

User Manual

Version 1.0

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Disclaimer

This user manual is meant to introduce users to the process of analyzing simulation data with MOSAICS. While we place some emphasis on the observables quantified, the main goal is to inform users of the options available to each program, thus enabling their engagement with the tools. This task is accomplished by describing the command line arguments and selection cards required to use each tool. We also describe the kind of data generated in each case. Examples of data generated are given in various figures. However, this data is intended for demonstrational purposes only and should not be interpreted as scientific reporting. We note that the user manual is an ongoing project, and your feedback is greatly appreciated. Please report any typos (or other mistakes) found in the manual, as well as bugs found in the source code, to nathan.bernhardt@nih.gov. Other comments and suggestions are welcome. Thank you for your help.

Chapter 1 Introduction

1.1 How to be Successful with MOSAICS

Making use of available resources is a key part of working with MOSAICS. The user manual is especially important since it contains information related to using the tools. On the other hand, the manual is quite large and parsing out which information should be considered and when is vital. We thus provide a few pointers to help users get started. To begin with, we recommend reading chapter 1 in its entirety. This chapter contains details that are common to most of the tools. For example, the MOSAICS Analysis Template (MosAT) is used when working with trajectory files. This topic is covered in detail in section 1.4. It is also common for MOSAICS tools to compute a spatially resolved time average of some observable and this topic is covered in section 1.11. The remaining contents of chapter 1 deal with the user interface and related topics. After completing this chapter, the user should be ready to use the tools. We recommend starting with the membrane thickness analysis measured with the program Z Coord (section 3.1). This analysis is suggested since Z Coord is simple to use, and many of the input arguments employed are common to the remaining tools. Of course, the user will need to prepare the trajectory before performing this analysis. We thus suggest reading section 2.1, which discusses several considerations that should be taken when prepping a trajectory. After measuring the membrane thickness, the user should have no problem using the remaining tools and may consult the remaining text as needed.

1.2 About MOSAICS

The relationship between lipid bilayers and their constituent protein is important in the field of biophysics. Yet the details of this relationship are, in many cases, unknown. Take, for example, the insertion of a protein into the cell membrane. It is well established that most membrane proteins have hydrophobic segments which span the bilayer interior. Despite this generality, the lengths of these regions do not always match the bilayer thickness perfectly. This difference is referred to as hydrophobic mismatch [1] and is hypothesized to affect protein structure. Given such a mismatch, one should expect a response from the solvating lipids. Indeed, molecular dynamics simulations have shown hydrophobic mismatch to disrupt the membrane thickness around proteins [2, 3]. In cases where the mismatch is asymmetric, the perturbations are highly localized [4-7] and are thought to coincide with an energetic penalty. If so, the system may find ways to eliminate the deformation, for example, by burying the problematic interface. This preference for eliminating the perturbation has been proposed as a force driving protein oligomerization in lipid bilayers [4], a phenomenon common to many membrane proteins [8]. In this model, the formation of the oligomer releases the solvating lipids from the defect, thus restoring their bulk characteristics. Testing this hypothesis can be aided with the use of molecular dynamics simulations. However, special tools are required to probe the lipids for changes in structure and dynamics as they maneuver around the protein. With this in mind, we have created Membrane Organization and Structure Analyzed and Interpreted with Computer Simulations or MOSAICS for short. MOSAICS is a collection of programs designed for characterizing molecular dynamics simulations of lipid bilayers. With these tools, many types of analysis are possible.

However, the primary focus of MOSAICS is on the characterization of lipid structure and dynamics based on the position of the lipids around an embedded protein.

The tools in MOSAICS are written in C++ and have been parallelized to take advantage of modern computing clusters. These are divided into two categories based on whether the tool reads a trajectory file or takes another form of data as input. For programs that work with trajectory files, the MOSAICS Analysis Template (MosAT) is used (see section 1.4). These programs are thus located in the MosAT directory, while the remaining tools are in the other-tools folder.

In the remainder of this chapter, we present prerequisite materials that should be viewed before using MOSAICS tools. The topics covered here include:

- Installing MOSAICS on a local cluster
- Reading and writing trajectory data with MosAT
- Conventions used for evaluating command line arguments
- Performing least squares fitting with MosAT
- Identifying lipids and sorting them based on the host leaflet
- Identifying other molecules such as the solvent or protein
- Making atom selections and interfacing data with an analysis tool using selection cards
- Computing averages using grid-interpolation
- Working with grid data
- Naming conventions used when multiple files are generated
- Focusing on a subset of the grid using a rectangular selection or masking file
- Identifying insignificant events using a noise filter

In Chapter 2, we discuss how trajectory files may be prepared for analysis and the tools used in this process. Then, in the remaining chapters, individual analysis tools are covered in detail. In Chapter 3, we focus on tools used for characterizing lipid structure. In Chapter 4, we switch to the characterization of lipid dynamics. And finally, Chapter 5 is dedicated to general-purpose programs such as those used when plotting data or averaging over multiple data sets.

1.3 Installation Instructions

Dependencies: MPI

MOSAICS may be installed on a computing cluster using the following CMake recipe:

```
$ cd MOSAICS
$ mkdir build
$ cmake src/MosAT/programs/ -S . -B build/
$ cmake --build build/
```

Alternatively, the tools may be installed using the `install_commands` file:

```
$ sh install_commands
```

Once completed, the user can check that the installation was successful by displaying the help options for one of the programs. This printing of help options is demonstrated in the following example.

```
$ mpirun -n 1 mosat_mpi -h
```

1.4 The Trajectory Reader/writer MosAT

MOSAICS uses publicly available libraries sourced from the GROMACS simulation package for reading/writing trajectory data and performing basic operations such as least squares fitting. These libraries are also used for calculations, such as measuring a molecule's root mean square deviation relative to a reference state. The basic trajectory reading routines are integrated into the workflow using object-oriented programming and are bundled together with other basic routines to form the MOSAICS Analysis Template (MosAT).

MosAT is a fully functional trajectory reading program, which may be built upon to create specialized analysis tools. For MOSAICS, all programs under the MosAT directory are built around MosAT and thus have a common foundation. With these tools and the MosAT program, a trajectory file may be read by specifying the file name using the -traj command line argument (see section 1.5 for details about command line arguments). Supported trajectory formats include xtc, trr, gro, and pdb. Because xtc files contain no information about the atom types, a reference file containing this information must be provided. This is done with the -ref tag. Currently supported reference files include the pdb and gro format. With a trajectory and reference file specified, MosAT will read each trajectory frame and extract the atomic coordinates as well as the atom names and numbers, etc. It should be noted that MosAT reads trajectory frames individually to minimize memory requirements. This design choice enables the analysis of very large trajectory files since the memory needed is independent of the number of frames. Additionally, all programs built around MosAT are fully parallelized using the message passing interface (MPI). To improve flexibility, MosAT supports two parallelization schemes, from which one was chosen when each tool was programmed. These include the "block parallelization" and "standard" schemes, the details of which we will discuss next.

For block parallelization, each MPI process is assigned a subset, also called a "block," of the trajectory for which it will analyze. These assignments are made sequentially such that MPI rank 0 might be responsible for reading the first ten frames while ranks 1 and 2 are responsible for 11-20 and 21-30, etc. (Figure 1-1). The "block" parallelization scheme is effective when each frame of the trajectory may be analyzed independently of the others. For example, when computing the average z-coordinate for the lipid head groups, each frame may be analyzed independent of the others so long as the data is collected afterward, and a proper average computed. In cases where each frame's analysis depends on another frame's result, the block parallelization scheme may be switched off, resulting in the "standard" scheme. With the standard scheme, each MPI process reads the complete trajectory. In this case, the workload may be distributed by a metric other than the trajectory frame. For example, each MPI process could be responsible for a subset of the grid when a grid-based analysis is performed or a subset of the lipids, etc. This flexibility enables the workload distribution to be tailored to each analysis tool based on the nature of the computation performed. Note that some analysis routines are difficult to parallelize, and MosAT may be set to run in serial.

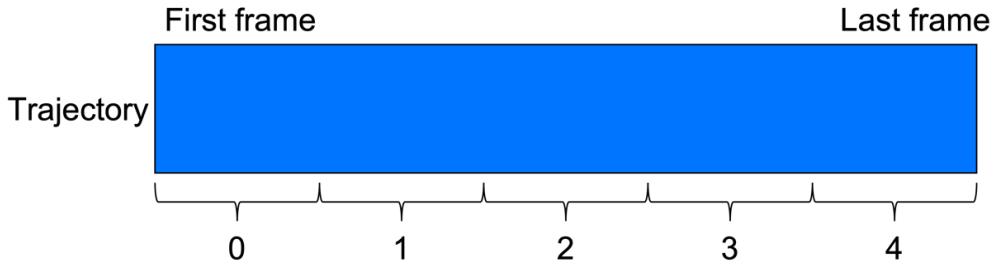


Figure 1-1 A schematic of the workload distribution for the block parallelization scheme. The blue rectangle represents frames in the trajectory and the numbers on the bottom the MPI processes. The brackets indicate the portion of the trajectory assigned to the MPI process.

MosAT has been programmed to allow for jumping between trajectory frames. This functionality is critical to the block parallelization scheme, where each MPI process skips to the first frame within its designated block. Unfortunately, the location of each trajectory frame is initially unknown. Because of this, the trajectory file must be analyzed, frame-by-frame, before the workload can be distributed. This initial examination of the trajectory results in a duplicate reading of the trajectory file, which may be justified when the benefits of offsetting the workload outweigh the cost of duplicate reading. Moreover, a workaround has been implemented that writes each frame's starting position to file upon first analyzing the trajectory. This data is written to a file containing the same name as the trajectory but succeeded with the .info extension. If a .info file already exists (upon repeated analysis), its contents are extracted, and the trajectory is no longer analyzed before distributing the workload. This workaround reduces the time spent analyzing the trajectory and becomes especially significant as the system size and the number of frames increase. It should be warned, however, that there is a level of risk associated with using info files. To see why, consider the following example where the user decides to extend their simulation after performing some initial analysis on a small trajectory. In this case, the user might go through the same process of preparing the extended trajectory for analysis, thus resulting in a new trajectory file with the same name as the previous one. In this case, the old .info file would be missing the frames added when the trajectory was extended. The user can check for this error by comparing the reported number of frames to that in their trajectory. Moreover, MosAT has been programmed to throw an error when unable to read a trajectory frame. With that said, we encourage the user to delete any .info files that may be questionable and let the analysis tool create a new one.

In addition to reading trajectories, MosAT can be used to write trajectory data. This option is selected by specifying the name of the output trajectory with the -o tag. Moreover, MosAT supports cross IO between the supported file types (xtc, trr, gro, and pdb). If the block parallelization scheme is selected, then a temporary trajectory file is written by each MPI process. These files are later spliced together. Furthermore, MosAT contains standard functionality such as a least-squares fitting procedure (see section 1.6), a stride for skipping frames, and a begin/end option for reading only a subset of the trajectory. These features are implemented so that frames falling between -b and -e are selected first, where -b and -e refer to the frame number. Then, frames that are a multiple of -stride are selected from this subset. And finally, MosAT will report any performance statistics before terminating. This information is typically broken down into several categories, as detailed below.

- **Analyze Trajectory:** Time spent analyzing the trajectory to get the position of each frame.
- **Main Loop:** Time spent in the main loop. This is where the main analysis is performed.
- **Finalize Trajectory:** Time spent splicing together temporary trajectory files.
- **Fin Ana:** Time spent collecting and processing data outside the main loop.
- **Total:** Total amount of time spent running the program.

It should be noted that the atom and residue numbering used inside MosAT does not follow that specified in the reference file. Instead, MosAT renames each atom and residue starting from 1, with each additional atom or residue increasing by one (Figure 1-2). This renaming guarantees each residue/atom has a unique identifier and is necessary since the reference file numbering can be redundant. For example, GROMACS indexes atoms up to 99,999 and residues to 9,999 before repeating the numbering. It is also possible to have a topology where the numbering begins with a positive integer other than 1. This potential difference in numbering is something to keep in mind when reporting data specific to an atom or residue that was generated with a MOSAICS tool. Similarly, when an analysis program takes a set of atom or residue numbers as input, the user must provide numbers that correspond to the internal numbering scheme. Examples include the program Protein Orientation (section 3.13), which takes a list of atom numbers that are used when defining the orientation vector. Another example includes the index provided when using the built-in least squares fitting procedure (see section 1.6). This index defines the atoms whose center is fixed during the rotation and must match the internal numbering scheme. One way to avoid confusion is to pass your reference file through MosAT using the -o option, as is shown in the example below:

```
$ mpirun -n 1 mosat_mpi -traj my_ref.gro -ref my_ref.gro -o my_ref_renumbered.gro
```

We note that the numbering in my_ref_renumbered.gro should match that used by MosAT internally and can be safely used to make an atom selection with tools like the GROMACS utility make_ndx.

Numbering Used In Sim

Gromacs Runs One Microsecond At Cannonball Speeds 99547						
6LEU	N	1	0.075	-3.270	1.454	
6LEU	H1	2	0.117	-3.360	1.463	
6LEU	H2	3	0.146	-3.199	1.460	
6LEU	CA	4	0.005	-3.259	1.321	
6LEU	HA	5	0.077	-3.275	1.242	
6LEU	CB	6	-0.084	-3.383	1.319	
6LEU	HB1	7	-0.149	-3.387	1.410	
6LEU	HB2	8	-0.022	-3.474	1.318	
6LEU	CG	9	-0.177	-3.385	1.199	
6LEU	HG	10	-0.247	-3.299	1.201	
6LEU	CD1	11	-0.085	-3.374	1.064	
6LEU	HD11	12	-0.065	-3.267	1.041	
6LEU	HD12	13	-0.145	-3.412	0.979	
6LEU	HD13	14	0.011	-3.430	1.073	
6LEU	CD2	15	-0.267	-3.510	1.282	
6LEU	HD21	16	-0.196	-3.595	1.211	
6LEU	HD22	17	-0.338	-3.517	1.117	
6LEU	HD23	18	-0.327	-3.501	1.295	
6LEU	C	19	-0.065	-3.124	1.292	
6LEU	O	20	-0.030	-3.054	1.202	
7TRP	N	21	-0.162	-3.086	1.378	
7TRP	HN	22	-0.188	-3.143	1.455	
7TRP	CA	23	-0.246	-2.961	1.364	
7TRP	HA	24	-0.274	-2.956	1.260	
7TRP	CB	25	-0.372	-2.985	1.449	
7TRP	HB1	26	-0.451	-2.915	1.413	
7TRP	HB2	27	-0.344	-2.961	1.554	
7TRP	CG	28	-0.439	-3.113	1.424	
7TRP	CD1	29	-0.449	-3.210	1.520	
7TRP	HD1	30	-0.433	-3.187	1.625	
7TRP	NE1	31	-0.505	-3.323	1.463	
7TRP	HE1	32	-0.549	-3.396	1.510	
7TRP	CE2	33	-0.532	-3.306	1.330	
7TRP	CD2	34	-0.492	-3.174	1.301	
7TRP	CE3	35	-0.502	-3.131	1.164	
7TRP	HE3	36	-0.488	-3.026	1.141	
7TRP	CZ3	37	-0.556	-3.214	1.069	
7TRP	HZ3	38	-0.589	-3.184	0.970	
7TRP	CZ2	39	-0.568	-3.392	1.233	
7TRP	HZ2	40	-0.571	-3.496	1.258	
7TRP	CH2	41	-0.585	-3.342	1.098	
7TRP	HH2	42	-0.621	-3.415	1.027	
7TRP	C	43	-0.172	-2.828	1.388	

Numbering Used In GromAT

Gromacs Runs One Microsecond At Cannonball Speeds 99547						
1LEU	N	1	0.075	-3.270	1.454	
1LEU	H1	2	0.117	-3.360	1.463	
1LEU	H2	3	0.146	-3.199	1.460	
1LEU	CA	4	0.005	-3.259	1.321	
1LEU	HA	5	0.077	-3.275	1.242	
1LEU	CB	6	-0.084	-3.383	1.319	
1LEU	HB1	7	-0.149	-3.387	1.410	
1LEU	HB2	8	-0.022	-3.474	1.318	
1LEU	CG	9	-0.177	-3.385	1.199	
1LEU	HG	10	-0.247	-3.299	1.201	
1LEU	CD1	11	-0.085	-3.374	1.064	
1LEU	HD11	12	-0.065	-3.267	1.041	
1LEU	HD12	13	-0.145	-3.412	0.979	
1LEU	HD13	14	0.011	-3.430	1.073	
1LEU	CD2	15	-0.267	-3.510	1.282	
1LEU	HD21	16	-0.196	-3.595	1.211	
1LEU	HD22	17	-0.338	-3.517	1.117	
1LEU	HD23	18	-0.327	-3.501	1.295	
1LEU	C	19	-0.065	-3.124	1.292	
1LEU	O	20	-0.030	-3.054	1.202	
2TRP	N	21	-0.162	-3.086	1.378	
2TRP	HN	22	-0.188	-3.143	1.455	
2TRP	CA	23	-0.246	-2.961	1.364	
2TRP	HA	24	-0.274	-2.956	1.260	
2TRP	CB	25	-0.372	-2.985	1.449	
2TRP	HB1	26	-0.451	-2.915	1.413	
2TRP	HB2	27	-0.344	-2.961	1.554	
2TRP	CG	28	-0.439	-3.113	1.424	
2TRP	CD1	29	-0.449	-3.210	1.520	
2TRP	HD1	30	-0.433	-3.187	1.625	
2TRP	NE1	31	-0.505	-3.323	1.463	
2TRP	HE1	32	-0.549	-3.396	1.510	
2TRP	CE2	33	-0.532	-3.306	1.330	
2TRP	CD2	34	-0.492	-3.174	1.301	
2TRP	CE3	35	-0.502	-3.131	1.164	
2TRP	HE3	36	-0.488	-3.026	1.141	
2TRP	CZ3	37	-0.556	-3.214	1.069	
2TRP	HZ3	38	-0.589	-3.184	0.970	
2TRP	CZ2	39	-0.568	-3.392	1.233	
2TRP	HZ2	40	-0.571	-3.496	1.258	
2TRP	CH2	41	-0.585	-3.342	1.098	
2TRP	HH2	42	-0.621	-3.415	1.027	
2TRP	C	43	-0.172	-2.828	1.388	

Figure 1-2 Gro files demonstrating how MosAT rennumbers atom/residue indices before any analysis can be performed. Left is an example of a numbering scheme used in a GROMACS simulation. Right is the numbering scheme used by MosAT on the resulting trajectory.

1.5 Command Line Arguments

The programs in MOSAICS take an approach to analyze command line arguments like that employed by GROMACS. Specifically, each argument is preceded by a tag containing a hyphen. To make this clear, consider the example where MosAT is used to extract the first 100 frames from a trajectory.

```
$ mpirun -n 1 mosat_mpi -traj my_traj.xtc -ref my_ref.gro -o my_traj_0_100.xtc -b 0 -e 100
```

In this example, the trajectory file to be read is preceded with the -traj tag. Similarly, the reference and output files are preceded by the -ref and -o tags and the first and last frames to be extracted with the -b and -e tags. Note, the user can view a list of possible command line arguments by running the program and including the -h tag as follows.

```
$ mpirun -n 1 mosat_mpi -h
```

This will print to screen all possible command line arguments as well as a short description of each, the required tag, whether the argument is required or optional, and the expected data type. We note that optional arguments are indicated with an O and required ones by an R. If an

argument is required but not provided by the user, then the program will report the error and terminate. Furthermore, the expected data types are labeled S, I, or F and correspond to string, integer, and a floating-point number, respectively. We note that MOSAICS screens any argument expecting an integer or floating-point number before the input is stored. Should the incorrect type be supplied, then an error message will be displayed, and the program terminated. This approach is taken to reduce errors by the user, especially those where the program continues to run but the output data is compromised. And finally, a short description of the program is also provided when the help option is included.

1.6 Least Squares Fitting

Each MOSAICS tool derived from MosAT contains a built-in least squares fitting procedure. This feature may be used on the fly since the rotations are performed on each frame before any observables are computed. Still, to avoid errors when using this feature, we must consider how the fitting procedure is related to the indexing scheme used by MosAT. Note, the indexing is explained in section 1.1 and the reader is encouraged to read that section before proceeding.

To use the least squares fitting procedure the user must provide an index file with a list of atom numbers. This specifies a group of atoms whose center is fixed during the rotation. Importantly, the atom numbers provided here should be consistent with the internal numbering scheme used by MosAT. An easy way to make this index is to first pass the reference file through MosAT. This will result in a new reference file whose indexing will be consistent with that seen by MosAT. This new reference file may be used with the `make_ndx` utility of GROMACS to make the final atom selection. This is demonstrated in the following example in which we select the alpha carbons from a protein molecule.

```
$ mpirun -n 1 mosat_mpi -traj ref.gro -ref ref.gro -o ref_renumbered.gro
```

```
$ printf 'a CA \n q\n' | make_ndx -f ref_renumbered.gro -o ca.ndx
```

Ca.ndx should contain a list of the protein alpha carbons which can be used when performing the least squares fitting. To use this data, we can copy the atom numbers from ca.ndx to a new file which we will call ca_lsq.ndx. Then, we provide ca_lsq.ndx to MosAT using the `-lsq` tag. In addition to this, least squares fitting requires a target structure for which the trajectory frames are to be fit. This structure is indicated using the `-lsq_r` tag. Currently, there are two options for the target structure. These are the structure contained in the reference file (`-lsq_r 0`) and the structure from the first trajectory frame (`-lsq_r 1`). In addition to this, the user can specify the dimension of the fit with the `-lsq_d` tag. For a full three-dimensional fit the user should use `-lsq_d 3`. However, GROMACS allows for a 2-dimensional fit (`-lsq_d 2`) in which the rotation is made around the z-axis only.

With the details of using the fitting routine out of the way, let us discuss some details of the procedure used by MosAT (Figure 1-3). To begin the process, the system is centered at the origin i.e., 0,0,0. This is accomplished by computing the center of mass $\vec{\mu}$ of the atom selection (`-lsq`) and translating the system such that this center sits at the origin. This step is done for both the target structure as well as the frame being fit. It should be noted that the results can vary depending on the format of the reference file used (`-ref`). This is because PDB files may contain

atomic mass data (taken as the B-factor) while gro files do not. This difference affects the computation of the center of mass. This is computed as:

$$\vec{\mu} = \frac{1}{M} \sum_{i=1}^n m_i \vec{r}_i \quad (1.1)$$

where m_i is the mass of atom i and M is the sum of mass over the selected atoms:

$$M = \sum_{i=1}^n m_i \quad (1.2)$$

When a gro file is provided as the reference file, the masses are assigned a value of 1 for all atoms. In this case, the mass cancels in the center of mass equation and a geometric center is computed instead:

$$\vec{\mu} = \frac{1}{n} \sum_{i=1}^n \vec{r}_i \quad (1.3)$$

Despite these differences, both options should result in a good fit. Once the system is centered at the origin a rotation matrix is applied and the system rotated. Then, in a final step, the center of the atom selection is moved to its final position. This is taken to be the original coordinates of the center which are taken from the first frame in the trajectory. This choice ensures that the center is located at the same position for all trajectory frames.

