can be determined on a site-by-site basis by direct measurements.

Few allometric equations exist that estimate belowground biomass on the basis of aboveground biomass, and those that do are based on studies conducted in North America (Valiela *et al*. 1976; Giroux & Bédard 1988; Darby & Turner 2008a). We recommend using the equations found in **Table 4.7** to estimate the total belowground biomass for a given plot size. These equations met with varying success in predicting *S. alterniflora* biomass at multiple sites, and further work is needed to derive allometric equations of belowground biomass for additional species and varying climatic and geomorphic settings.

Table 4.7 Equations to predict belowground root and rhizome biomass on the basis of aboveground biomass of *Spartina alterniflora* (Gross *et al*. 1991)

EQUATION	ABOVE-GROUND COMPONENTS INCLUDED (FOR AN ENTIRE PLOT)	r^2
ln (Live belowground biomass, g) = 0.718 x ln (Live Aboveground biomass, g) + 2.646	Living leaves and stems	0.86
ln (Live Belowground biomass, g) = 0.700 x ln (Live Aboveground biomass, g) + 3.051	Living leaves only	0.85
ln (Live Belowground biomass, g) = 0.713 x ln (Total Aboveground biomass, g) + 2.235	All live and dead aboveground	0.86

Direct sampling will be the most accurate method for determining belowground biomass. To do this, samples are collected by extracting a core (see Chapter 3 for details). Most studies have taken relatively shallow cores for the estimation of belowground root biomass. However, we recommend sampling at 1 m depth because this measure correlates with established soil sampling techniques in Chapter 3, and there is evidence that salt marsh species can tap fresh water to this depth (Arp *et al*. 1993). Common protocols recommend using cores of 10 cm diameter and sampling segments 2.5 cm wide at 2.5, 5, 7.5, 12.5, 15, 22.5, and 25cm depth, and then sampling 5 cm wide segments cut at depths 35, 45, 55, and 65 cm (Saunders *et al*. 2006). we suggest adding 75 and 95 cm. Segments are washed over a 1 mm screen and visually separated into root, rhizome, and dead litter components. Living and

dead material are separated on the basis of color and texture (Saunders *et al*. 2006). Living root and rhizome materials are then oven-dried at 60 ºC to constant dry weight. Belowground biomass is determined as follows:

- Belowground biomass for each sample segment (g) = dry mass of sample (g) / wet mass of sample (g);
- Belowground biomass for each sample area (entire core, g/cm^2) = (Biomass of segment #1 + biomass of segment #2 + …….. biomass of segment #n) / area sampled (based on coring diameter, cm^2);
- Average belowground biomass (g/cm^2) = (Biomass of sample #1 + biomass of sample #2 + …….. biomass of sample #n) / n.

Determining the carbon within the belowground biomass (kg/m^2): The belowground carbon component is determined by multiplying the biomass (kg)—either calculated from cores or from allometric equations—by a carbon conversion factor for grass species specific to that region.

If the biomass used was determined for the entire plot using allometric equations, simply multiply that number by the carbon conversion factor to determine the total belowground carbon component for your plot.

If the biomass used was determined through core sampling, the average belowground biomass for the sample area is multiplied by the carbon conversion factor and then scaled up to the plot size.

The carbon conversion factor for belowground biomass of tidal salt marsh grasses can be determined either through laboratory analysis using an elemental analyzer (Chapter 3) or by a conversion factor of 0.34 for belowground biomass of seagrasses (Duarte 1990).

EXAMPLE (BIOMASS DETERMINED BY ALLOMETRIC EQUATIONS)

- Carbon in the belowground component (kg C/m^2) = (Estimated biomass of the belowground component (kg) * carbon conversion factor (0.34)) / area of the plot (m^2)

EXAMPLE (BIOMASS DETERMINED BY SAMPLING)

- Belowground carbon component = Average below ground biomass (g/cm^2) * carbon conversion factor (0.34)
- Because the numbers here represent the below ground carbon component found in the sample area, you can simply scale this up to your plot size

LITTER

Leaf litter is defined as recently fallen, non-wood, dead organic material on the soil surface. It may include tidal salt marsh vegetation, seagrass, or marine algal wrack. In most tidal salt marshes, the biomass of this component is relatively small due to rapid decomposition and removal by herbivores or the tide.

Litter in most studies is destructively sampled (see the section on mangrove litter above for more details). All litter easily removed from the surface is collected from a quadrat (e.g., 50 x 50 cm),

taken back to the laboratory in a sturdy bag or container, where the sample, or a representative sample thereof, is dried to a constant weight to determine biomass (kg) and converted to carbon using an estimated carbon conversion factor of 0.45.

EXAMPLE

- Carbon in the leaf litter component (kg C/m^2) = (Average biomass of the litter * carbon conversion factor (0.45)) / area of the plot (m^2)

Figure 4.21 Tidal salt marsh leaf litter. (© Neil Saintilan, OEH NSW)

DEAD AND DOWNED WOOD

Dead and downed wood is rarely a significant component of tidal salt marsh aboveground biomass, but it may be incorporated into a tidal salt marsh carbon stock as allochthonous material. Where this occurs in a plot, the dead wood should be sampled using the line intersect technique. Dead and downed wood biomass (kg/ha) is calculated as the volume multiplied by its mean wood density. Finally, convert downed wood biomass to carbon mass. Acceptable default values based upon carbon content (wt %) of dead wood in tropical forests are 50%, corresponding to a carbon conversion factor of 0.50. See the section above for methods of measuring dead/downed wood in mangroves.

SEAGRASS MEADOWS

Seagrass meadows are coastal marine ecosystems that are dominated by flowering plants and are completely submerged or in intertidal flood areas. There are about 60 seagrass species in four families (**Fig. 4.22**), and they are generally restricted to places with unconsolidated sediments and high light availability. Unlike mangroves, which are generally restricted to the tropics/subtropics, and tidal salt marshes, which are much more common in temperate climates, seagrass meadows are distributed globally, occurring on every continent with the

exception of Antarctica (Green & Short 2003). Seagrass meadows are valued as habitats for numerous economically and ecologically important species as well as for their high rates of primary production, enhancement of fisheries, nutrient cycling, sediment stabilization, attenuation of storm waves (Orth *et al*. 2006), and importance in the carbon cycle (Duarte *et al*. 2005; Duarte *et al*. 2010; Kennedy *et al*. 2010; Fourqurean *et al*. 2012a).

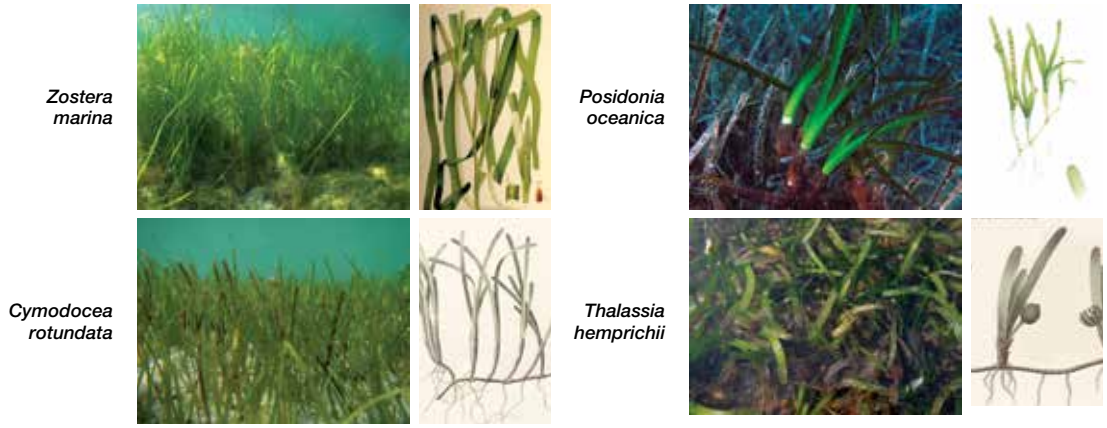

Figure 4.22 Examples of common seagrass species (© Peter Macreadie, UTS and wikicommons)

By reducing the stresses exerted by waves and currents on the soils, seagrasses reduce the amount of sediment re-suspension and enhance the trapping of suspended particles (Hendriks *et al*. 2008). They not only trap and store organic carbon generated by plants and other sources within the seagrass meadow, but also trap and bury carbon originating elsewhere (allochthonous carbon). Typically, 50% of the sediment carbon in seagrass meadows is allochthonous (Kennedy *et al*. 2010). Not all of the net production within the seagrass meadows is retained; part of seagrass ecosystem-derived carbon is also exported to other coastal and marine ecosystems where it may be consumed or buried (Heck Jr *et al*. 2008).

Saegrass meadows are also among the world's most threatened ecosystems. Rates of loss of known seagrass habitat are accelerating from a median of 0.9% per year before 1940 to 7% per year since 1990 (Waycott *et al*. 2009). Globally, the main reasons for seagrass loss are water quality degradation (e.g., eutrophication) and dredging and filling operations.

The soils that underlie seagrass meadows contain more organic carbon than terrestrial forest ecosystems (Fourqurean *et al*. 2012a). Thus the potential impact of loss of seagrass ecosystems to global CO_2 budgets is significant. In fact, the amount of organic carbon stored in seagrass meadows is roughly equal to the total amount of organic carbon stored in the world's marine tidal salt marshes and mangrove forests combined (Chmura *et al*. 2003; Donato *et al*. 2011). These estimates make seagrass meadows global hotspots for carbon storage.

Sufficient data are not available to definitively comment on regional patterns in carbon storage in seagrass ecosystems, but the data currently available are summarized by region in **Table 4.8**.

Table 4.8 Carbon stocks for a range of seagrass beds (Fourqurean *et al*. 2012b) .

REGION	LIVING SEAGRASS BIOMASS (MgC/ha)		SOIL ORGANIC CARBON (MgC/ha)	
	n	Mean ± 95% CI	n	Mean ± 95% CI
Northeast Pacific	5	0.97 ± 1.02	1	64.4
Southeast Pacific	0	ND	0	ND
North Atlantic	50	0.85 ± 0.19	24	48.7 ± 14.5
Tropical Western Atlantic	44	0.84 ± 0.17	13	150.9 ± 26.3
Mediterranean	57	7.29 ± 1.52	29	372.4 ± 56.8
South Atlantic	5	1.06 ± 0.51	5	137.0 ± 56.8
Indopacific	47	0.61 ± 0.26	8	23.6 ± 8.3
Western Pacific	0	ND	0	ND
South Australia	40	2.32 ± 0.63	9	268.3 ± 101.7
*ND = no data				

Field Sampling Considerations

Seagrass biomass varies seasonally, particularly in temperate and boreal climates. In some locations seagrass aboveground vegetation may be lost during winter. Therefore, we recommended measuring biomass when standing stocks are greatest and repeated sampling in subsequent years occurs at the same time of year. Seagrass can be submerged or intertidal. Sampling of intertidal seagrass can be done at low tide (as little as 3 to 4 hours), but sampling of sub-tidal seagrass meadows must be done using snorkeling or SCUBA equipment. Seagrass plants are herbaceous and relatively small; thus, small plots (0.25–1 m^2) are appropriate to obtain estimates of carbon stocks. Seagrass beds vary in structure along a depth gradient due to hydrodynamic energy, reduced irradiance, and other environmental gradients such as salinity, which may affect the species composition and their ability to capture carbon (Serrano et al., 2014). Thus, stratification of the study area typically aligns with depth. For example, strata can be based on intervals of known depth to obtain a reliable representation of biomass variation with water depth. At each depth interval, determine the biomass by assessing it at random positions (Chapter 2) (Short & Coles 2001).

Biomass Estimates and Carbon Content

This section provides guidance on how to measure aboveground biomass across a range of vegetation types likely to be encountered in the field. Some differences in sampling procedures are required to accommodate differences in growth forms.

LIVING BIOMASS

In most seagrass systems, the living aboveground and belowground component of biomass can be collected by inserting a large-diameter core tube (10–25 cm diameter) into the sediment through the aboveground plant material, with care not to cut the leaves, and into

the upper root dominated soils (also called the rhizosphere, typically 40 cm in depth). The tube is then capped and removed (**Fig. 4.23**). These cores are transferred to screens or mesh bags, washed free of sediment, and separated into living aboveground and belowground components (**Fig. 4.24**).

Figure 4.23 Biomass sampling/coring in seagrass meadows in Ceara, Brazil (© Kcrishna Barros, UFC)

Figure 4.24 Above and belowground biomass for *Ruppia maritime* (© Margareth Copertino, FURG)

Separation of living root and rhizome material from dead structures is often ambiguous. Some of the living belowground tissues are obvious as they are typically light in color and turgid, but it can be difficult to tell if some of the older, darker-colored structures are living or dead. Local knowledge and experience are often required to ensure consistent separation of the living material from the dead. For most seagrass species, the aboveground portion of the plants are all green leaves and are easy to separate from non-green, belowground tissues. Note that in many locations, these green leaves support a considerable epiphyte load that must be removed and analyzed separately (see next section).

Determining the Carbon within the Living Vegetation Component (kg C/m^2): In the laboratory, determine plant biomass by oven-drying the living biomass to a constant weight (approximately 72 h at 60 ºC). The living vegetative component is determined by multiplying the biomass (kg) of a sample of plant material for a given area (m^2) by a carbon conversion factor. Carbon contents are then scaled up to determine the carbon pool for the given plot size. To determine the carbon content, samples can be analyzed indirectly by determination of organic matter by loss on ignition (LOI) and use of allometric equation to estimate organic carbon or directly using an elemental analyzer (with inorganic carbon having been previously removed or accounted for by methods in Chapter 3. If this is not practical, a value of 0.34 can be used (Duarte 1990).

EXAMPLE

- Carbon in the living biomass component (kg C/m^2) = (Estimated biomass of the plant * carbon conversion factor (0.34)) / area of the plot (m^2)

EPIPHYTE BIOMASS

Seagrass epiphytes are organisms that grow on the blades of seagrasses, including algae, diatoms, and other encrusting organisms (**Fig. 4.25**). Even though epiphytes are generally a minor component of the organic carbon in a seagrass ecosystem, it is common practice to remove the epiphytes from the seagrass blades and analyze them as a separate carbon pool. Careful records must be kept regarding whether epiphytes were separated from leaf material or not to make comparison across sites possible.

Figure 4.25 Epiphytes. (A) example of coralline algae, (B) Example of filamentous algae (© Justin Campbell, Smithsonian Institute)

There are two main techniques used for removing epiphytes from seagrass leaves: manual scraping and acid-washing (**Fig. 4.26**). Neither method is perfect. Scraping well-adhered epiphyte material can either result in incomplete removal of the epiphytes or it can abrade the surface of the leaves and contaminate the epiphyte component with seagrass material. Acid-washing seagrasses efficiently removes carbonate epiphytes, but fleshy epiphytes may still require scraping and the acid can leach soluble organic compounds from both the seagrass tissue and the epiphytes. If the aim is to quantify the epiphyte pool, then the scraping method is recommended.

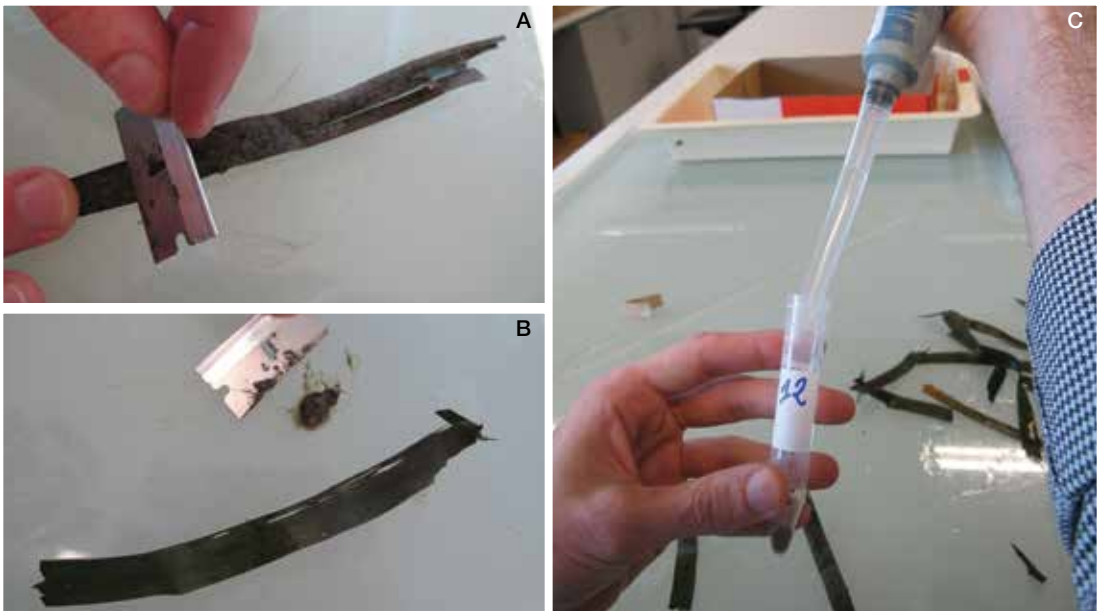

Figure 4.26 Epiphyte removal and processing. (A) Removal of epiphytes from both sides of the seagrass leaf using a blade, (B) Epiphytes (algae) and a clean seagrass sample, and (C) Dried epiphytes undergoing an acid wash (© Oscar Serrano, ECU)

Determining the Carbon within the Epiphyte Component (kg C/m²): Epiphyte removal is conducted at the laboratory. Therefore, if using a permanent plot system the samples must be taken from an area of known size outside of the plot. Some types of epiphytes will contain high levels of calcium carbonate; thus, determining the inorganic carbon in the epiphyte component is needed. Procedures are similar as those described for soils with high levels of carbonate described in detail in Chapter 3 and briefly below.

1) Epiphytes are removed from both sides of the leaf by scrapping with straight edge blade (**Fig. 4.26**).

2) Epiphytes dried at 60 ºC to a constant weight.

3) When dry, homogenise the sample and determine the weight of sample.

4) Inorganic carbon is determined in one of two methods:
   a. Acidification:
      - Soaking the sample in 1N HCL for approximately 18 hours, followed by washing the sample three times in distilled water; dry to constant weight, and analyze carbon content using an elemental analyzer or LOI Test.
      - Note that acidification can leach organic matter.
   b. Elemental Analyzer:
      - Determine the total carbon content of a dry subsample via an elemental analyzer; ash a second subsample at 500 ºC; weigh the remaining ash; determine the inorganic carbon content using an elemental analyzer; and subtract the inorganic content from the total carbon content.

Always keep track of sample masses at all stages of the procedure so you can calculate the % inorganic and organic carbon. The carbon content is corrected for the loss of inorganic carbon to produce a carbon conversion factor. The epiphyte sample biomass is multiplied by the corrected carbon conversion factor and then averaged across all samples to determine the carbon pool for the given plot size (m^2).

LITTER

The litter in seagrass meadows typically consists of dead leaves, rhizomes, fruits, and algae. In most seagrass settings, the amount of litter (and therefore the carbon stock) only constitutes a very small component of the carbon pool due to the high remineralization efficiency of leaves as well as export through hydrodynamic energy.

If it is measured, litter is destructively sampled through collection from microplots (e.g., 0.5 x 0.5 m in size). All organic surface material is collected into a bag or container. The bags are labelled with the location, date, plot, and sample number.

The samples need to be transported to the laboratory and dried to constant mass (72 h at 60 ºC). Due to constraints of carrying bulky samples and the often-limited oven drying space—especially in rural field settings—we recommend measuring the wet weight of the entire sample and then extracting a well-mixed representative subsample for transportation to a laboratory, and determining the ratio between wet and dry mass of the subsample for scaling-up the biomass of the bulk sample.

EXAMPLE

- Biomass of litter (kg) = (dry mass of subsample (g) / wet mass of the subsample (g)) * wet mass of all the litter in the sample plot (kg)

The carbon content in the litter biomass (kg) can then be estimated for the given plot size (m^2). The conversion factor for living biomass (0.34) can also be used to estimate the mean carbon content of seagrass litter dry mass.

EXAMPLE

- Carbon in the litter component (kg C/m^2) = (Average biomass of the litter * carbon conversion factor (0.34)) / area of the plot (m^2)

TOTAL CARBON STOCK

The total vegetative carbon stock within a project area is determined by adding the carbon content of each vegetative component (tree, litter, grass, shrub, etc.) within a defined area. The total carbon stock in a project area can be determined as follows:

Step 1: Determine the carbon content of each component for your plot size (see above).

Step 2: Convert the carbon content of each component for your plot size into the units commonly used in carbon stock assessment (Mg C/ha) using the following unit conversion factors (There are 1000 kg per Mg and 10 000 m^2 per ha)):

Vegetative component carbon pool (Mg C/ha) = Carbon content (kg C/m^2) * (Mg/1000 kg) * (10 000 m^2/ha).

REPEAT FOR EACH VEGETATIVE TYPE MEASURED

Step 3: Sum all the vegetative components present within a single plot.

Total vegetative carbon in a plot (Mg C/ha) = component #1 (Mg C/ha) + component #2 (Mg C/ha) + component #3 (Mg C/ha) + …

REPEAT FOR ALL PLOTS

Step 4: Determine the average amount of vegetative carbon in a plot and calculate the associated standard deviation to determine variability/error.