Note, it is generally a good idea to test that the fit has produced the desired result. This can be accomplished by taking a short snippet from the resulting trajectory and inspecting the fit using a graphics program such as PyMOL (Schrödinger, LLC) or VMD [9]. An example is given below.

```
$ mpirun -n 1 mosat_mpi -traj my_traj.xtc -ref ref.gro -lsq ca_lsq.ndx -lsq_r 0 -lsq_d 3 -o my_traj_fit.pdb -e 100
```

In the example above MosAT will read the first 100 frames of the trajectory and fit each frame to the reference structure. MosAT will then write out a PDB of the rotated system which can be used for visual inspection.

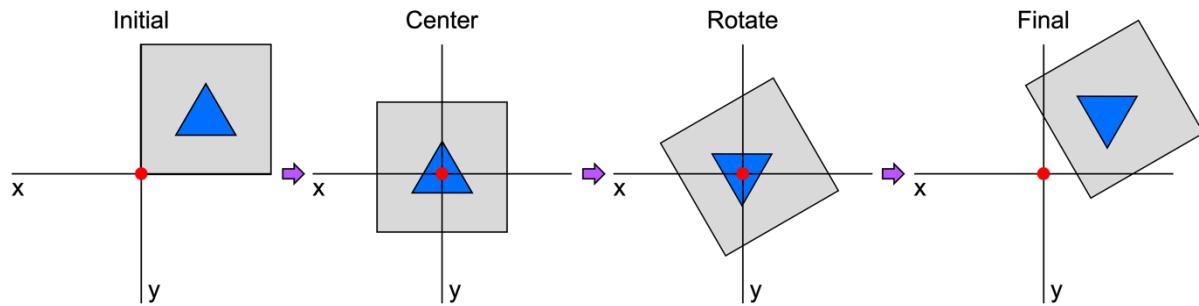


Figure 1-3 Least Squares Fitting protocol used by GROMACS. Panels moving from left to right are the initial system, the system with the protein centered at the origin, the rotated system, and the final system. The protein is shown as a blue triangle and the origin (0,0) as a red circle.

1.7 Selection Cards

For most types of analysis, it is necessary to define an atom selection before any observables can be measured. To facilitate this task, we have implemented into MOSAICS a parameter file reader. With this routine, data from a structured text file, which we refer to as a selection card, can be processed and instructions interpreted by the analysis tool. This makes it possible to define an atom selection for a lipid type without having to specify the individual lipids. This is accomplished by creating a selection card with N rows and M columns as is shown in the following example.

#lipid_t	#atom_i	#atom_j	#k(kJ/mol)	#r
POPC	D2A	C3A	1250	0.47
DLPC	C2A	C3A	1250	0.47

In the example considered here we have selected a pair of bonded atoms for a Martini POPC molecule as well as the corresponding pair for DLPC. We have also specified for each the spring constant and equilibrium bond position. With this information, a MOSAICS analysis tool could compute the potential energy of this bond for each POPC and DLPC molecule. It should be noted that entries beginning with the hashtag symbol (#) are ignored by the selection card reader. This lets the user label the rows and columns in their cards. An exception to this rule is the index file used for performing least squares fitting (-lsq) which uses a different routine for reading in data. We note that the format of each card is dependent on the analysis tool. For this reason, the details are provided for each tool separately in later sections.

The selection card approach may be sufficient when the necessary atom selections are simple. However, some analysis requires greater flexibility than others. For example, the computation of the lipid-solvent contacts lets the user define the lipid atoms participating in contacts. In this case, each lipid type might require a unique set of atoms to be used for analysis. When such complex atom selections are desired, a network of selection cards may be used. In this case, a single primary card is provided using the command line arguments. This card, like an ordinary selection card, should contain pieces of information like the lipid and atom types as well as other parameters. However, a single column of this card is reserved for filenames of other secondary selection cards. In the case of the lipid-solvent contacts, the atoms included in the contact analysis may be specified for each lipid type using these secondary selection cards. We note that the number of columns in each card is unique to the analysis performed and is

determined during the software development. In contrast, the number of rows may generally vary giving the user full control over the lipid types included in the analysis.

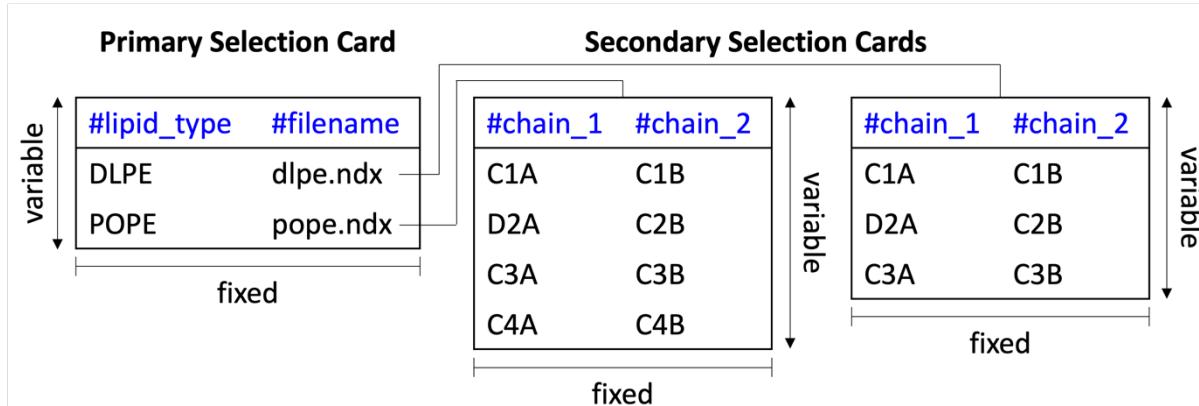


Figure 1-4 Example of a network of selection cards. In the example provided here the user has specified the atoms making the acyl-chains for POPE and DLPE Martini lipids. The network of cards approach thus enables the atoms making each acyl chain to be uniquely defined.

1.8 Leaflet Finder

Many MOSAICS tools require the selection of lipid molecules from which an observable is computed. Importantly, these calculations may be performed on each leaflet separately. To aid in this task, MOSAICS contains a built-in leaflet finder used for sorting lipids. This routine works by analyzing the membrane structure contained within the reference file. The program then assumes that no lipids flip over the course of the trajectory. If this is not the case an iterative procedure may be required (not yet supported).

The leaflet finder routine sorts lipids into an upper or lower leaflet. This is accomplished by comparing the z-coordinates of the lipid head group to that of the tail atoms. For lipids in the upper leaflet, taking this difference should result in a positive number. On the other hand, lipids in the lower leaflet should have a negative ΔZ (Figure 1-6). This approach works if the membrane is relatively flat and will be prone to error for highly curved membrane patches or vesicles. Still, because leaflet finder uses the structure contained within the reference file, the user can choose this structure so that the assignments are made correctly. For example, the user may provide a pre-equilibrated reference structure where the membrane patch is still highly ordered, and leaflet finder is likely to work. Of course, it is best practice to confirm that the leaflets have been correctly identified. This may be done by including the `-lf_pdb` command line argument which will have the analysis program write a PDB of the reference structure with the leaflets indicated by their B-factor (upper:1, lower:-1, non-lipids:0, see Figure 1-5). For most programs, the leaflet assignments may also be inspected by writing out a PDB of the trajectory using the `-o` tag. Like with the `-lf_pdb` tag, the leaflets will be indicated by the B-factor in the output trajectory.

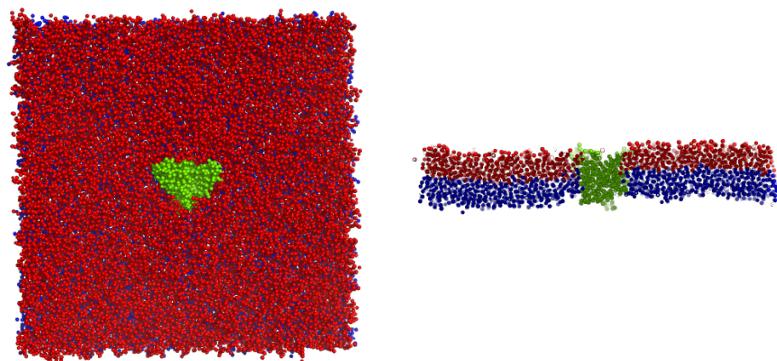


Figure 1-5 A PDB generated using the `-lf_pdb` tag indicating the selected leaflets (upper shown in red and the lower blue). The protein, which belongs to neither leaflet, is colored green. Colors were set according to the atom's B-factor.

It is also standard for programs using leaflet finder to report a summary of the leaflet selection. An example is given below.

There are 4 lipid types in the selected leaflet (upper) as summarized below:

index	type	count
0	POPE	423
1	POPG	210
2	DLPE	487
3	DLPG	238

In the example above leaflet finder selected the upper leaflet, identified the types of lipids making the leaflet and counted the number of lipids for each type. This information can be used to check that leaflet finder is working correctly. For example, if leaflet finder fails to identify one of the lipid types in your system, then that lipid will not be included in the report. We note that leaflet finder comes with several common lipid types pre-programmed and additional lipids may be added by construction of a parameter file. The new lipid types are provided by the user using the `-lf_prm` command line argument. An example is provided below.

```
-lf prm
#lip_t  #head_1  #tail_1  #head_2  #tail_2
POPE    PO4       C4A      PO4       C4B
POPG    PO4       C4A      PO4       C4B
```

In the example above we have added martini POPE and POPG lipid types to the leaflet finder. Here, the first column gives the lipid residue name while columns 2 and 3 give the atom names for the head and tail atoms respectively (tail 1). Columns 4 and 5 give the head and tail atoms for the second tail of the lipid. This brings us to an important point. With leaflet finder, both tails are analyzed, and ΔZ measured for each. To make the final assignment, the ΔZ of larger magnitude is used. This helps to reduce errors caused by splaying of the tails where it is possible for one of the tails to curl back toward the lipid head group. In this case the z-coordinate could become greater than that of the head atom, for example in the upper leaflet. This would cause an

improper leaflet assignment. However, it is unlikely for both tails to do this and the less curled tail would likely have a ΔZ of greater magnitude. Thus, the ΔZ with greater magnitude is more likely to make the correct assignment (Figure 1-6).

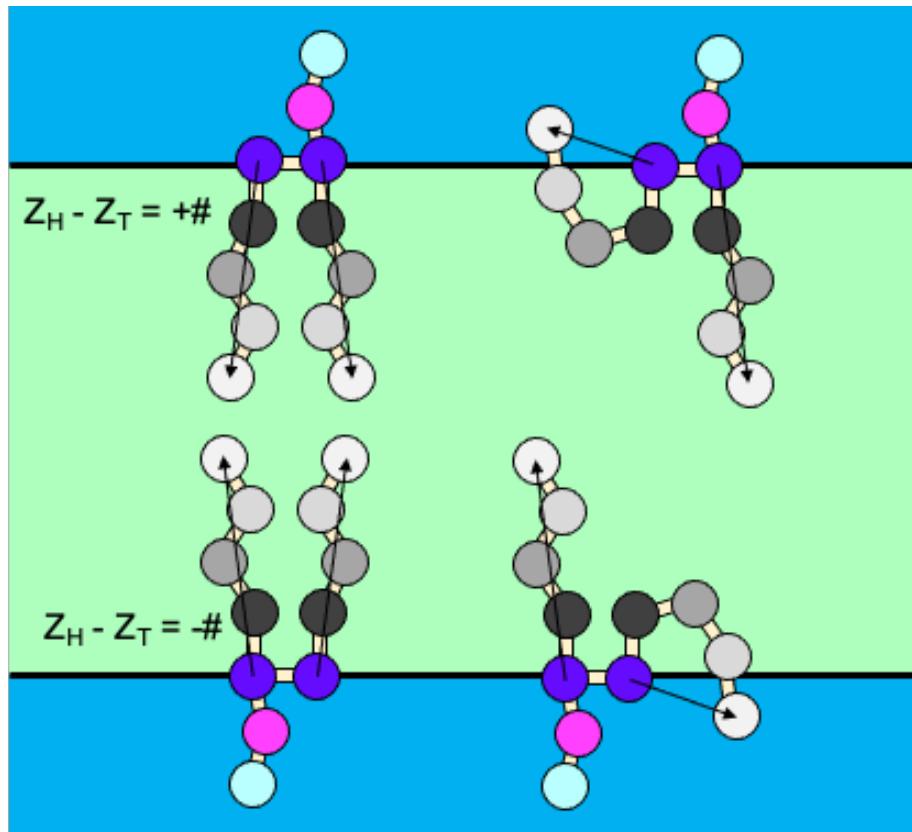


Figure 1-6 Schematic showing how lipids are assigned to a leaflet-by-leaflet finder. Taking the difference in z of the head and the tail atoms will give a positive number. This is opposed to the lower leaflet where the same difference gives a negative number. While a delta z is computed for both tails of a lipid the one with the greater magnitude is more likely to produce the correct assignment. This is demonstrated by the two lipids on the right.

As a final note, leaflet finder is included only in select MOSAICS programs. To check if a MOSAICS tool includes leaflet finder the user can type the program name along with the `-h` tag. This will print to screen a list of the command line arguments for that program. Then, any program using leaflet finder will contain a `-leaf` and `-lf_pdb` argument. These arguments are used to specify the leaflet used in the analysis (0:both, 1:upper, 2:lower) and the name of the output PDB file with the leaflets indicated by the B-factor.

1.9 Protein Finder

For some computations it is necessary to identify the protein atoms within a system. For example, the MOSAICS program Lipid Contacts (section 3.8) lets the user quantify the number of contacts formed between the lipids and other molecules such as the protein. In this case, Lipid Contacts will execute a nested loop over the lipid and protein atoms while computing the distance between each. To create a list of the protein atoms, Lipid Contacts uses the built-in protein finder routine. Protein finder works by comparing the residue names for the system with a list of pre-programmed amino acid names. Then, if a residue name matches one of the names in this list,

then the atom is assumed to belong to the protein. Still, the user is encouraged to check that the protein assignment is made correctly. This may be done by including the -pf_pdb command line argument which instructs the program to write a PDB of the reference structure with the protein indicated by its B-factor (1:protein, 0:non-protein). Additionally, programs using protein finder will report a summary of the protein selection. An example is given below.

There are 879 atoms, 430 residues and 20 residue types in the protein as summarized below:

index	type	count
-------	------	-------

index	type	count
0	THR	25
1	PRO	19
2	LEU	61
3	ALA	52
4	ILE	36
5	PHE	27
6	MET	17
7	VAL	30
8	GLY	57
9	ASP	9
10	LYS	10
11	TRP	6
12	GLN	11
13	ASN	11
14	ARG	16
15	HIS	6
16	TYR	9
17	CYS	3
18	SER	11
19	GLU	14

Output included in the report includes the residue types and how many of each type are present. If a system contains residue types not recognized by protein finder, additional parameters may be added using the -pf_prm tag and a text file containing the new residue types. An example follows.

```
-pf_prm
#res_type
HSE
HSP
HSD
```

In the example here several histidine types are added to the protein finder.

1.10 Solvent Finder

As was mentioned in section 1.9 the MOSAICS tool Lipid Contacts (section 3.8) lets the user measure the number of contacts between the lipids and other molecules. One possibility is to use this program to quantify solvent accessibility at the level of the lipid head groups or tails etc. For this type of analysis, Lipid Contacts must identify the water molecules and uses a built-in solvent finder routine. Solvent finder works like protein finder and compares residue names with a list of pre-programmed water types. If a residue name matches one of the names in the list, then the molecule is assumed to be a water. As always, it is good practice to check that the waters have been identified correctly. This may be done by including an output filename with the -sf_pdb tag and checking the resulting PDB which will have the water molecules indicated by their B-factor (sol:1, non-sol:0). In addition to this, programs using solvent finder will report a summary of the solvent selection as is demonstrated in the example below.

There are 879 atoms, 430 molecules and 20 molecule types in the solvent as summarized below:

index	type	count
0	W	2545
1	WF	282
2	ION	62

This report includes the number of solvent molecules for each component detected as well as the number of atoms overall. If a system contains a solvent type not recognized by solvent finder, additional parameters may be added using the -sf_prm tag and a text file containing the new solvent types. An example is now provided.

```
-sf_prm
#sol_type
W
TIP3
WF
```

Here we have added several common water residue names to the solvent finder.

1.11 Computing Averages using Grid Interpolation

Here we briefly introduce the grid-based averaging procedure used by MOSAICS. This theory is provided in greater detail in ref? and the reader is encouraged to read that paper. However, we add to that description important details for using the tools. To begin let us consider the characterization of a lipid bilayer in relation to an embedded protein. More specifically, we are interested in any observable f which can be computed for the individual lipids and that changes as one moves around the protein. Examples include the lipid tilt angle, or the number of contacts formed with the lipids on the opposing leaflet, etc. For these calculations, the reference frame must be fixed about the protein such that its position is unchanging with time. With this constraint, a lipids location around the protein may be described by a set of coordinates in the

XY plane. To facilitate calculations on a computer, this plane is discretized by construction of a 2-dimensional lattice. The bilayer is then characterized at each grid point i,j where i and j are the indices for the x and y dimensions (Figure 1-7 A). This process is repeated for each trajectory frame and is followed by an averaging over the frames (Figure 1-7 B):

$$\langle F^{ij} \rangle = \frac{1}{\rho^{ij}} \sum_{t=0}^T F_t^{ij} \quad (1.4)$$

where F_t^{ij} is the property of interest for frame t , $T+1$ is the total number of trajectory frames, and ρ^{ij} is a normalizing factor (more on this later). We note that the approach taken here indexes the trajectory frames from 0 to T . This assumes that the initial snapshot is the starting configuration of the simulation. As such, the simulation time may be found as $t\Delta t$ where Δt is the time step between trajectory frames.

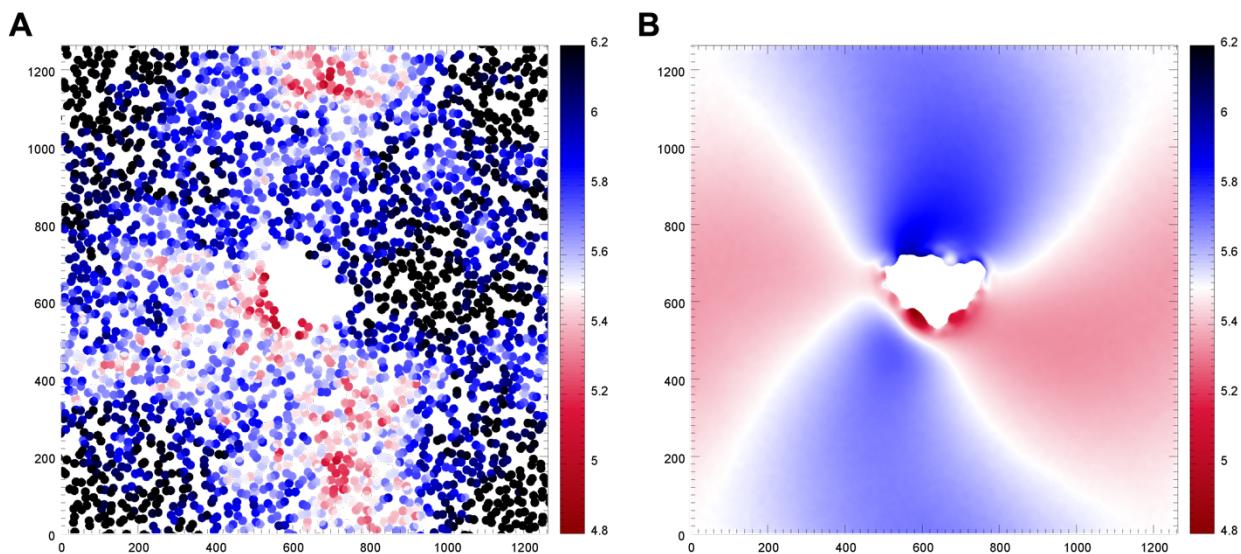


Figure 1-7 Example of an observable f characterized for a single trajectory frame. (Panel A). The same observable is then averaged over the trajectory frames. (Panel B). Units for the x/y axis are grid points. Since the observable measured was the z-coordinate of the ester atoms of a POPX bilayer the units of the color bar are nm.

For MOSAICS tools, single frame estimates of F_t^{ij} are computed using grid interpolation (Figure 1-7 A). This allows the lipid bilayer, which is particulate in nature, to be modeled as a continuous band. In this approach, F_t^{ij} is computed as a weighted sum over the lipids:

$$F_t^{ij} = \frac{\sum_{k=1}^N w_{k,t}^{ij} f_{k,t}}{\sum_{k=1}^N w_{k,t}^{ij}} \quad (1.5)$$

where N is the total number of lipids in the system and $f_{k,t}$ is the observable computed for lipid k , and $w_{k,t}^{ij}$ weights lipid k at lattice point i,j . Equation 1.5 holds for all $\sum_{k=1}^N w_{k,t}^{ij} > 0$ and the lattice point is otherwise left undefined. Moreover, $w_{k,t}^{ij}$ is given as a function of the distance

between the lattice point i,j and lipid k . While the form of this function may vary, it is common to use a gaussian:

$$w_{k,t}^{ij} = A \exp\left(-\frac{dx^2}{2\sigma_x^2} - \frac{dy^2}{2\sigma_y^2}\right) \quad (1.6)$$

where dx and dy refer to the x and y component of the separating distance. Similarly, A is the amplitude of the gaussian and the σ terms designate the distribution width in each direction. Taking this approach, the membrane may be characterized for a single frame after $N*G$ distance calculations where G is the total number of lattice points and N is the number of lipids. For high resolution analysis of biologically significant systems this computation is nontrivial. Fortunately, the cost can be reduced by recognizing that $w_{k,t}^{ij}$ drops off exponentially with distance. Because of this, only nearby lipids contribute significantly to F_t^{ij} and we may ignore lipids beyond a threshold distance (r). If this method is implemented by scanning over each grid point, then a neighbor list must be kept if any reduction in computational cost is to be realized. Alternatively, the algorithm can be implemented by looping over the lipids while simultaneously selecting lattice points within the cutoff distance and adding weighted data accordingly. For this approach, the upper and lower bounds for potential lattice points can be directly computed thus reducing the number of distance calculations for each lipid to $4r^2/\Delta^2$ where r is the cutoff distance and Δ is the spacing between lattice points. If r is taken as $1/2\sqrt{APL}$, where APL is the area per lipid, then the number of distance calculations required to characterize a single frame of the trajectory reduces to G (see Figure 1-8). For MOSAICS tools, this latter approach is taken thereby boosting the performance. In addition to this, we let the functional form of $w_{k,t}^{ij}$ be unity for lattice points within the cutoff distance and nil otherwise. The result is a partial interpolation of the grid data that is highly efficient in its implementation.