Average vegetative carbon in a plot = Total vegetative carbon for plot #1 (Mg C/ha) + Total vegetative carbon for plot #2 (Mg C/ha) + Total vegetative carbon for plot #3 (Mg C/ha) +…. n) / n

Standard Deviation (σ) determines how closely the data are clustered about the mean, and is calculated as follows:

$$\text{Standard Deviation} \quad (\sigma) = \left[\frac{(X_1 - \overline{X})^2 + (X_2 - \overline{X})^2 + \ldots (X_n - \overline{X})^2}{(N-1)} \right]^{1/2}$$

- $\overline{X}$ = average vegetative carbon in a plot
- X_1 = individual result for plot #1, in MgC/hectare; X_2 = individual result for plot #2, in MgC/hectare, etc.,
- N = number of plots.

Step 5: Obtain the total amount of carbon in the ecosystem.

Estimate of vegetative carbon in a stratum (Mg C) = Average vegetative carbon in a plot (Mg C/ha) * area of stratum (ha)

Sum the MgC values for each stratum to determine the total soil carbon stock.

Total carbon in a project area (MgC) = Estimate of vegetative carbon in stratum #1 (Mg C) + Estimate of vegetative carbon in stratum #2 (Mg C) + Estimate of vegetative carbon in stratum #3 (Mg C) + …

Step 6: To report a value for the variability/error associated with these measurements, calculate the total uncertainty in the data. First, calculate the standard deviation of the average Mg C for each stratum. [Multiply the standard deviation carbon value (MgC/hectare) for each core determined in step 5 above by the area of each stratum (in hectares).] Then propagate the uncertainty through the calculations by combining the standard deviations of the average MgC for each stratum as follows:

$$(\sigma_T) = \sqrt{(\sigma_A)^2 + (\sigma_B)^2 + \ldots (\sigma_N)^2}$$

- Where σ_T = the total variability associated with the measurements
- σ_A = standard deviation of the core average MgC for stratum A * area of stratum
- σ_B = standard deviation of the core average MgC for stratum B * area of stratum
- σ_N = standard deviation of the core average MgC for remaining stratum * area of each individual stratum.

This approach can be used when adding average values, as is done when combining the data from the individual strata.

Step 7: The final vegetative carbon stock will be presented in an average value ± the total uncertainty. Alternatively, a minimum and maximum carbon stock can be used by multiplying by the project area by the minimum and maximum carbon densities.

Total organic carbon in a project area (calculated in Step 5) ± the standard deviation (calculated in Step 6)

QUICK GUIDE

Step 1: Plot design

- Accessibility and safety are the number one priorities when choosing plot areas.
  - Remember each site may only be accessible for a few hours.
- Stratify the study site.
- Decide if the sampling plots will be permanent or temporary.
  - Permanent plots are used if the same location will be assessed in the future to determine change.
  - Temporary plots are used when sampling will only be done once or when permanent plots are not feasible.

Step 2: Measure vegetative components (above- and belowground as well as living and dead components)

- Determine the relevant components for the study site and the scale of the sampling area needed for each.
  - Mangrove forests
    - Live trees
    - Scrub mangrove trees
    - Standing dead trees
    - Lianas
    - Palms
    - Pneumatophores
    - Leaf litter
    - Dead and downed wood
    - Belowground tree biomass (if an allometric equation is available)
  - Tidal Marshes
    - Shrubs
    - Grasses and reeds
    - Leaf litter
    - Dead and downed wood
  - Seagrass meadows
    - Living plants
    - Epiphytes
    - Leaf litter
- Determine biomass either by laboratory analysis or by allometric equations.
- Determine the carbon content for a known plot size.

Step 3: Calculate Total Vegetative Carbon Stock

- Sum the relevant component carbon pools for each plot.
- Determine the average and standard deviation for plots within a stratum and multiply by the stratum area. Repeat for each stratum.
- Determine total vegetative carbon stock for the project area by summing carbon stocks within each stratum and calculating the error.
- You must include the variability associated with the measurements and the total soil depth assessed when reporting results.

4

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How to Estimate Carbon Dioxide Emissions

INTRODUCTION

Understanding how blue carbon ecosystems contribute to climate change mitigation requires an understanding of ecosystem extent (Chapter 2), the amount of carbon present in each pool (Chapters 3 and 4), and the how much carbon is emitted or sequestered over time (presented here). The amount of carbon emitted to the atmosphere or sequestered can be determined directly using methods that detect gas exchange, or more commonly using carbon stock change as a proxy for gas exchange. However, other greenhouse gases (GHGs) such as methane (CH_4) and nitrous oxide (N_2O) can be measured only by using gas exchange methods. Assessing changes in blue carbon stocks and GHG emissions can be used to establish baselines for tracking sequestration rates and emission trends, identifying activities that increase emissions, assessing progress of restoration efforts, communicating with stakeholders, and more. Together, this information enhances countries' and institutions' ability to meet current and future reporting needs, participate in carbon-based markets, and inform mitigation policy and management practices at local to national scales.

APPROACHES TO MEASURING EMISSIONS

To determining changes in carbon stocks (as a proxy for CO_2 emissions), two approaches can be used:

1) Stock-difference method: This method estimates the difference in carbon stocks measured at two points in time and results in IPCC Tier 3 estimates (Kauffman *et al*. 2014).

2) Gain-loss method: This method estimates the difference in carbon stocks based on emissions factors for specific activities (e.g., plantings, drainage, rewetting, deforestation) derived from the scientific literature and country activity data and results in Tier 1 and 2 estimates (GOFC-GOLD 2009; IPCC 2013).

In addition, a third method exists, for the direct measurement of GHG emissions:

3) Flux method: This method estimates the GHG flux between the soil and vegetation and the atmosphere/water column through direct measurements or by modeling results in Tier 2 and 3 estimates.

Often, a mix of methods is used, but ultimately the method selected will be based on the needs of the project, the resources available, and the level of accuracy required.

Stock-difference Method

The stock-difference method requires carbon measurements for each significant carbon pool at least two points in time (T1 = initial assessment and T2 = subsequent assessment). Results from each inventory are then compared and analyzed for changes. This approach not only tracks changes to carbon stocks as a whole, but also changes in the individual carbon pools. For example, if the living biomass pool decreases but the dead biomass pool increases, it can be assumed that the ecosystem was degraded between the two time points. However, this technique alone cannot determine the cause of the degradation (natural or human caused).

INITIAL ASSESSMENT (T1)

The most accurate method is to establish permanent plots at the time of the first inventory so that subsequent assessments are done at the exact same location, for the same size plot, with the same protocols. However, if establishing permanent plots is not feasible, future measurements can be made from a similar location with higher uncertainty. If not using permanent plots, it is important to ensure that the area of land being assessed at T1 and T2 are identical to avoid confounding stock change estimates with area changes (IPCC 2013).

Carbon pools are measured according to the methods laid out in Chapters 2–4. The initial assessment will be used as a reference point (datum) for future change. This is particularly important for soils in blue carbon ecosystems because much of the stock change occurs when the soil accretes (gains volume, such as through elevation gain) or erodes. Examples of possible reference points for a change in soil volume include depth to the organic soil base or depth to an underlying substrate (i.e., bedrock) (**Fig. 5.1**). A constant soil volume reference datum, such as the underlying substrate, is useful because any change could only have come from soil accretion or erosion since the substrate is constant (baseline scenario). Other reference points, such as carbon pool measurements, will be based on the results of the initial assessment (project scenario). A separate datum must be established for each parameter being tracked.

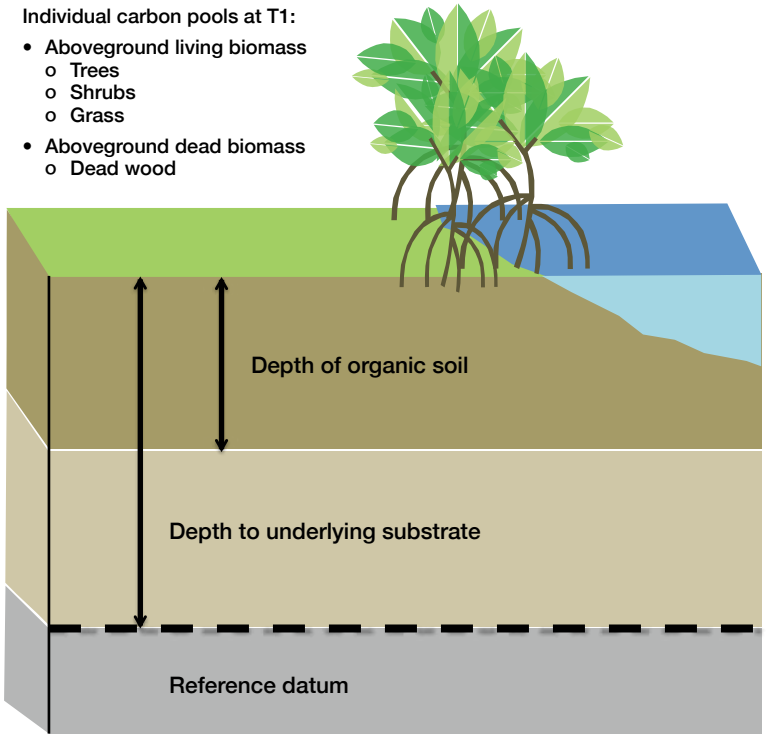


Figure 5.1 Establishing a soil volume reference datum

TIME BETWEEN ASSESSMENTS

The time recommended between measurements depends on the project site, intensity of activities, and the time needed to see a measurable change. If the site is pristine, and no land use change has been observed, the timing between measurements should relate to the time required to see natural changes in the carbon stocks. For example, seasonal changes

in aboveground biomass associated with loss of leaves in the cooler months is balanced out by regrowth in the spring, so the size of that biomass carbon pool is relatively stable. Thus, annual measurements are usually not needed; instead, measurements every 5–10 years might be more valuable. Similarly, soils accrete very slowly (rate of millimeters per year), and the carbon found in the deeper sediments (beneath the top meter) is relatively stable and not likely to change if the ecosystem is not altered. Therefore, adequate soil carbon measurements may also be taken at 5–10 years or longer (10– 20 years). However, if the project area has undergone a land use change, or if land-use changes nearby are impacting the site, measurements should be taken more regularly to record any significant impacts.

SUBSEQUENT ASSESSMENTS (T2)

Return to the same location analyzed at T1, or an alternative location that is representative of the size and current species density of the original location, and repeat the blue carbon inventory. Vegetative carbon pools can be measured according to the steps outlined in Chapter 4. The carbon inventory from T1 can be used as a reference to determine changes in the vegetative carbon pools.

Measuring the soil carbon pool is more challenging due to soil accretion/erosion that impact soil volume or carbon density. For example, if significant soil accretion/erosion has occurred at the study site, a core taken from 0–100 cm below the soil surface at T1 will not cover the same absolute depth range as a core taken at 0–100 cm from the soil surface at T2 (**Fig 5.2**). In order to correct for this, the the change in soil volume between T1 and T2 must be determined by establishing a reference datum as described above. The concept is that a reference datum is a horizontal line below the influence of plant activity (i.e., all carbon added by root growth or surface deposition occurs above the reference). One approach for establishing a reference datum is to locate a feature in the soil profile that can be clearly identified over time, such as bedrock or a sharp transition from organic-rich to organic-poor soil (**Fig 5.1**). For example, Kauffman *et al*. 2014 used the boundary between organic soils and marine sands as the reference datum in Honduras. Some commonly used methods for establishing a reference datum do not capture the full soil profile, leaving open the possibility that soil volume below the feature changed. For example, the position of the soil surface in 1963 can be established by locating a peak in cesium-137. Soil Elevation Tables (SETs) are an expensive but accurate method for assessing change soil volume change across the full soil profile in cases where there is no clear boundary.

To obtain high-resolution measures of wetland soil elevation, a portable, mechanical device called a Surface Elevation Table (SET) can be used (**Fig 5.3**). The SET device has an accuracy of 1.5 mm and can be used to determine elevation trends in any wetland setting (Cahoon *et al*. 2002). It is more accurate to measure soil elevation seasonally every year between T1 and T2 to establish trends and determine an average gain/loss per year. However, if it is not possible to routinely make measurements, soil elevation can be determined from a different but similar location where routine sampling is easier, but the uncertainty will increase. Rates of soil elevation gains/losses over time may also be available from government agencies or researchers.

A detailed explanation of the SET marker horizon (MH) methodology, a list of the scientists using it, and the locations where it is being used can be found on the US Geological Survey (USGS) web site: www.pwrc.usgs.gov/set. Briefly, the SET-MH methodology comprises a

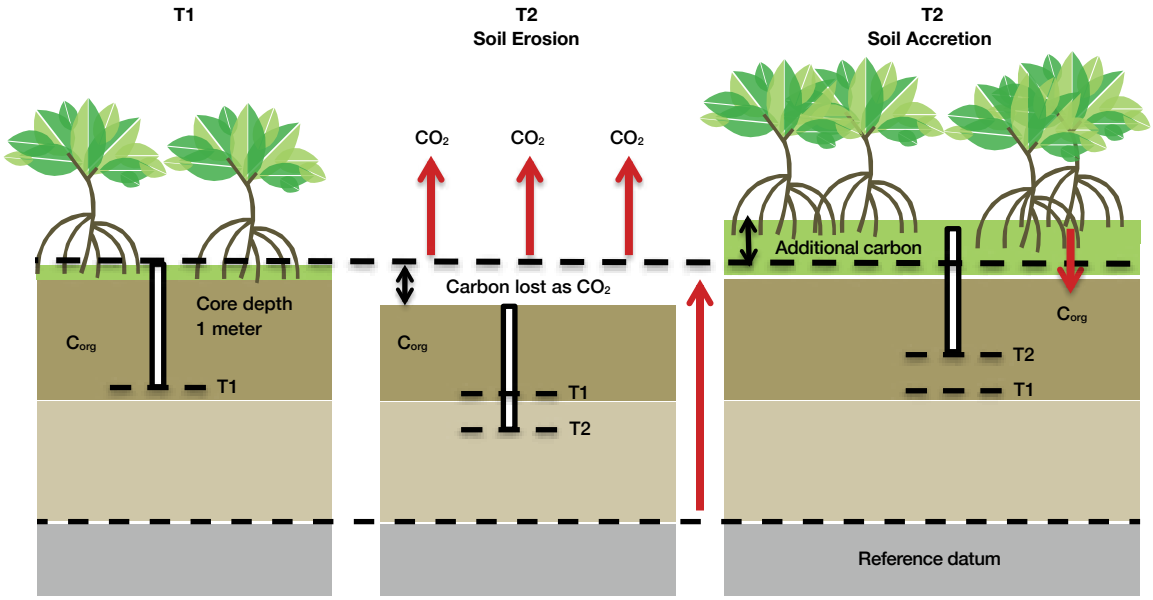


Figure 5.2 Effect of accretion and erosion on soil samples. A reference datum is usually given the designation "0," and future measurements should refer to it. For example, soil accretion of 3 cm at T2 would be reported as "+3 cm relative to the datum" where the initial datum would also be reported. Note that the "0" reference datum is this example is established by measuring the distance to a second, primary reference datum below the soil surface. The primary reference may be bedrock or a distinct soil layer such as clay (**Fig. 5.1**), or the bottom of an SET benchmark (**Fig. 5.3**).

platform that attaches to a benchmark (pole driven permanently into the ground that does not move and remains from T1 to T2). The platform is suspended above the ground, and rods of known length are fed through openings in the platform and rested on the surface of the soil (**Fig 5.3**). The height from the top of the platform to the top of the rod is measured. The

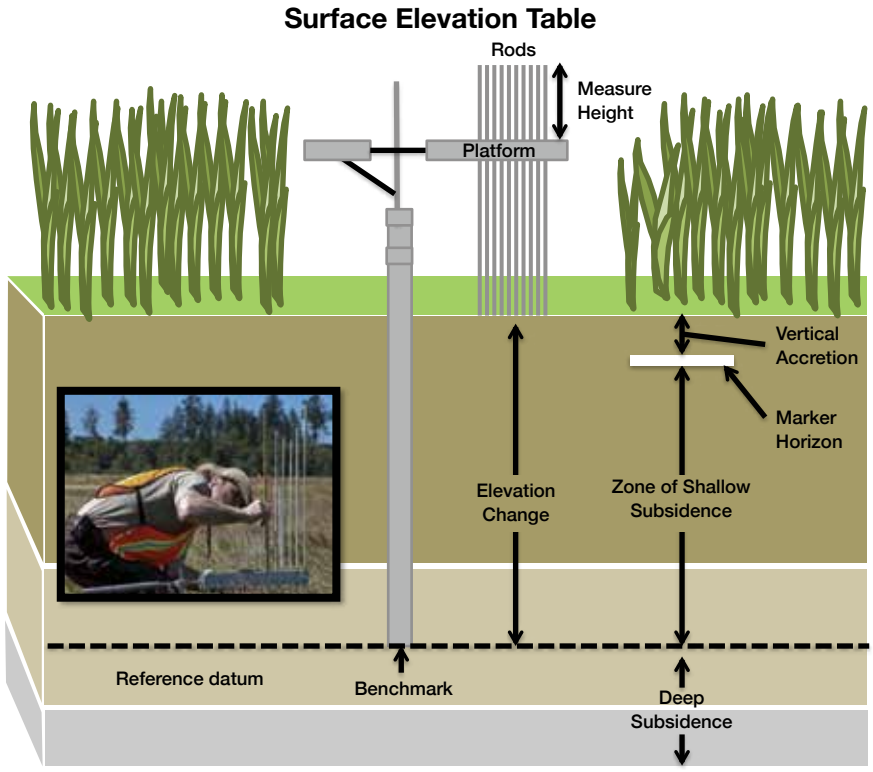


Figure 5.3 Diagram of a Surface Elevation Table (SET) marker horizon (MH) method used to detect changes in soil surface elevation. Inset image: USFWS biologist Bill Bridgeland taking measurements on a SET (© Roy Lowe, U.S. Fish and Wildlife Service).

height of the rod changes as the soil surface changes, representing change in soil surface elevation, which corresponds to a change in soil volume. Change in soil volume is the only parameter required for calculating stock change. However, the associated marker horizon can give additional insights that are interesting for research purposes, namely a calculation of shallow subsidence (e.g., due to root decomposition) (Cahoon *et al*. 2002).

EXAMPLE

- Rod height at T1 = 100.46 cm
- Rod height at T2 = 100.98 cm
  - Elevation change = 100.98 cm – 100.46 cm = 0.52 cm = 5.2 mm (common unit used for describing elevation changes in soil and sea level)
- Depth of the marker horizon at T1 = 0
  - The marker horizon is established during the first assessment
- Depth of the marker horizon at T2 = 8.6 mm
  - Vertical accretion = 8.6 mm – 0 mm = 8.6 mm
- Shallow subsidence = Vertical accretion – elevation change
  - 8.6 mm – 5.2 mm = 3.4 mm

In the case of soil accretion, the top sub-sample is used to determine the change in the soil carbon due to change in the soil volume (Lovelock *et al*. 2014).

EXAMPLE

- T2 soil core top 10 cm = 0.195 g/cm^3
- Soil elevation rate (determined by SET-MH measurements) = 0.52 cm per year
- Time between T1 and T2 = 10 years
- Total soil accretion occurring in that time = 5.2 cm
- 5.2 cm * 0.195 g/cm^3 = 1.014 g/cm^2 additional soil carbon due to accretion (in a single core)

This number can then be extrapolated to the hectare scale:

- Total carbon content in the core due to accretion = 1.014 g/cm^2
- 1 Mg = 1 000 000 g
- 1 hectare = 100 000 000 cm^2
- 1.014 g/cm^2 * (Mg/1 000 000 g)*(100 000 000 cm^2/ha) = 101.4 Mg C/ha
- **101.4 Mg C/ha was added to the system**

If the area has undergone erosion, a one meter soil core at T2 will go deeper than the sample taken at T1, and the new surface that was once buried is now exposed to the air/water. In this case, a change in the soil carbon at T2 is determined by doing an assessment of carbon content for the overlapping depth (**Fig. 5.4**).

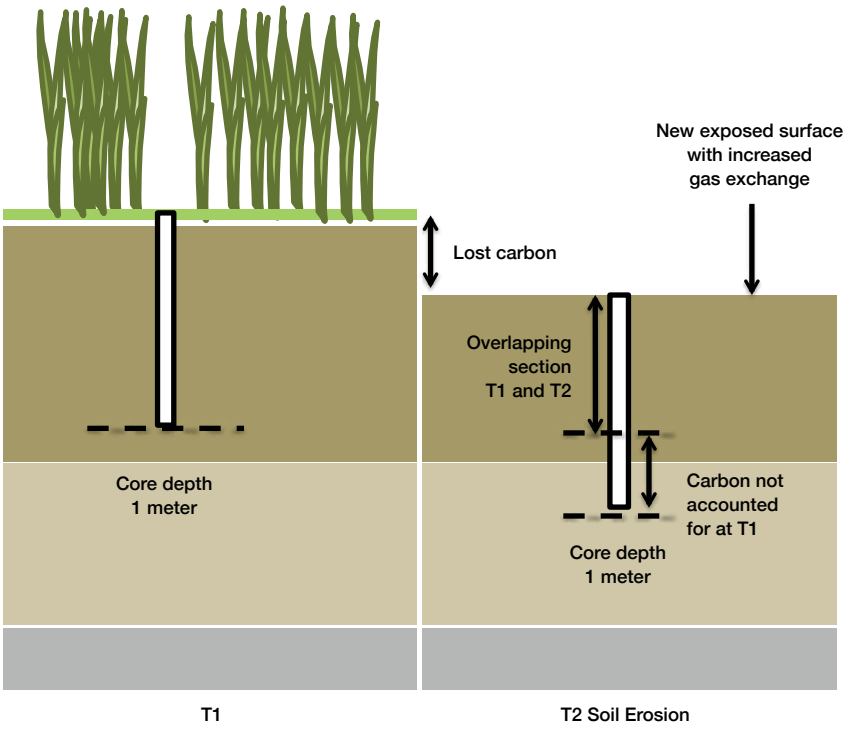


Figure 5.4 Diagram of how carbon losses may be accounted for where the soil surface has subsided or been eroded.