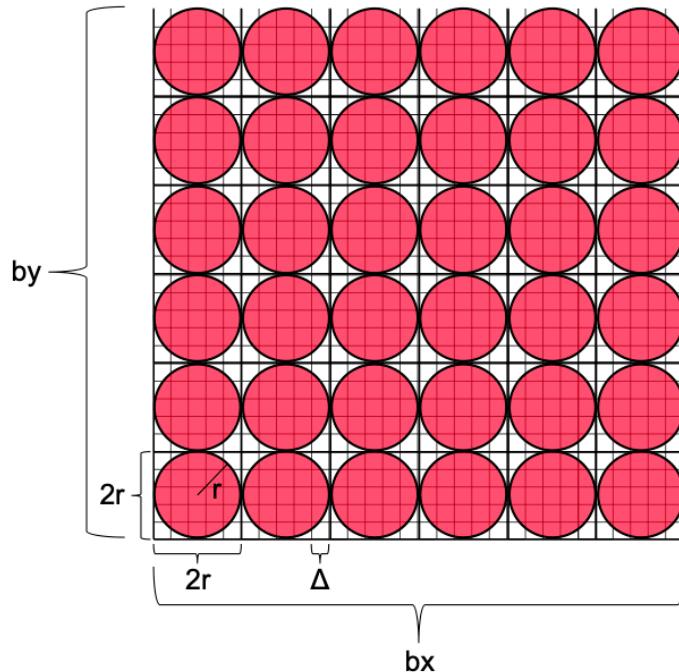


Figure 1-8 Schematic used for determining how many distance calculations are required to characterize the membrane for a single trajectory frame. Lipids are represented as red circles. Furthermore, the upper and lower bounds for which grid point distances must be measured for each lipid are indicated with bold lines. Similarly, the lattice points are represented by the intersection of weaker lines. We note that the number of distance calculations for each lipid is $4r^2/\Delta^2$. Setting $r = 1/2\sqrt{APL}$ gives APL/Δ^2 distances per lipid. There are thus $N*APL/\Delta^2$ distances in total. Of course, $N*APL$ gives the area of the box $A_{box} = bx*by$. And finally, A_{box}/Δ^2 gives the number of grid points G .

As was mentioned previously, weighted data is deposited to the grid around each lipid. It is therefore necessary to select a point in XY to represent the lipids position. This could be taken as the center of mass of the lipid or one of the atoms making the head group such as the phosphate of a phospholipid. If a set of atomic coordinates is used, then the selected atom is referred to as a mapping atom. Continuing with this idea, one could choose multiple mapping atoms such that data is deposited around each of them. This is the approach taken by MOSAICS and we recommend the carbon atoms of the glycosidic linkages or their coarse-grained counterparts. In addition to this, the user must specify a cutoff radius (r) using the -r command line argument. Currently a single cutoff distance is allowed. It should be noted that using a hard cutoff for the weighting function results in some grid points being left uncharacterized for a given trajectory frame. Because of this, the normalizing factor (ρ^{ij}) of equation 1.4 will vary from one grid point to another. This factor counts the number of trajectory frames for which a lipid was local to the grid point, i.e., how frequently the lattice point was characterized in the single frame interpolation data. Because of this, ρ^{ij} is loosely referred to as the sample count. Moreover, most grid-based MOSAICS tools make available ρ^{ij} in a separate output file. This file is named like the averaged observable but contains the “rho” appendage. Furthermore, in addition to serving as a normalizing factor, the sample count may be used to distinguish between significant and insignificant data. This is because, ρ^{ij} contains the number of data points the average is taken over. If this number is too small, then the average is not meaningful. To determine which lattice points are insignificant ρ^{ij} is first averaged across the grid:

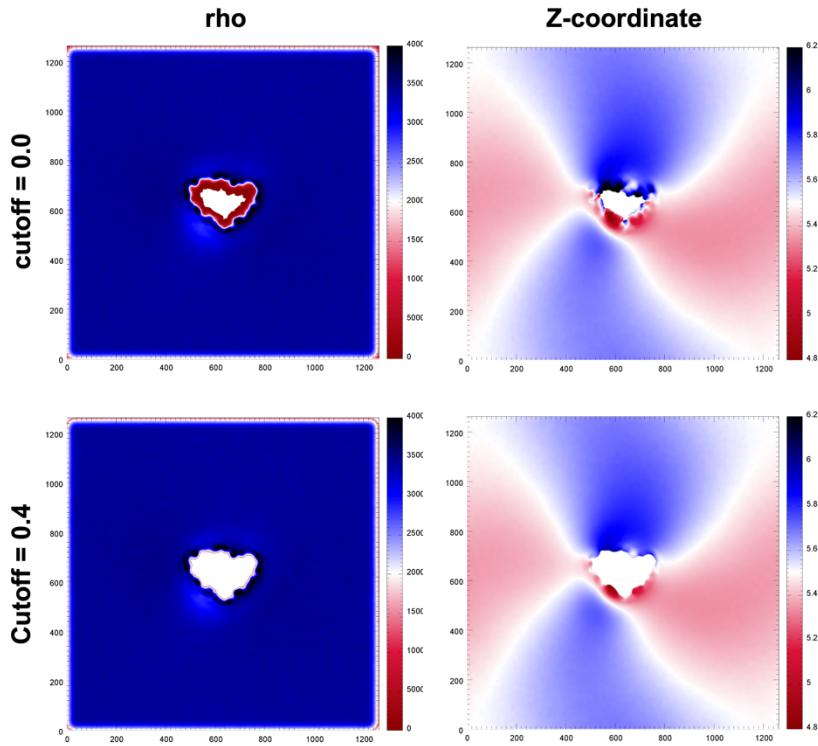


Figure 1-9 Sample count (left panels) and time average z-coordinate measured for the ester atoms of POPX lipids (right panels). For the upper panels a cutoff of 0.0 was used for χ . For the lower panels a cutoff of 0.4 was used resulting in the exclusion of some data points. Units for the x/y axis are grid points. For the color bars the units are the number of lipids (left panel) or nm (right panel).

$$\langle \rho \rangle = \frac{1}{G} \sum_i \sum_j \rho^{ij} \quad (1.7)$$

Then, a threshold count α is defined. This is the value that ρ^{ij} must exceed for $\langle F^{ij} \rangle$ to be considered significant and is defined as a percentage of the mean sample count:

$$\alpha = \chi \langle \rho \rangle \quad (1.8)$$

where χ is specified by the user using the -cutoff tag. If the user wishes to exclude no data, then a value of 0 may be chosen. In this case, insignificant data may be excluded later using the MOSAICS tool Grid Data Excluder which is described in section 1.13. In either cases, insignificant lattice points are marked with a value of “NaN” in the resulting grid data.

With the basics out of the way, let us shift our focus towards any input parameters that are needed to build the grid. For example, the grid resolution may be set with the -APS tag which is short for “area per square”. This parameter is aptly named since the grid spacings in the x and y directions are equal. This results in a lattice that is composed from smaller squares. With this choice, the grid spacing is fully determined by a single parameter. In general, high-resolution grids are preferred with a spacing between lattice points of 1 Å or less. Moreover, the overall size of the grid may be taken from the initial box at frame zero or the user may specify the dimensions with the -bx and -by tags. Upon running the analysis, details concerning the lattice dimensions and resolution are reported as is shown in the example below.

Using box at frame zero to construct the grid as follows:

box_x	box_y	cell_size	num_g_x	num_g_y
15.126637	15.126637	0.070711	215	215

In the example here, “box_x” and “box_y” give the size of the grid (nm) in the x and y directions respectively. Likewise, the distance (nm) between lattice points is given by “cell_size” while “num_g_x” and “num_g_y” give the number of grid points in the x and y directions. In addition to these settings, there are 2 formats available for writing out grid data. These include the matrix and vector formats (set using the -odf tag). Focusing on the matrix format, each data point is written to file row-by-row. This format contains no information about the spacing between lattice points. Consequently, any plots of data using the matrix format will have units of grid points for the x- and y-axis. On the other hand, the vector format writes grid data in addition to the x and y value corresponding to the lattice point. Data plotted using the vector format will have units of distance (nm) for the x- and y-axis.

In addition to averages, many of the tools included in MOSAICS allow for the computation of the standard deviation of F_t^{ij} over the frames:

$$\sigma_F^{ij} = \sqrt{\frac{1}{\rho^{ij} - 1} \sum_{t=0}^T (F_t^{ij} - \langle F^{ij} \rangle)^2} \quad (1.9)$$

In addition to this, the standard error of the mean may be computed as:

$$\sigma_M^{ij} = \sqrt{\frac{1}{\rho^{ij}(\rho^{ij} - 1)} \sum_{t=0}^T (F_t^{ij} - \langle F^{ij} \rangle)^2} \quad (1.10)$$

These options may be selected by inclusion of the -stdev 1 argument at runtime. Compatible programs will then write the single frame grid data F_t^{ij} to an output file like that shown in Figure 1-7. Following the computation of $\langle F^{ij} \rangle$, this single frame data is re-examined and the deviation from the average is recorded until finally the standard deviation and standard error of the mean is found (Figure 1-10). Because single frame grid data is written to an output file for each frame there will likely be many of these. The user can choose to remove these files after computation of the standard deviation by including the -clean 1 argument or choose to leave the files intact for later processing.

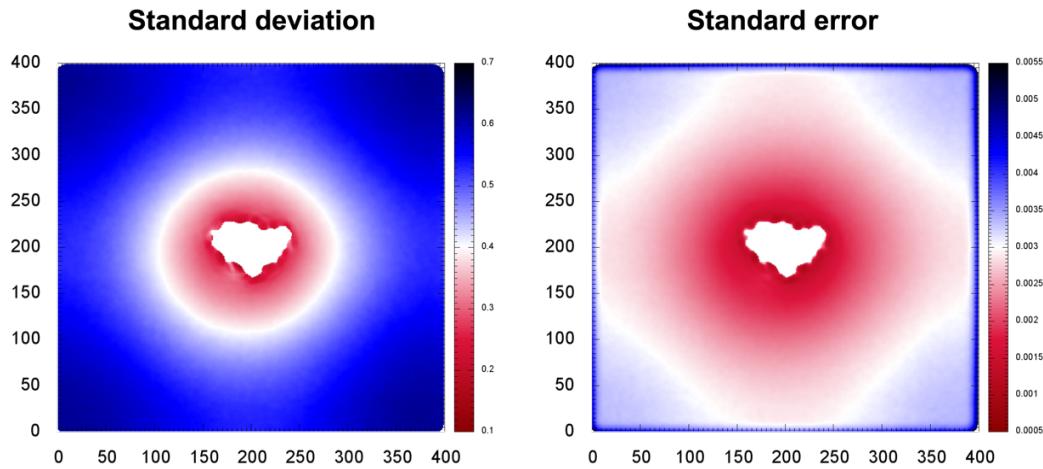


Figure 1-10 The standard deviation across the frames for the z-coordinate of the ester atoms of martini POPX lipids (A). The standard error of the mean z-coordinates (B). Units for the x/y axis are grid points. For the color bars, the units are nm.

As a last note it is worth mentioning that the lower left corner of the grid corresponds to the origin (0,0,0). This convention follows a GROMACS simulation with periodic boundary conditions where the origin is placed in the lower left corner of the box. This can be different for other simulation packages where the origin is sometimes placed in the center of the box. For such a trajectory, the user will need to translate the system such that the lower left corner of the box sits at the origin before using MOSAICS (see Figure 1-11). This can be done using trjconv as follows.

```
$ gmx trjconv -f my_traj.xtc -s ref.gro -o my_traj_centered.xtc -center -pbc res
```

In the above example the user will be prompted to select a group of atoms whose center will be placed in the box center. By including -pbc res the center of mass of each residue will also be placed inside the box. This will prevent the occurrence of broken lipids. Similarly, if the protein is placed in the box center, then it is also unlikely that the protein will be broken across the boundaries.

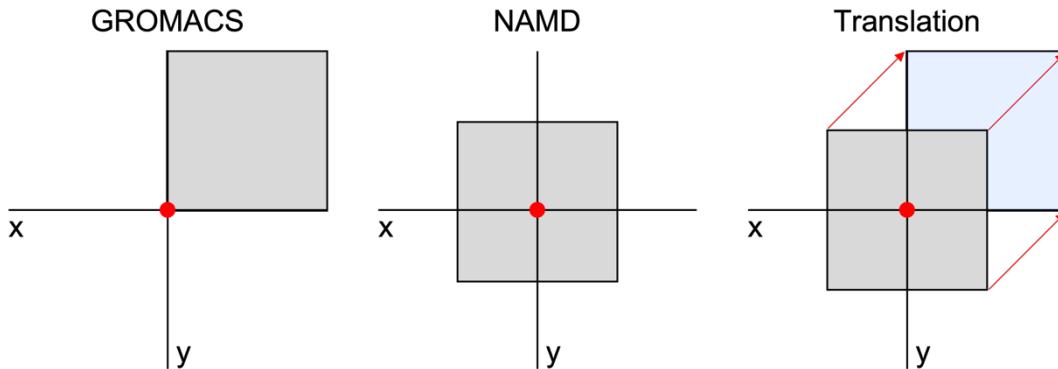


Figure 1-11 2-dimensional representation of system boxes. Left panel contains a GROMACS box and the middle panel a NAMD box. The origin is shown as a red dot. To use MOSAICS on a trajectory produced with NAMD the system must be translated (right panel) to agree with a GROMACS box.

1.12 Plotting Grid Data

As was mentioned in section 1.11, MOSAICS supports two formats for working with two-dimensional grid data. These include the matrix and vector formats, and the user may plot either using Gnuplot. At the current time, we recommend using the matrix format since Gnuplot better handles insignificant data with this format. That is, MOSAICS assigns insignificant lattice points a value of “NaN” in the grid data (section 1.11) and these points are ignored by Gnuplot when the matrix format is used. Conversely, the user will need to replace “NaN” with a numerical value when using the vector format. On the other hand, the vector format contains the x and y coordinates of the lattice points resulting in each axis having units of distance. This is compared to the matrix format where the resulting units are given in lattice point. Of course, it is possible to convert the units to distance for the matrix format if the spacing between lattice points is known but this adds extra work. Each format thus has some strengths and weaknesses. With that said, we have obtained good results using the matrix format and have included a Gnuplot script for this format called “heatmap_template.gnu” that is located in the “scripts” folder. We note that this script uses command line arguments and requires Gnuplot version 5 or newer. An example is now given for plotting grid data with Gnuplots.

```
$ gnuplot -c heatmap_template.gnu [0:400] [0:400] [:] example_grid_data.dat 2d_plot.png
```

In the example given here, we have included the heatmap_template.gnu followed by three entries enclosed in brackets []. These give the upper and lower bounds for the x-axis, y-axis, and the color bar respectively. In the case of the color bar, we have included “[:]” without specifying a range. This choice instructs Gnuplot to choose the range based on the minimum and maximum values found in the grid data (the same option may be used for setting the x- and y-axis range).

1.13 Extended Analysis of Grid Data

Once a lipid observable has been projected onto the XY plane and a time average acquired, it may be desirable to perform additional analysis on this data. For example, the user might wish to report the average over a subset of the grid. Similarly, we could average the lattice points based on their distance from the protein surface. With this, we could report the average observable f as a function of distance from part of the protein. Still more, we could use a mask (section 1.15) to select some lattice points in combination with the single frame grid data F_t^{ij} and compute a probability distribution for the observable. In this section we will introduce additional MOSAICS tools which make possible these analyses and more.

To begin, let us revisit the problem of excluding insignificant grid data. Recall from section 1.11 it was noted that data could be excluded later if a cutoff value of 0.0 (χ) was provided when computing $\langle F^{ij} \rangle$. This may be done using the MOSAICS tool Grid Data Excluder. To use the program, the user must provide the file for which data is to be excluded as well as the sample count data. These files are specified using the command line arguments -d and -rho respectively as is shown in the example below.

```
$ grid_data_excluder -d upper_z.dat -rho upper_rho.dat -o upper_z_0.4.dat -cutoff 0.4 -odf 0
```

In this example, grid points with a sample count less than 40 percent of the average count will be excluded (see Figure 1-9).

In addition to this, there is a tool called Grid Region Integrator that can be used to sum or average data over a subset of the lattice. This subset may be specified using a rectangular selection (section 1.15) or alternatively with a rectangular selection in combination with a mask (section 1.15). We note that the mask filename is specified with the -mask tag and the rectangular selection is defined using the usual -x, -y, -rx, -ry, and -invert tags. The average over the selection is thus computed as:

$$\text{avg} = \frac{\sum_i \sum_j m^{ij} b^{ij} \langle F^{ij} \rangle}{\sum_i \sum_j m^{ij} b^{ij}} \quad (1.11)$$

Where m^{ij} is the value for lattice point i,j in the masking data and b^{ij} tells if the lattice point is inside the rectangular selection; both m^{ij} and b^{ij} are restricted to the values of 0 or 1. An example is now provided in which the average membrane thickness is computed for a patch of the membrane from the bulk:

```
$ grid_region_integrator -d mem_thickness.dat -x 200 -y 200 -rx 100 -ry 100 -odf 0
```

In the example above, the average membrane thickness is computed over a rectangle that is 200 grid points in both length and height and that is centered on grid point 200,200 (see Figure 1-12). Output from grid region integrator includes the number of lattice points summed over (count), the sum of data over these points (sum), and the average (equation 1.11). In addition to this, a mask is generated that highlights the selected area. This data is written to an output file that is given the “selection.dat” filename. We note that, Grid Region Integrator has the option of writing $\langle F^{ij} \rangle$ for each lattice included in the average to a list. This option may be used by including a filename for the resulting data using the -list tag.

Having acquired the bulk membrane thickness with Grid Region Integrator, we could shift the thickness data. This would make quantification of changes in thickness relative to the bulk much easier. For this type of modification, we have the MOSAICS tool Grid Addition. In the following example we subtract the bulk membrane thickness from the grid data containing the membrane thickness.

```
$ grid_addition -d mem_thickness.dat -o mem_thickness_shifted.dat -add -3.196 -odf 0
```

Note, in the example above, the bulk thickness of 3.196 nm was subtracted from each point in the grid (see Figure 1-12).

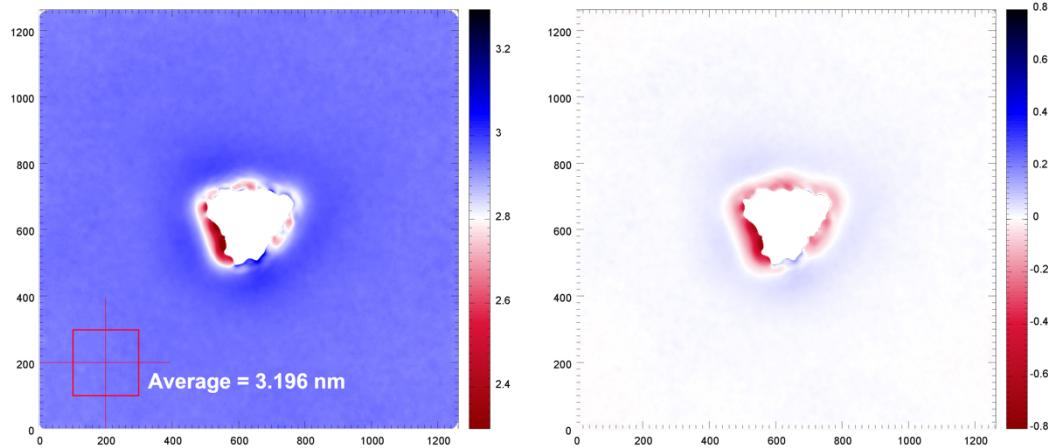


Figure 1-12 Average membrane thickness in the bulk (left panel). Red square centered at 200,200 encloses the grid points included in the average. Right panel shows the membrane thickness after subtracting the thickness of the bulk. Units for the x/y axis are grid points and nm for the color bar.

In some cases, it may be desirable to average a quantity over both leaflets. This can be done using Leaflet Averager. With Leaflet Averager, each leaflet is weighted by the sample count ρ_{ij}^l for that leaflet. For example, the average interdigitation at a particular grid point is computed as:

$$I_{avg}^{ij} = w_l^{ij} * I_l^{ij} + w_u^{ij} * I_u^{ij} \quad (1.12)$$

Where I_l and I_u are the interdigitation for the lower and upper leaflets. The weights are computed from the sample counts by:

$$w_l^{ij} = \frac{\rho_l^{ij}}{\rho_l^{ij} + \rho_u^{ij}} \text{ and } w_u^{ij} = \frac{\rho_u^{ij}}{\rho_l^{ij} + \rho_u^{ij}} \quad (1.13)$$

where ρ_l^{ij} and ρ_u^{ij} are the sample counts for grid point i, j in the lower and upper leaflets. In addition to this, Leaflet Averager can exclude insignificant data. This is done by summing, for each grid point, the sample count for the upper and lower leaflets. This gives the total sample count ρ_T^{ij} . Data is then excluded by computing the average of ρ_T^{ij} over the grid $\langle \rho_T \rangle$. Following the procedure described in section 1.11, insignificant data is excluded by computation of $\alpha = \chi \langle \rho_T \rangle$ and checking each value of a ρ_T^{ij} against this. Then, if ρ_T^{ij} is less than α , the data point is excluded. An example follows.

```
$ leaflet_averager -d1 upper_i.dat -d2 lower_i.dat -rho1 upper_rho.dat -rho2 lower_rho.dat -o average_i.dat -cutoff 0.4 -odf 0
```

An example of data generated with Leaflet Averager is shown in Figure 1-13.

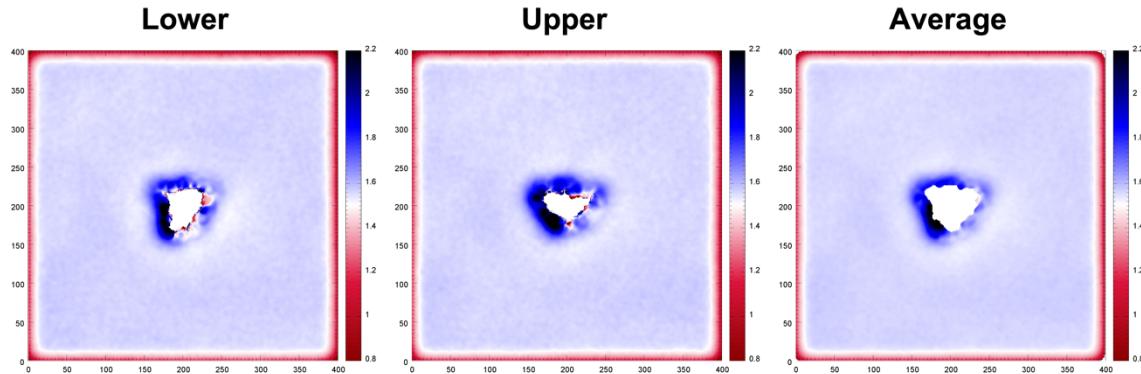


Figure 1-13 Interdigitation in the lower (left) and upper (middle) leaflets. Average interdigitation over the two leaflets is shown in the right panel. Data taken from martini coarse-grained simulation of the CLC-ec1 protein [10]. Units for the x/y axis are grid points. The color bar is unitless.