EXAMPLE

- Soil elevation rate (determined by SET measurements) = -0.86 cm per year
- Time between T1 and T2 = 10 years
- Total soil erosion occurring in that tim. = -8.6 cm
  - So the one-meter core at T2 went 8.6 cm deeper than a one-meter core at T1, and, thus, the bottom 8.6 cm cannot be used to determine change
- The carbon content of the remaining top 91.4 cm (as determined by methods in outlined in Chapter 3) = 69.2 g/cm^2

This number can then be extrapolated to the hectare scale:

- Total carbon content in the top 91.4 cm of the core = 69.2 g/cm^2
- 1 Mg = 1 000 000 g
- 1 hectare = 100 000 000 cm^2
- 69.2 g/cm^2 * (Mg/1 000 000 g)*(100 000 000 cm^2/ha) = 6920 Mg C/ha
- T1 soil carbon pool = 7205 Mg C/ha
- T2 soil carbon pool – T1 soil carbon pool = change in soil carbon
  - 6920 Mg C/ha – 7205 Mg C/ha = -285 Mg C/ha
- **285 Mg C/ha was lost from the system**

DETERMINING CHANGES IN CARBON STOCKS/POOLS

The change in carbon stocks for a given blue carbon ecosystem over a specific area can be determined as follows:

- Change in carbon stock (Mg C) = total carbon stock at T2 (sum of all carbon pools) – total carbon stock at initial measurement T1 (sum of all carbon pools)
- For example in a degraded salt marsh:
  - Measurements taken from a salt marsh site in 2002 (T1) estimated the total carbon stock (sum of all relevant pools) to be 34 667 Mg C.
  - Measurements taken for the same salt marsh site using the same methods in 2012 (T2) estimated the total carbon stock (sum of all relevant pools) to be 25 167 Mg C.
  - 25 133 Mg C – 34 667 Mg C = -9534 Mg C (carbon lost from the ecosystem)

If we assume all carbon lost is emitted to the atmosphere/water column (see Additional Considerations for Carbon Accounting below) then the total change in carbon can be used to estimate the annual rate of change and potential CO_2 emissions.

- Annual change in carbon stock (Mg C/year) = (total carbon stock at T2 – total carbon stock at initial measurement T1) / (T2 – T1)
- Using the example above
  - (25 133 Mg C – 34 667 Mg C) / (2012 – 2002) = -953 Mg C/year.
  - Associated CO_2 emissions = 953 Mg C * 3.67 (conversion factor) = 3498 Mg CO_2

The stock-change approach is used to make Tier 3 estimates of carbon stock/pool change. It is applicable in countries that have national inventory systems, or ongoing monitoring programs, where the stocks of different biomass pools are measured at periodic intervals (IPCC 2013). However, the stock-change approach requires measurements of each carbon pool and significant resources, making this approach not feasible in all situations.

Gain-loss Method

This method is used following an initial blue carbon inventory when subsequent inventories from direct measurements are not feasible. Instead, the gain-loss method uses activity data to account for the change in carbon stock between T1 and T2. There are numerous activities that result in carbon stock gains (transfer between pools, restoration activities, annual plant growth, soil accretion, etc.) and losses (transfer between pools, land-use changes, extractive activities, natural disturbances such as storms, etc.) in coastal marine ecosystems, the impacts of various activities are complex and as yet not well characterized. Thus, this method yields Tier 1 and 2 results. Tier 1 results are calculated based on the general conversion factors obtained from globally compiled databases (i.e., 2013 IPCC Wetlands Supplement), Tier 2 results are based on country-specific conversion factors. Both Tier 1 and Tier 2 estimates require country-specific or project-specific activity data.

If we assume all carbon lost is emitted to the atmosphere/water column (see Additional Considerations for Carbon Accounting below), using the Gain-Loss method change in carbon stocks for a given blue carbon ecosystem over a specific area can be determined as follows:

- Change in carbon stock (Mg C) = Carbon stock at T1 – (carbon losses (land use change, natural disasters, erosion, etc.) + carbon gains (soil accretion, growth, restoration, etc.))

FOR EXAMPLE

- Measurements taken from a 1000 ha salt marsh site in 2002 (T1) estimated the total carbon stock to be 34 667 Mg C.
- In 2007: 200 ha were drained resulting in an emissions rate = 7.9 Mg C/ha/yr (IPCC 2013, **Table 4.13**, p. 31)
- In 2010: 50 ha were rewetted resulting in an emissions rate = -0.91 Mg C/ha/yr (IPCC 2013, **Table 4.12**, p. 29)
- Second assessment conducted in 2012 (T2)
- Assuming all else remained the same carbon stock change can be estimated as follows:
  - (200 ha * 7.9 Mg C/ha/yr) * (2010-2007) = 4740 Mg C
  - (150 ha * 7.9 Mg C/ha/yr) * (2012-2010) = 2370 Mg C
  - (50 ha * -0.91 Mg C/ha/yr) * (2012-2010) = -91 Mg C
- Total carbon difference = 4740 Mg C + 2370 Mg C + -91 Mg C = 7019 Mg C lost
- Associated CO_2 emissions = 7019 Mg C * 3.67 (conversion factor) = 25 739 Mg CO_2

Additional Considerations for Carbon Accounting

When calculating carbon emissions, it is important to consider where any additional ecosystem carbon originated from and where any loss of carbon ends up. The calculations above apply when the difference in carbon between T2 and T1 is released to the atmosphere/water column as CO_2, providing an upper estimate of carbon emissions. However, in some cases carbon is not emitted but redeposited to adjacent habitats or the deep ocean. Corrections must be made to include carbon that is lost but not emitted. However the science to support choosing the correction factor describing the proportion of carbon emitted to the atmosphere is still developing.

Autochthonous organic carbon (carbon originating or forming in the place where it is accumulated) can be included in carbon accounting. Allochthonous organic carbon (carbon originating outside the project boundary and being deposited in the project area) can be challenging to account for at the project level because the carbon was lost from an adjacent location but not emitted. For example, in tidal wetlands erosion may reduce carbon stocks at one location, but some of this carbon may be deposited and trapped in a neighboring system. Indeed, up to 50% of seagrass soil carbon is allochthonous (Kennedy *et al*. 2010). In this case, it is incorrect to presume that the carbon lost at the first site resulted in carbon emissions.

In the case of live vegetation, any carbon additions are due to increased plant biomass from growth (autochthonous) and any loss is from degradation (e.g., deforestation or weather event); therefore, this carbon is transferred to the non-living vegetative carbon pool. Carbon in the non-living vegetation pool either gets released to the atmosphere/water column, buried in the soil as the plant material decomposes, or is removed to a neighboring ecosystem by wind/currents. Carbon lost to the atmosphere/water column can be counted as emissions, carbon buried in the soil will be accounted for during the soil analysis, and carbon that is moved to a new location can either be ignored (if < 5%) or estimated based on conservative conversion factors or modeling. Similarly, deposited allochthonous vegetative carbon (i.e., leaf litter, branches) can either be ignored (if < 5%), estimated based on conservative conversion

factors or modeling, or—if it is obvious (i.e., tree branches in low marsh area)—can be calculated as a separate carbon pool.

In the case of soil, organic carbon additions result from either increased belowground biomass or sedimentation. Where soil organic carbon accumulates on the project site due to sedimentation, a compensation factor must be applied. This factor is based on a percentage of the carbon stock that is derived from allochthonous soil organic carbon and may be derived from published values, field-collected data (e.g., stable isotopes of carbon can be used in seagrass beds to establish the origin of accreted carbon (Kennedy *et al*. 2010), proposed conservative default factors (e.g., 50% allochthonous carbon) or modeling. When soil carbon is lost due to erosion, the fate of the lost carbon (e.g., redeposited or oxidized to CO_2) determines if this loss can be accounted as CO_2 emission. Therefore, a compensation factor is needed here, too. The science on this subject is not sufficiently developed to propose a default factor, and, therefore, must be based on either field data or modeling. Studies to date have used a range e.g., 25–100% of organic carbon in the top meter of the soil as being emitted to the atmosphere (Fourqurean et al. 2012a; Pendleton *et al*. 2012; Siikamäki *et al*. 2012).

Gas Flux Techniques for Directly Assessing Carbon Flux

To understand how coastal wetlands influence climate, there is a need to understand how much GHG actually leaves coastal ecosystems and enter the atmosphere. While the methods described above can be used as a proxy to estimate the carbon emissions from coastal ecosystems, flux measurements are the only way to measure the emission of methane (CH_4) and nitrous oxide (N_2O), which do not accumulate in the system. Flux measurements can also be used to measure CO_2 emissions directly, as an alternative to the stock change methods described above.

In general, management activities resulting in extraction of soils, such as construction of aquaculture ponds, can result in large CO_2 emissions in mangroves and tidal salt marshes (IPCC 2013), N_2O emissions originate mostly from aquaculture activities and upland agricultural practices, and rewetting areas increases CH_4 from drained freshwater tidal systems. The mass of CH_4 and N_2O exchanged between wetlands and the atmosphere is smaller than the exchange of CO_2 by orders of magnitude; however, they are far more effective greenhouse gases than CO_2, with global warming potentials 25 and 298 times higher than CO_2 over a 100-year time horizon. For this reason, relatively low emissions of these gases have the potential to significantly change calculations on the climate benefits of conserving, creating, or restoring wetlands.

N_2O emissions are generally negligible unless the ecosystem has a source of nitrate loading, such as from fertilizer runoff. CH_4 production is directly related to salinity (Purvaja & Ramesh 2001; Poffenbarger *et al*. 2011). Systems with salinity levels greater than 18 ppt can assume a zero value for CH_4 emissions. For the purposes of monitoring emissions from blue carbon ecosystems, N_2O emissions will most likely be minimal to nonexistent in pristine areas but may be significant in degraded areas. Methods below use CH_4 as the example, but the principles of measurement are the same for all three gases.

Static Chamber Method

The most common technique for measuring CH_4 (or any GHG) flux is to trap the gases inside a closed chamber that is open only to the soil and plants rooted therein, then measure

the rate at which gas concentrations change over time (Megonigal & Schlesinger 2002; Lovelock *et al*. 2011; Sidik & Lovelock 2013). Flux measurements require the construction or purchase of static chambers and boardwalks to avoid disturbing the site where fluxes will be measured. Static chambers are relatively inexpensive to construct, can be designed to function with a wide range of plant types and site conditions, and can be deployed in groups to save field time (**Fig. 5.5**). However, static chambers may also change the temperature and light environments of plants, they do not capture CH_4 escaping from the soil as bubbles (as opposed to diffusion), and assumptions are needed in order to scale short-term emissions data to annual fluxes.

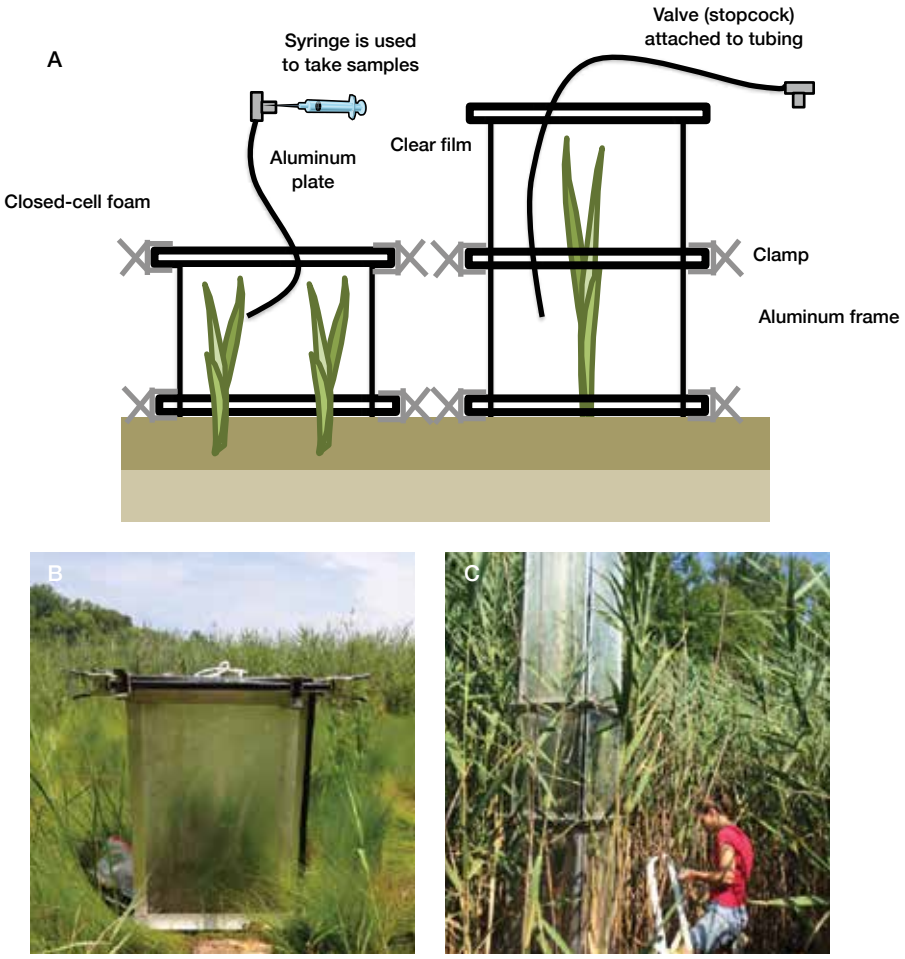


Figure 5.5 Chamber volume must be adjusted to enclose plants of different stature (B & C). (B, © Rachel Hagar, SERC; C, Eric Hazelton, SERC)

The basic design of the chamber requires a base that extends into the soil or the floodwater and a chamber that can be placed over the plants and sealed to the base. Once the chamber is in place, the headspace concentration of gases being emitted or consumed by the plant-soil system will begin to change. These changes can be quantified by taking samples from the headspace. The sensitivity of the method is determined largely by the ratio of the chamber headspace to the area of soil from which CH_4 is emitted. CH_4 emissions are often quite low from tidal wetlands (compared to CO_2), so it may be necessary to enclose fairly large areas (minimum 0.25 m^2). The measurement period can be lengthened to increase sensitivity, but longer time periods increase the risk of the chamber heating up in the sun and impacting the results.

The chambers must be constructed of gas-impermeable material. The side of the chambers can be transparent or opaque depending on the vegetation. Transparent chambers are used when enclosing the foliage of plants with pressurized gas transport systems (e.g., *Phragmites*), but are not necessary for plants that have diffusion transport (e.g., *Spartina*). Opaque chambers are easier to use because they do not require temperature regulation and can be made easily from common items like plastic trashcans. If long incubation times are needed because of low CH_4 flux rates, it may be necessary to cool the air inside transparent chambers in order to maintain temperature within about 2 ºC of ambient.

Additional issues to consider include the time of day measurements are taken and approaching the chamber properly. If CH_4 emissions are affected by light, then both light and dark emissions should be measured in order to estimate a 24-hour rate. It may be easier to measure emissions when the site is not flooded; a few studies report that flooding does not affect CH_4 emissions, but there is at least one exception (Van der Nat & Middelburg 2000). Care must be taken not to step or stand on the soil surface within 1–2 m of the chamber; the pressure may cause CH_4 bubbles trapped in the soil to escape and artificially increase emission rates. For this reason it is useful to build a simple boardwalk elevated 5–10 cm off the soil surface for approaching the chamber, and to install the chamber base several days before taking the first samples. The headspace CH_4 concentration is typically quantified by drawing a headspace sample into a gas-tight syringe, injecting it into a leaf-free vial for storage (e.g., Exetainer vial), and analyzing against standards using gas chromatography.

Water motion affects seagrass photosynthesis, gas exchange, sediment transport, and epiphyte load. Therefore, estimates of flux that constrain or alter water motion will confound the estimate of flux. Benthic chambers used in seagrass ecosystems rely on the temporal measurement of water properties, but do not replicate the physical conditions of the aquatic environment (flowing water).

CALCULATIONS

The rate of gas flux is determined by measuring changes in headspace concentration over time as determined by the slope of a best-fit line through the data (**Fig. 5.6**). To do this, samples are taken at several consecutive points in time (e.g., 2 min, 15 min, 35 min, 45 min,

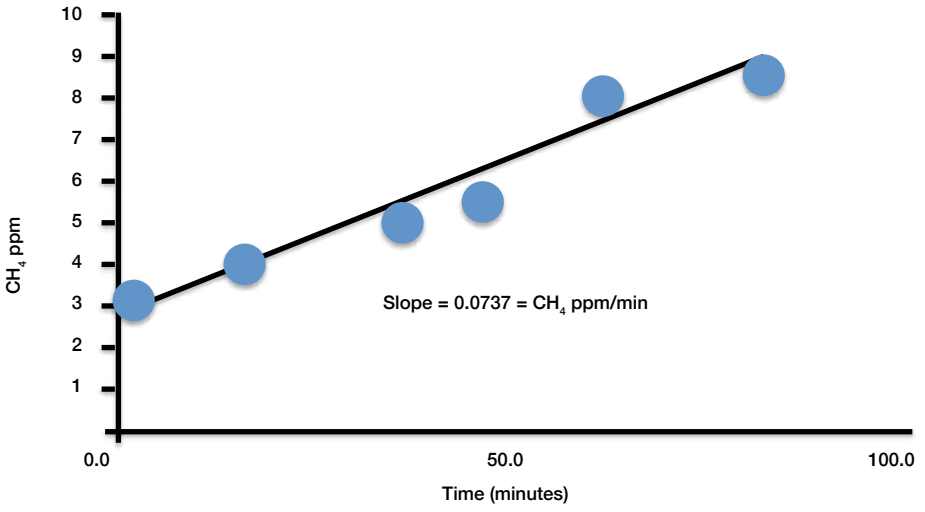


Figure 5.6 Proportion of CH_4 gas in the chamber is determined by plotting gas concentration per samples against the time between closing the chamber and collecting the sample. The slope is determined by calculating a best-fit line.

60 min, and 80 min) and the amount of CH_4 is determined for each sample. The slope is converted from concentration to a mass-based flux of CH_4 by accounting for the total mass of all gases in the chamber headspace. Mass flux can then be divided by the area of soil inside the chamber in order to express emissions on an area basis.

Measurements needed to determine CH_4 flux (**Table 5.1**):

- Atmospheric Pressure (assume this value = 1 atmosphere)
- Volume of the chamber (Liters)
- Universal gas constant = 0.0820 (L*atmos/K*mol)
- Temperature in Kelvins at the time of each measurement (K = 273 + temp in °C)

Table 5.1 Example of how to determine moles of gas molecules within a flux chamber

PRESSURE (atmos)	VOLUME OF CHAMBER (L)	GAS CONSTANT (L*atmos/ K*mol)	TEMPERATURE (Kelvins)	GAS MOLECULES IN THE CHAMBER (moles)
P	V	R	T	n = PV/RT
1	515	0.0820	288	21.8072

Run each sample through a gas chromatograph or similar instrument to determine the proportion of the total gas in the chamber that is CH_4; use the results of each sample to calculate the slope of the data (**Fig. 5.6**).

CALCULATE FLUX

- n (total amount of gas in the chamber) is in moles, but the CH_4 is measured is in parts per million (ppm) so these need to be converted to similar units
  - Amount of gas molecules in the chamber = 21.8072 moles
  - Amount of CH_4 = 0.0737 ppm/minute
  - 1 ppm = 1 µmole CH_4 per mole of gas
  - 0.0737 ppm = 0.0737 µmole/mole/minute
  - So for every mole of gas, 0.0737 µmole is CH_4
    - Thus, the total amount of CH_4 being emitted in the chamber is:
    - (0.0737 µmole/mole/minute) * 21.8072 moles = 1.6072 µmole/minute
- To determine the amount of CH_4 emitted per minute per unit area, divide by the area of the chamber (assumed to be 0.5 m^2 in this example)
  - (1.6072 µmole/minute)/0.5 m^2 = 3.2 µmole/m^2/min
- Extrapolate to Mg/ha/day
  - 1440 min = 1 day
  - 1 ha = 10 000 m^2
  - 1 mole = 1 000 000 µmole
  - 1 mole of methane = 16.042 g (according to molecular weight of methane)
  - 1 Mg = 1 000 000 g
  - 3.2 µmole/m^2/min * 10 000 m^2/ha * 1mole/1 000 000 µmole *16.042 g/mole * 1Mg/1 000 000 g = $51*10^{-8}$ Mg CH_4/ha/min
  - $51*10^{-8}$ Mg CH_4/ha/min * 1440 min/day = 7.4*10–4 Mg/ha/day

- If the project area was 1500 ha then the total CH_4 emissions for the project area is
  - 1500 ha * ($7.4*10^{-4}$ Mg/ha/day) = **1.11 Mg CH_4/ha/day**

When extrapolating these measurements to an annual scale (Mg/ha/year), assumptions must be made about the level of variation of emissions over time, or variation over time must be measured and emissions scaled up using modelling techniques. Confidence decreases when scaling up to longer time scales as emissions may vary over seasons and with short- and long-term variation in temperature, salinity, and other factors (Poffenbarger et al. 2011), among other factors.