With Grid Distance Projection, the user can project the grid data as a function of distance from the protein surface (Figure 1-16). This is done by averaging over lattice points within a thin shell around the protein whose midplane lies some distance d from the protein. This average is computed as:

$$\langle F(d \pm tol) \rangle = \frac{\sum_{ij} \langle F^{ij} \rangle m^{ij}}{\sum_{ij} m^{ij}} \quad (1.14)$$

where $\langle F^{ij} \rangle$ is the grid data of interest and m^{ij} is the mask that takes on the values of 0 or 1 and defines the shell. We note that the shell is given a thickness of $2*tol$ where tol is typically a small number. To use Grid Distance Projection, the user must supply a masking file indicating which grid points correspond to the protein. This mask can be produced from the sample count data. For this, we assume that regions of the grid with a low count represent the location of the protein. The user can convert the ρ^{ij} data into a protein mask in two steps (Figure 1-14). First, the places with a low sample count are indicated using the program Protein Mask as shown in the following example:

```
$ protein_mask -d upper_rho.dat -o prot_mask0.dat -cutoff 0 -odf 0
```

Protein Mask works like Grid Data Excluder. To be clear, the average sample count $\langle \rho \rangle$ is first computed over the grid. Then a threshold count α is determined such that $\alpha = \chi \langle \rho \rangle$. And finally, if ρ^{ij} is greater than α , then the grid point is set to 0. Otherwise, the lattice point is set to 1. This highlights regions where the sample count is low, such as where the protein is positioned but also the boundaries of the box. In the second step, these outside boundaries are removed using the MOSAICS tool Grid Editor as is shown in the following example.

```
$ grid_editor -d prot_mask0.dat -o prot_mask1.dat -x 225 -y 0 -rx 225 -ry 25 -val 0
$ grid_editor -d prot_mask1.dat -o prot_mask2.dat -x 225 -y 400 -rx 225 -ry 25 -val 0
$ grid_editor -d prot_mask2.dat -o prot_mask3.dat -x 0 -y 225 -rx 25 -ry 225 -val 0
$ grid_editor -d prot_mask3.dat -o prot_mask_fin.dat -x 400 -y 225 -rx 25 -ry 225 -val 0
```

In this example we have selected the outside edges of the box and changed the values to zero. This is done using a rectangular selection (section 1.15) centered at $-x$, $-y$ with a half width of $-rx$, $-ry$. We note that the rectangular selection can be coupled with a masking file if the $-mask$ tag is included; in this case, grid points are edited that are inside both the rectangular and the masking selections.

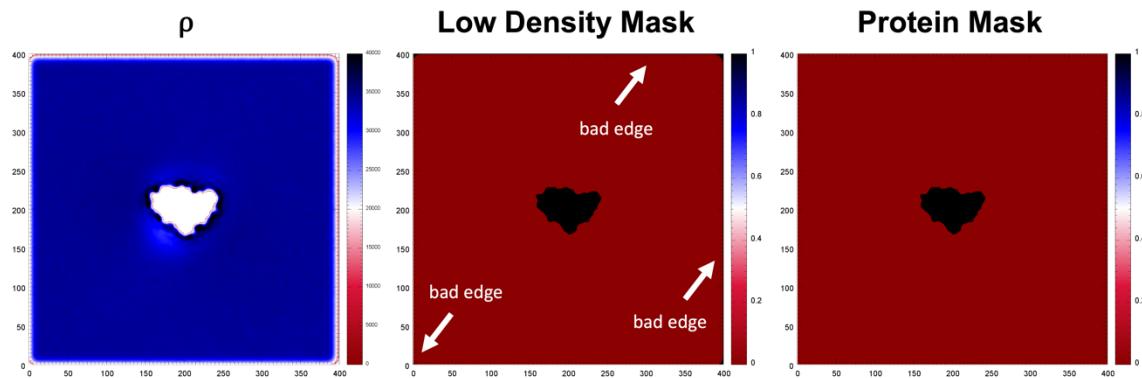


Figure 1-14 Sample count (left panel). Regions of the grid with a low sample count ($\rho^{ij} < \alpha$) are indicated with a value of 1 (middle panel). Box edges with low sample count are also indicated. Note that all box edges have sufficiently low counts but only a few are noticeably visible (look closely). After removing the bad edges, the resulting mask is of the protein only (right panel). Units for the x/y axis are grid points. For the color bars, the units are the number of lipids (left panel). The color bars for the 2 masks (middle and right panels) are unitless.

As an alternative to Grid Editor, this previous step of removing the grid edges could have been performed using Protein Mask Grower. With this tool, the user must specify a grid point ($-x$, $-y$) for which a mask will be grown from. This grid point is called a seed. With a seed specified, a mask is grown from it by finding all grid points that can be connected back to the seed via a path consisting only of 1's. This is accomplished by finding all lattice points with a value of 1 that neighbor the seed. Note, the seed should also have a value of 1. These points are then set to 1 and the process repeated until no new grid points are found with a value of 1. For our example, placing a seed somewhere inside the protein region of the grid will result in the selection of the protein but not the edges of the box. This is demonstrated in the following example:

```
$ protein_mask_grower -d prot_mask0.dat -o grown_prot.dat -x 200 -y 200 -odf 0
```

Similarly, the program NaN Selector can be used to select a region of the grid composed of NaN values. This program works the same way as Protein Mask Grower but selects NaN instead of 1. With NaN Selector, a protein mask can be made in a single step as is demonstrated in the following example:

```
$ nan_selector -d upper_rho_0.4.dat -o prot.dat -x 210 -y 210 -odf 0
```

We note that "upper_rho_0.4.dat", as specified in the above example via the $-d$ tag, is a sample count data where χ was set to 0.4. The seed is thus set to the central region of this grid data where the lattice points have already been excluded; this region represents the protein's location.

Regardless of which program is used to generate a protein mask, Grid Distance Projection will use this mask to create an additional mask m^{ij} (Figure 1-15) which defines the shell whose midplane lies the desired distance d from the protein. An example is now given.

```
$ mpirun -n 100 grid_distance_projection_mpi -d monomer_digi_upper.dat -mask prot_mask_fin.dat -o digi_dist_proj.dat -x 105 -y 105 -rx 105 -ry 105 -iter 100 -res 0.1 -range 0.5 -invert 0 -APS 0.005 -odf 0
```

In the example here, a shell is made with a half-width that is specified using the -range tag. We note that the shortest distance between the grid point and the protein mask is computed when determining which lattice points belong to the shell. Furthermore, a rectangular selection has been used thus enabling the selection of a subset of the protein surface (section 1.15). With this selection, only grid points inside the rectangle (-x, -y -rx, -ry) are considered in the average, i.e., m^{ij} is set to 0 if the grid point falls outside the rectangular selection. Of course, the rectangular selection can be inverted using the -invert 1 tag.

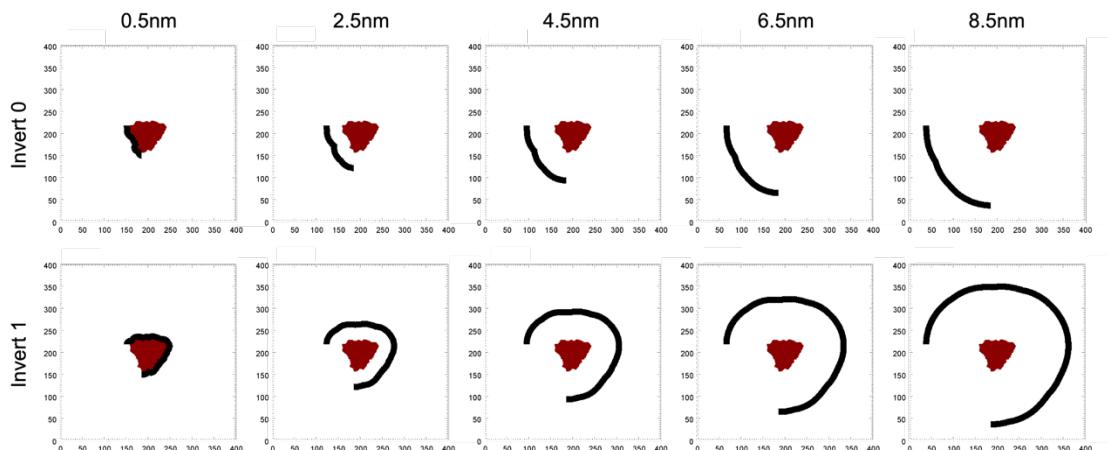


Figure 1-15 Masks used for computing grid averages as a function of distance from the protein interface. Top panels show masks in the rectangular selection (lower left corner) and bottom panel shows masks outside the rectangular selection. The regions for which data is averaged over is shown in black. For clarity, the protein mask is also shown (burgundy). The protein mask may be included in the resulting mask data files by including the -incl_p 1 command line argument. In this case, the protein is indicated by a value of -1. Units for the x/y axis are grid points. The color bars are unitless. We note that the masks m^{ij} can be obtained using the provided script “get_mask_plots.sh” located in the “scripts” folder. Try using something like: \$ sh/get_mask_plots.sh base_filename 0 100 dir/ where base_filename is the base filename (section 1.14) of the resulting masks. Here, 0 and 100 are used to set the range, i.e., which masks are plotted (there were 100 of them in the example), and “dir/” tells what directory to store the resulting .png files in.

Once a mask is made, the grid points included in the mask are averaged according to equation 1.14. This gives the average value for a particular distance d . This process is then repeated -iter times and the distance between the shell midplane and the protein surface is increased by -res each time. The set of averages is then written to an output file as specified via the -o tag. We note that the selection masks, defining which lattice points included in the average, are also written to file, one for each iteration (Figure 1-15). These files are given the same name as specified via -o but with the “_i_mask” appendage where i specifies the iteration. An example of data generated with Grid Distance Projection is shown in Figure 1-16

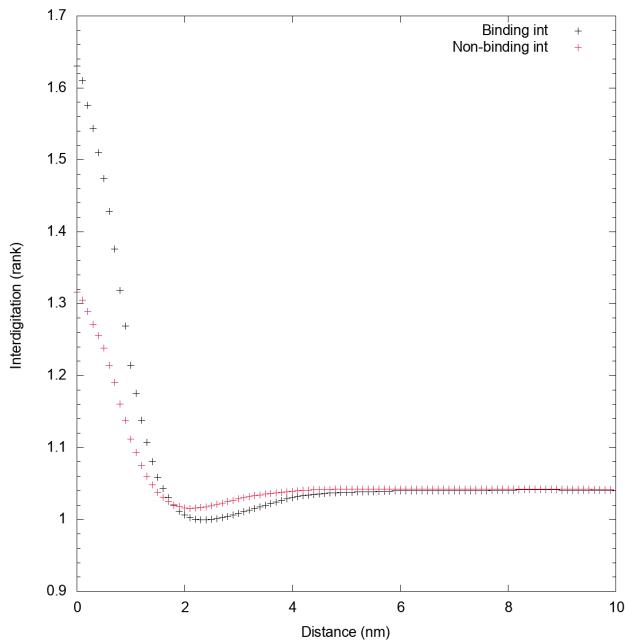


Figure 1-16 Lipid interdigitation projected as a function of distance from the protein interface (either the binding or non-binding interface of CLC-ec1).

Like the tools Protein Mask, Protein Mask Grower, and NaN Selector, a general use mask can be built around the protein using the program Mask Maker. The program works by reading in a mask for the protein (-d) and then finding grid points that are within a cutoff distance (-dist) from the protein surface. The program also uses a rectangular selection (section 1.15) centered at -x,-y with a half width of -rx,-ry. With the -invert 0 option, only grid points inside the rectangle will be included in the output mask and with -invert 1 only grid points outside the rectangle will be included. With the -copy 1 tag, the protein will also be included in the final mask. An example of the run commands used with Mask Maker are now given:

```
mask_maker -d prot.dat -o out_mask.dat -x 85 -y 108 -rx 100 -ry 110 -invert 0 -dist 1.0 -APS 0.005
-copy 0 -odf 0
```

In the example provided here, a region of the lipids extending 1 nm from the protein surface has been selected. We note that the area per lattice square must be given via the -APS tag so that the distance between grid points can be computed. An example of several masks created with Mask Maker can be seen in Figure 1-17.

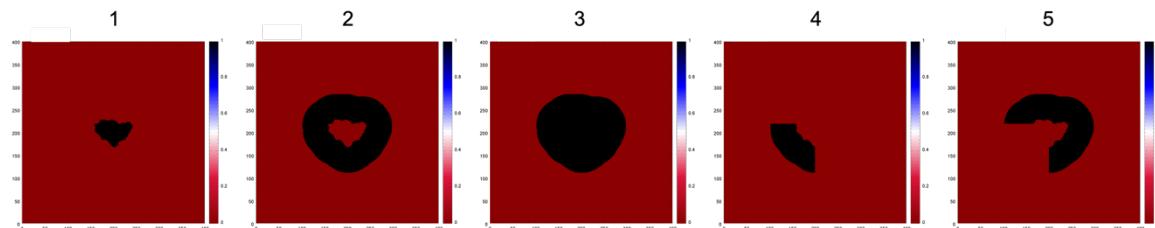


Figure 1-17 1 shows a protein mask used for input. 2 is a mask around the protein letting the rectangle cover the entire box. 3 is the same setup as 2 but the protein is also stamped onto the mask. 4 is a mask created with a rectangle in the lower left corner of the box. 5 is the same as 4 but using the -invert 1 option. Units for the x/y axis are grid points. The color bars are unitless.

With Mask Maker, the user can make a mask for a region of interest around the protein or in the bulk. This masking data can be used with Single Frame Distributions and the single frame data F_t^{ij} generated with one of the grid-based analysis tools to plot a probability distribution for the observable within the region of interest. For example, we could make a probability distribution for the rank 2 order parameter averaged over the lipid tails. We could then compare how the distribution changes moving from the protein interface to a region in the bulk. To use the program, the user must specify the base file name (-base) for the single frame data (this is the file name up to but not including the frame number, see section 1.14). In addition to this, a masking file is provided with the -mask tag. Other input commands include the number of single frames to be read (-frames) and the bin width (-width) to be used when making a histogram. An example is shown next.

```
$ mpirun -n 100 single_frame_distributions_mpi -mask upper_dimer_int.dat -base upper_p2 -o upper_p2_dimer_histo.dat -frames 83325 -odf 0 -width 0.01
```

An example of data generated using Single Frame Distributions is provided in Figure 1-18.

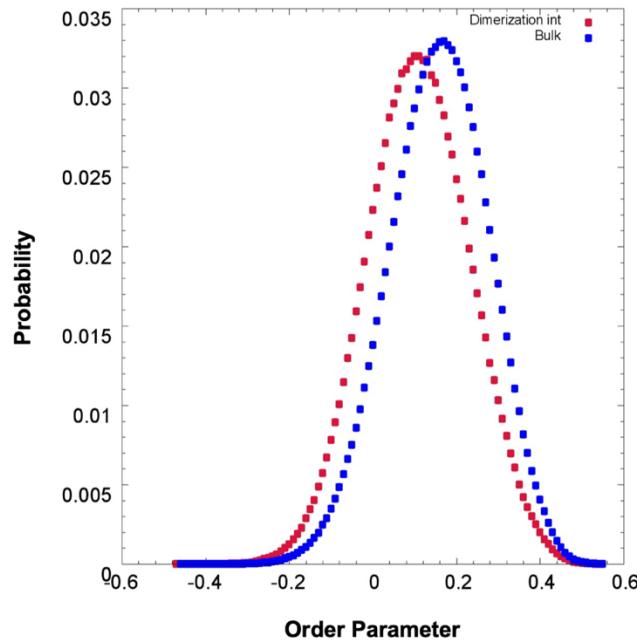


Figure 1-18 Shown is the distribution of the order parameter (averaged down the lipid tails) for lipids at the dimerization interface and in the bulk. For the dimerization interface a 1 nm mask was created using Mask Maker. To generate a mask of the bulk lipids required 2 steps. First a 1 nm mask was made around and including the protein. Then, this mask was used as input to generate a second mask, which was 10s of nm thick around that. The resulting mask defines the bulk lipids.

1.14 File Naming Conventions

Here we discuss some of the details regarding the file naming convention used within MOSIACS. To begin, MOSIACS regulates the file extensions used throughout. For reference files and trajectories, filenames must contain one of the xtc, trr, gro, or pdb extensions. For other input files, such as indexes and selection cards, a ndx or crd extension is required. Grid data and other formatted data, like histograms or timelines, are given the dat extension. Likewise, parameter files, like those used to specify parameters for leaflet finder, receive the prm extension. We note

that the required extension is typically displayed for each command line argument when the help options are displayed (-h). Moreover, an error message is displayed, and the program terminated should the wrong extension is used.

It should also be noted that, in some cases, an analysis tool will use an input or output filename to generate additional output filenames. This is done to reduce the amount of input required from the user. For example, most grid-based programs compute the average of an observable f but also write to file the sample count ρ^{ij} (section 1.11) used during the averaging. This additional information is written to a file with the same name as the averaged observable but is given an additional “_rho” tag. Furthermore, in some cases, an analysis tool will generate many files. For example, most grid-based analysis tools have the option to write single-frame data, i.e., F_t^{ij} , to file. This data receives the same filename as the averaged observable but with the global frame number tagged onto the end. Other programs which generate many files include Grid Distance Projection (section 1.13) which generates a masking file for each distance evaluated. 2d Kinetics (section 4.3) also generates many binding events files (be), one for each lattice point. When these files are used as input for further analysis, the “base” filename is required as input. This is the filename up to the indexing tag. Note, the indexing tag could be the global frame number or the grid index (i,j) for single-frame data and binding events files, etc. Command line arguments that require a base filename are indicated in the argument description which may be viewed by including the -h tag.

1.15 Rectangular Selections and Masking Files

For some analysis tools it is possible to focus on a subset of the grid data. In these cases, the user must indicate which region of the grid is of interest. The simplest approach for making such a selection is with a rectangular selection tool. To use the rectangular selection tool, which is built into many of the MOSAICS tools, the user must specify the rectangle center and the half width of each side. This is done with the -x, -y, -rx and -ry tags respectively. Note, the units for each input argument are grid points for the matrix format (section 1.11) and nm otherwise. For example, if assuming a matrix format then the rectangle center is found by moving right of the origin (0,0) by -rx grid points and then moving up -ry points. The rectangle width is then twice -rx and the height is twice -ry (Figure 1-19 A). Moreover, the selection can be inverted by setting the -invert 1 command line argument. In this case, everything outside of the rectangle is selected.

An alternative to using a rectangular selection is to specify the selection using a masking file. A masking file contains grid data whose values are used to indicate which lattice points are of interest. For example, a protein mask is often used where the protein is indicated with a 1 and other regions with a 0 (Figure 1-19 B).

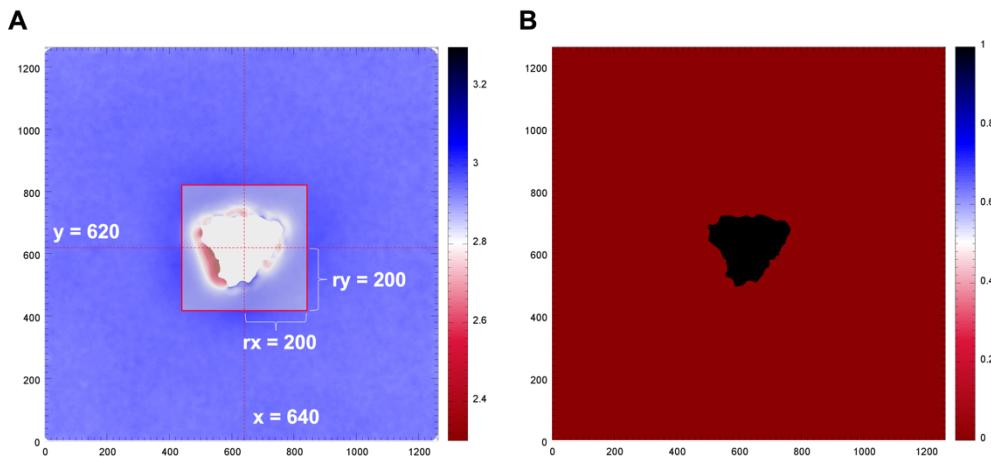


Figure 1-19 Schematic of the rectangular selection (A). Here we have selected a region around the protein for some membrane thickness data. In this example, the parameters -x 640, -y 620, -rx 200, -ry 200 and -invert 0 were used. The resulting rectangle is drawn in red, and the area selected is shaded. If the -invert 0 would have been chosen, then everything outside of this rectangle would have been selected. A mask used to indicate the location of the protein position (B). Lattice points with a value of 1 indicate the protein while the remaining points are set to 0. Units for the x/y axis are grid points. The color bars have units of nm (A) or are unitless (B).

1.16 Noise Filters

A noise filter is a device used to determine if an event occurring at time $t\Delta t$ is significant based on how frequently it occurs in other local frames. This heuristic follows from defining a significant event as one which is present in multiple trajectory frames as opposed to just one. In some cases, this means the lifetime of the event is sufficiently long. For example, the binding of a lipid may be deemed insignificant if the residence time is too small. In this case, a noise filter can be used to remove such events by assigning a binding state b_t (0:unbound, 1:bound) to the lipid for each trajectory frame t . Then, b_t is analyzed over a series of $2N+1$ frames centered on t and the filtered state b_t^* is computed as:

$$b_t^* = \begin{cases} 1 & \text{if } f_1 \geq \omega(2N + 1) \\ 0 & \text{if } f_1 < \omega(2N + 1) \end{cases} \quad (1.15)$$

where f_1 is the number of times b_t had a value of 1 in the $2N+1$ frames examined by the filter and ω is a significance threshold (ranging between 0 and 1) set by the user. A typical value used for ω is 0.5. However, this is not a hard rule and other values are used when appropriate. Noise filters are used in many of the analysis tools concerning dynamics computations. These include the Lipid Mixing program and many of the tools centered around 2d Kinetics. See Figure 1-20 for an example of a noise filter.