Eddy Covariance Method

The eddy covariance (EC) method is an alternative, more sophisticated, method for directly measuring the exchange of CO_2 between ecosystems and the atmosphere. It is non-intrusive, *in situ*, and measures fluxes of the entire ecosystem. However, EC is expensive because it requires buying flux towers and paying personnel to perform complex data processing. For more information on how to set up an EC system, please see Aubinet *et al*. (2012) and Burba (2013).

Mangroves and tidal salt marshes can use standard terrestrial EC towers (Barr *et al*. 2010). However, these systems were originally designed for terrestrial forests, and the close proximity of large water bodies will impact the readings and require an additional layer of data processing to correct. Underwater EC systems used for seagrasses currently only measure O_2 because CO_2 sensors are not fast enough to measure CO_2 in turbulent water (Chipman *et al*. 2012).

When conducting flux measurements it is important to consider the form of carbon being lost. Carbon may be lost from coastal wetlands in the form of dissolved inorganic carbon, organic carbon, or particulate organic compounds that move into adjacent ecosystems (mangrove-seagrass-coastal ocean). Typically, the loss of dissolved organic compounds is only a small fraction of the of the carbon budget (Wetzel & Penhale 1979), but losses of DIC from mangroves may be large (Bouillon *et al*. 2008). Horizontal fluxes of carbon are generally missed by atmospheric measurements of flux, which supports why carbon flux between the atmosphere and the soil surface may not be equivalent to changes in the coastal wetland storage of carbon. Assessment of the magnitude of different pathways of carbon fluxes from mangroves, salt marsh and seagrass ecosystems is an active area of research that will reduce uncertainty around the carbon budgets of these ecosystems.

Remote Sensing and Mapping

INTRODUCTION

Access to coastal ecosystems such as mangroves, salt marshes, and seagrasses for field surveys can be very expensive, difficult, and/or hazardous. In addition, they do not lend themselves well to conventional manual sampling regimes, are so large they may not be able to be studied within project time constraints, or are in need of a change analysis with no previous on-site sampling having been conducted. Given these impediments, remote sensing can provide unique and valuable information on coastal vegetation structure and areal coverage that could not easily be obtained otherwise.

For the purposes of measuring blue carbon, remote sensing is vital for determining ecosystem extent, stratification and plot design, biomass measurements, and analyzing land use and carbon stock change over time for national carbon accounting. Remotely sensed measurements can be made at different spatial resolutions and, depending on the sensor, can identify various biophysical and structural characteristics of the coastal vegetation communities. Also, once in service, satellites are usually a continuous source of information for many years, providing decade long and large scale monitoring of natural and man-made changes in ecosystems.

Here we provided guidelines on the possibilities and limitations of different remote sensing approaches. This chapter is not intended to describe how to carry out remote sensing. We recommend that experts be brought into the project to aid with the actual data collection and analysis. Instead, the goal of this chapter is to provide information so that the reader is familiar enough with the procedures and options to communicate their needs more effectively to remote sensing experts.

BASICS OF REMOTE SENSING

Here we briefly describe the basic concepts of remote sensing; there are numerous books and reviews devoted to this topic that provide more detailed background information and possible applications (Green *et al*. 2000; Klemas 2010; Kuenzer *et al*. 2011; Giri 2012; Rees & Rees 2012).

Passive vs. Active Techniques

Remote sensing systems can be categorized as either passive or active, depending on the source of the energy being detected. Passive sensors record reflected sunlight (optical) and emitted temperature (thermal) from the Earth's surface. Optical and thermal imagery are currently the most commonly available datasets for monitoring coastal ecosystems. Optical imagery is easy to use and interpret but images can be hindered due to persistent cloud cover, common in tropical regions where many of these ecosystems are located. By contrast, active systems transmit their own energy pulses and measure the time of travel and intensity of the pulse that gets reflected from the surface back to the sensor. Active remote sensing can be more expensive but it can penetrate clouds and thick canopies thus providing more information.

Each sensor works in specific bands of the light spectrum (e.g., colors) to create images. Bands are a set of similar wavelengths or frequencies. For example, visible light is composed of blue, green, and red bands of the electromagnetic spectrum; other bands include radio,

microwaves (radar), and infrared waves. Visible (blue, green, and red), near-infrared (NIR), and microwaves are primarily used in coastal vegetation studies.

Both active and passive techniques offer unique advantages and disadvantages (**Table 6.1**). Combining data from both techniques is a viable option and can often offer unique information that is not detectable through one method alone.

Table 6.1 Advantages and disadvantages of remote sensing techniques.

	ADVANTAGES	DISADVANTAGES
Passive	Data is usually easier to interpret as it produces images similar to a camera; the reflection from different spectral bands can be used to classify land cover types and vegetative species; vegetative health can be inferred using near-infrared and infrared reflections.	Requires sunlight for imaging; changes in season need to be considered (polar regions with large variation in length of day light by season, for example); cloud cover can limit imaging capabilities because clouds scatter and absorb light (equatorial regions that often have persistent cloud cover year round).
Active	Transmit their own energy pulses that are often weather- and daylight independent; possible to directly compare images done using the same parameters (mode, incidence angle, polarization and processing level).	Can be more expensive; image analysis is more difficult and can differ dramatically depending on the parameters used.

Resolution

In remote sensing, the fundamental unit of data collection is known as a pixel and is defined in terms of ground dimensions. It is usually presented as a single value that represents the length of one side of a square. For example, a spatial resolution of 30 meters means that one pixel represents an area 30 meters by 30 meters on the ground. The resolution of an image is an indication of its potential detail, where the smaller the pixel the finer the detail (**Fig. 6.1**). In other words, 30 meters resolution data could identify any earthly feature that is 30 meters by 30 meters (useful for mapping ecosystem extent). Anything smaller than 30 meters by 30 meters requires a coarser resolution (10-meter resolution can be used to monitor encroachment by agriculture). We recommend starting with higher-resolution satellite

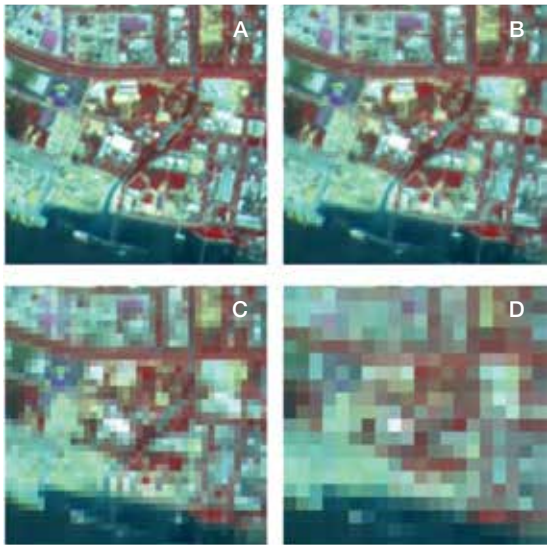


Figure 6.1 Effect of pixel size on the visual appearance of an area.
(A) 10 m pixel size,
(B) 20 m pixel size,
(C) 40 m pixel size,
(D) 80 m pixel size
(© Centre for Remote Imaging, Sensing & Processing)

imaging to obtain a general view of the ecosystem extent and overall health. Then follow with higher-resolution imaging just for areas of particular interest. In most cases, freely available 30 meter resolution imaging will be sufficient for blue carbon ecosystem mapping.

AVAILABLE DATA SETS

Satellites equipped with instruments for monitoring the earth's surface have been launched into orbit by a host of Nations since the 1970s creating a huge archive of data. However, this wealth of data can be overwhelming and not all data are freely available to the public. In addition to the satellite data, aerial photography has also been used for coastal monitoring, especially after natural or anthropogenic disasters (such as hurricanes or oil spills), but those data sets are very limited, research oriented, and not readily available. Currently Landsat, MODIS, SRTM, PALSAR and ICESat/GLAS datasets are appropriate and freely available for the operational purpose of coastal ecosystem studies at global scales. All are described in detail below.

Landsat

Description: Landsat is the most popular and longest running series of civilian Earth-observing satellites. The first Landsat was launched in 1972, and the latest satellite in the series, Landsat-8, launched in 2013. Landsat data is by far the most widely used dataset to map and monitor tidal wetlands. All missions carried multispectral sensors operating from the visible to the near infrared (NIR) portion of electromagnetic spectrum. Landsat 8 was augmented with a new band (true-blue) to facilitate measurements in coastal waters. It is a passive sensor and provides both optical (30 m pixel size) and thermal (60 m pixel size) imagery. Optical bands of this satellite record blue, green, red, near-infrared, and mid-infrared regions of the reflected sunlight. Different combinations of these bands are used to detect vegetation health, seasonal variability, leaf area index, land cover change, deforestation, and afforestation. Detailed information on Landsat is available on the following web site: http://landsat.usgs.gov/.

Figure 6.2 Example image from LANDSAT-8 data (© NASA)

Where to find the data: Landsat data can be viewed and downloaded from multiple places, but the most reliable place is the US Geological Survey (USGS) web portal. The Landsat Look Viewer site (http://landsatlook.usgs.gov/) is for viewing data availability and downloading a pseudo-color jpeg image. The GloVis site (http://glovis.usgs.gov/) is for browsing and downloading individual images. For searching and downloading multiple images of an area covering all available imagery, the EarthExplorer site (http://earthexplorer.usgs.gov/) is appropriate. All of these sites are intuitive, self-explanatory, and they make browsing and downloading data easy.

The EarthExplorer site allows a user to filter data based on different criteria, such as range of dates, percentage of cloud cover in an image, different Landsat satellites, etc.

Potential applications: For blue carbon purposes, Landsat images can be used to produce vegetation index (VI) products, which indicate the presence/absence and abundance of vegetation. A detailed review of different VIs and their usage is given in Bannari *et al*., (1995). Normalized Difference Vegetation Index (NDVI) and Enhanced Vegetation Index (EVI) are the two most robust and widely used Vis; however, each has its own limitations. NDVI values easily saturate at moderate to high vegetation density leading to underestimates in very dense ecosystems. EVI does not saturate, but the blue band reflectance can add significant noise due to the atmospheric scattering of blue light. Recent studies have shown that for coastal ecosystems, a modified EVI, called EVI2, is better suited to provide accurate estimates of vegetation intensity. Unlike NDVI, EVI2 does not saturate, and due to the lack of the blue band, it does not add significant noise.

The VI data calculated from Landsat imagery can estimate intensity of vegetation cover in coastal mangrove, tidal salt marsh, and in some seagrass ecosystems. Higher VI values indicate denser vegetation with higher leaf area index. Correlating the VI values to field-observed vegetation density produces spatially explicit maps of biomass at 30 meter pixel size for the entire area of study.

MODIS

Description: NASA's Moderate Resolution Imaging Spectroradiometer (MODIS) is a sensor aboard NASA's Terra and Aqua satellites. Terra was launched in 1999 and orbits around the Earth from north to south crossing the equator in the morning. Aqua was launched in 2002; it passes south to north over the equator in the afternoon. Terra MODIS and Aqua MODIS view Earth's entire surface every 1 to 2 days, acquiring data in 36 spectral bands, at a spatial resolution of 250 m, 500 m, and 1 km. Detailed descriptions about MODIS can be found in NASA's http://modis.gsfc.nasa.gov/ website.

Where to find the data: MODIS data of different processing and product levels can be downloaded from NASA's MODIS websites (such as Land Processes Distributed Active Archive Center LPDAAC site: https://lpdaac.usgs.gov/products/modis_products_table). However, each MODIS data granule covers a vast area, which may cover too large an area compared to what is needed for blue carbon projects. A more suitable place to download areal subsets of MODIS data is NASA's Distributed Active Archive Center (DAAC) at Oak Ridge National Laboratory (ORNL) (http://daac.ornl.gov/MODIS/). This site is intuitive and it provides step-by-step instructions for downloading data. Another advantage of using this site is that the data can be downloaded as geo-tiff files in the latitude-longitude format, which can be easily opened by any image processing or geographic information system (GIS) software program.

Potential applications: Similar to the Landsat data, VI images from MODIS data can be used to identify vegetation density of coastal ecosystems, and when correlated with biomass of field plots, these data can be used to map large areas of biomass. Another important use of MODIS data is the change detection of coastal areas. MODIS sensors have collected data from global land and ocean surfaces since 2000, making daily time series data available for studying changes in the vegetation cover of coastal areas. Since the highest spatial resolution of MODIS is 250 m, EVI2 index images produced from these datasets are appropriate tools

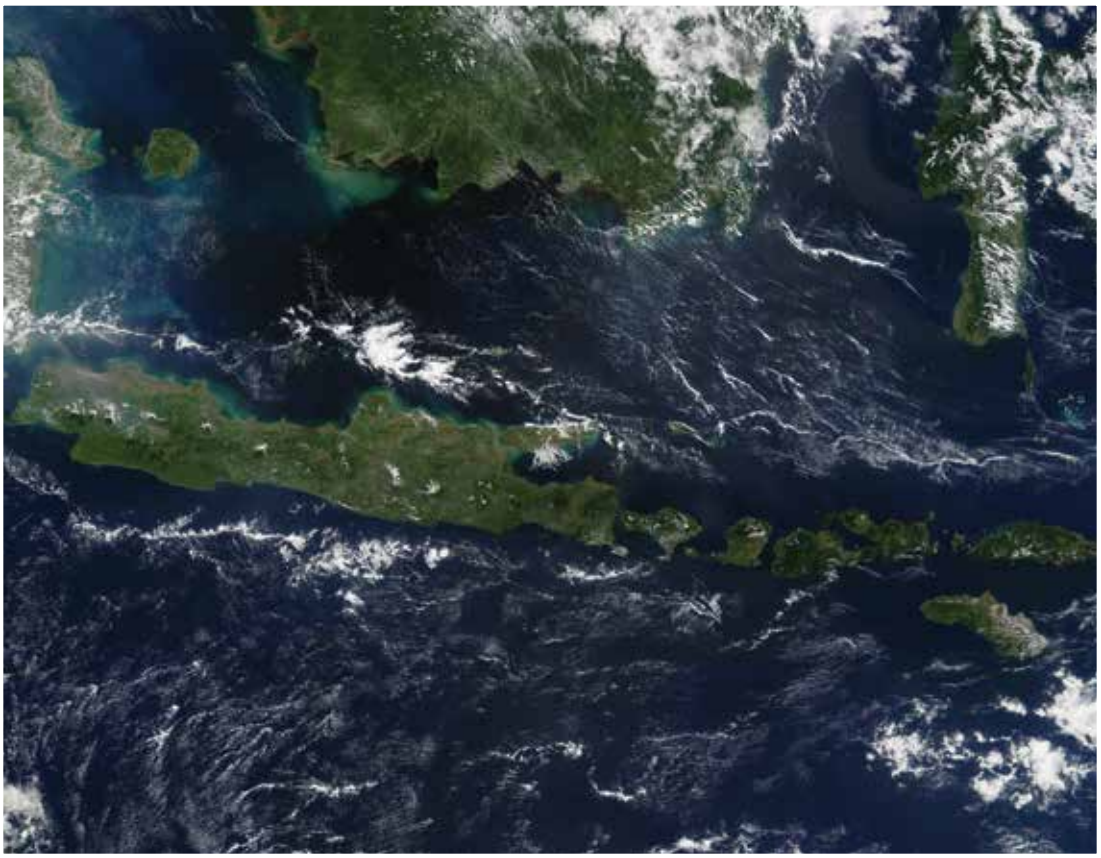


Figure 6.3 Example image from MODIS data (© NASA)

for studying temporal changes of coastal mangroves and salt marshes. A detailed method for using MODIS data to quantify mangrove destruction of a large area is given in Rahman *et al*. (2013).

SRTM

Description: The Shuttle Radar Topography Mission (SRTM) was flown aboard the space shuttle Endeavour February 11–22, 2000. Single-pass radar interferometry was used for these datasets, which acquired two signals at the same time by using two different radar antennas. An antenna located on board the space shuttle collected one dataset; the other was collected by an antenna located at the end of a 60 meter mast that extended from the shuttle. Differences between the two signals allowed for the calculation of surface elevation. The processed data are available in 1-arc-second (approximately 30 meter) resolution elevation only for the United States and 3-arc-second (approximately 90 meter) resolution elevation for global coverage.

Where to find the data: USGS EarthExplorer (http://earthexplorer.usgs.gov/) sites provide the SRTM data for the U.S. Global coverage of SRTM data can be downloaded from the Consultative Group on International Agricultural Research (CGIAR) Consortium for Spatial Information (http://srtm.csi.cgiar.org/) website. NASA has released version 3 of the SRTM data, which exhibits well-defined water bodies and coastlines. The version 2 directory also contains the vector coastline mask derived by National Geospatial Intelligence Agency, called the SRTM Water Body Data (SWBD), in ESRI Shapefile format. The data may be obtained through the http://dds.cr.usgs.gov/srtm/ website. All versions are distributed with

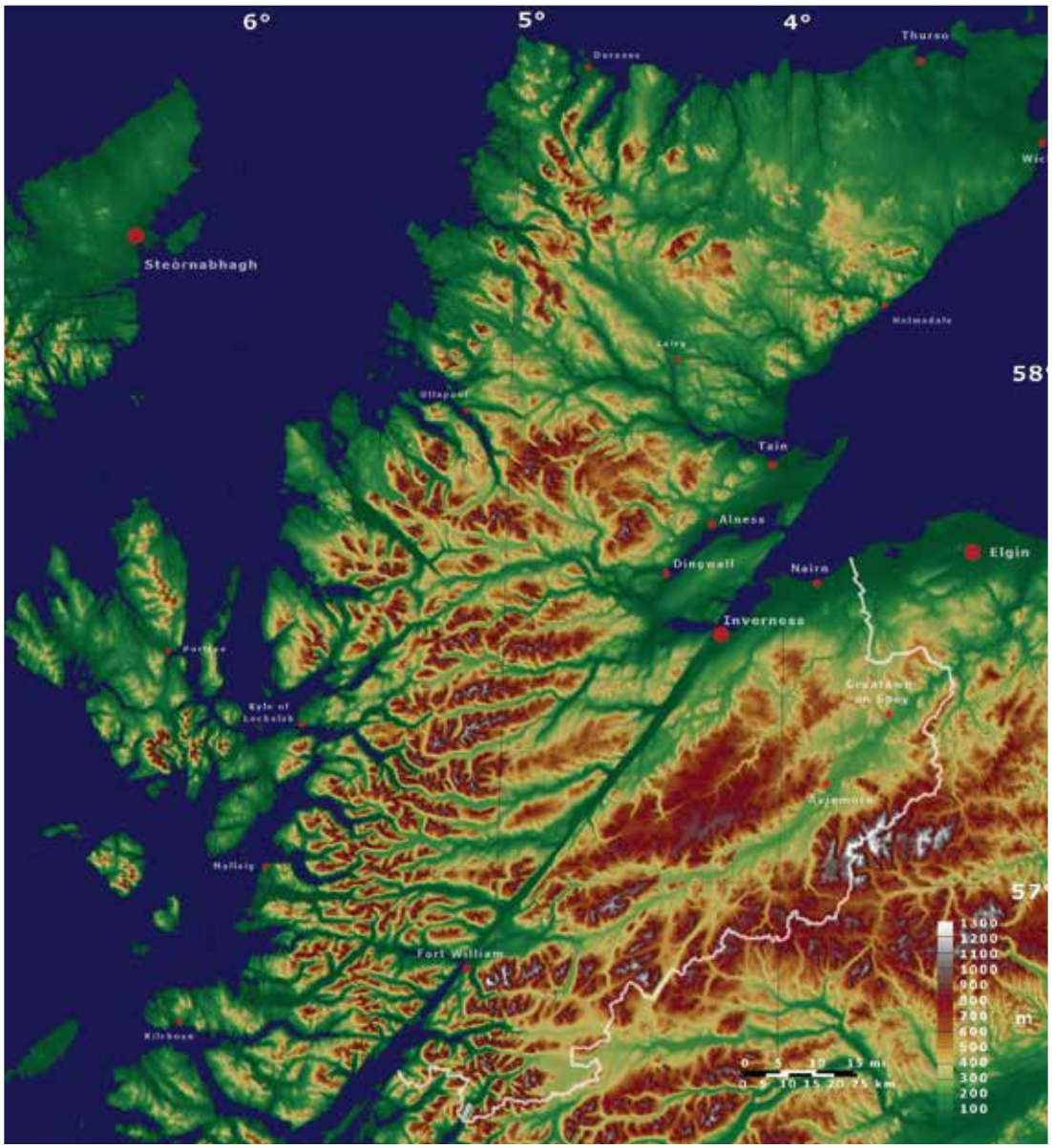


Figure 6.4 Example of a falsely colored image from SRTM elevation data (© PawelS Wikimedia Commons)

the appropriate metadata documentation.

Potential applications: The application of SRTM data for blue carbon estimation is mainly to map watersheds and mangrove environments. Since mangroves grow at sea level, the SRTM data can potentially provide vegetation heights of mangrove stands. Although the data are from 2000, mangroves grow very slowly and these data can still be utilized to assess the vegetation height of mangrove stands that have remained undisturbed. Using allometric equations that correlate aboveground biomass with canopy height and diameter-at-breast height (dbh) measurements, SRTM data can provide biomass of mangrove stands. Data from the areas that have been disturbed or deforested since 2000 can be used to estimate the loss of above ground biomass (Simard *et al*. 2006; Simard *et al.* 2008).