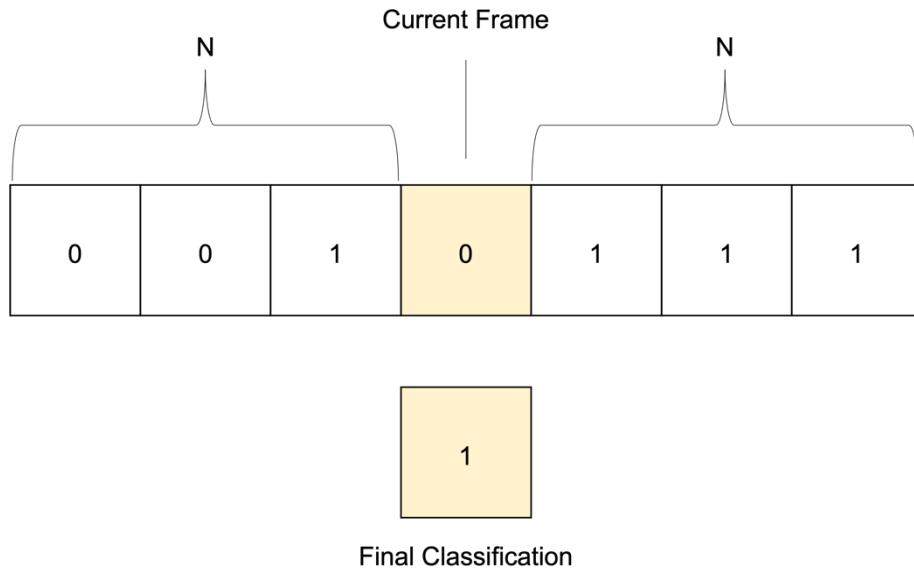


Figure 1-20 An example of a noise filter. Here the filtered property is the binding state as described in the main text above. The filter half-width is set to $N = 3$ frames and a cutoff of 0.5 is used. This means a total of $0.5 * (2 * 3 + 1) = 4$ frames must be bound for the binding state to be classified as 1.

1.17 Discretized Voronoi Diagrams

MOSAICS uses discretized Voronoi tessellations also called a Voronoi diagram to represent the lipids position in the XY plane when estimating residence times as well as for computations of the area per lipid. Such discretized tessellations are derived by finding for each lattice point the lipid whose lipid-to-lattice-point distance is the smallest. Once determined, the lattice point is assigned the res id of the winning lipid. The Voronoi cell is thus defined by the collection of lattice points with a common res id.

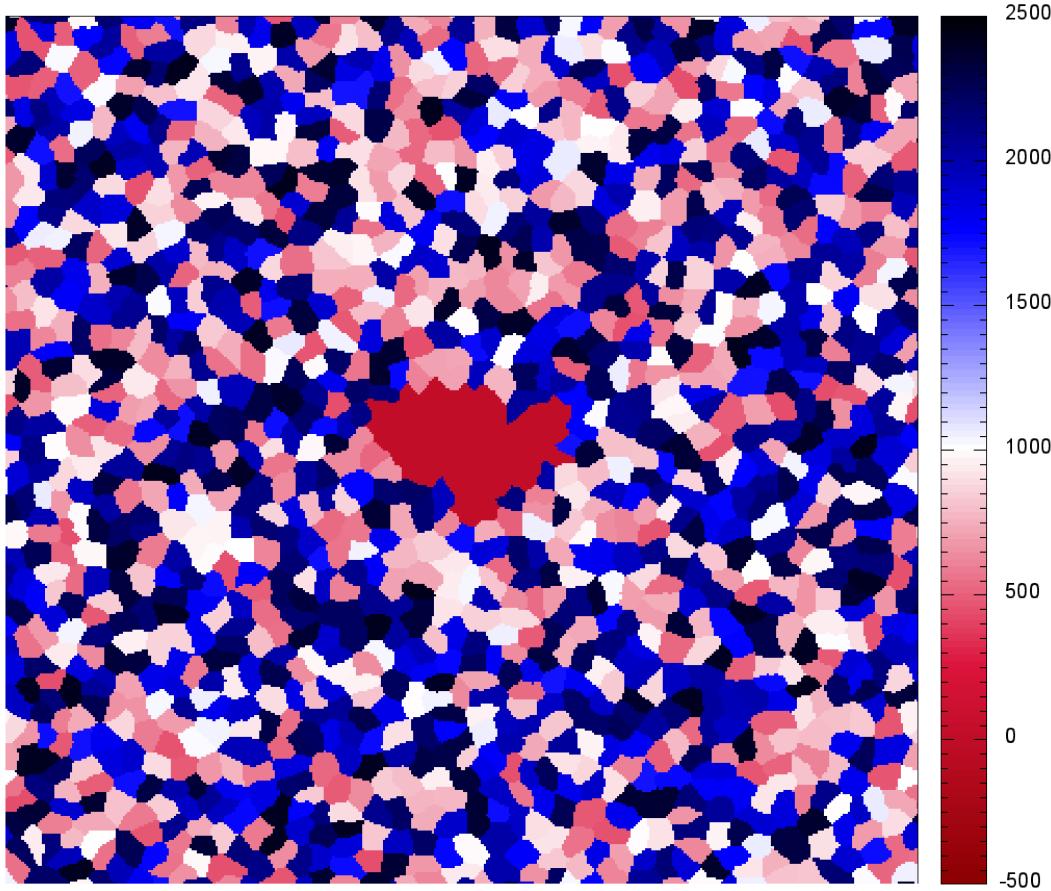


Figure 1-21 Discretized Voronoi diagram. In this case, the color bar indicates the res id. We note that the protein is also included in the tessellation and is shown here in red, i.e., the res id is set to -1. MOSAICS also accounts for periodic boundary conditions when computing tessellations and has the option to account for varying box dimensions.

The computation of a discretized Voronoi diagram thus requires $N*L$ distance calculations where N is the number of lipids and L the number of lattice points. On the other hand, the diagram may be obtained more efficiently using a variation of the stamping method introduced previously. To see how, let each lipid stamp its res id to the grid such that the stamping radius is identical for each lipid (Note that the stamping performed here is a little different in that a list is created for each lattice point that holds the res id of the stamping atoms). Focusing on a grid region with some coverage the lipid whose lipid-to-lattice-point distance is shortest will be contained in the list. It then reasons that the lipid-to-lattice-point distance must be computed and compared only for these lipids. For lattice points missed by the stamping procedure the distance must be measured between all lipids. However, the stamping radius may be increased to minimize the number of such points. In practice, the stamping radius is increased until the efficiency is maximized. We find this to occur with a stamping radius of approximately 0.8 nm and the default stamping radius is thus set to this value. Given this approach, we observe an increase in computational efficiency of discretized Voronoi diagrams by nearly a factor of 100.

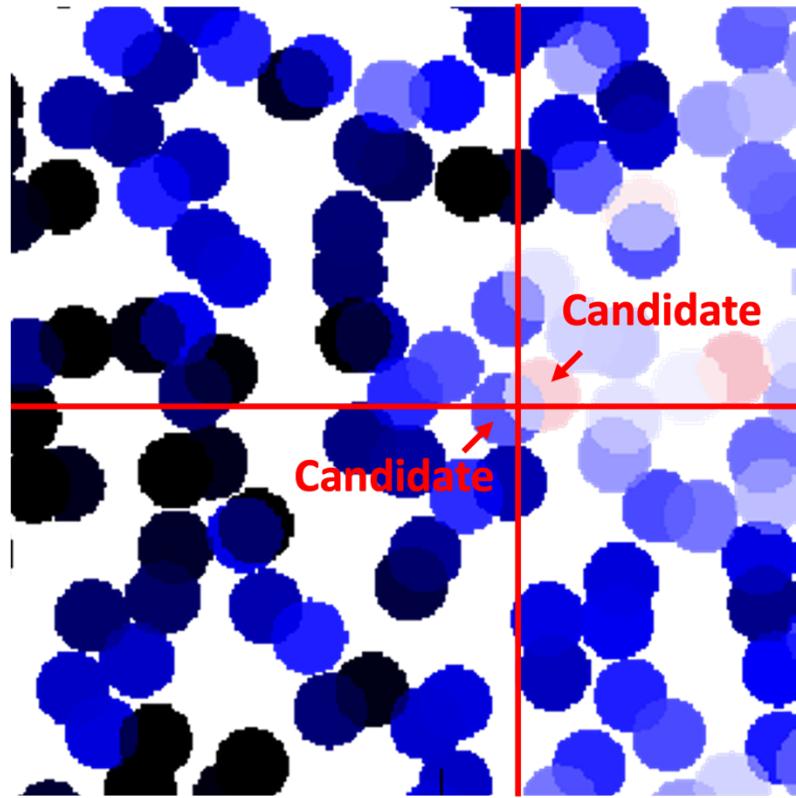


Figure 1-22 Example of the stamp assisted Voronoi tessellation where each lipid stamps its res id to the lattice. In the example shown here we focus on a single lattice point as indicated by the intersection red lines. In this case, the lipid contains overlap with two lipids. The discretized Voronoi diagram is thus characterized at this lattice point by one of these two lipids.

Chapter 2 System Preparation

2.1 Guidelines for Preparing Your System for Analysis

In this section we discuss the various considerations one should make when preparing their trajectory files for use with a MOSAICS tool. Here we briefly discuss each known issue and list any MOSAICS tools that can be used to overcome these challenges. Then, in the remaining sections the individual tools are described in greater detail. Note the recommendations provided here are not a recipe that should be strictly followed. It is up to the user to determine the best procedure for preparing their trajectories.

- **Converting the trajectory to a supported format:** If a simulation is run using a platform other than GROMACS there is a good chance that the trajectory files produced will be in an unsupported format. This problem may be overcome by converting the trajectory files to one of the supported formats (.xtc, .trr, .gro or .pdb). This may be done with VMD [9]. See traj.tcl in the scripts folder for more details. Note, the user will also need to generate a reference file. For example, a good reference file can be acquired by extracting the first frame of the trajectory as a .pdb file.
- **Least squares fitting:** The user must determine if least squares fitting is required for their trajectory. Recall that the protein position, if the membrane simulation contained one, must remain fixed throughout the simulation if one wishes to perform grid-based analysis. If a simulation is performed and the proteins position is restrained, (maybe using colvars or plumed, etc.) then no least squares fitting is required. However, the user may want to use the MOSAICS tool System Translator (section 2.2) to better pin the protein's location especially if the restraints included some wiggle room (flat bottom potential). On the other hand, if no restraints are included in the simulation, then the protein will be free to move about the system. This motion includes translations as well as rotations. If this is the case, least square fitting should be performed before using MOSAICS. This may be done with GROMACS using trjconv or with the fitting routine built into MosAT. If one of these tools are used, then the user will have the option of performing either a 2- or 3-dimensional fit. If a 2-dimensional fit is chosen, then the rotations are performed around the z-axis only. With this approach the protein can vary its tilt within the bilayer but not rotate. In contrast, when a 3-dimensional fit is performed the protein tilt is removed but replaced by a tilting of the bilayer. We should note that for simulations, which were performed with an MD engine other than GROMACS, least squares fitting may be performed using another platform such as VMD. This allows the user to make the rotation while converting the trajectory format thus saving time and disk space.
- **Fixing broken molecules:** An important step in preparing a trajectory for analysis is to fix any molecules that are broken across a periodic boundary. Not doing so will result in serious inaccuracies for many of the computations performed with MOSAICS. This problem arises in computations that require the center of mass (equation 1.1) or

geometric center (equation 1.3) of a lipid be known. For such cases, a broken lipid will result in an incorrect calculation of the center. Some tools prone to this error include Nearest Neighbors, Lipid Mixing, Lipid MSD, Membrane Thickness, and Lipid Salt Bridges. Broken molecules may be fixed using trjconv with the -pbc whole option. Note, a .tpr file is required for repairing broken molecules. A .gro file will not work here since these files do not contain molecule definitions. If a simulation was performed using an engine other than GROMACS then the molecules should be repaired before performing any conversions of the trajectory. It should be noted that it is not always obvious whether a trajectory contains broken molecules or not. It is therefore a good idea to use the MOSAICS tool Check Broken Mols before performing any analysis (section 2.6).

- **Fixing periodic boundary problems in z:** One problem that can occur with simulations of membrane systems involves the jumping of lipids in the z direction across a periodic boundary. This has the effect of separating the lipids, either from the host leaflet or separating the 2 leaflets altogether (see Figure 2-2). This problem can occur when the bilayer fluctuations in the z-direction are large compared to the z-component of the box. This problem can be fixed by looking for lipid jumps and making corrections accordingly. This can be done using the MOSAICS tool PBC Z (section 2.4).
- **membrane only systems:** For simulations of lipid bilayers, in the absence of any embedded protein, only translations in the z-direction need be removed. This can be done using the MOSAICS tool Bilayer Z (section 2.3).
- **Diffusion coefficients and periodic boundary conditions:** For calculations of the lipid diffusion coefficient, the MOSAICS tool Lipid MSD (section 4.2) is used. Because this tool computes the mean square displacement of the lipids as a function of time the periodic boundary conditions in the x and y directions should be removed. This can be accomplished using the MOSAICS tool PBC XY (section 2.5).
- **Comparing grid data from multiple simulations:** It is sometimes necessary to compare grid data from 2 or more simulations. For example, the user might vary the lipid composition within a set of N simulations and look for changes to $\langle F^{ij} \rangle$ between them. To facilitate such comparisons, the user will want to fit each of the N-1 trajectories to the remaining simulation. More specifically, the first simulation is fit in the usual manner using least squares fitting, etc. Following this, each of the remaining simulations is fit to the same structure that was used for the first simulations. To facilitate this task, we have provided the MOSAICS tool System Translator (section 2.7).
- **Adjusting the trajectory time:** For analysis concerned with the lipid dynamics the trajectory time is often needed. If the simulation was performed outside of GROMACS and later converted to .xtc or .trr using VMD then there is a good chance that the time for each frame is not preserved. In this case, the user may fix the time values for each frame using the MOSAICS tool Traj Time (section 2.8).

- **Back mapping from a coarse-grained to an atomistic model:** The task of preparing an all-atom membrane system for simulation is one that is challenging to this day. One technique is to select a structure from a coarse-grained simulation, which can be thoroughly equilibrated, and use back mapping to create an all-atom representation. When this approach is taken, the user may wish to select a frame from the trajectory that is as close to the equilibrium state as possible. To facilitate this task, we have included the MOSAICS tool Single Frame Error (section 2.9).

2.2 Simulations with Constrained motions

Protein Translator is an analysis tool used for preparing a trajectory for analysis. This tool is specifically useful for simulations in which a membrane protein is included and whose position is restrained to prevent translations and rotation. In such a simulation, the user typically picks two atom selections for which the center of mass (equation 1.1) is found for both to give centers C1 and C2. Restraints are then applied to these centers so that they are locked into the XY plane. Following this, a third center C3 is found as the center of C1 and C2. Then, C3 is restrained to the YZ plane. With these restraints, the protein should be free to tilt but translations and rotations are suppressed. Because of this, the resulting trajectory may be prepared without performing least squares fitting. Instead, the user only needs remove minor motions arising from wiggle room in the restraints, etc. Protein Translator thus takes two atom selections from the user and computes the center of mass (equation 1.1) for each. Note, it makes sense to use atoms from C1 and C2 for this. The program then finds the center of these two centers, i.e., C3, and translates that center to a user specified point in space.

To use the program the user must specify two atom selections. This is done using the -n1 and -n2 tags. An example follows.

-n1

```
#group_1    1    2    3    4    5
```

-n2

```
#group_2    6    7    8    9   10
```

In addition to this, the user must specify the mass of the atoms via the B-factor in the reference PDB file (-ref). We note that, the masses are set to 1 when a gro file is used as the reference. In this case, the geometric center (equation 1.3) is used in place of the center of mass. And finally, the user must specify the position in space, i.e., the coordinates, that center C3 should be translated to. This is accomplished with the -x, -y and -z tags. Note that Protein Translator moves atoms that are translated outside of the box back inside. This can result in broken molecules which can be fixed in a later step using a program like trjconv and -pbc whole (requires a tpr file and a GROMACS installation). An example of the run commands for Protein Translator is now given:

```
mpirun -n 50 protein_translator_mpi -traj traj.xtc -ref ref.pdb -o traj_translated.xtc -n1
center_1.crd -n2 center_2.crd -x 14.5 -y 14.5 -z 4.5
```

Output from Protein Translator is a trajectory file with the translated system. A snapshot from a trajectory prepared with Protein Translator is shown in Figure 2-1.

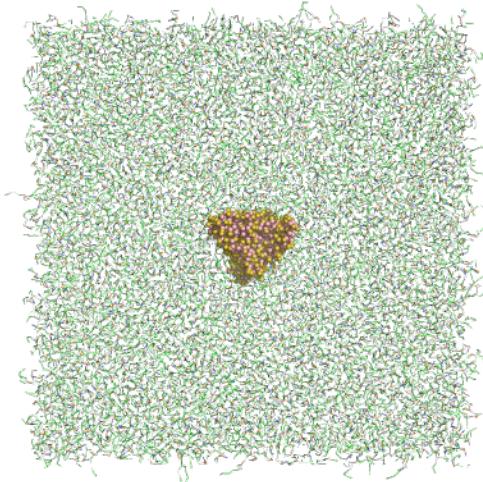


Figure 2-1 CLC-ec1 [10] system prepared with Protein Translator. Many of the heatmaps shown throughout this manual were generated using trajectories prepared with this tool.

2.3 Bilayer Simulations

Bilayer Z is an analysis tool used to prepare membrane simulations for analysis. Specifically, the program is used to remove bilayer translations in the z direction. To accomplish this, Bilayer Z computes the center of mass (equation 1.1) of the membrane and then shifts the system so that this center's z-component resides at a user specified z-coordinate. We note that the mass data is taken from the B-factor specified in the reference PDB file (-ref). If a gro file is used for the reference file, then the masses are set to 1 and a geometric center (equation 1.3) is used instead. Otherwise, the z-component of the bilayer center of mass is computed as:

$$C_z = \frac{1}{M} \sum_i^N m_i z_i \quad (2.1)$$

where M is the mass summed over the bilayer atoms, i.e., $M = \sum_i^N m_i$, N is the number of atoms in the bilayer selection, and z_i is the atomic z coordinate of atom i. To use the program, the user must specify the z coordinate for which the bilayer center of mass is to be translated to. This is done with the -z tag. The user must also specify the leaflet to be included in the computation. This is done with the -leaf tag where the entire bilayer may be chosen with -leaf 0. Note that Bilayer Z relocates any atoms that are moved outside the box during the translation back inside. This can result in broken molecules which can be fixed in a second step using trjconv and -pbc whole (requires an installation of GRAMACS and tpr file). An example of the run commands used by Bilayer Z is now given:

```
$ mpirun -n 50 bilayer_z_mpi -traj traj.xtc -ref ref.pdb -o traj_bilayer_z.xtc -z 4.5 -leaf 0
```

In the example given here, the bilayer center of mass was translated so its z-coordinate was positioned at 4.5 nm. Since our system had a box that was approximately 9 nm in this direction, the bilayer was thus placed in the box center.

2.4 Fixing Periodic Boundary Conditions in Z

PBC Z is an analysis tool used for preparing membrane protein trajectories for analysis. Specifically, the program is used to fix fragmented leaflets. This is a periodic boundary problem caused by bilayer fluctuations in the z-direction combined with a short box height. Under these conditions, atoms from a subset of the lipids can cross the boundary. If the molecules are made whole, this could result in the lipids being placed on the opposite side the box relative to the remaining leaflet lipids (see Figure 2-2).

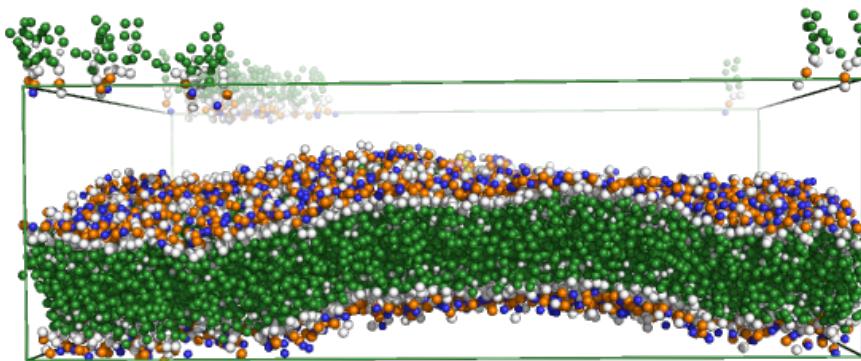


Figure 2-2 Snapshot showing a fragmented leaflet where some lipids have jumped across the periodic boundary in the z-direction.

PBC Z works to correct fragmented leaflets by removing periodic boundary jumps in the z-direction. More specifically, a jump number $J_{i,t}$ is recorded for each atom i at each trajectory frame t. This jump number is history dependent and is computed as:

$$J_{i,t} = J_{i,t-1} \pm 1 \quad (2.2)$$

where 1 is added to $J_{i,t-1}$ when a jump is detected across the upper boundary and 1 is subtracted from $J_{i,t-1}$ when the lower boundary is crossed; otherwise, $J_{i,t} = J_{i,t-1}$ if no jump is detected. The corrected atomic coordinates $\vec{r}_{i,t}^c$ are thus computed as:

$$\vec{r}_{i,t}^c = \vec{r}_{i,t} + J_{i,t} b_{z,t} \quad (2.3)$$

We note that jumps are detected by looking for atoms whose z-coordinate changes by a large amount between neighboring frames. Specifically, the program looks for jumps that are greater in magnitude than a user specified percentage (-cutoff) of the box z-component (b_z). This strategy only works if the first frame in the trajectory is not fragmented. What's more, the probability that the strategy works increases with b_z . The program can fail if the box size is small such that the cutoff value times b_z approaches the size of atomic fluctuations. This suggests the use of large cutoff values. However, one must also consider fluctuations in membrane curvature which also change the lipids z coordinate between neighboring frames. For example, it could happen that a lipid is positioned close to the middle of the box in one frame. Then, in the next frame the bilayer

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curves up placing the lipid outside the box. The lipid is then placed on opposite side of the box. Taking the delta z , we get a value closer to $\frac{1}{2} b_z$ rather than b_z . This is because the curvature of the bilayer has canceled some of the change caused by the periodic boundary shift. To minimize errors caused by this type of effect and those discussed previously, we suggest using a cutoff value of 0.5. An example of the run commands used by PBC Z is now given:

```
$ mpirun -n 1 pbc_z_mpi -traj traj.xtc -ref ref.pdb -o traj_pbc_z.xtc -cutoff 0.5
```

We note that PBC Z shifts atoms for the protein and membrane atoms only. That is, the atoms detected by solvent finder are not shifted. Consequently, the user must make certain that the solvent atoms are identified correctly. If not, then these atoms will be shifted with the protein and lipids which can lead to long term drift, i.e., diffusion, in the z -direction.

It should be noted that detecting a fragmented bilayer is not always an easy task. In the example shown in Figure 2-2, less than one percent of the 50,000 trajectory frames contained a fragmented leaflet. In-fact, upon visual inspection of the trajectory where every 1000 frames were written to PDB, not a single frame contained a fragmented leaflet. Cases where a small percentage of trajectory frames are fragmented can be detected by analysis of a free energy profile created using Membrane Thickness (section 3.1). In this case, even a small fraction of fragmented lipids can lead to a second minimum in the profile (Figure 2-3).

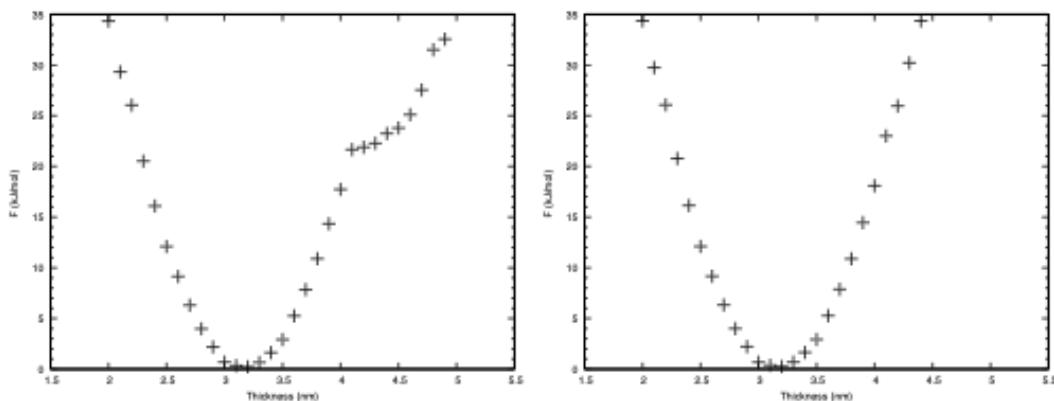


Figure 2-3 Free energy as a function of membrane thickness. Thickness is measured between pairs of lipids in opposing leaflets. Thus, the energy is kJ/mol where a mol refers to the number of lipid pairs. Left panel shows results on a trajectory containing a small number of frames with fragmented leaflets. Right panel shows the same analysis after fixing the fragmented leaflets with PBC Z.

Furthermore, PBC Z will report the trajectory frame and atom number for the first 100 jumps detected. The program can also be set to report the jump number $J_{i,t}$ for each atom and for each frame t . This option is specified using the -record tag and the resulting data can be plotted as a heatmap like that shown in Figure 2-4.

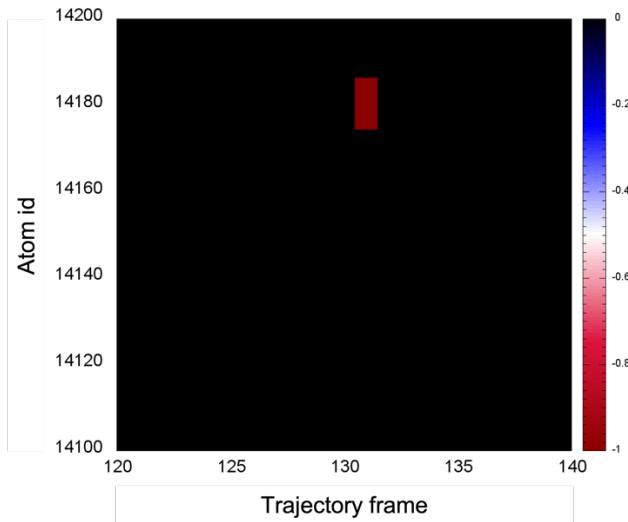


Figure 2-4 The jump number $J_{i,t}$ for each atom as a function of the trajectory frame number. Here we focus on a single lipid that jumped across the lower boundaries of the box. This event lasted for a few frames and the lipid then jumped back across the upper boundary. The jump number for this event is shown in red.

2.5 Diffusion Coefficients and Removing Boundary Conditions in XY

As is discussed in section 4.2, the lipid diffusion coefficient may be computed using the MOSAICS tool Lipid MSD. However, before using this tool the periodic boundary conditions should be removed from the x and y dimensions in a process commonly referred to as “unwrapping” the system. There are many tools available for unwrapping molecular systems that are included in popular platforms like VMD and GROMACS. However, most unwrapping tools are ad hoc developments and were not necessarily created with the computation of diffusion coefficients in mind. Problematically, these methods fail to account for non-diffusive motions arising from changes in the box volume thus leading to exaggerated motion of lipid molecules in constant pressure simulations, a problem that grows as the simulation time increases [11]. To avoid these errors, specialized unwrapping tools have been proposed [11, 12]. Here we introduce an unwrapping tool called PBC XY that mirrors the method proposed by Smith and Lorenz [12].

PBC XY works by comparing the wrapped coordinates $\vec{r}_{i,t}^w$ for each atom i between trajectory frames t and $t-1$. Then, if the position of atom i moves in, for example the x direction, by more than $\frac{1}{2}$ the corresponding box dimension $b_{x,t}$, then it is concluded that the atom crossed the boundary and was reflected inside the box on the opposite side. PBC XY monitors these jumps and makes corrections to remove them by adding a corrective term that depends on the box dimensions at the time the jump occurred. More specifically, the unwrapped x-coordinate $\vec{r}_{i,t,x}^u$ at time t is given in relation to the wrapped coordinate as:

$$\vec{r}_{i,t,x}^u = \vec{r}_{i,t,x}^w + \sum_{\tau=0}^t J_{i,\tau} b_{x,\tau} \quad (2.4)$$

where $J_{i,\tau}$ is either a 0 if no jump was detected at time τ or a plus or minus 1 depending on which direction the jump occurred in. Unwrapping in the y-direction may be achieved using the same approach but replacing x with the y-coordinates.

To use PBC XY, the user must provide the percentage of the box dimension required before counting an atom as having jumped. This is provided with the -cutoff tag. An example of the run commands used with PBC XY is now given:

```
$ mpirun -n 1 pbc_xy_mpi -traj traj.xtc -ref ref.pdb -o traj_pbc_xy.pdb -cutoff 0.5
```

For an example of data generated with PBC XY, see Figure 2-5.

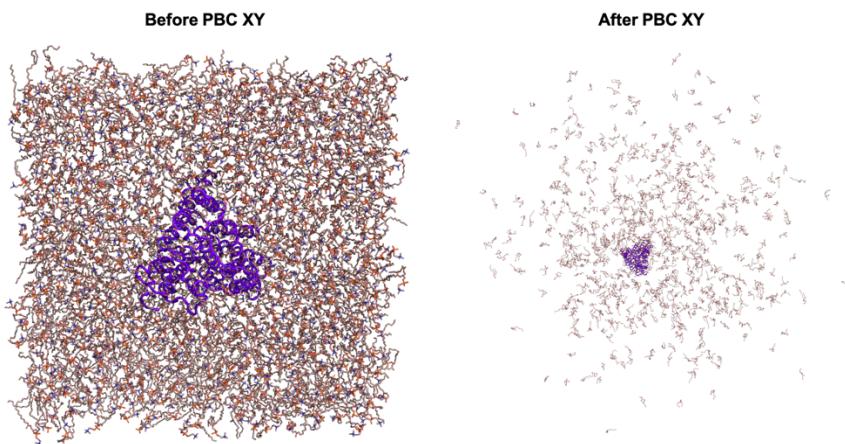


Figure 2-5 Snapshot of a system before (left) and after (right) removing the periodic boundary conditions in XY.

2.6 Checking for Broken Molecules

Check Broken Mols is an analysis tool used for checking a trajectory for broken molecules. More accurately, the program looks for broken residues. To facilitate this task, each residue must be distinguishable from the others. Because MosAT renames the residues, we can be certain that each identifier is unique. With this information, a broken residue can be identified by analyzing the distance matrix for the atoms making the residue. For a broken residue, one or more distances will be unnaturally large. In principle, the same method could be used to check for broken molecules. However, we lack a unique identifier for the atoms of each molecule. Despite this difficulty, many molecules are composed of a single residue like waters, ions, and lipids. Thus, the tool is used to identify broken molecules of this type. The program works by measuring the distance between each atom in the residue and every other atom in the same residue. This gives $N^2/2$ distances for each residue where N is the number of atoms in the residue. If any of these distances is greater than $\frac{1}{2}$ of any of the box dimensions, then the residue is reported as being broken. To use the program, the user only needs to provide a trajectory and reference file as shown in the following example:

```
$ mpirun -n 1 check_broken_mols_mpi -traj traj.xtc -ref ref.pdb
```

Output from Check Broken Mols includes a list of broken residues and the trajectory frame. A PDB can also be written if the -o tag is included. Here, the broken molecules will be indicated by

their B-factor (Figure 2-6) making for easy identification with PyMOL (Schrödinger, LLC) or VMD [9].

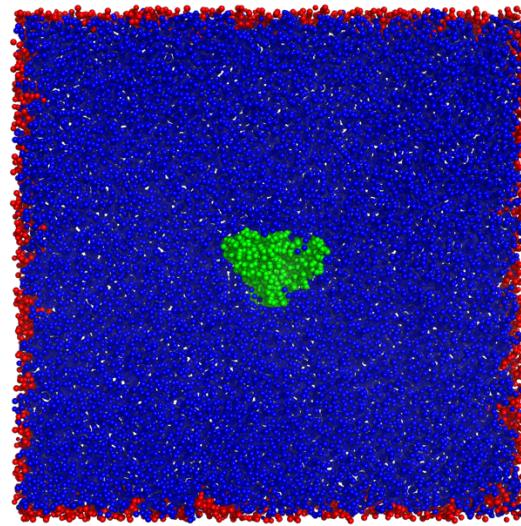


Figure 2-6 Broken molecules detected by Check Broken Mols. The broken molecules are colored red and non-broken molecules blue. The protein is shown in green.

2.7 Comparing Multiple Simulations

System Translator is a system prep tool used for aligning a membrane protein system on top of another membrane protein system. To see why this is useful, consider the following example. Suppose we have performed 2 simulations (A and B) of the CLC-ec1 protein (**Error! Reference source not found.**). In simulation A we have CLC in a pure POPC bilayer and in simulation B a mixture of POPC and DLPC. Suppose then that we wish to compare the solvation structure of the 2 simulations. This can be done by projecting a property of the lipids onto a 2-dimensional grid for each system and then using Delta Plot (section 3.1) to compare how the simulations differ. Such analysis will therefore require alignment of protein A onto protein B such that the grid point i,j correspond to the same region of space for both systems. To accomplish this, let us assume that simulation B has been prepared already, i.e., the protein has been fit to a reference structure and placed in the center of the grid. We will thus prepare trajectory A to match the rotation and position of the protein in trajectory B. This is done with System Translator. First, let us focus on fitting protein A to match that of protein B. For this example, we assume that protein B was fit to a reference structure called ref_b. So, it will be enough to use the least squares fitting routine of MosAT (also part of System Translator) if we provided ref_b using -ref, -lsq_r 0 and an index for doing the fit with -lsq (see 1.6). Of course, ref_b is not compatible with trajectory A since the lipid types differ, so we cannot use this reference structure. However, if there are no mutations made to the protein, then we can copy the protein coordinates from ref_b onto ref_a, where ref_a is the reference file corresponding to trajectory A. With this, the least squares fitting routine will align the protein in trajectory A to the same reference structure that was used for trajectory B. With the fitting part out of the way, all that remains is to translate the center of protein A onto protein B. This is necessary since the least squares fitting routine of MosAT simply translates the system back to its original location (taken from the first trajectory frame) after performing the

rotation. To accomplish this, we provide System Translator with trajectory A and B as well as reference files A and B. With this, the program can determine the original position of trajectory B in the first frame. These files are provided with the -traj, -ref, -traj_b and -ref_b tags. Moreover, we must also include an index for each system. This index tells which atoms to use when determining the center of the protein. An example is given below.

```
$ mpirun -n 10 system_translator_mpi -traj traj_a.xtc -ref ref_a.gro -traj_b traj_b.xtc -ref_b  
ref_b.gro -ind ca_a.ndx -ind_b ca_b.ndx -lsq ca_a.ndx -lsq_d 2 -lsq_r 0 -o  
system_a_aligned_to_b.xtc
```

For a quick check that the fitting and alignment procedure worked as desired, use Mean Protein Coords to compare the mean protein coordinates from both trajectories. An example is shown in Figure 2-7.

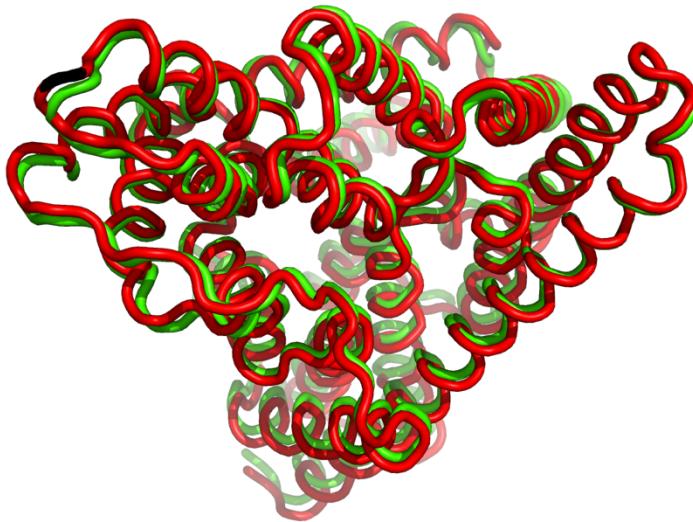


Figure 2-7 Time average protein coordinates from trajectory A (red) and B (green) in the example described above. Note that the proteins from each trajectory are now pinned to each other. With this, the grid-based analysis should be directly comparable.

2.8 Fixing Mistakes in the Trajectory Time

Traj Time is an analysis tool used for correcting the time in a trajectory file. For example, there are some instances when the time stored for each trajectory frame can become erroneous. For example, trajectories collected with the Anton machine can be converted into .trr format using VMD [9] and then to .xtc with MosAT. However, in this process the time is not correctly preserved. This matter is made worse if the original trajectory is too big to be handled by VMD. In this case the user can convert the trajectory in chunks and splice them together with trjcat. This will ultimately result in duplicate times within the trajectory file. This problem will later manifest itself, for example, if the user tries to measure the RMSD vs time. To get around this problem the user can manually override the time in the trajectory file with Traj Time. To use Traj Time the user only needs to specify the time step between frames. This is provided with the -dt tag as is demonstrated in the following run commands:

```
$ mpirun -n 1 traj_time_mpi -traj traj.xtc -ref ref.pdb -o traj_100ps.xtc -dt 1000
```

In the example provided here, the output trajectory file with the corrected time is specified with the `-o` tag. We note that *Traj Time* does not support *gro* or *pdb* for the output file type, i.e., the time will not be modified from the input trajectory for these types.

2.9 Finding a Structure for Back Mapping

Single Frame Error is an analysis program designed to select the frame from the trajectory that best matches the average behavior of the system. The program works by analyzing single frame grid data F_t^{ij} (produced with Z-coord, P2, Lipid Distances, etc.) and comparing this with a grid containing the average of the same observable $\langle F^{ij} \rangle$.

To use the program, the user must specify the data file containing $\langle F^{ij} \rangle$. This is done using the `-d` command line argument. Similarly, the single frame data is specified by giving the base file name (section 1.14) for the single frame data. For example, if the user has single frame z-coord data `upper_z0.dat` the user would use `-base upper_z`. The program will add the frame number and `.dat` to this base for each file to be opened. Additionally, the user must specify how many single frame files there are to analyze. This is done with the `-frames` tag. And finally, the user must provide a masking file (section 1.13) that tells how the error of each grid point should be weighted. This is done with the `-mask` tag. Note the masking file should contain only zeros and ones. With this, only grid points with a 1 will be counted when comparing each frame to the average. This allows the user to compare regions near the protein while ignoring those far away. An example of the run commands used with Single Frame Error is now given.

```
$ mpirun -n 10 single_frame_error_mpi -d upper_z.dat -mask prot_mask.dat -base upper_z -frames 50000 -odf 0 -o upper_error.dat
```

In searching for the frame which best matches the average, an error function is computed. Here the error is defined as:

$$E = \sqrt{\frac{1}{N} \sum_i \sum_j (F_t^{ij} - \langle F^{ij} \rangle)^2 m^{ij}} \quad (2.4)$$

where F_t^{ij} is the value of grid point i,j from the single frame and $\langle F^{ij} \rangle$ is the corresponding time average. Furthermore, N is the number of grid points where there exists data for both the single frame and the average and where the mask i.e., m^{ij} , has a value of 1. Put simply, the error function is the average deviation of the single frame data compared to the average. It should be noted that each deviation is squared so that error does not cancel. Moreover, the square root ensures that the units are the same as the quantity being analyzed. For example, if the error is computed for the z-coord then the units will be nm. This should give the user intuition about the results such that a result of 0.1 nm would mean the z-coord is on average 1 angstrom from the average, a result that would be reasonably good. Output from Single Frame Error includes a list of each frame and the error as well as the frame with the lowest error (Figure 2-8). To extract a PDB from this frame the user can use MosAT and the `-b` `-e` tags to dump the frame. For example, if frame 36,289 had the lowest error, then the user could get a PDB of this frame with:

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```
$ mpirun -n 1 mosat_mpi -traj traj.xtc -ref ref.gro -o best_frame.pdb -b 36389 -e 36289
```

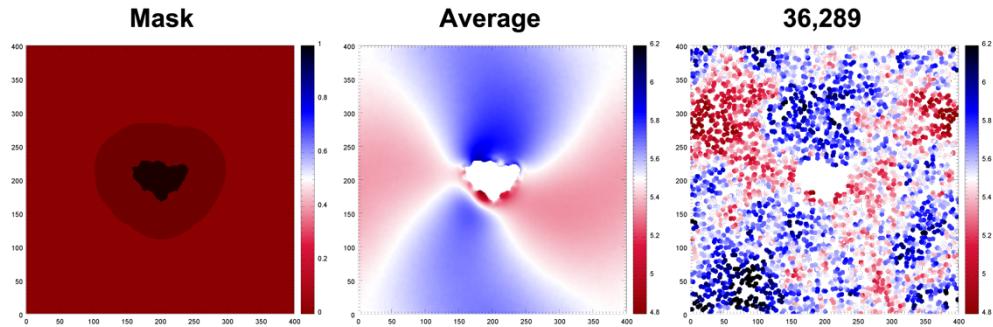


Figure 2-8 Mask highlighting grid points to include in error analysis (left panel). The protein mask is shown as well (dark black). The average z-coordinate (nm) for which the error is based (middle panel). The z-coordinate (nm) for the frame with the lowest error (right panel). Results show the best frame (36,289) is within 1.5 angstroms from the mean on average. Units for the x/y axis are grid points. The color bars have units of nm (middle and right panels) or no units at all (left panel).

Chapter 3 Analysis of Lipid Structure

3.1 Membrane Curvature and Thickness

Observables like the membrane thickness and curvature are often affected by the presence of an embedded protein and can even affect its stability. For this reason, these properties are often characterized early in the analysis phase of a research project. With MOSAICS tools there are 2 programs used to characterize the bilayer thickness. These include the programs Z Coord and Membrane Thickness. One of the benefits of using Membrane Thickness is that individual thickness measurements are made thus making it possible to create a free energy profile for compressing a pair of lipids. On the other hand, Z Coord can also be used to extract a profile of the membrane curvature. Thus, each program has a unique set of capabilities. In the remainder of this section, we examine each of these programs in greater detail.

Z Coord is an analysis tool used for computing the average z-coordinate for a user specified group of atoms. This data is then projected onto the XY plane and the time average computed. The program works by selecting atoms from the designated lipid types and adding their z-coordinates to the grid around the mapping atom. Use of the program therefore requires specifying this information. This is done by feeding Z Coord a selection card using the -crd tag. The contents of this card could be something like the following:

```
-crd
#lip_t  #z  #map
POPE   GL1  GL1
POPE   GL2  GL2
POPG   GL1  GL1
POPG   GL2  GL2
```

In the example above the user has specified the lipid types POPE and POPG with the ester atoms GL1 and GL2 selected for z-coordinate measurements. This information is then added to lattice points around the mapping atoms GL1 and GL2 respectively. The selection card, as structured this way, makes it possible to measure the z-coordinate of specific lipids within a complex mixture and probe specific atom types like the tail or head atoms. In addition to this, Z Coord can be used to compute the membrane thickness. To do so the user must measure the average z-coordinate for the upper and lower leaflets as shown below:

```
$ mpirun -n 50 zcoord_mpi -traj traj.xtc -ref ref.pdb -crd popx.crd -z upper_z.dat -APS 0.005 -r 0.26 -cutoff 0.4 -leaf 1
```

```
$ mpirun -n 50 zcoord_mpi -traj traj.xtc -ref ref.pdb -crd popx.crd -z lower_z.dat -APS 0.005 -r 0.26 -cutoff 0.4 -leaf 2
```

In the examples provided here, the target leaflet is selected with the -leaf tag and the output filename containing the time averaged z-coordinates is indicated using the -z tag. Once both leaflets have been analyzed the user can use the MOSAICS tool Delta Plot to find the difference between the two surfaces as shown in the following example:

```
$ delta_plot -d2 upper_z.dat -d1 lower_z.dat -o delta_z.dat -odf 0
```

Similarly, the user can find the midplane between the upper and lower leaflets using the MOSAICS tool Midplane. Midplane works the same way as Delta Plot but instead of taking the difference, an average of the leaflet z-coordinates is computed. An example is given below.