PALSAR

Description: The Japan Aerospace Exploration Agency's (JAXA) Phased Array L-band Synthetic Aperture Radar (PALSAR) produced data from 2006-2011, and a new sensor was launched in 2014. PALSAR is an active microwave sensor used to achieve cloud-free and day-and-night land observation. It is a fully polarimetric instrument, meaning it measures the polarization of transverse electromagnetic waves. PALSAR can emit and receive horizontal (H) or vertical (V) transverse waves in various combinations, fine-beam mode with single polarization of HH (horizontal transmitting, horizontal receiving) or VV (vertical transmitting, vertical receiving), dual polarization (HH+HV or VV+VH), or full polarimetry (HH+HV+VH+VV). The scattering patterns measured from the different polarizations provide information to the structure of the vegetation. It also features wide-swath ScanSAR mode, with single polarization (HH or VV). Spatial resolution of the fine-beam mode HH or VV polarization is approximately 12 m, and that of the ScanSAR mode is 100 m.

Where to find the data: PALSAR data can be downloaded from the Alaska Satellite Facility (ASF) https://ursa.asfdaac.alaska.edu/cgi-bin/login/guest/ website. Data can be imported using ASF MapReady software to produce geo-tiff format images. Background noise that may lower the quality of the image can be removed using Lee filter, and the images can be mosaicked for each polarization (HH with HH, HV with HV).

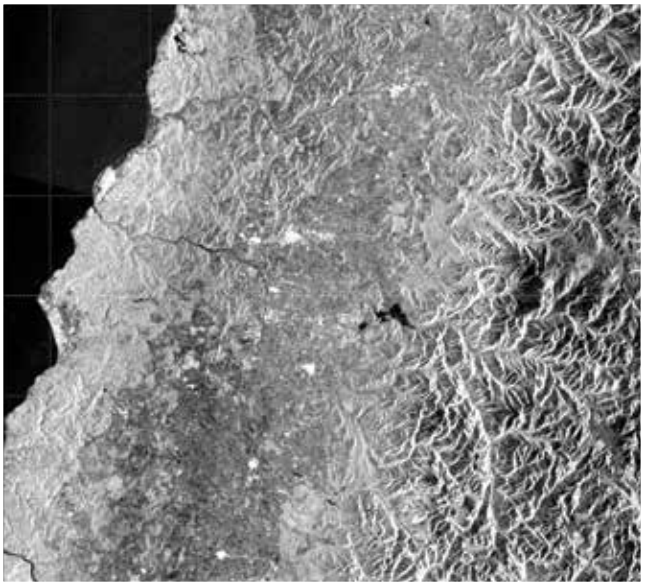


Figure 6.5 Example Image from PALSAR data (© JAXA)

Potential applications: Fine-beam PALSAR data can be used to make digital elevation maps (DEM), extract topography data, or estimate biomass of coastal ecosystems. In order to extract the coastal vegetation information, we suggest a Principal Component Analysis (PCA) be performed. The first step is to create a Radar Forest Degradation Index (RFDI) from the HH and VV images. The RFDI is able to assess the strength of double-bounce scattering, which is the scattering of radar waves off a horizontal (ground) and a vertical (grass, tree trunks, etc.) surface. This double-bounce scattering has the potential to differentiate between distinct types of vegetation. Next, the RFDI, HV, and HH layers are stacked to create a three-band image. Studies have shown that PCA-1 can clearly distinguish between water and vegetation, thus allowing mapping of coastal deforestation (mainly of mangroves). PCA-2 can potentially be used to estimate vegetation height at ~12 m resolution. This can be combined with field data to estimate biomass of coastal ecosystems.

A document explaining ALOS Palsar data from JAXA is available at www.eorc.jaxa.jp/ALOS/en/doc/fdata/ALOS_HB_RevC_EN.pdf. Section 7 of that document has detailed description of PALSAR data, including the steps required to process data at different levels according to the user requirement. JAXA has also released global mosaics of ALOS/PALSAR data that can be found at www.eorc.jaxa.jp/ALOS/en/palsar_fnf/fnf_index.htm and can be used to monitor

land use changes as well as biomass.

ICESat/GLAS

Description: Launched on 12 January 2003, after seven years in orbit and 18 laser-operations campaigns, the Ice, Cloud, and land Elevation Satellite (ICESat)'s science mission ended due to the failure of its primary instrument. The main objective of the Geoscience Laser Altimeter System (GLAS) instrument was to measure ice sheet elevations and changes in elevation through time. Secondary objectives included measurement of cloud and aerosol height profiles, land elevation and vegetation cover, and sea ice thickness. Lidar datasets from ICEsat/GLAS (Geoscience altimeter system) contain global data points collected over a period of seven years (from 2003 to 2009).

Where to find the data: The National Snow and Ice Data Center (NSIDC) distributes 15 Level-1 and Level-2 data products from the GLAS instrument that was aboard the ICESat satellite. For information please consult the NSIDC website http://nsidc.org/data/icesat/data.html.

Potential application: GLAS data allows estimate of canopy height with accuracies of a few meters (Simard *et al*., 2011; Simard *et al*., 2008; Fatoyinbo & Simard 2012). The new ICESat-2 is set to launch in 2016, given favorable conditions the data produced may provide more dense spatial coverage.

It is important to note that there are many other data sets that are not highlighted here. Data from European satellites, such as SPOT, have also been used for coastal ecosystem studies, but these data are of limited access. Image data from commercially available satellites, such as IKONOS, GeoEye, and QuickBird have also been applied to study the coastal ecosystems, but these data are of limited spatial and temporal scopes, and not freely available. Sonar data are mainly useful in the mapping of seagrass meadows regardless of water clarity, but are very rarely available. The applications, limitations, and potentials of sonar data for blue carbon estimation are still in the active research phase and not yet operational on a global scale.

Developing clear goals and working with a trusted remote sensing expert to determine which sensor and method of data analysis is most appropriate and practical for your project will ensure that the end product meets the project goals (Chapter 2: Conceptualizing the Project and Developing a Field Measurement Plan).

DATA ACQUISITION AND PROCESSING

Once the data type, resolution, and scale needed for your project have been determined, the resulting images will need to be processed (**Fig. 6.6**). This should be done by a remote sensing specialist, but the extent of processing that has been done to an image should be documented in the image's metadata (Chapter 7: Data Management).

Remote sensing data sets are extensive, span decades, and most require expertise and professional software to download and analyze; therefore, it can take 10 to 32 weeks (realistically) to implement a project. This time frame largely depends on an organization's experience, if any additional images had to be requested beyond the freely available archives, and/or the number of steps that have been provided by others (e.g., data providers, software

programs). The appeal of raw data, which may be faster to obtain, is the ability to apply one's own calibration/navigation formulae to it, in contrast to using standard algorithms from some data provider. The disadvantage of this approach is that the user must possess the hardware, software, and personnel resources to perform these steps before the data is usable.

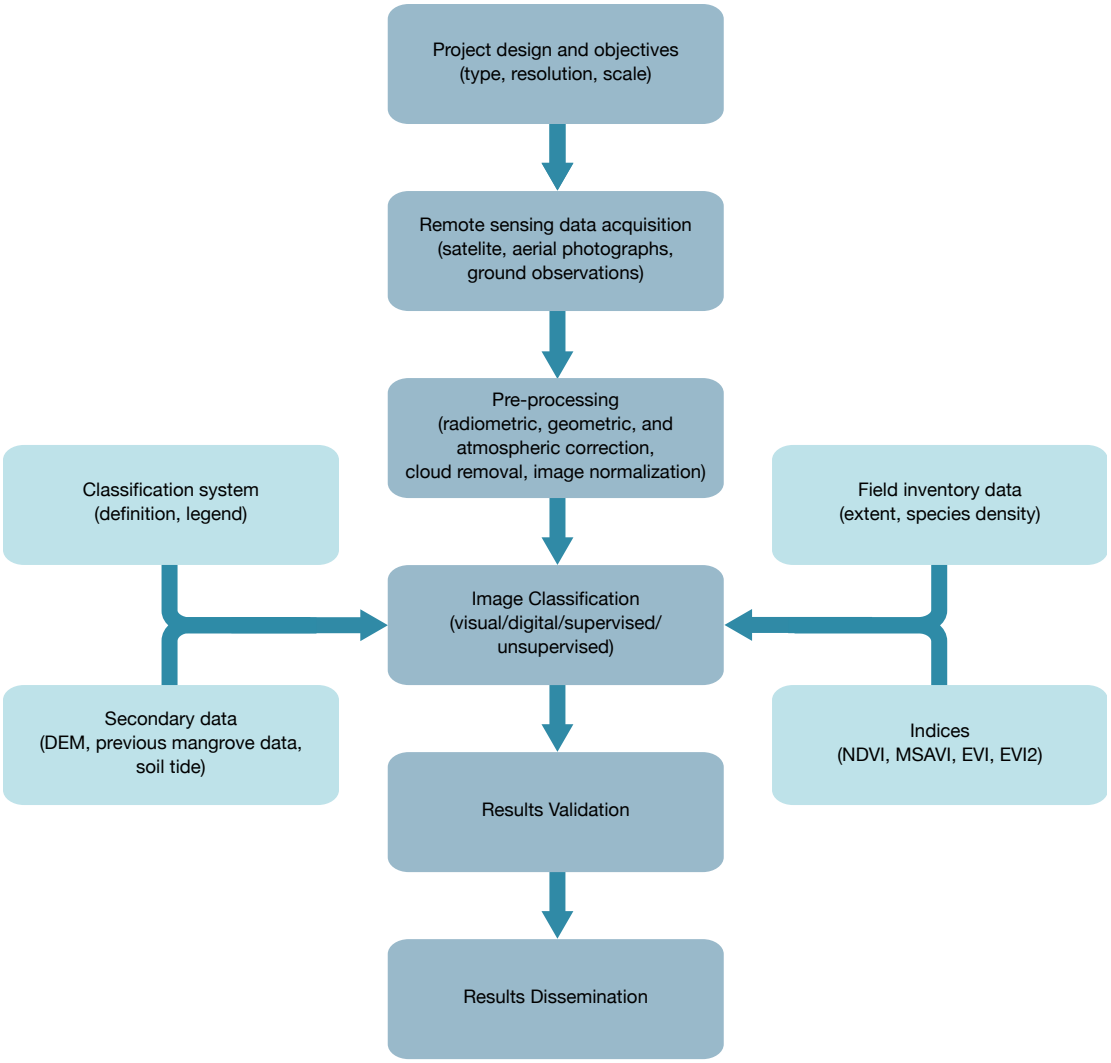


Figure 6.6 Steps for processing remotly sensed images

Pre-processing of remotely sensed data involves correction of distortion, degradation, and noise introduced during the imaging process and produces a corrected image removing these anomolies. Typically, images will need to be processed to correct radiometric (haze and atmospheric scattering) and geometric (Earth's rotation and satelite location) issues. Persistant cloud cover is a major issue in most of the tropical regions where blue carbon ecosystems exist. When Radar data is not available or is too expensive, cloud free pictures can be assembled using individual images collected over time. The time frame used depends on the rate of change. For example, if the ecosystem being mapped is relatively stable, then it might be possible to piece together images spanning serval years without loosing any infomration. However, if the ecosystem is being distroyed at a rate of 10% every 5 years you might want to limit your search to images produced in the last year to create a current map

that is as accurate as possible. Image normalization is needed to analyzse multi-temporal data or data covering large areas.

Once the images are acquired and pre-processed to create a usable image, the image will need futher processing to extract the project relevant data. Processing involves classification of the image components (i.e., trees, shrubs, water, mud flats, etc.) and may also include data on vegetative species composition, density, and biomass. Any additional information, such as known vegetation cover, density, species composition, management history, and past disturbances will be useful during image classification. Secondary data such as tide information, elevation maps, published and unpublished ecosystem maps and reports are also highly beneficial. The United Nation's Land Cover Classificaiton System (LCCS) is the recommended standard for defining mapping classes (Di Gregorio & Jansen 2000; Di Gregorio 2005). Validation should be performed using higher resolution remote sensing data or ground truthed data.

POTENTIAL USES OF REMOTELY SENSED DATA

Mapping

Habitat mapping and classification by means of remote sensing are performed by correlating a cluster of numerical pixel values with verified features, such as vegetative cover, open water, tidal flats, inland marshes, forested wetlands, or bare soil type. Remote sensing techniques coupled with on the ground validation and modeling has led to the development of spectral signatures that can be used to map ecosystem extent, type, and in some cases provide information at the species level (see Appendix E for a general protocol for mapping mangroves and salt marshes). However, challenges such as distinguishing coastal wetland vegetation from the neighboring inland vegetation, accounting for areas where the vegetation is sparse, and routine detection of individual species remain difficult to overcome (Heumann 2011).

There are many techniques for creating coastal ecosystem maps. For example, mangroves and marshes are found within the intertidal range (up to two meters above sea level); thus it is possible to use ground elevation and tidal range data to determine the potential location of these systems. However, accurate DEM data at large scales are rare, so your area of interest may require additional local knowledge to design the proper limits. Another technique that can be employed is to map tidal extent using a time-series of images that coincide with high and low tide (Murray *et al*. 2012). Synthetic aperture radar (SAR) data have been increasingly used for mapping, and monitoring at local and regional scales. SAR is particularly useful as it can penetrate the forest canopy and interact with larger vegetative components (branches, trunks and above ground roots) (Lucas *et al*. 2007a; Souza-Filho *et al*. 2011; Nascimento Jr *et al*. 2013). SAR data has also been used to identify mangrove forest structural parameters such tree density, basal area, height, biomass, age distribution, and forest structure (Aschbacher *et al*. 1995; Mougin *et al*. 1999; Held *et al*. 2003). Most recently, UNEP-WCMC updated the "World Atlas of Mangroves" in 2010 (Spalding *et al*. 2010), and Giri *et al*. (2011) generated an updated global mangrove baseline map primarily using Landsat data (**Fig. 6.7**). UNEP-WCMC has also accumulated global observations of salt marsh distribution and extent; that map should be available fall of 2014.

Seagrasses is particularly difficult to map using remote sensing. The water turbidity and color, sun glint, and the epiphytes that cover the blades of grass may dilute the spectral reflectance signal of sea grasses and hinder the instruments ability to "see" through the water. They are also found to grow in a wide range of densities which can affect the return signal to the sensors. Under clear water conditions, remote sensing images have been able to detect a wide variety of seagrass densities ranging from < 25% to 100% cover (Roelfsema *et al*. 2009; Pu *et al*. 2012) and, when combined with local knowledge, aerial photography, and observations in the field, rough outlines of seagrass extent can be successfully mapped.

Figure 6.7 Global distribution of mangroves prepared using Landsat satellite data at 30 m spatial resolution of year 2000 (Giri *et al*. 2011)

Canopy Height and Biomass

Besides mapping ecosystem extent, remote sensing can be used to map biomass that can later be used to estimate the amount of carbon in the aboveground vegetative pool. Biomass is mapped based on the species composition and height, and several datasets are available and serve this purpose. GLAS data allows estimate of canopy height with accuracies of a few meters (Simard *et al*. 2008; Simard *et al*. 2011; Fatoyinbo & Simard 2013). The new spaceborne interferometric system launched by the German Space Agency: TanDEM-X could potentially be used to measure height and biomass within salt marshes and canopy height in mangrove forests. In February 2000, STS-109 successfully fulfilled its SRTM mission (Shuttle Radar Topography Mission) and gathered topographic data over 80% of the land surfaces of the Earth as well as radar backscatter (HH, VV polarization) and interferometric coherence. SRTM elevation measurements can be used to estimate canopy height with an accuracy of 2–4 meters but unfortunately this makes it insufficient for mapping the height of salt marshes (Simard *et al*. 2012). Space Shuttle Endeavour was equipped with two radar antennas used interference patterns between the two radar signals to derive terrain height.

Baseline maps of mangrove height and biomass have been generated for several regions, including Africa (Fatoyinbo *et al*. 2008; Fatoyinbo & Simard 2013), Florida (Simard *et al*. 2006) and Colombia (Simard *et al*., 2009). A global map should be published in 2015. When used in combination with SAR data, the height maps not only improve the accuracy of biomass estimates, but also can be used to map mangrove species (Held *et al*. 2003; Lucas *et al*. 2007b). For example, Held and Ticehurst (2003) and Lucas *et al*. (2007) noted that where extensive root systems occurred (e.g., in mangroves dominated by *Rhizophora* species), a subsequent decrease in the backscatter occurs in proportion to increases in biomass. Such decreases typically occur once mangroves have attained a height threshold, with Lucas *et al*., (2007) suggesting 10 meters as an appropriate threshold. By exploiting these observed characteristics of mangroves, new mangrove mapping techniques have been developed that can differentiate between mangrove type (with/without aboveground roots) and species.

Monitoring Ecosystem Change

Monitoring impacts from blue carbon ecosystem restoration/conservation efforts and rates of degradation/deforestation from land use change is valuable for national carbon accounting programs and for monitoring climate mitigation and adaptation strategies. However, despite their importance, no systematic maps of coastal ecosystem change at regional to global scales currently exist.

For regional assessments, the use of moderate (< 30 meters) spatial resolution optical (e.g., Landsat) data has been successfully demonstrated and is generally recommended (Spalding *et al*. 1997; Giri *et al*. 2011). However, routine detection of change has proved difficult, partly because persistent cloud cover in the tropics prevents regular observation. In some cases, this has been overcome using SAR (e.g., Souza-Filho & Paradella 2003; Nascimento Jr *et al*. 2013). SAR can be used to detect change within and away from existing baselines (when baseline maps are available), and that data can further be complimented with corresponding biomass data. Exploiting the dense time-series of Landsat sensor data, including data provided by the recently launched Landsat-8, can also increase the level of change detected.

Carbon Estimations

Combining maps of ecosystem extent, species, and biomass with average carbon stock values derived from field data enables national, regional, and global estimates of carbon stocks (DeFries *et al*. 2007). Similarly, this data can then be used to monitor changes to the carbon stock and estimate carbon emissions (emitted or removed) based on degradation, conservation, and restoration of blue carbon ecosystems. Historic remote sensing data can be used to construct a history of carbon stocks and emissions that can be used for reference scenarios (Gibbs *et al*. 2007). This technique has been done with mangroves, but research into expanding the technique to include salt marshes and seagrasses is a high priority.

Numerous books and reviews devoted to creating carbon stock maps and measuring carbon emissions using remote sensing are available and can provide more detailed background information and methods.

VALIDATING WITH FIELD DATA

To make use of remote sensing data for inventories, and in particular to relate land cover to land use, it is good practice to complement the remotely sensed data with ground reference data (often called ground truth data). Land uses that are rapidly changing or that are easily misclassified should be more intensively ground-truthed than other areas, preferably from actual ground surveys collected independently. High-resolution aerial photographs or satellite imagery may also be useful.

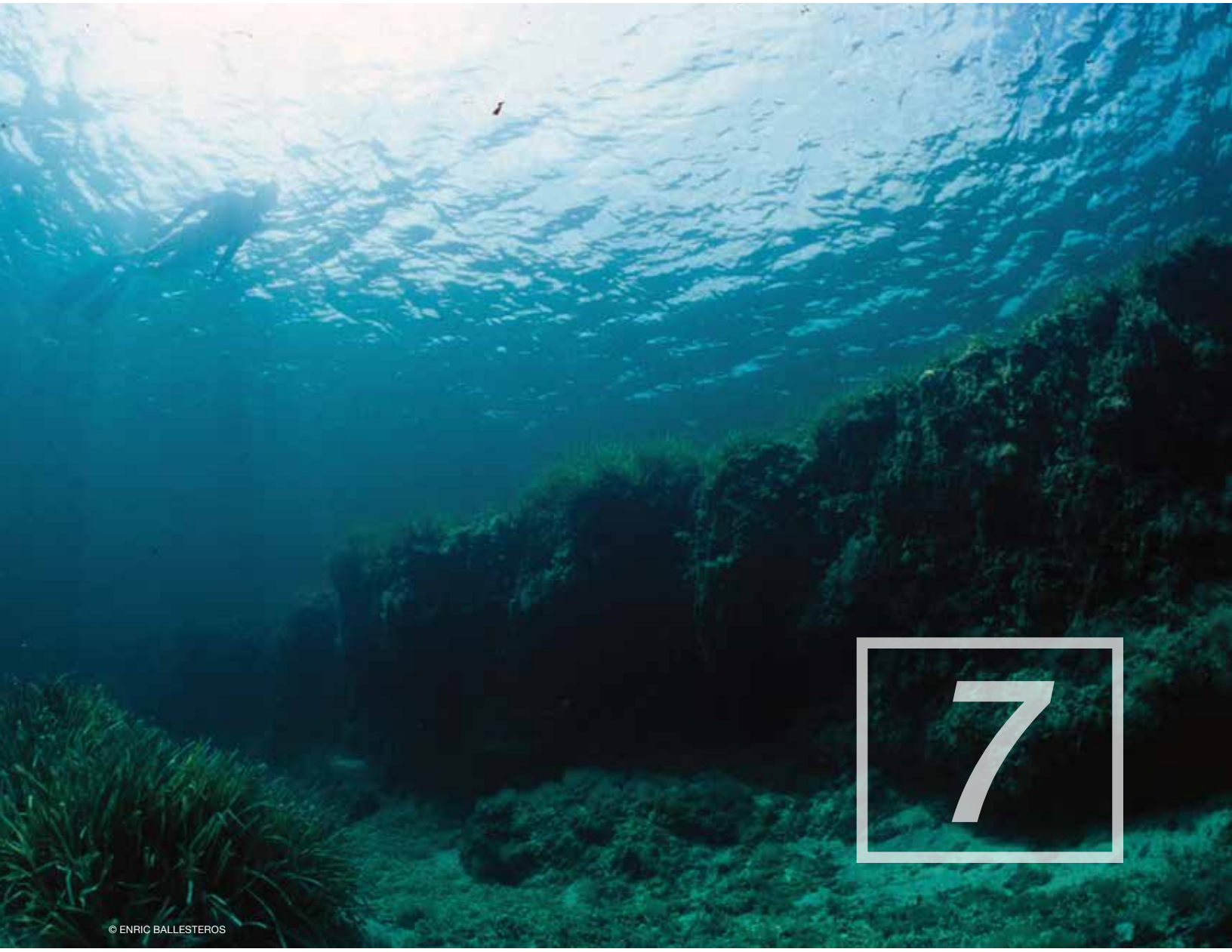
Parameters that can currently be measured remotely are presence and absence of the ecosystem, species, leaf area and canopy cover, canopy height, and vegetative biomass. Therefore, similar parameters should be measured for ground truthing. Ideally, each field plot should encompass a similar area to that of a single pixel and span the entire range of the ecosystems macro-structural attributes. In wetland ecology, typical plots are in the orders of meters and may not be appropriate for remote sensing applications. Instead, plots

of 1 hectare would enable accurate characterization of canopy heterogeneity and provide excellent calibration for remote sensing measurements. If that is not feasible, plot sizes of a few tens of meters can be used if they are representative of the local (hectare scale) structure.

The opportunity exists to involve local communities in the collection of ground truthed data. While significant progress has been made toward building capacity and communicating the importance of blue carbon ecosystems to local, national, and international decision makers, these efforts are still in their infancy and continued work is needed to ensure the ongoing success and implementation of these and other related projects. Opportunities for field work increase relationships within the community and create a sense of ownership that stimulates continued support for conservation and restoration efforts long after the study has been completed.

CONCLUSION

When remote sensing systems are used wisely, including complementary combinations of different satellite and airborne sensors, they can provide data that enhances the research and management of coastal ecosystems. Each type of sensor has its own unique measurement in which to record subtle and obvious changes to coastal environments. Therefore, combining data from various sensors have and will continue to provide pertinent information regarding the biophysical and structural components of the coastal landscapes. One of the key advantages of remote sensors is that they can monitor and assess long-term trends and short-term changes of vegetation and hydrology faster, more completely and at lower cost per unit area than field or ship surveys alone (Klemas 2013).



Data Management

INTRODUCTION

Effectively recording, managing, and archiving the data collected using the methods described in Chapters 2–5 is essential for accurate and reliable analysis and use. This not only includes scientific analysis of carbon in coastal ecosystems, but also using carbon values in the design and implementation of coastal conservation and management practices or the inclusion of coastal ecosystems in greenhouse gas accounting. Further, making ecosystem data widely accessible will support broader application.

Unfortunately coastal ecosystem carbon data sets are often not widely accessible, and/or the specific data in them is incomplete or of insufficient resolution to support broader use. The data that is available is often collected using different parameters, units of measurement, time scales, and more, making comparisons across studies exceedingly difficult or impossible. To alleviate this situation, a uniform structure and format of data collection and management is recommended here to allow data inter-compatibility.

REASONS TO MANAGE AND PUBLISH YOUR DATA

Management of coastal carbon data should be a priority for any project no matter the scale or scope. Effective data management is beneficial for (MITLibraries):

- **Documentation:** Effectively documenting data ensures that proper descriptions of your data are maintained to support future use. Doing so also ensures that other users can properly acknowledge the data source and authors.
- **Meet reporting requirements:** Certification and/or funding for carbon projects—including scientific research, conservation, and policy actions—now require some form of data management plan to be in place to ensure project integrity.

In addition to the direct benefits of proper data recording and management, it is strongly recommended that data is publically available for use by others. This can mean publishing data in scientific papers, giving seminars, acting as advisors to other projects or programs, and submitting your data to open access data repositories, such as the Global Costal Carbon Data Archive described below. Benefits of data dissemination include:

- **Facilitation and support for other projects or research:** Enabling other users, including researchers, to use your data prevents duplication of effort, supports projects that might not have the capacity or resources to collect data, and allows for broader and synthetic analysis and comparison.
- **Dissemination:** Enabling a data repository to house and disseminate your data alleviates the work required to respond to requests for your data and time spent creating a system (like a personalized website) to house your data yourself.
- **Increased visibility:** Making your data useable and available to other users (such as policy-makers, project developers, or scientific researchers) through broadly accessible repositories increases the visibility and relevance of your program.

Access to quality controlled data, based on transparent and standardized protocols for interoperability, will result in profound decisions across sectors with regard to blue carbon habitats. For example:

- **Blue Carbon:** Improved assessments of blue carbon to support inclusion of coastal ecosystems in national climate mitigation and adaptation strategies;
- **Ecosystem Services:** More complete assessments of the ecosystem benefits provided to coastal communities and other beneficiaries;
- **Finance:** Support for viable market-based instruments for conserving coastal ecosystems;
- **Vulnerability Assessment:** Stronger representation of coastal ecosystems in environmental impact assessments and risk assessments for development activities; and
- **Increased Capacity:** Strengthened capacity to effectively incorporate appropriate coastal management measures into national management and protection strategies.

DATA COLLECTION

The data generated through assessments of carbon in coastal ecosystems is collected as measurements in the field and the laboratory. All data should come with metadata that describes the conditions, location, and other details of how the measurements were made. See **Table 7.1** for examples.

Table 7.1 Examples of the types of data collected in a typical coastal blue carbon project

EXAMPLES	*REMOTE SENSING*	*FIELD WORK*	*LABORATORY*
Data	Hectares of mangrove habitat	Tree diameter at breast height	Carbon content of a soil sample
Associated Metadata	Satellite information (organization, type, ID), sensor used, dataset used, parameters, proxies, etc.	Date of measurement, species of the tree, location of the tree (latitude and longitude), tool used to measure diameter, description of where on the tree the measurement was taken, etc.	Date, type of tool used (make and model of the elemental analyzer, or furnace for LOI), sample ID and description, controls used, protocol used, etc.

Written Descriptive Data

To ensure that all needed data and associated metadata are recorded, it is essential to record the data during, or soon after, collection. There are several methods for recording data (written notes, audio taping, videotaping, etc.). However, written notes are the most cost effective method of data collection. Establishing a data collection plan prior to conducting field work should be a top priority. Ensure that the plan takes into account the exact variables and the extent of data collected during any field study or laboratory analysis. The compilation of field and lab notes may appear to be straight forward; however, predetermining what to write down, how to write it down, and when to write it down will ensure all needed data is collected.

What to write down: It is often helpful to have premade worksheets that are used by all personnel in the field or laboratory. This will not only keep the data organized in a similar

fashion but also will ensure that all relevant field and laboratory data and associated metadata is consistently collected. The type of information collected depends on what is being sampled and the wider project goals (**Appendices F–H**).

How to write it down: It is useful to predetermine the level of detail required for data and metadata descriptions, units of measure to use, and types of data to be recorded. If codes/shorthand is to be used, be sure to have a predetermined reference list that defines codes or abbreviations. In the field, it is productive to have a single person in each team tasked with recording measurements and taking notes so that the recorded data are consistent. This also allows the team members taking measurements to be more efficient in moving on to the next task rather than stopping to take notes. In the lab, each researcher should have a personal lab notebook and be responsible for his or her own note taking.

When to write it down: In principle, one should aim to make notes as soon as possible after a measurement is taken (e.g., record core length at the time that the core is removed) and rely on memory as little as possible. The importance of meticulous note taking cannot be overemphasized; thus, it is imperative that the amount of time needed to accurately record data be integrated into the schedule.

Photographic Data

In addition to written descriptive data, it is valuable to establish a photographic record, especially in the field. Photos of the field site, soil cores, vegetation, people involved in the study, sampling processes as they happen, etc. are all useful for establishing a record and documenting data.

Protocols are usually established so that photographic data is consistent for all sampled plots.

- For example, in mangroves, it is common practice to take four photos—one in each cardinal direction (N, S, E, W)—from the plot center (Hall 2001a, b) (**Fig. 7.1A**).
- For seagrasses and tidal salt marshes, it is sufficient to take photos looking down on the plot. The number of photos needed per plot depends on how many it requires to get a representative idea of the health of the entire plot (**Fig. 7.1B**).

Metadata such as the name and affiliation of the photographer, location (GPS coordinates if available), and date of each photograph should be recorded in the written notes as the photos are taken to assist in easy photo identification later. All photos should be stored electronically with other project data. It is important to back up photos as well as record the metadata associated with each.

Photography can be a useful and simple mechanism for monitoring changes in an ecosystem. A photo point monitoring system can be established by taking photographs at the same location with the same field-of-view at different points in time. Such photo point monitoring is an easy and inexpensive, yet effective, method of tracking vegetation and ecosystem change. With appropriate site marking and documentation, photos can be precisely replicated by different people many years apart (Hall 2001a, b).

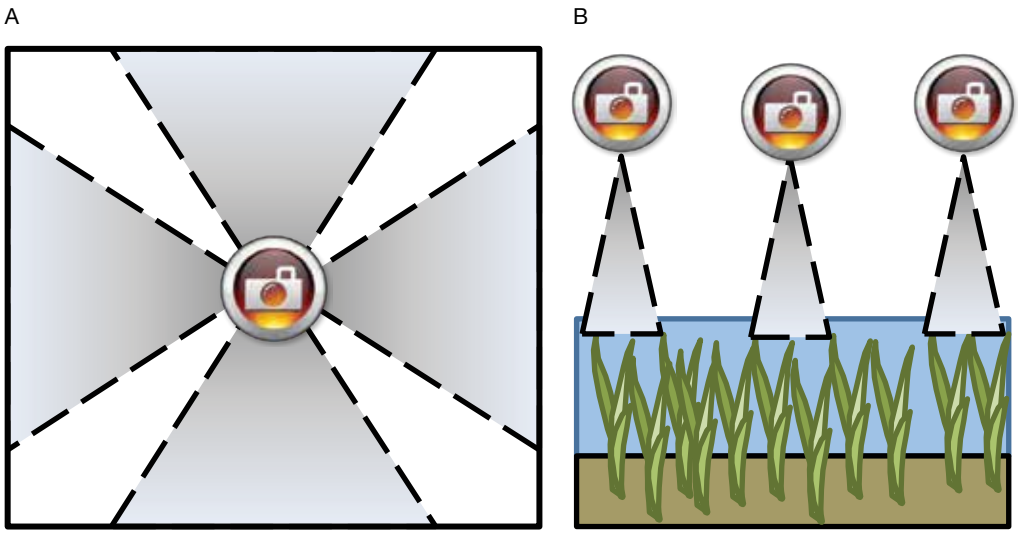


Figure 7.1 Photo point monitoring. (A) For mangrove plots, the person taking the photos stands in the center of the plot and takes a photo in each direction. (B) For seagrasses and salt marshes, photos are taken looking straight down and in various locations to get a general idea of the appearance of the site.

DATA QUALITY ASSURANCE AND QUALITY CONTROL

At the end of every day of field research, all data records should be collected by a data reviewer. The data reviewer should immediately review all the data for completeness, legibility, and accuracy in the presence of the person who recorded the data in case there are any inconsistencies or questions. Once satisfied by the quality of data recorded, the reviewer should write his or her name and the date of the review, along with any notes on issues that were noticed during the review so that they can be prevented in the future (**Fig. 7.2**).

Laboratory data should be recorded in notebooks which should stay in the lab at all times to prevent them from being lost or damaged. Upon completion of a study, photocopies of the relevant pages should be made and stored in the lab for future reference. Regular lab meetings can serve as venues to discuss results and address any issues.

Once the field and laboratory data have been vetted by the data reviewer, it needs to be entered into a computer to aid in analysis and for uploading to a database (see next section). Once the person entering data has checked the computer entry against the data sheet and corrected any errors, he or she should write his or her name at the bottom of the data sheet and the date of data entry (**Fig 7.2**). Any issues should be noted so that they can be corrected in the future. In addition, a subsample of data sheets (~ 10%) should be compared to the computer entry by someone other than the person who entered the data. The data entry reviewer should also write his or her name and the date of the data review along with any notes on issues that were apparent or corrections that were made (**Fig. 7.2**). It is important that the field supervisor be made aware of all issues noted on the data sheets so that preventative measures can be taken.

The procedure for data quality assurance and quality control is as follows:

- Collect field data using a predetermined worksheet (**Appendices F–H**)
  - Each day submit worksheets to data reviewer
  - Data reviewer checks and signs off on each worksheet
- Collect laboratory data and record it in a lab notebook
  - Review data each week at a lab meeting
- Enter data into a computer
  - Data reviewer enters all data into a predetermined program (e.g., Excel spreadsheet)
  - A different person reviews what has been entered to ensure that the data is accurate and understandable

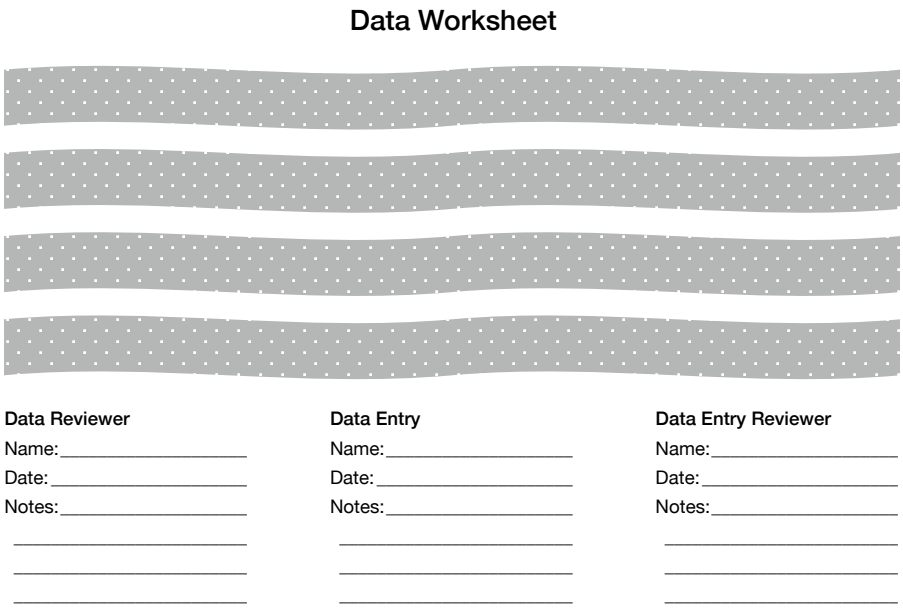


Figure 7.2 Example of a data sheet with a method for recording who was responsible, quality control, and quality assurance for each step in data recording.

REPORTING

Reporting may be as simple as presenting the total carbon stock for an ecosystem of a certain area with all of the components combined into a single measurement. Reporting can be more specific by breaking down the portion of the total carbon stock that can be contributed to each specific pool (soil, trees, shrubs, grasses, litter, etc.). Partitioning ecosystem pools allows for clearer interpretation and more accurate determination of shifts in carbon stocks through time that may occur due to changes in land management, land use, or climate change. It also facilitates reporting of statistical analyses, which can test for changes in the pool size of individual components as well as changes in total ecosystem stocks through time.

Graphical displays are useful tools for illustrating the different carbon pools (e.g., bar or pie charts). Photos accompanying carbon stock results may assist in interpreting how plant composition and structure relates to ecosystem carbon pools. Graphical data are valuable for rapid interpretation of the size of individual carbon pools and how they compare to other structural components of the ecosystem. They are also valuable for comparing structural

differences between like ecosystems (i.e., tidal marshes in different locations) or between different ecosystems (i.e., mangroves and other forest types).

DATA SHARING AND ACCESSIBILITY (DATABASES)

Open access to high quality data is viewed by many as a public good. Sharing data encourages scientific inquiry and debate, promotes innovation, leads to new collaborations between data users and data creators, reduces the cost of duplicating data collection, provides credit to the researcher that collected the data, and provides resources for project development, policy, education, and training. One of the most efficient ways to share data is through an open access database.

Uploading Data

Recognizing that the value of data often depends on its timeliness, if you choose to upload your data to a database or repository, it is best to do it as soon as possible after the study is complete and the results have been published or used for their project purpose. Hence, data from small studies can be analyzed and submitted relatively quickly. However, data from large studies that are collected over several time periods could be released as it becomes available or as specific analyses and results are finalized and published.

Criteria for Selecting a Database

In general a database is considered useful and reputable if it meets the following criteria:

- Provides common data and metadata standards and formats;
- Allows for data submission by any group in the world in generic format;
- Is well recognized and referenced by the scientific community; and
- Addresses data ownership issues by assigning a digital object identifier (DOI) number to each submission in order for it to become instantly citable;

Currently, there is no coordinated data infrastructure to support blue carbon research and monitoring efforts globally. Local datasets do exist, but many are difficult to access, subject to license restrictions, and/or being developed using incompatible approaches. The International Blue Carbon Scientific Working Group has identified management of coastal carbon data as a priority activity necessary for supporting the conservation, effective management, and creation of incentives for blue carbon coastal ecosystems through research, policy development, and field implementation. As a unifying community initiative, they have decided to establish a Global Costal Carbon Data Archive (hereafter referred to as Data Archive, see next section) to support better data management practices and standardization, and to bring all the available carbon data for coastal ecosystems together in a common format.

International Blue Carbon Initiative's Global Costal Carbon Data Archive

Blue carbon has considerable and growing support within multiple sectors throughout the international community. However, the degree to which our general understanding of blue carbon ecosystem spatial distributions and carbon stock levels has limited our ability to incorporate blue carbon issues into local and national policy. Carbon stock and flux data for coastal ecosystems are extremely patchy globally, and those that are available have not yet been integrated. In addition, considerable field work is being done around the world to collect these data, but there are limited pathways for sharing it. The International Blue Carbon Initiative hopes to improve on these issues by creating a blue carbon data archive. The archive will also increase the accuracy of and confidence in global estimates of carbon storage and emissions of blue carbon ecosystems. This data archive will serve as a central foundation upon which the coastal blue carbon science community can continue to grow. The archive will be sub-divided into three categories, one for each of the blue carbon coastal ecosystems (mangroves, tidal salt marshes and seagrasses), and each will be tailored to accommodate ecosystem-specific needs. The entire collection as well as pre-determined sub-collections of the full dataset and links to metadata will be permanently stored at the data archive and will be freely available to the public. A DOI number will be assigned to each data set upon submission making each individual data set citable, thus resolving data ownership issues.

The data archive aims to:

- Increase cost-efficiency of projects by designing them based on known spatial, temporal and process-related data coverage;
- Create a platform for modeling studies based on maximized input quality and quantity;
- Allow for the possibility of web-based visualization of data (i.e., mapping); and
- Provide a strong base for more accurate predictions, which in turn will strengthen policy recommendations at the community to national level.

Development plans for the data archive are already underway. The Initiative hopes to have the database fully functional by 2015.

Appendices

A

APPENDIX A
Additional Guidance Documents

This table provides a list of guidance documents for measuring blue carbon and for obtaining carbon credits. The documents listed either refer to the need, or attempts to provide, internationally accepted measurement and monitoring procedures for greenhouse gas accounting. This manual is meant to compliment the currently available methodologies and result in data that meet the criteria for relevant standards.

TITLE	ORGANIZATION[2]	DATE RELEASED	CATEGORY	GOAL OF THE DOCUMENT
Greenhouse Gas Offset Methodology Criteria for Tidal Wetland Conservation	Restore America's Estuaries	2015 (expected)	Carbon accreditation	Outlines the Verified Carbon Standard (VCS)-approved procedures to estimate net GHG emission reductions and removals resulting from conservation of tidal wetlands. The conservation activities intend to protect environmental benefits, including emission reductions and the net sequestration of GHGs.
Methodology for Tidal Wetlands and Seagrass Restoration	Restore America's Estuaries	2014 (expected)	Carbon accreditation	Outlines the VCS-approved procedures to estimate net GHG emission reductions and removals resulting from restoration of tidal wetlands and seagrass beds along the entire salinity range. The restoration activities intend to protect and re-establish environmental benefits, including emission reductions and the net sequestration of GHGs.
2013 Supplement to the 2006 IPCC Guidelines for National Greenhouse Gas Inventories: Wetlands	Intergovernmental Panel on Climate Change (IPCC)	2014	Carbon accreditation	Updates default data for estimation of carbon stock changes in mangroves, living biomass and dead wood pools for coastal wetlands, CO_2 emissions and removals from organic and mineral soils (for extraction, drainage and rewetting, and revegetation activities), and default data for the estimation of anthropogenic CO_2 emissions and removals from wetland soil. It addresses N_2O emissions from aquaculture and CH_4 emissions from rewetting and revegetation of mangroves and tidal salt marshes.
Methodology for Coastal Wetland Creation (VM0024)	Louisiana Coastal Protection and Restoration Authority	2014	Carbon accreditation	This methodology quantifies the greenhouse gas benefits of wetland creation activities. The scope of this methodology includes two primary project activities—substrate establishment and vegetation establishment—typically implemented in combination in order to create new wetlands (e.g., to restore wetlands that have degraded to open water). The methodology also allows for implementation of either project activity individually.

2 Only the lead organization is listed

TITLE	ORGANI-ZATION[2]	DATE RELEASED	CATEGORY	GOAL OF THE DOCUMENT
Restoration of Degraded Deltaic Wetlands of the Mississippi Delta	Tierra Resources LLC	2013	Carbon accreditation	Details procedures for GHG emission reduction accounting from wetland restoration activities implemented on degraded wetlands of the Mississippi Delta. The modular format provides flexibility for numerous types of wetland restoration projects (including those that require hydrologic management), and allows the user to decide whether wetland loss will be included in the baseline.
Afforestation and Reforestation of Degraded Mangrove Habitats (AR-AM0014	Clean Development Mechanism (CDM)	2013	Carbon accreditation	Outlines CDM-approved procedures to estimate net GHG emission reductions and removals resulting from afforestation or reforestation of mangroves. Project activities applying this methodology may choose to exclude or include accounting of any of the carbon pools of dead wood and soil organic carbon, but cannot include the litter carbon pool.
Simplified Baseline and Monitoring Methodology for Small Scale CDM Afforestation and Reforestation Project Activities Implemented on Wetlands (AR-AMS000)	CDM	2013	Carbon accreditation	Outlines CDM-approved procedures to estimate net GHG emission reductions and removals resulting from afforestation or reforestation of wetlands following the simplified modalities for small-scale projects under the CDM.
REDD+ Methodology Modules (VM0007)	Avoided Deforestation Partners	2010–2015	Carbon accreditation	Intends to cover the entire range of project activities eligible under three VCS project categories (reducing emissions from deforestation and forest degradation (REDD), reforestation and revegetation activities (ARR), wetlands restoration or conservation (WRC)), or combinations of these, providing maximum flexibility in the use of accounting procedures in complex settings where conservation and rehabilitation are combined, as well as in single category interventions. Under the WRC banner, peatland conservation and rewetting procedures are included in 2014, while coastal wetlands will be added in 2015.

TITLE	ORGANI-ZATION[2]	DATE RELEASED	CATEGORY	GOAL OF THE DOCUMENT
Guiding Principles for Delivering Coastal Wetland Carbon Projects (working title)	United Nations Environment Programme (UNEP), Center for International Forestry Research (CIFOR)	Expected 2014	Guidance on blue carbon measurement and project design	Draws together experience in carbon project and coastal wetland project development to demonstrate best practice principles in enacting blue carbon interventions. These interventions may range from policy activities leading to improved management of coastal resources recognizing climate change mitigation along with other ecosystem service, through to projects supported by carbon finance. The guidance is based upon experience developed by the project team supplemented by field missions and interviews.
Blue Carbon Practice Manual (working title)	RAE, Silvestrum	Expected 2014	Guidance on blue carbon measurement and project design	Provides detailed guidance on how to apply RAE's Methodology for tidal wetlands and seagrass restoration and develop a blue carbon project under the VCS standard.
Building Blue Carbon Projects: An Introductory Guide	Abu Dhabi Global Environmental Data Initiative (AGEDI)	2014	Guidance on blue carbon measurement and project design	Aims to stimulate discussion regarding projects that support the conservation and restoration of coastal ecosystems based on a Blue Carbon approach. It serves as a snapshot of potential common blue carbon project elements based on existing projects and an introduction of key issues for consideration. The guide is intended to complement existing blue carbon reports and initiatives and potentially stimulate support for further project development.
Protocols for the measurement, monitoring and reporting of structure, biomass and carbon stocks in mangrove forests	CIFOR	2012	Guidance on blue carbon measurement and project design	Describes the approaches necessary for the measurement, monitoring and reporting of structure, biomass and carbon stocks in mangrove forests. Because of their value as carbon stocks and sinks and their numerous other benefits, mangroves could be excellent candidates for carbon mitigation programs including REDD+ and Enhancing Forest Carbon Stocks in Developing Countries.

APPENDIX B
Equations

Chapter 1

Total Carbon (MgC/ha) * Area (ha) = Tier 1 total carbon stock for the project site (Mg)

- Where Total Carbon = the mean carbon stock for a given ecosystem (from **Table 1.2**)
- Area = the area of the ecosystem being investigated

Total potential CO_2 emissions per hectare (Mg CO_2/ha) = Conversion factor for the CO_2 that can be produced from the carbon present in the system * carbon in the system

- Conversion factor = 3.67, the ratio of the molecular weights of CO_2 (44) and carbon (12)
- Carbon in the system = the mean carbon stock for a given ecosystem

Chapter 3

Compaction correction factor = length of the sample recovered (cm) / length of core penetration (cm)

Corrected sampling of a compressed core = depth interval * compaction correction factor

Dry bulk density (g/cm^3) = Mass of dry soil (g) / Original volume sampled (cm^3)

Pre-dried volume of soil sample = [π * (radius of core barrel)^2] * (height of the sample, h)

% Loss on Ignition (% LOI) = [(dry mass before combustion (mg) – dry mass after combustion (mg)) / dry mass before combustion (mg)] * 100

% Inorganic Carbon (% IOC) = [((dry mass before acid treatment (g) – dry mass after acid treatment (g)) * 0.12) / dry mass before acid treatment (g)] * 100

- Where 0.12 is derived from the contribution of carbon to carbonate's molecular weight (12%)

Organic carbon content of a sample = Total carbon content (elemental analyzer or LOI %) – (Inorganic carbon content of ashed subsample * (Weight of subsample after ashing/Dry weight before ashing)

Soil carbon density (g/cm^3) = dry bulk density (g/cm^3) * (% C_{org}/100)

Amount carbon in core section (g/cm^2) = Soil carbon density (g/cm^3) * thickness interval (cm)

Core #1 carbon content = Amount carbon in core section A (g/cm^2) + Amount carbon in core section B (g/cm^2) + Amount carbon in core section C (g/cm^2) + …. all the samples from a single core

Total core carbon (MgC/hectare) = Summed core carbon (g/cm^2) * (1 Mg/1 000 000 g) * (100 000 000 cm^2/1 hectare)

Average carbon in a core = Carbon content for core #1 (determined in step 4) + Carbon content for core #2 + Carbon content for core #3+…. n) / n

Standard Deviation between Cores $$(\sigma) = \left[\frac{(X_1 - \overline{X})^2 + (X_2 - \overline{X})^2 + \ldots (X_n - \overline{X})^2}{(N-1)} \right]^{1/2}$$

- $\overline{X}$ = average carbon in a core
- X_1 = individual result for core #1, in MgC/hectare; X_2 = individual result for core #2, in MgC/hectare, etc.,
- N = total number of results

Total organic carbon in a project area (MgC) = (average core carbon from Statum A (MgC/hectare) * area Statum A (hectares)) + (average core carbon from Statum B (MgC/hectare) * area Statum B (hectares) + …

Standard Deviation between Strata $$(\sigma_T) = \sqrt{(\sigma_A)^2 + (\sigma_B)^2 + \ldots (\sigma_N)^2}$$

- Where σ_T = the total variability associated with the measurements,
- σ_A = standard deviation of the core average MgC for stratum A * area of stratum,
- σ_B = standard deviation of the core average MgC for stratum B * area of stratum, and
- σ_N = standard deviation of the core average MgC for remaining stratum * area of each individual stratum

Chapter 4

General Biomass equation for mangroves (Americas) = $0.168*\rho*(D)^{2.471}$

- ρ = wood density (g/cm^3)
- D = diameter at breast height

General Biomass equation for mangroves (Asia) = $0.251*\rho*(D)^{2.46}$

- ρ = wood density (g/cm^3)
- D = diameter at breast height

General Biomass equation for mangroves = $0.0509*\rho*(D)^2*H$

- ρ = wood density (g/cm^3)
- D = diameter at breast height
- H = height

Biomass for lianas (kg) = (Diameter 130 cm from the soil surface (cm))$^{2.657}$ * $e^{0.968}$ x ln (Diameter 130 cm from the soil surface (cm))

Biomass for pneumatophores (kg) = Average dry mass of sampled pneumatophores * number of pneumatophores in the microplot

Biomass of litter (kg) = (dry mass of subsample (g) / wet mass of the subsample (g)) * wet mass of all the litter in the sample plot (kg)

Downed wood biomass (kg/ha) = volume (m^3/ha) * average wood density (kg/m^3)

Belowground tree biomass (kgC) = 0.199 * ((wood density (g/cm^3)$^{0.899}$) * (tree diameter at breast height (cm))$^{2.22}$

Carbon content of vegetation (kg C) = biomass (kg) * carbon conversion factor

- Conversion factor mangrove trees = 0.46–0.5
- Conversion factor scrub mangrove trees = 0.46–0.5
- Conversion factor dead standing mangrove trees = 0.5
- Conversion factor lianas = 0.46
- Conversion factor palm fronds = 0.47
- Conversion factor palm trees = 0.47
- Conversion factor pneumatophores = 0.39
- Conversion factor litter (mangroves/marshes) = 0.45
- Conversion factor litter (seagrass) = 0.34
- Conversion factor downed wood = 0.5
- Conversion factor belowground tree components = 0.39
- Conversion factor marsh grass = 0.45
- Conversion factor marsh shrubs = 0.46–0.5
- Conversion factor seagrass = 0.34

Carbon in total vegetation component (kg C/m^2) = (carbon content of plant #1 + carbon content of plant #2 + ……. Plant #n) / area of the plot (m^2)

Elliptical crown area = (W1 * W2/2)^{2}*π

- W1 = Widest length of canopy
- W2 = Canopy width perpendicular to W1

Crown Volume = Elliptical crown area * crown depth

Estimating the top-diameter of a broken-topped dead tree (cm) = the measured basal diameter (cm) – [100 * tree height (m) * ((the measured basal diameter (cm) – diameter at breast height (cm) / 130)]

Dead tree volume (cm^3) = [π * (100 x tree height (m)) / 12] * [base diameter (cm)^2 + top diameter (cm)^2 + (base diameter (cm) x top diameter (cm))]

Decay Status 3 dead tree biomass (kg) = Volume of the dead tree (cm^3) * wood density (g/cm^3)

Wood density (g/cm^3) = Dry weight (g) / volume of fresh wood (cm^3)

Quadratic mean diameter (cm) = $\sqrt{(\sum}$diameter of each piece of wood^2) / number of pieces sampled

Wood volume for fine, small, and medium classes per unit of ground area (m^3/ha) = (π^2 x [number of samples x quadratic mean diameter for the size class (cm))^2 / (8 x transect length (m))]

Wood volume of large (> 7.6 cm diameter) down wood per unit of ground area (m^3/ha) = π^2 * [$\sum$diameter of each piece of wood^2 / (8 x transect length (m))]

Vegetative component carbon pool (Mg C/ha) = Carbon density (kg C/m^2) * (Mg/1000 kg) * (10 000 m^2/ha)

Total vegetative carbon in a plot (Mg C/ha) = component #1 (Mg C/ha) + component #2 (Mg C/ha) + component #3 (Mg C/ha) + …

Average vegetative carbon in a plot = Total vegetative carbon for plot #1 (Mg C/ha) + Total vegetative carbon for plot #2 (Mg C/ha) + Total vegetative carbon for plot #3 (Mg C/ha) +…. n) / n

Standard Deviation between plots $$(\sigma) = \left[\frac{(X_1 - \overline{X})^2 + (X_2 - \overline{X})^2 + \ldots (X_n - \overline{X})^2}{(N-1)} \right]^{1/2}$$

- $\overline{X}$ = average vegetative carbon in a plot
- X_1 = individual result for plot #1, in MgC/hectare; X_2 = individual result for plot #2, in MgC/hectare, etc.,
- N = number of plots

Estimate of vegetative carbon in a stratum (Mg C) = Average vegetative carbon in a plot (Mg C/ha) * area of stratum (ha)

Total carbon in a project area (MgC) = Estimate of vegetative carbon in stratum #1 (Mg C) + Estimate of vegetative carbon in stratum #2 (Mg C) + Estimate of vegetative carbon in stratum #3 (Mg C) + …

Standard Deviation between strata $$(\sigma_T) = \sqrt{(\sigma_A)^2 + (\sigma_B)^2 + \ldots (\sigma_N)^2}$$

- Where σ_T = the total variability associated with the measurements
- σ_A = standard deviation of the core average MgC for stratum A * area of stratum
- σ_B = standard deviation of the core average MgC for stratum B * area of stratum
- σ_N = standard deviation of the core average MgC for remaining stratum * area of each individual stratum

Stock-difference method = total carbon stock at T2 (sum of all carbon pools) – total carbon stock at initial measurement T1 (sum of all carbon pools)

- T1 = initial assessment
- T2 = subsequent assessments

Gain-loss method = Carbon stock at T1 – (carbon losses at T2 (land use change, natural disasters, erosion, etc.) + carbon gains at T2 (soil accretion, growth, restoration, etc.))

Soil elevation changes = vertical accretion based on the marker horizon – elevation changes based on SET measurements

Annual change in carbon stock (Mg C/year) = (total carbon stock at T2 – total carbon stock at initial measurement T1) / (T2 – T1)

Gas molecules in the static chamber (moles) = (Pressure (atmos) * Volume of the chamber (L)) / (Gas Constant (L*atmos/K*mol) * Temperature (Kelvins))

- Pressure = 1 atmos
- Gas constant = 0.0820 L*atmos/K*mol
- Temperature = 273 + temp in ºC

Gas flux of a specific GHG (µmole/minute) = µmole/mole/minute of GHG * moles of gas molecules total in the chamber

Amount of specific GHG emitted per minute per unit area (µmole/m^2/min) = Gas flux of a specific GHG (µmole/minute) / Chamber area

Specific GHG emitted over time (Mg/ha/day) = Amount of specific GHG emitted per minute per unit area (µmole/m^2/min) * (10 000 m^2/1 ha) * (1 mole/1 000 000 µmole) * (molecular weight of GHG (g)/1 mole) * (1 Mg/1 000 000 g) * (1440 min/1 day)

C

APPENDIX C
Example

I have a project area located in a salt marsh and it is comprised of 3 strata (76, 186, and 253 hectares respectively). I want to know the total amount of blue carbon found in the top one meter of soil and vegetation and the potential emissions that could be released if I convert this area to waterfront hotels.

In each stratum I took soil core samples from 3 plots, 3 cores per plot; each core was a total of 1 meter in length, using a highly aggregated sampling scheme, entire core sections were homogenized and subsamples were removed for a total of 5 subsamples. I am sending the samples to an outside lab for elemental analysis and will determine inorganic carbon content using the acidification technique.

FOR STRATUM #1, PLOT #1, CORE #1, SAMPLE A

Dry bulk density

- Volume of the sample = 125 cm^3
- Dry mass of the sample = 100 g
- 100 g /125 cm^3 = **0.8 g/cm^3**

Organic carbon content (using a subsample of sample A)

- Dry mass of subsample = 150 mg
- Elemental analyzer results = 25% C_{org}
- Organic carbon content
  - 150 mg * 0.25 = **37.5 mg**

Inorganic carbon content (using a subsample of sample A and acid technique)

- Dry mass of subsample = 150 mg
- Dry mass of subsample after acid treatment = 116 mg
- Mass of carbonate (inorganic carbon is in the form of carbonates such as calcium carbonate, $CaCO_3$)
  - 150 mg – 116 mg = 34 mg
- Mass of inorganic carbon component of carbonate (carbon makes up 12% of the molecular weight of calcium carbonate ($CaCO_3$)
  - 34 mg * 0.12 = 4.08 mg
- Percent inorganic carbon
  - (4.08 mg /150 mg)*100 = **2.72%**

Organic carbon content, correcting for inorganic carbon component

- Elemental analyzer determined organic carbon content = 37.5 mg
- Percent carbon that originated from carbonate = 2.72%
- Amount of the carbon content estimated by the elemental analyzer was carbon from carbonate
  - 37.5 mg * 0.0272 = 1.02 mg
- Actual organic carbon content
  - 37.5 mg – 1.02 mg = 36.48 mg
  - (36.48 mg /150 mg) * 100 = **24.32%**

Soil carbon density

- Dry bulk density = 0.8 g/cm^3
- Organic carbon content = 24.32%
- 0.8 g/cm^3 * (0.2432) = **0.195 g/cm^3**

Carbon content per sample

- Soil carbon density = 0.195 g/cm^3
- Sample thickness = 5 cm
- 0.195 g/cm^3 * 5 cm = **0.975 g/cm^2**

REPEAT FOR ALL SUBSAMPLES FROM CORE #1

Estimated carbon per core

- Sample A = 0.975 g/cm^2; Sample B = 0.865 g/cm^2; Sample C = 0.659 g/cm^2; Sample D = 0.510 g/cm^2; Sample E = 0.452 g/cm^2
- Total length of the core = 100 cm
- (0.975 g/cm^2 + 0.865 g/cm^2 + 0.659 g/cm^2 + 0.510 g/cm^2 + 0.453 g/cm^2) / 5 = 0.692 g/cm^2
- 0.692 g/cm^2 * 100 cm = **69.2 g/cm^2**

Convert Soil carbon density to MgC/ha

- Total carbon content in the core = 69.2 g/cm^2
- 1 Mg = 1 000 000 g
- 1 hectare = 100 000 000 cm^2
  - 69.2 g/cm^2 * (Mg/1 000 000 g)*(100 000 000 cm^2/ha) = **6920 Mg/ha (for the top meter of soil)**

REPEAT FOR ALL CORES

Average carbon stock per stratum

- Core #1 = 6920 Mg/ha
- Core #2 = 5018 Mg/ha
- Core #3 = 6111 Mg/ha
- (6920 Mg/ha + 5018 Mg/ha + 6111 Mg/ha)/3 = **6016 Mg/ha**

Standard deviation in carbon stock measurements

- Average carbon content per core = 6016 Mg/ha
- Number of cores taken per stratum = 3
- $[((6920\ Mg/ha - 6016\ Mg/ha)^2 + (5018\ Mg/ha - 6016\ Mg/ha)^2 + (6111\ Mg/ha - 6016\ Mg/ha)^2) / (3-1)]^{1/2}$ = **954 Mg/ha**

REPEAT FOR ALL STRATA

Total organic carbon in the project area

- Stratum #1 = 6016 Mg/ha; area = 76 ha
- Stratum #2 = 5342 Mg/ha; area = 186 ha
- Stratum #3 = 5826 Mg/ha; area = 253 ha
- (6016 Mg/ha * 76 ha) + (5342 Mg/ha * 186 ha) + (5826 Mg/ha * 253 ha) = **2 924 806 Mg C**

Standard deviation in carbon stock measurements

- Stratum #1 = 6016 ± 954 Mg/ha C
- Stratum #2 = 5342 ± 1265 Mg/ha C
- Stratum #3 = 5826 ± 1227 Mg/ha C
- $(954^2 + 1265^2 + 1227^2)^{1/2}$ = **2004**

THE SOIL CARBON POOL FOR MY PROJECT AREA IS:

2 924 806 ± 2004 MgC

The vegetation in all three strata consists of grasses, roots and rhizomes, and leaf litter. I spent all my funding sending soil samples to a lab for analysis by elemental analyzer. So the carbon content will be based on carbon conversion factors found in the literature. The most accurate numbers I could find are based on a study done about 600 km south with similar species.

Three plots of 20 m x 20 m were set up per strata, and each plot had six microplots of 30 cm x 30 cm.

FOR STRATUM #1, PLOT #1, MICROPLOT #1

Grass Component:

Develop an allometric equation

- 110 grass stems were collected
- The height of each stem (living portion) was measured and the biomass after heating was determined

STEM ID	HEIGHT (cm)	BIOMASS (g)
1	15	0.36
2	23	0.51
3	46	1.17
… n		

- Results were plotted with height on the x-axis and biomass on the y-axis
- Regression analysis was done to determine a relationship between height and biomass using the Microsoft Excel program
  - $Y = -0.006 (height) + 0.0002 (height)^2$
  - $R^2 = 0.91$
  - Y = biomass
- The biomass for all other stems in all the other microplots can now be found based on height alone

Carbon content of the grass

- Sum the biomass of each stem (as determined by allometric equation)
  - Stem #1 Biomass (g) + Stem #2 Biomass (g) Stem #3 Biomass (g) + …… Stem #n Biomass (g) = Biomass of the grass in the microplot
  - 0.36 g + 0.51 g + 1.17 g + ……. n = 74.8 g
- Carbon in the grass component (g/cm^2) = (Total estimated biomass * carbon conversion factor (0.45)) / area of the microplot (cm^2)
  - (74.8 g * 0.45) / (30 cm * 30 cm) = 0.0374 g/cm^2

Root and Rhizome component:

- Vegetative material was collected from a 1 meter soil core washed over a 1 mm screen, weighed and was found to be 27.8 g, dried to a constant weight and weighed again and was found to be 14.3 g
- Biomass (g) = dry mass (g) / wet mass (g)
  - 14.3 g / 27.8 g = 0.51 g

- Biomass of roots and rhizomes per core (g/cm^2) = biomass (g) / area sampled (based on core diameter)
  - Biomass = 0.51
  - Core diameter = 10 cm
  - Area = πr^2
    - 3.14 * 52 = 78.5 cm^2
  - 0.51 g / 78.5 cm^2 = 0.006 g/cm^2

Carbon content in the roots and rhizome component

- Carbon in the root and rhizome component (g/cm^2) = Biomass per core (g/cm^2) * carbon conversion factor (0.34)
  - 0.006 g/cm^2 * 0.34 = 0.002 g/cm^2

Leaf litter component:

- Biomass of leaf litter (g) = (dry mass of subsample (g) / wet mass of subsample (g)) * wet mass of all the litter in the microplot
  - Subsample wet weight = 13 g
  - Subsample dry weight = 9.8 g
  - All leaf litter in the microplot wet weight = 40.3 g
  - (9.8 g / 13 g) * 40.3 g = 30.4 g

Carbon content in the leaf litter

- Carbon in the leaf litter (g/cm^2) = (leaf litter biomass * carbon conversion factor (0.45)) / area of the microplot (cm^2)
  - (30.4 g * 0.45) / (30 cm * 30 cm) = 0.015 g/cm^2

Total vegetative carbon

- Total carbon = grass carbon component (g/cm^2) + root and rhizome carbon component (g/cm^2) + leaf litter carbon component (g/cm^2)
  - 0.0374 g/cm^2 + 0.002 g/cm^2 + 0.015 g/cm^2 = 0.054 g/cm^2

REPEAT FOR ALL MICROPLOTS

Average carbon per plot (6 microplots per plot)

- Microplot #1 = 0.054 g/cm^2
- Microplot #2 = 0.124 g/cm^2
- Microplot #3 = 0.982 g/cm^2
- Microplot #4 = 1.222 g/cm^2
- Microplot #5 = 1.450 g/cm^2
- Microplot #6 = 0.073 g/cm^2
- (0.054 g/cm^2 + 0.124 g/cm^2 +0.982 g/cm^2+ 1.222 g/cm^2+ 1.450 g/cm^2+ 0.073 g/cm^2) / 6 = 0.651 g/cm^2

Convert vegetative carbon to Mg/ha

- Total vegetative carbon = 0.651 g/cm^2
- 1 Mg = 1 000 000 g
- 1 hectare = 100 000 000 cm^2
- 0.651g/cm^2 * (Mg/1 000 000 g)*(100 000 000 cm^2/ha) = **65.1 Mg/ha**

REPEAT FOR ALL PLOTS

Average vegetative carbon per stratum

- Plot #1 = 65.1 Mg/ha
- Plot #2 = 76.9 Mg/ha
- Plot #3 = 79.3 Mg/ha
- (65.1 Mg/ha + 76.9 Mg/ha + 79.3 Mg/ha) / 3 = 73.7 Mg/ha

Standard deviation between plots in stratum #1

- Average vegetative carbon per plot = 73.7 Mg/ha
- Number of plots per stratum = 3
- $[((65.1 \text{ Mg/ha} - 73.7 \text{ Mg/ha})^2 + (76.9 \text{ Mg/ha} - 73.7 \text{ Mg/ha})^2 + (79.3 \text{ Mg/ha} - 73.7 \text{ Mg/ha})^2) / (3-1)]^{1/2}$ = **7.6 Mg/ha**

REPEAT FOR ALL STRATA

Total organic carbon in the project area

- Stratum #1 = 73.7 Mg/ha; area = 76 ha
- Stratum #2 = 85.9 Mg/ha; area = 186 ha
- Stratum #3 = 103.6 Mg/ha; area = 253 ha
- (73.7 Mg/ha * 76 ha) + (85.9 Mg/ha * 186 ha) + (103.6 Mg/ha * 253 ha) = **47 789 Mg C**

Standard deviation in carbon stock measurements

- Stratum #1 = 73.7 ± 7.6 Mg/ha C
- Stratum #2 = 85.9 ± 10.4 Mg/ha C
- Stratum #3 = 103.6 ± 18.3 Mg/ha C
- $(7.6^2 + 10.4^2 + 18.3^2)^{1/2}$ = **22.4**

THE VEGETATIVE CARBON POOL FOR MY PROJECT AREA IS:

47 789 ± 22.4 MgC

Total Carbon in the Ecosystem

- Total carbon = Soil carbon + vegetative carbon
  - Conservative estimate = (Soil carbon – standard deviation) + (vegetative carbon – standard deviation)
    - (2 924 806 MgC – 2004 MgC) + (47 789 – 22.4 MgC) = 2 970 569 MgC
  - High estimate = (Soil carbon + standard deviation) + (vegetative carbon + standard deviation)
    - (2 924 806 MgC + 2004 MgC) + (47 789 + 22.4 MgC) = 2 974 621 MgC
  - The total carbon stock for my project area is 2 972 595 ± 2026 MgC

Potential CO_2 emissions

- Potential CO_2 emissions = total carbon stock * 3.67 (conversion factor)
- 2 972 595 MgC * 3.67 = 10 909 420 ± 7435 Mg CO_2

THE TOTAL CARBON STOCK / CO_2 EMISSIONS FOR MY PROJECT AREA IS:

2 972 595 ± 2026 MgC

10 909 420 ± 7435 Mg CO_2

APPENDIX D
% LOI in Mangroves, Tidal Salt Marshes, and Seagrasses

D

% LOI IN MANGROVES

A positive, yet not particularly strong relationship (r^2 = 0.59) between the organic matter determined via % LOI and the carbon content (% C_{org}) has been found for mangrove soils (Kauffman *et al*. 2011), showing that roughly 40% of the organic matter (% LOI) was organic carbon (% C_{org}) (**Fig. D1**). In other locations a slightly different relationship may exist.

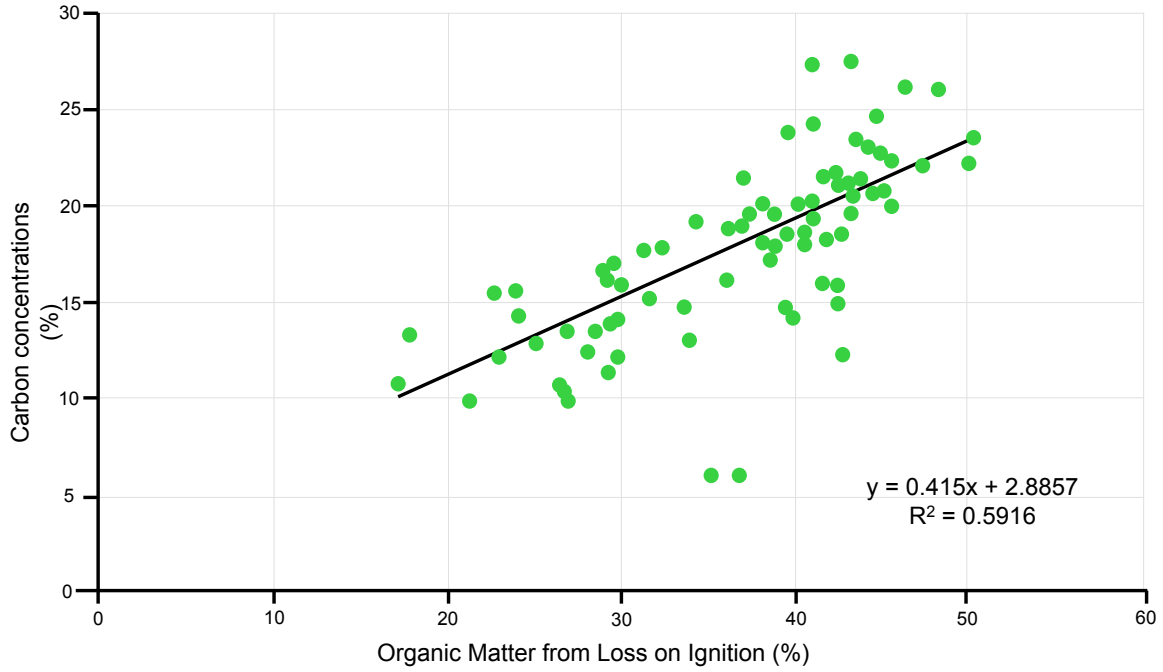


Figure D1 The relationship of organic matter calculated via loss on ignition to carbon concentration (percent) calculated via dry combustion for mangrove soil samples from the republic of Palau (Kauffman *et al*. 2011).

% LOI IN TIDAL SALT MARSHES

In tidal marshes in Maine, Craft et al. (1991) determined that in both mineral and organic rich marsh soils, pretreated for carbonate removal, and containing less than 11% clay, % LOI, as determined by heating samples for eight hours at 450 ºC, could successfully predict the organic carbon content (% C_{org}) in the soil (Craft *et al*. 1991). However, more recent studies conducted in similar tidal salt marshes in Maine reveal that there is some variability in the relationship between % C_{org} and % LOI. This new evidence shows that roughly 46% of the organic matter in marsh soils is organic carbon (**Fig. D2**). Because of the variability measured in various studies, we recommend whenever possible creating your own curve in the region of interest.

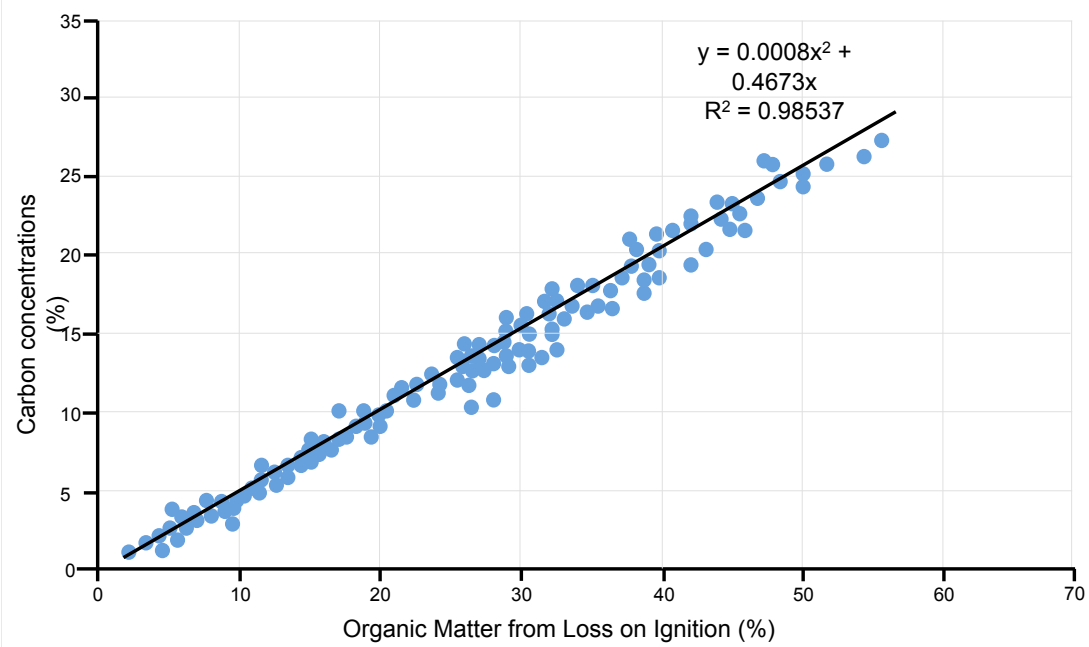


Figure D2 The relationship of organic matter (% LOI) with organic carbon (% OC) for tidal salt marsh samples in Maine (Johnson *et al*. in prep).

% LOI IN SEAGRASSES

In seagrass meadows, Fourqurean et al. reported that % LOI, as determined by heating samples for at least 3 hours at 550 ºC, was a good predictor of the % OC content in the soil (Fourqurean *et al*. 2012b). To improve the predictive capacity of the % LOI measurements two different linear equations were developed for samples with % LOI higher or lower than 0.2 % (**Fig. D3**).

For seagrass soils with % LOI < 0.20 % OC = -0.21 + 0.40 (% LOI);

For seagrass soils with % LOI > 0.20 % OC = -0.33 + 0.43 (% LOI).

Note that each of these equations has intercepts significantly different from zero (e.g., there is some losson ignition even in soils with no C_{org} content). It is likely that this loss represents the loss of water from mineral phases or oxidation of non-organic compounds. For the entire range of the data, the slope of the relationship between LOI and organic carbon was 0.43 and the intercept was -0.33 ± 0.02; indicating that samples with no organic carbon content would have a calculated LOI of 0.77% dry weight. In addition, Fourqurean et al. (2012a) observed that LOI is a less accurate proxy for organic carbon for soils with very low organic carbon contents.

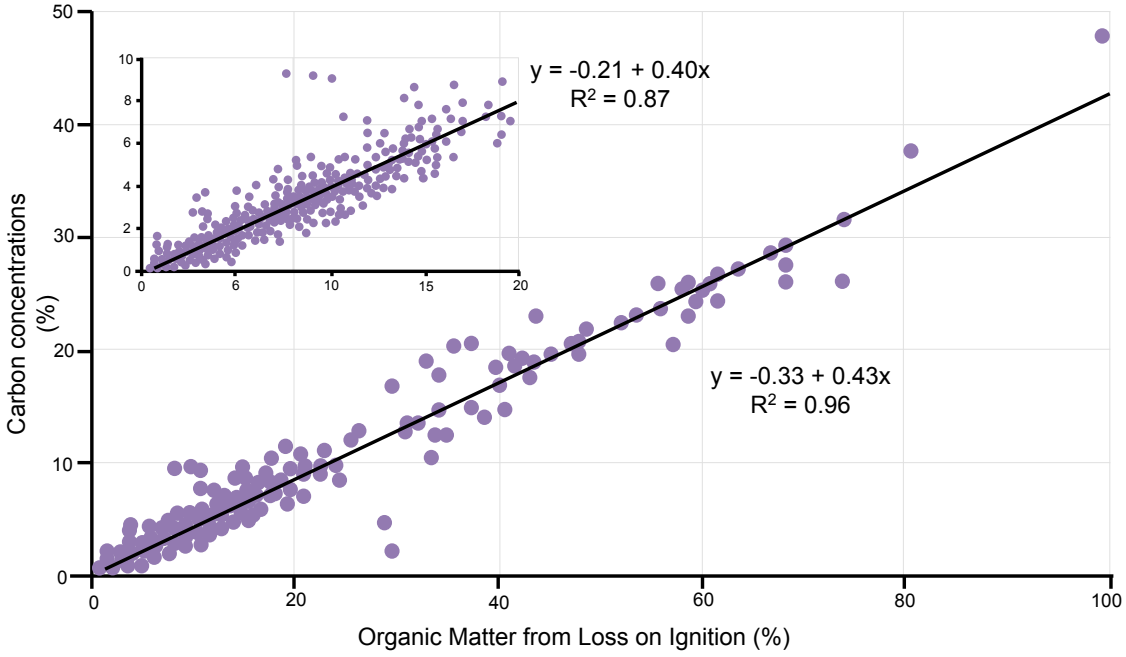


Figure D3 The relationship of organic matter (% LOI) with organic carbon (% OC) for seagrasses (Fourqurean *et al*. 2012b).

E

APPENDIX E

General Steps for Mapping Mangroves and Tidal Salt Marshes

Below is a streamlined methodology to map extent, height, and biomass of mangrove forests (Simard *et al*. 2006; Simard *et al*. 2008; Fatoyinbo & Simard 2013). However, the proposed method can be limited by current abilities to distinguish between tidal and adjacent inland ecosystems such as fresh water marshes and tropical forests.

STEP 1: IDENTIFY REGION POTENTIALLY INFLUENCED BY TIDAL INTRUSION.

A land cover map can be produced using optical instruments like Landsat and/or any other data in hand to identify the extent and geomorphology of the tidal wetlands. First, a mask of elevation should be produced to identify regions within the tidal range. Using SRTM elevation data with a threshold of about 40 meters (or expected maximum mangrove canopy height) or less than 2–5 meters for tidal salt marshes is sufficient. No tidal marshes exist above the tidal range.

STEP 2: MAP ECOSYSTEM EXTENT

Using Landsat imagery within the potential tidal range (Step-1), perform an isodata classification using any commercial or open-source software, or simply hand draw contours through visual interpretation.

STEP 3: MAP CANOPY HEIGHT

Produce a mask of mangrove or salt tidal marsh from the land cover map and crop the SRTM or TanDEM-X elevation map. Generally, both SRTM and TanDEM-X are referenced to sea level. To verify, identify any salt flat or bare ground areas within or adjacent to the mangrove or tidal flat so that elevation is within tidal range. Otherwise validate with field data as described later. Canopy height can be obtained from interferometric radar (inSAR) or Lidar data assuming ground elevation can be obtained from other datasets or assuming height is relative to mean sea level. For mangrove forest, Step-3 already provides a means of estimating canopy height. In the case of tidal salt marshes, one should use TanDEM-X relative to mean sea level and using neighborhood elevation measurement over salt flats and bare ground. Airborne Lidar may also be used to estimate ground elevation accurately.

STEP 4: MAP BIOMASS

There are several ways of estimating biomass through allometry relating biomass with a) radar backscatter, b) canopy cover from optical imagery, or c) lidar or inSAR-derived canopy height.

a) Radar backscatter (intensity of reflected microwave) can be used to estimate biomass within the wetlands. Assuming backscatter increases with biomass, it is possible to identify regions of low and high biomasses. However, radar backscatter tends to saturate at high biomass. The biomass level at which saturation occurs depends on the wavelength. At X-band, saturation can occur at very low biomass ~ 25t/ha, at C-band around 50t/ha, and L-band around 100t/ha. Other mechanisms related to flood level impact the backscatter. At microwave frequencies water acts are a mirror, enhancing reflection through the so-called double-bounce scattering mechanism. In other words, the radar pulse reflects both on the water and vertical component of the vegetation. This is just like throwing a ball to the foot of a wall to bounce on the floor (water) and then a wall (trunk). This phenomenon changes with water level and may complicate time-series analysis. However, given a single snapshot in time, a preliminary but spatially explicit map of biomass can be obtained using radar backscatter if current field estimates of biomass are available. It is recommended to use radar data obtained during low tide to maximize interaction with plants.

b) Vegetation cover is the fraction of land covered with plants. It can be derived from optical remote sensing given the spectral signature of vegetation and soil differ.

c) To obtain biomass estimates from remote sensing derived-height, allometry relating height and biomass must be available from field measurements.

APPENDIX F
Data Recording Worksheet for General Ecosystem Status

FIELD	Person/Institution (and contact information)			
	Date			
	Hour and tide information			
		Minimum	**Optimal**	**Ideal**
	Area	In hectares	% cover per area	Detailed distribution maps
	General Condition	Impacted/good/ pristine	Type of impact	Level, location, and description of impacts
	Substrate	Muddy, sandy, calcareous, etc.		Grain size per slice
	Water and sediment nutrient conditions	Oligotrophic/ eutrophic/cultural euthroficated	Mean value [N] Mean value [P]	Mean value [N] and mean value [P] with methods, time of the measurement
	Bathymetry	Position in the intertidal zone— low → high	Position relative to LAT or mean sea level (some fixed point)— method (unit)	Local bathymetry— digital elevation model with ecosystem mapped onto the bathymetry.
	Temperature	Average air temperature at a meteorological station close to the site of measurement		Water/air temperature at the site
	Salinity	Estuarine/marine	Single measurement of salinity at site	Multiple measurments of salinity at site over time

NOTES/COMMENTS

APPENDIX G
Data Recording Worksheet for Soil Samples

FIELD	Person/Institution (and contact information)	
	Date	
	Hour and tide information	
	Core ID	
	General location (area, country)	
	GPS position	
	Depth of water column (if applicable)	
	Coring device material	
	Internal diameter of the core	
	Total length of the corer (cm)	
	Coring system	
	Corer-end (cutting head/hypodermic)	
	Coring vertically (Y/N)?	
	Total length of corer outside sediment after core insertion (cm)	
	Total length of soil core (cm)	
	Sliced in X cm-slices (whole core or hemi-core?)	
	Total number of samples	

NOTES/COMMENTS
E.g., Coring issues? Sealing correct? Study site: Plant density/cover? Additional pictures of sampling site? Presence of shells, gravel, mud, plant debris, etc.
Visual description of the core (high-resolution digital picture)

CREATE A SHEET FOR EACH SAMPLE

LAB		
	Person/Institution	
	Date	
	Core ID	
	Sample ID	
	Slice depth (cm)	
	Slice thickness (cm)	
	Dry bulk density (g/cm^3)	
	Carbonate present (Y/N)?	
	Method used to determine inorganic carbon content	
	Inorganic carbon content (%)	
	Organic carbon content (corrected for inorganic portion, g)	

NOTES/COMMENTS
E.g., Any deviations from standard operating protocols? Any machinery malfunctions?

CREATE A SHEET FOR EACH CORE

LAB		
	Person/Institution	
	Date	
	Core ID	
	Corresponding sample IDs	
	Total carbon in core (MgC)	

G

FINAL CARBON ANALYSIS

Person/Institution	
Date	
Location	
Number of cores taken	
Average carbon content of the cores (MgC)	
Total area of strata (ha)	
Total soil carbon (per top X meters) of the strata (MgC/ha, in top X meters of soil)	

APPENDIX H
Data Recording Worksheet for Vegetation

FIELD		
	Person/Institution (and contact information)	
	Date	
	Hour and tide information	
	General location (area, country)	
	GPS position	
	Ecosystem (mangrove, marsh, seagrass)	
	Depth of water column (if applicable)	
	Vegetation	
	Mono or mixed	
	Dominant species	
	Ranked list of all species	
	Number of trees sampled	
	Number of shrubs sampled	
	Description of other components sampled	
	Lianas	
	Grasses	
	Pneumatophores	
	Litter	
	Deadwood	
	Other	
	Total number of samples	

NOTES/COMMENTS
E.g., Coring issues? Sealing correct? Study site: Plant density/cover? Additional pictures of sampling site? Presence of shells, gravel, mud, plant debris, etc.
Visual description of the core (high-resolution digital picture)

CREATE A SHEET FOR EACH SAMPLE

LAB		
	Person/Institution	
	Date	
	Sample ID	
	Sample type (wood, shrub, leaf litter, etc.):	
	Allometric equation used	
	Dry bulk density (g/cm^3)	
	Sample parameters (if applicable)	
	Height (m)	
	Diameter at breast height (cm)	
	Width (cm)	
	Volume (cm^3)	
	Decay status	
	Biomass (kg)	
	Organic carbon content (g)	

NOTES/COMMENTS
E.g., Any deviations from standard operating protocols? Any machinery malfunctions?

CREATE A SHEET FOR EACH PLOT

LAB		
	Person/Institution	
	Date	
	Plot ID	
	Corresponding sample IDs	
	Total carbon in plot	

FINAL CARBON ANALYSIS

Person/Institution	
Date	
Location	
Components being included	
Average vegetative carbon in plot (MgC)	
Total area of strata (ha)	
Total soil carbon in the strata (MgC/ha)	

A

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