```
$ midplane -d1 upper_z.dat -d2 lower_z.dat -o midplane.dat -odf 0
```

For both Delta Plot and Midplane, grid points are excluded where one of the leaflets has no data (NaN). An example of the output from Z Coord, Delta Plot and Midplane is given in Figure 3-1.

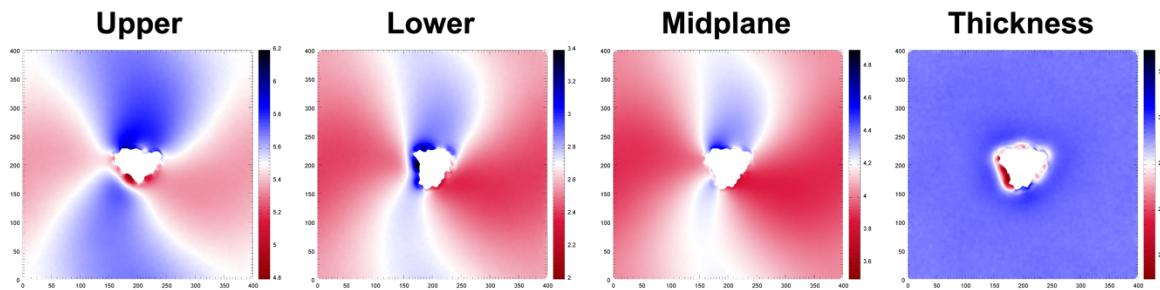


Figure 3-1 The average z-coordinate for the upper leaflet (far left panel) and lower leaflet (middle left panel). The middle right panel shows the midplane between the upper and lower leaflets. The far-right panel shows the membrane thickness. Z-coordinates were measured for the ester atoms in a POPE/POPG bilayer. Units for the x/y axis are grid points and nm for the color bars.

Like Z Coord, Membrane Thickness is an analysis tool designed to probe the membrane thickness within a membrane simulation and project this information onto the XY plane. The program works by measuring the instantaneous bilayer thickness for individual lipid pairs. This is done for every frame of the trajectory as follows. First, Membrane Thickness looks for lipids of a given type in the target leaflet. Once identified, a group of atoms is selected from the lipid and the geometric center (equation 1.3) is calculated. This process is then repeated for the opposing leaflet such that the target lipids are again selected, and a geometric center computed for each. The distance in XY is then measured between the target leaflet lipid center and the centers in the opposing leaflet. Membrane Thickness looks for the opposing lipid with the shortest distance in XY while requiring this distance be less than a user specified cutoff (-xy). If such a pair is identified, then the distance in the z-direction is measured and stamped to the grid around the mapping atoms of the target leaflet lipid. To use the program, the user must specify the target lipids as well as the atoms used for geometric center calculations. This information is provided using a network of selection cards as specified with the -crd tag (Figure 3-2).

-crd			
#lipid_type	#map_1	#map_2	#filename
POPE	GL1	GL2	gl_12.crd
POPG	GL1	GL2	gl_12.crd
DLPE	GL1	GL2	gl_12.crd
DPLG	GL1	GL2	gl_12.crd

#center_atom
GL1
GL2

#center_atom
GL1
GL2

#center_atom
GL1
GL2

#center_atom
GL1
GL2

Figure 3-2 Selection card structure used by Membrane Thickness. Here the POPE, POPG, DLPE, and DPLG lipids are included in the computation. The geometric center of the GL1 and GL2 atoms is then used to represent these lipid's positions in XY. Once a thickness measurement is made, the value is stamped to the lattice around the mapping atoms GL1 and GL2.

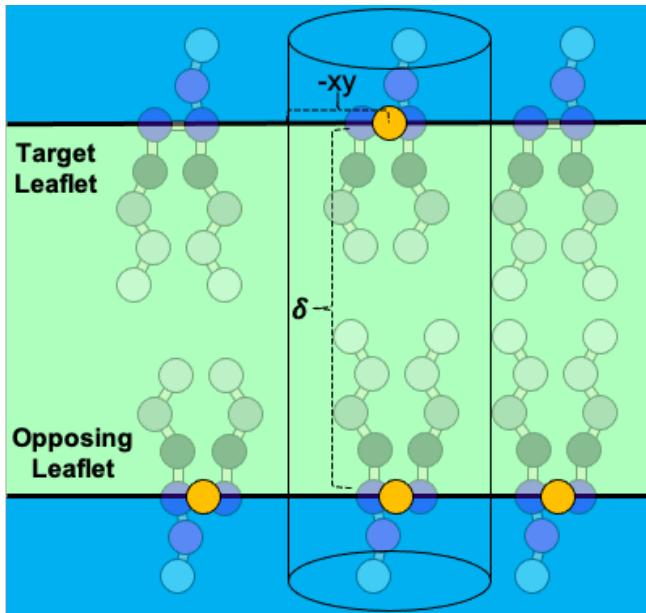


Figure 3-3 Membrane thickness measured for a lipid pair. Geometric centers for ester atoms are shown in orange. The maximum cutoff distance for making a pair is represented as a cylinder.

We note that the lipid types included in the calculation are identical for the target and opposing leaflets as specified in the selection card, i.e., pairs are found from the given types. However, it is possible to use a subset of the lipid types (given via the selection card) in the target leaflet while using all types (irrespective of what is specified in the selection card) in the opposing. This option may be used by inclusion of the -all 1 tag.

In short, Membrane Thickness measures the thickness for lipid pairs which are the closest to each other in XY (Figure 3-3). One benefit of measuring bilayer thickness this way as opposed to using Z Coord is that it lets the user quantify the spread in distances over the simulation. This can be computed by inclusion of the -stdev 1 tag as shown in the following example:

```
$ mpirun -n 50 membrane_thickness_mpi -traj traj.xtc -ref ref.pdb -crd podl.crd -thk upper_thk.dat -APS 0.005 -r 0.26 -cutoff 0.4 -leaf 1 -xy 0.5 -width 0.1 -temp 303.15 -stdev 1 -clean 1
```

Note we have specified the name of the output grid data containing the membrane thickness using the -thk tag. This filename is used to generate additional filenames, like the sample count, and, as we will see, several other files containing useful data. This approach is taken to reduce the amount of input required by the user. An example of data generated with Membrane Thickness is shown in Figure 3-4.

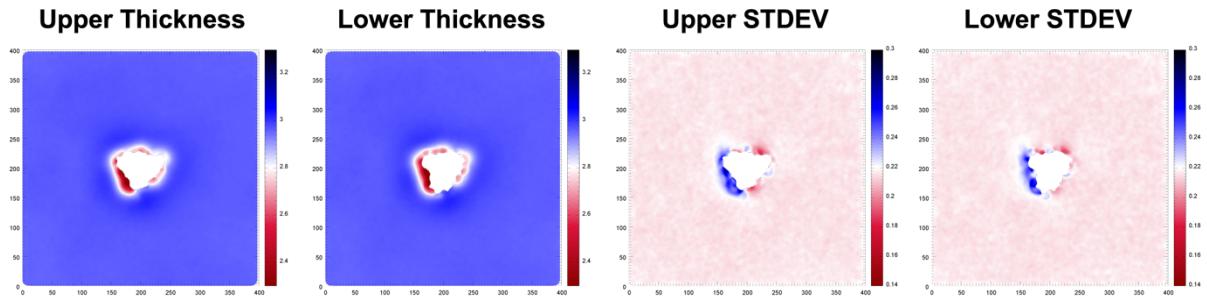


Figure 3-4 Membrane thickness (left 2 panels) and the standard deviation of the thickness (right 2 panels). Analysis was run twice with the upper or lower leaflet considered as the target leaflet. Results should be very similar and can be averaged using Leaflet Averager. Units for the x/y axis are grid points and nm for the color bars.

We note that Membrane Thickness checks each thickness measurement for consistency and reports an error when the sign of dz is inconsistent. Receiving this warning may indicate that a lipid was incorrectly assigned to a leaflet or that jumps are occurring for lipids in the z-direction (section 2.4), etc. It should also be noted that if the $-xy$ parameter is too small then some target lipids will fail to find a partner from the opposing leaflet for which a thickness measurement can be taken. When this happens the target lipid will not deposit a thickness measurement to the grid. Furthermore, this will lead to a sample count (ρ^{ij}) that is incomplete. Keep in mind that ρ^{ij} is used to compute the average thickness, i.e., the average thickness is found by dividing by the number of samples which contributed to the grid point. However, a second sample count is computed which includes all target lipids including those that could or could not find a partner from the opposing leaflet and is used for excluding insignificant data. To check if a larger $-xy$ value is needed, Membrane Thickness computes the percentage of lipids that found a partner and projects this information onto the XY plane along with the two sample counts (Figure 3-5). These files are given a name derived from the membrane thickness grid data but with the “_rho”, “rho_t”, and “_pairs_percent” appendages.

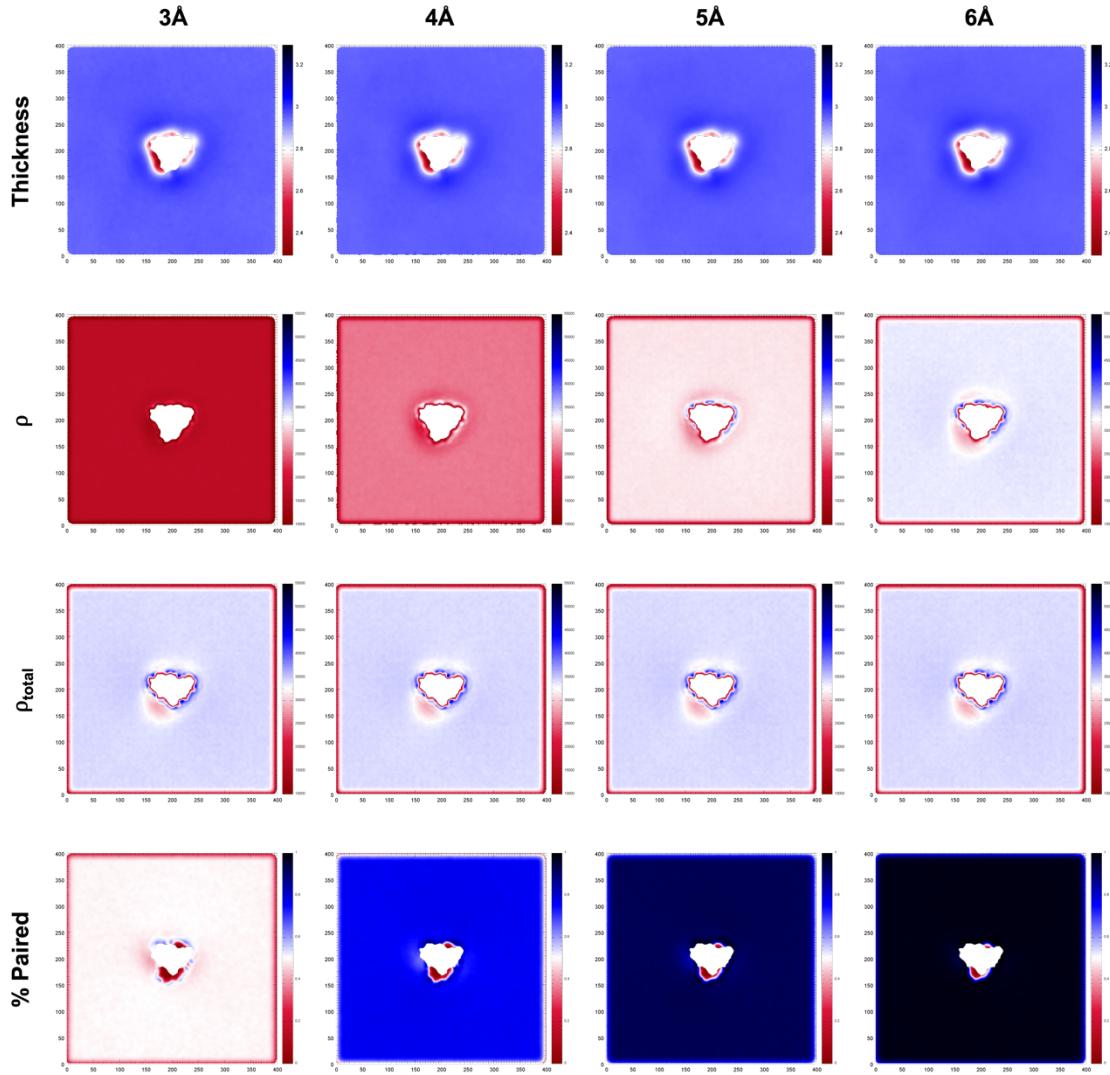


Figure 3-5 Exploration of increasing $-xy$ values. Top row shows the average thickness (nm). The second row from the top shows the sample count for lipids that successfully found a pair in the opposing leaflet while the second panel from the bottom shows rho for all target lipids. The bottom panel shows the percentage of target lipids that found a partner. Units for the x/y axis are grid points. The color bars have units of nm (thickness row), number of lipids (ρ and ρ_{total} rows) or no units (% percent paired row).

In addition to projecting the average thickness onto XY, Membrane Thickness also computes a free energy profile as a function of thickness. This is done by combining thickness measurements into a single list. Then, after all frames of the trajectory have been analyzed the list is sorted into bins whose width is set via the $-width$ tag and the free energy computed by:

$$A(\delta) = -\frac{1}{\beta} \ln[P(\delta)] \quad (3.1)$$

where $P(\delta)$ is the probability of a lipid pair having a thickness δ and $\beta = 1/k_B T$ such that k_B is the Boltzmann constant and T is the temperature (set with the $-temp$ tag). Furthermore, the free energy profile is shifted such that the minimum has a value of zero. This is computed as:

$$A(\delta) = -\frac{1}{\beta} \ln \left[\frac{P(\delta)}{P(\delta)_{max}} \right] \quad (3.2)$$

where $P(\delta)_{max}$ is the thickness with the largest probability. We note that the free energy profile data is written to a file named like the membrane thickness data but containing the “_free_energy” appendage. A free energy profile generated by Membrane Thickness can be seen in Figure 3-6.

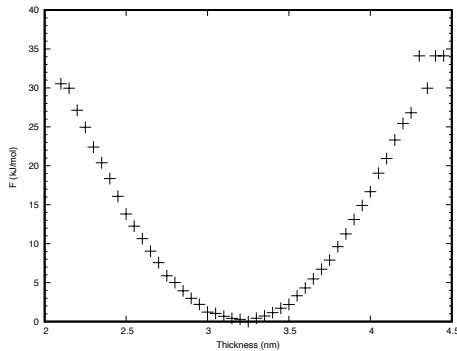


Figure 3-6 Free energy profile as a function of membrane thickness. The free energy is for a mole of lipid pairs. Data comes from martini simulations of a POPE/POPG bilayer.

To help investigate thickness measurements, Membrane Thickness can be set to dump frames from the trajectory containing thickness measurements falling in a user specified range (set with -dump_l and -dump_u, i.e., $-dump_l < \delta < -dump_u$). The resulting DPB will highlight the lipid pair by the B-factor (Figure 3-7).

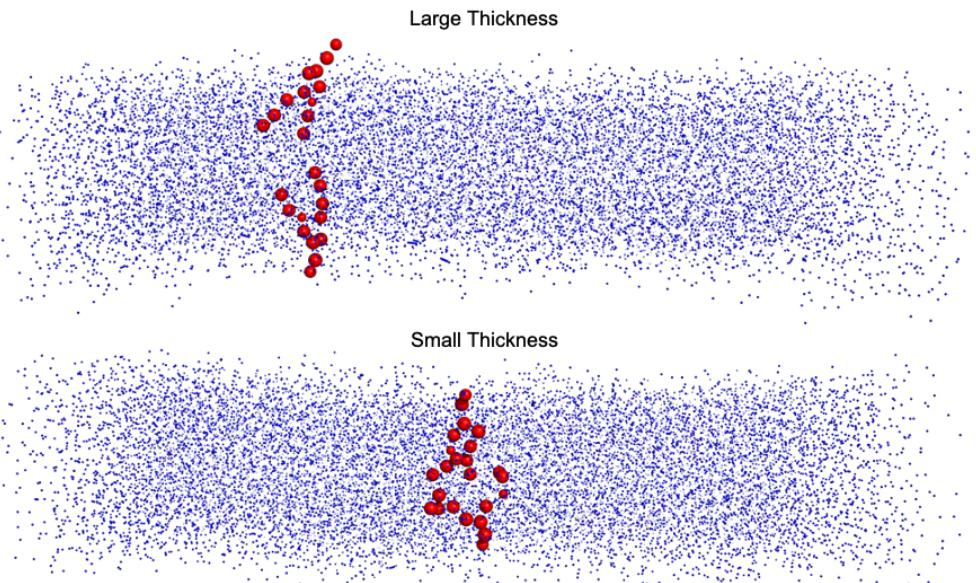


Figure 3-7 PDB showing an unusually large thickness measurement (upper panel) and an unusually small thickness measurement (lower panel).

Using a free energy profile such as that shown in Figure 3-6 one can estimate the energetic costs of a membrane deformation as caused by the presence of a membrane protein. This can be done using the MOSAICS tool Bilayer Free Energy. Bilayer Free Energy works by analyzing thickness data from a grid (Figure 3-4) and looking up the cost of placing a pair of lipids at that thickness from a free energy profile (typically generated by analyzing a membrane simulation with no embedded protein) (Figure 3-6). The free energy contribution for grid point i,j is thus computed as:

$$A^{ij} = \#lip^{ij} A(\delta) \quad (3.3)$$

where $\#lip^{ij}$ is the number of lipids at grid point i,j and is taken from the simulation data. The average number of lipids $\#lip^{ij}$ can be computed with Lipid Density (section 3.2). With each component ready, a free energy profile may be generated using something like the following:

```
$ bilayer_free_energy -d upper_thk.dat -rho upper_rho_norm.dat -fe upper_thk_free_energy.dat
-o upper_fe.dat -odf 0
```

An example of a free energy estimate generated using Bilayer Free Energy is shown in Figure 3-8. Once each grid point has a free energy component the total cost of the deformation can be found by adding up all the grid points around the defect using Grid Region Integrator (section 1.13). This approach is possible since the bulk thickness should equal the free energy minimum which is shifted to have a free energy value of zero. We note that Bilayer Free Energy will also generate grid data for the free energy cost per pair of lipids, i.e., $A(\delta)$. This data is given the same filename as for the free energy penalty data describing equation 3.3 but receives the “_pair” appendage.

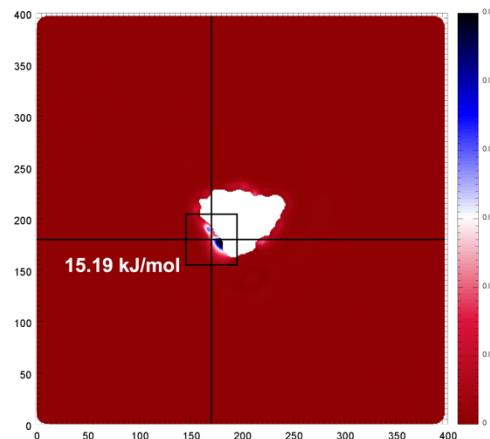


Figure 3-8 Free energy cost of deforming the membrane when the CLC-ec1 protein [10] is present. Grid points were assigned a free energy cost with Bilayer Free Energy and the total cost found by summing over the defect using Grid Region Integrator. Units for the x/y axis are grid points and kJ/mol for the color bar.

We note that the free energy analysis described in this section is meant to give a rough estimate of the penalty associated with a thinning defect as these types of analysis remain an active area of research.

3.2 Normalized Lipid Density

It is sometimes useful to have a normalized lipid density which can be used for analysis. For example, a normalized lipid density was used to count the number of lipids around the protein in section 3.1 when we used Bilayer Free Energy to estimate the free energy penalty of solvating a protein. To aid in such computations, we have the MOSAICS tool Lipid Density. Lipid Density is an analysis tool designed to compute the lipid density in the XY plane. To use the program the user must specify the lipid types and the atoms for which density is deposited to the grid. This information is provided using a network of selection cards and the -crd tag (Figure 3-9).

-crd

#lipid_type	#filename	#target_atom	#target_atom
POPE	gl_12.crd	GL1	GL1
POPG	gl_12.crd	GL2	GL2

Figure 3-9 Selection card structure used by Lipid Density. In this example, the POPE and POPG lipids are selected, and density added to the grid around the GL1 and GL2 atoms of each lipid.

We note that the density computed by Lipid Density differs from the sample count ρ^{ij} computed by other analysis programs such as Z Coord in that this density is normalized. Specifically, Lipid Density deposits density in a circle around the target atoms. However, the density is normalized such that adding up the density over all grid points gives a value of 1 for each lipid. The density is also normalized to account for the number of frames in the analysis. The final density can be summed over the grid to give the number of lipids in the box (for the leaflet analyzed). Note, that the density sum only gives the number of lipids in the box if the lattice is large enough to encompass all the lipids and the system positioned inside the boundaries before performing the analysis. An example is now given:

```
$ lipid_density_mpi -traj traj.xtc -ref ref.pdb -crd podl.crd -rho upper_rho_norm.dat -APS 0.005 -r 0.26 -cutoff 0.0 -leaf
```

We note that the normalized lipid density is written to a file as specified using the -rho tag. An example of data generated by Lipid Density is given in Figure 3-10.

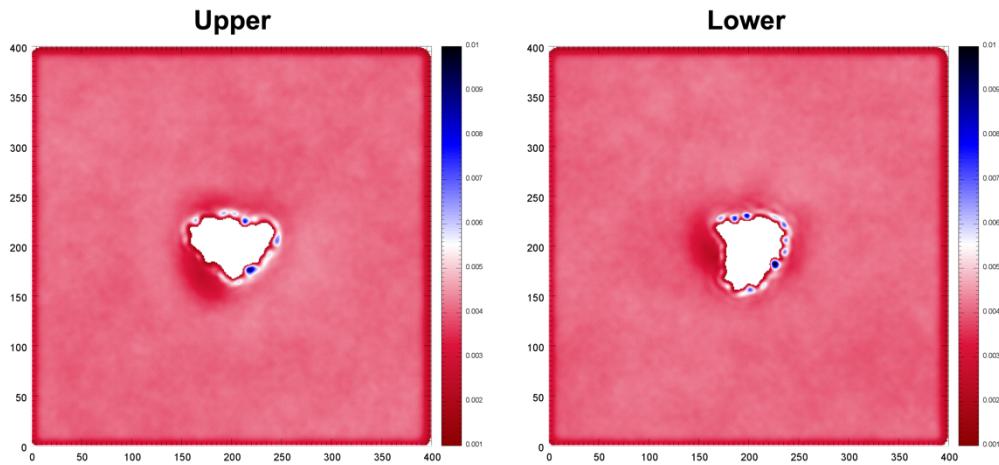


Figure 3-10 Normalized lipid density for the upper (left panel) and lower (right panel) leaflets. Data is taken from coarse-grained simulations of the CLC-ec1 membrane protein [10]. Units for the x/y axis are grid points and lipids for the color bar. Note summing over the grid will give the number of lipids in each leaflet.

3.3 The Rank 2 Order Parameter

The rank 2 order parameter is a commonly reported value in experimental and computational studies of lipids. This observable provides insight into the lipid tilt angle and can be used to detect variations across the grid. To perform this type of analysis, one can use the MOSAICS tool P2. P2 is designed to compute the rank 2 order parameter averaged over a chemical group, i.e., a subset of the lipid atoms or the full molecule, and project this data onto the XY plane. The rank 2 order parameter is defined as:

$$P2 = 0.5(3\cos^2(\theta) - 1) \quad (3.4)$$

where θ is the angle made by a pair of bonded atoms and the z-axis (Figure 3-11).

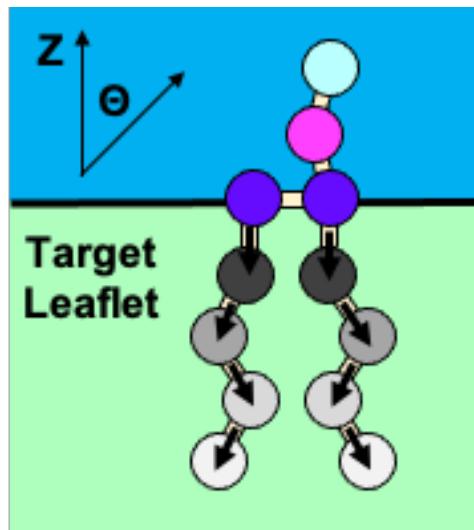


Figure 3-11 Rank 2 order parameter measured for each bond of the lipid tails. The angle is relative to the z-axis.

It then follows that the order parameter averaged over the chemical group is given by:

$$\overline{P2} = \frac{1}{N} \sum_{n=1}^N P2_n \quad (3.5)$$

where N is the number of bonds contained within the chemical group and n indexes these bonds.

To use P2 the user must specify which lipid types to include in the analysis, the bonds for which the angles are measured relative to the z-axis and a pair of mapping atoms for adding $\overline{P2}$ to the grid. This is accomplished using a network of selection cards specified with the -bond tag. An example follows:

-bond			
#lipid_type	#map_1	#map_2	#filename
POPE	GL1	JNK	chain_1.crd
POPE	GL2	JNK	chain_2.crd
POPG	GL1	JNK	chain_1.crd
POPG	GL2	JNK	chain_2.crd

#atom_1	#atom_2
GL2	C1B
C1B	C2B
C2B	C3B
C3B	C4B

#atom_1	#atom_2
GL1	C1A
C1A	D2A
D2A	C3A
C3A	C4A

#atom_1	#atom_2
GL2	C1B
C1B	C2B
C2B	C3B
C3B	C4B

#atom_1	#atom_2
GL1	C1A
C1A	D2A
D2A	C3A
C3A	C4A

Figure 3-12 Selection card structure used by p2. Here we measure $\overline{P2}$ for each acyl chain of POPE and POPG lipids. We note that the second mapping atom of the primary selection card has been set to “JNK”. This is a nonexistent atom type. As such, each measurement of $\overline{P2}$ is mapped to a single atom. This allows us to probe each acyl chain separately. However, both chains could be analyzed simultaneously (treated as a single chemical group) by including a second mapping atom and combining the contents of chain_1.crd and chain_2.crd into a single card. See Figure 3-20 for an example taking this approach.

In the example above, P2 selects lipids with the correct type as specified in the first column of the primary selection card. Then, the secondary selection card corresponding to the current row of the primary card is analyzed. This secondary card defines the bonding atoms whose angles are measured against the z-axis. That is, each row specifies a bonding pair where the first column gives the first bonding atom and the second column the second. We note that each row in the secondary card specifies a value for $P2_n$. Once the secondary selection card has been analyzed, $\overline{P2}$ is computed using equation 3.5. In the example provided here, we measure $\overline{P2}$ for each lipid acyl chain and map this value to the ester atom (GL1 or GL2) in which the lipid tail is connected to. We now give an example of the required run commands:

```
$ mpirun -n 50 p2_mpi -traj traj.xtc -ref ref.gro -bond bonds.crd -p2 upper_p2.dat -APS 0.005 -r 0.26 -cutoff 0.4 -leaf 1
```

Here, the output grid data containing the time average of $\overline{P2}$ is specified using the -p2 tag. An example of this data is shown in Figure 3-13.

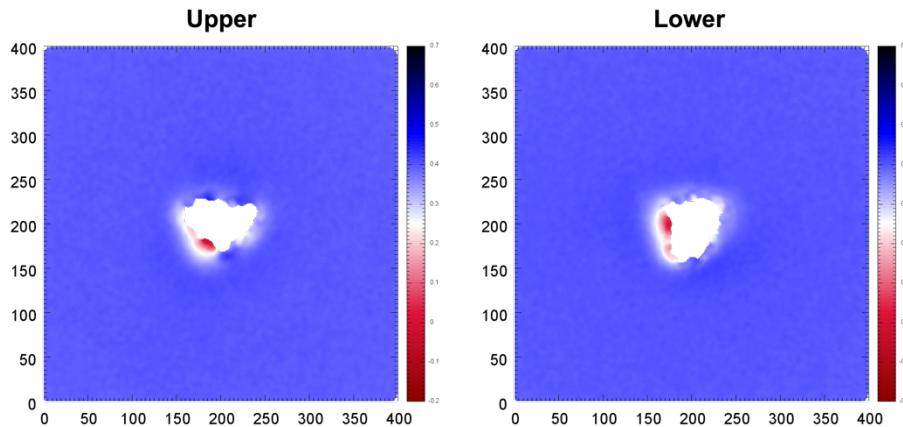


Figure 3-13 The time average of the spatially resolved rank 2 order parameter (\bar{P}_2) averaged over the tails of POPE and POPG lipids in coarse-grained simulations of CLC-ec1 protein [10]. Units for the x/y axis are grid points. The color bars are unitless.

We note that P2 also computes the time average of the spatially resolved observable $\bar{\Theta}$, where $\bar{\Theta}$ is defined as:

$$\bar{\Theta} = \frac{1}{N-1} \sum_{n=1}^N \Theta_n \quad (3.6)$$

i.e., the angle (formed between the pair of bonding atoms and the z-axis) averaged over the specified bonds. This data is given the same filename as the spatially resolved time average of \bar{P}_2 but with the “_theta” appendage.

3.4 Internal Lipid Distances

Researchers are often interested in the distance between a pair of atoms within a lipid molecule. For example, the end-to-end alkyl chain distance can be used to detect compression within the lipid tails. Such compressions could occur in cases where the lipid bilayer becomes unnaturally thin like near the surface of an embedded protein. In addition to this, one could measure the lipid splay distance which could also increase as the bilayer thins. To enable these types of calculations we have the MOSAICS tool Lipid Distances. Lipid Distances is designed to measures the average distance between a pair or pairs of atoms on the lipids and project this data onto the XY plane.

To use the program the user must provide a selection card using the -pairs tag that tells Lipid Distances which lipid types to include, what atoms make the pairs and the mapping atoms used for adding distances to the grid. An example is provided below.

-pairs

#lip_t	#atom_1	#atom_2	#map
POPE	GL1	C4A	GL1
POPE	GL2	C4B	GL2
POPG	GL1	C4A	GL1
POPG	GL2	C4B	GL2

In the above example, Lipid Distances measures the distance between the ester atoms and the C4 tail atoms for both tails of POPE and POPG lipids (Figure 3-14).

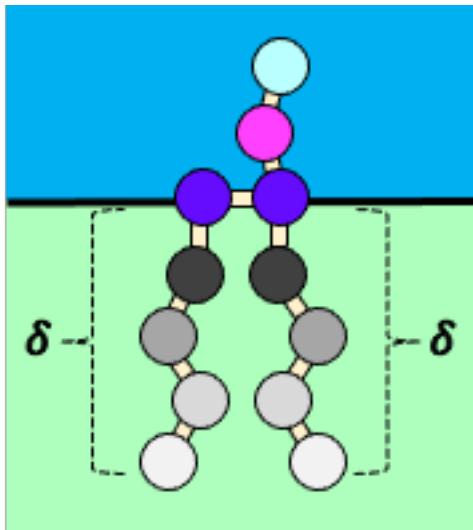


Figure 3-14 End-2-End distance as measured with Lipid Distances. While it is typical to measure the distance for each tail the program is flexible such that other distances can be computed.

The columns of the selection card are organized as follows. First, column one contains the lipid type. Columns two and three contain a pair of atoms in which a distance is to be measured and column four is the atom in which this distance is to be mapped to in the XY plane. We now give an example of the run commands needed to use Lipid Distances:

```
$ mpirun -n 50 lipid_distances_mpi -traj traj.xtc -ref ref.gro -pairs pairs_po.crd -ld upper_dist.dat  
-APS 0.005 -r 0.26 -cutoff 0.4 -leaf 1
```

In this example, the output file containing the time averaged projection of the distance measurements is specified using the -ld tag. An example of output from Lipid Distances is shown in Figure 3-15.

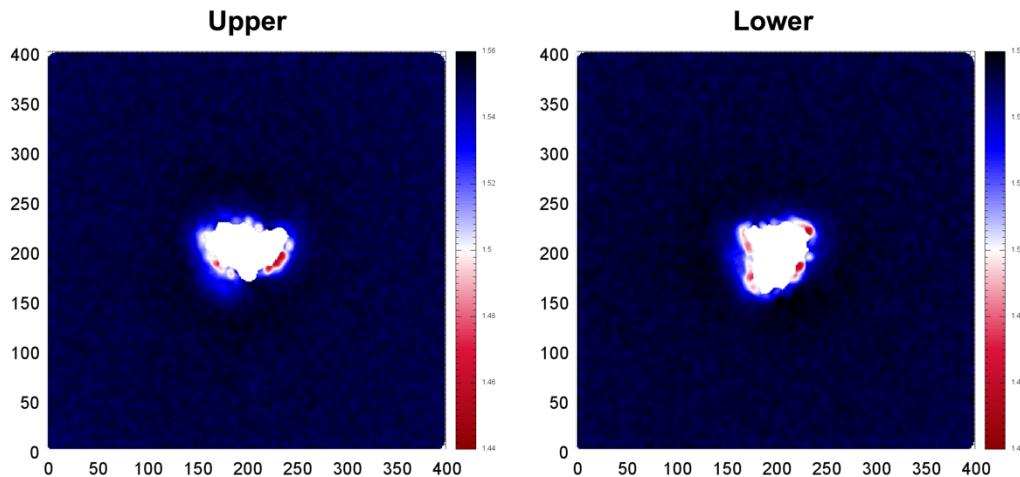


Figure 3-15 The end-to-end distance (nm) for POPE and POPG lipids in coarse-grained simulations of the CLC-ec1 protein [10]. The end-to-end distance is taken to be the distance between the ester atoms (GL1/GL2) and the tail atoms (C4A/C4B). Units for the x/y axis are grid points and nm for the color bars.

3.5 The Lipid Tilt Angle

Preferences in the lipid tilt angle can be probed using the MOSAICS tool Lipid Orientation. Lipid Orientation is designed to probe the preferential tilt angle of the lipids and project this data onto the XY plane. This program works by computing an orientation vector for the lipids, a record of which is kept for each grid point. This vector is usually the one connecting the ester atoms (GL1/GL2) with the corresponding tail atoms (C4A/C4B) (Figure 3-16).

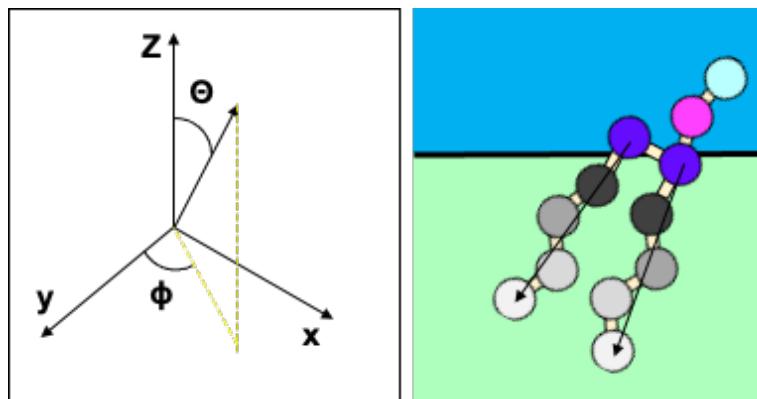


Figure 3-16 Right panel shows a martini lipid and how the orientation vector is defined in the above example. The arrows represent the orientation vectors for each lipid tail. The left panel shows the two angles θ and ϕ which are eventually computed thus characterizing the time average orientation vector. These angles range from 0-180° and 0-360° respectively.

By computing the time average orientation vector, it is possible for the lipid tilt in one direction to cancel if the lipids tilt in the opposite direction with the same frequency. This is typically the case for lipids in the bulk. However, near the protein, the lipids may have a preferred tilt which does not cancel. This preference for tilting can be probed using Lipid Orientation.

To use the program the user must provide the lipid types to be analyzed, the atoms making the orientation vector and the atoms in which the vector is to be mapped to in the XY plane. This information is provided with the -pairs tag. An example is given below.

-pairs

#lip_t	#atom_1	#atom_2	#map
POPE	GL1	C4A	GL1
POPE	GL2	C4B	GL2
POPG	GL1	C4A	GL1
POPG	GL2	C4B	GL2

In the example above, Lipid Orientation probes the POPE and POPG lipid tilt angle such that the orientation vectors are those connecting the ester (GL1/GL2) atoms to the tail atoms (C4A/C4B). Moreover, the orientation vectors are mapped to the XY coordinates of the ester atoms. The columns of the selection card are as follows. Column one gives the lipid type. Columns two and three give the atoms making the orientation vector and column 4 is the atom for which this vector is to be mapped to. The scheme used here allows the user to probe the orientation of both tails of the lipid simultaneously (see Figure 3-16). Once the time average orientation vector is computed (for each lattice point), Lipid Orientation characterizes this vector. Specifically, two angles Θ and Φ are measured (see Figure 3-16). An example of the run commands for Lipid Orientation is now given:

```
$ mpirun -n 50 lipid_orientation_mpi -traj traj.xtc -ref ref.gro -pairs popx.crd -p2 upper_tilt.dat -APS 0.005 -r 0.26 -cutoff 0.4 -leaf 1
```

In this example, the output data file containing the preferred tilt angle is specified with the -p2 tag. We note that this file name is used to generate filenames for the preferred tilt angle Θ as well as other characterizations of the time averaged orientation vector. For example, the preferred tilt angle Θ (Figure 3-16) is given the same name as specified with -p2 but a “_theta” tag is added to the filename. Similarly, the direction of tilt ϕ (Figure 3-16) is computed and stored in a data file containing the “_phi” appendage. Other characterizations of the time averaged orientation vector include its x, y, and z components (given the “_x”, “_y”, and “_z” appendages respectively) as well as the vector length (“_dist”) and the order parameter P2 (computed using the angle given in “_theta” and given the appendage “_p2”). The sample count used for computing the time average orientation vector and for excluding insignificant data points within the grid data is given the “p2_rho” appendage. An example of data generated by Lipid Orientation is given in Figure 3-17.

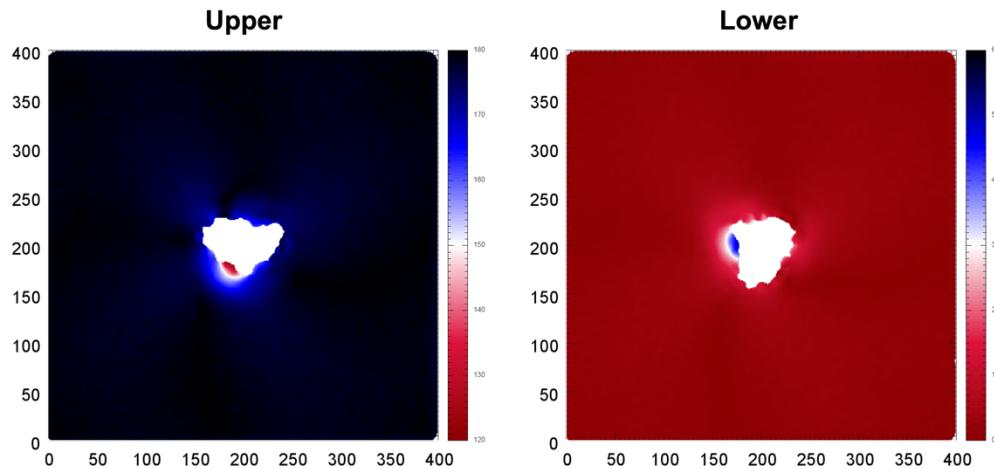


Figure 3-17 The average angle θ for the POPE and POPG lipids in coarse-grained simulations of the CLC-ec1 protein [10]. Units for the x/y axis are grid points and degrees for the color bars.

3.6 Leaflet Interdigitation

It is possible to probe the degree of interdigitation between opposing leaflets using a couple different metrics. For example, the number of interleaflet contacts should correlate with the interdigitation. Similarly, one could examine these contacts and note which atoms in the opposing leaflet the contacts are with. With this metric, contacts with atoms closer to the head groups correspond to a high degree of interdigitation while those near the tails correspond to low interdigitation (Figure 1-1). In this section, we examine 2 tools used to characterize the lipid interdigitation. These are the MOSAICS tools Interleaflet Contacts and Interdigitation.

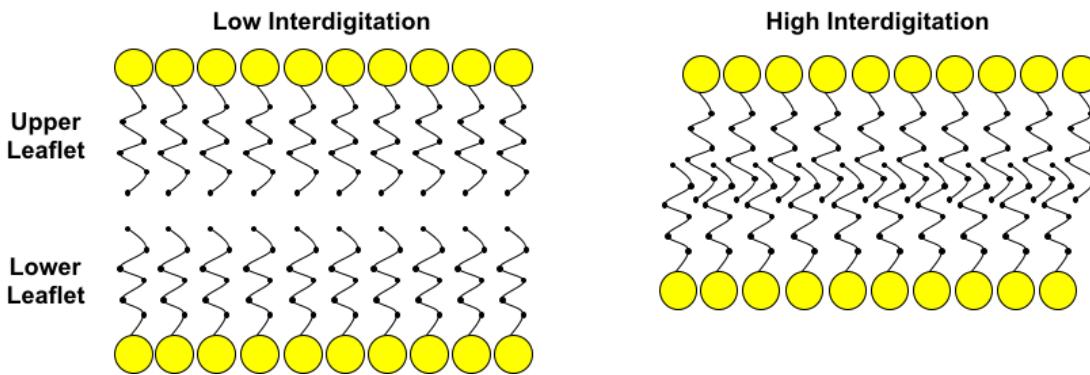


Figure 3-18 Cartoon bilayer showing low levels of interdigitation (left) and high levels of interdigitation (right).

Interleaflet Contacts is an analysis tool designed for counting the number of interleaflet contacts for a given chemical group (possibly a full lipid molecule but maybe a subset of the atoms like an acyl chain) and mapping this information onto the XY plane. This is accomplished by measuring the number of contacts formed between a group of atoms (making the chemical group) from the selected lipid (in the target leaflet) and a group of atoms from the lipids in the opposing leaflet (see Figure 3-19).

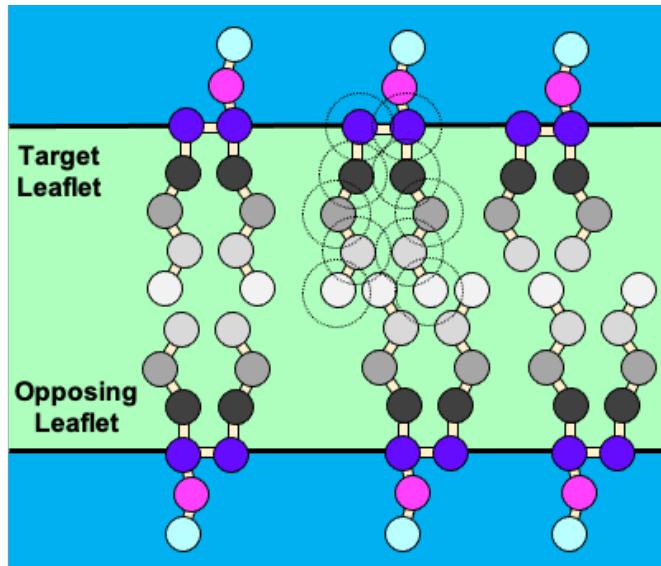


Figure 3-19 Schematic of lipids from the target and opposing leaflets. The circles enclosing the target atoms indicate which atoms to include in contact analysis and the cutoff distance for counting contacts with opposing lipids. A similar selection can be made for the opposing leaflet.

This is expressed mathematically as:

$$ILC_k = \sum_{n=1}^N ILC_n \quad (3.7)$$

where ILC_k is the number of interleaflet contacts for the chemical group k, N is the number of atoms making the chemical group, and ILC_n is the number of interleaflet contacts formed by a single atom in the chemical group. In short, Interleaflet Contacts computes the time average of the spatially resolved ILC_k .

To use the program the user must select the lipids to be included in the calculation for both the target and opposing leaflet. This information is specified using two networks of selection cards as specified with the -crd_1 tag for the target leaflet and the -crd_2 tag for the opposing leaflet. By including two networks, the user is given full control over the lipids and atoms included in the contact analysis for each leaflet. Moreover, the atoms used in the contact analysis (defining the chemical groups) are provided in the secondary files (see Figure 3-20 for an example).

Analysis of Lipid Structure

-crd_1 (Target leaflet)

#lipid_type	#map_1	#map_2	#filename	#contact_atom	#contact_atom
POPE	GL1	GL2	po.crd	GL1	GL1
POPG	GL1	GL2	po.crd	C1A D2A C3A C4A GL2 C1B C2B C3B C4B	C1A D2A C3A C4A GL2 C1B C2B C3B C4B

-crd_2 (Opposing leaflet)

#lipid_type	#filename	#contact_atom	#contact_atom	#contact_atom	#contact_atom
POPE	pope.crd	GL0	NH3	GL0	NH3
POPG	popg.crd	PO4	PO4	PO4	PO4
DLPE	dlpe.crd	GL1	GL1	GL1	GL1
DLPG	dlpg.crd	C1A C2A C3A GL2 C1B C2B C3B	C1A C2A C3A GL2 C1B C2B C3B	C1A D2A C3A C4A GL2 C1B C2B C3B	C1A D2A C3A C4A GL2 C1B C2B C3B C4B

Figure 3-20 An example of selection cards used to measure the interleaflet contacts between POPE/POPG lipids in the target leaflet and POPE/POPG/DLPE/DLPG lipids in the opposing leaflet. In this case, each contact is mapped to the ester atoms (GL1, GL2) for the target lipids. These contacts are additive for the mapping atom such that each mapping atom stamps the total number of contacts for the atoms contained in the secondary card, i.e., the full acyl chain for the example given here. To get the contacts per chain, the user could provide a nonexistent atom for one of the mapping atoms and reduce the atoms in the secondary selection card to those making a single chain. This approach will require duplication of the lipid type in the main card; one for each chain. See Figure 3-12 for an example where the individual acyl chains are characterized.

To provide a clear understanding of the computation at hand, we discuss the internal workings of the program. To begin, a lipid is selected from the target leaflet if its type matches one of the entries in the primary selection card (-crd_1). With a target lipid identified, the secondary selection card, specific to this type, is examined one row at a time. In this step, ILC_n is computed for each atom in the list. The computation of ILC_n is accomplished by looping over the opposing leaflet and finding lipids whose type matches an entry from the second selection card (-crd_2). The secondary cards of -crd_2 specify the atoms participating in the contact analysis for a given ILC_n . Once ILC_n is computed for each atom in the list (-crd_1), then ILC_k is computed using equation 3.7 and stamped to the lattice around the two mapping atoms.

An example of the run commands for Interleaflet Contacts is now given:

```
$ mpirun -n 100 inter_leaflet_contacts_mpi -traj traj.xtc -ref ref.gro -crd_1 target_leaf.crd -crd_2 opposing_leaf.crd -lfc upper_ilc.dat -APS 0.005 -r 0.26 -cutoff 0.4 -leaf 1 -cdist 0.6
```

In the example here, the cutoff distance for counting contacts is set with the -cdist tag. Similarly, the output data file containing the spatially resolved time average of ILC_k is specified via the -lfc tag. An example of output data from Interleaflet Contacts is given in Figure 3-21.

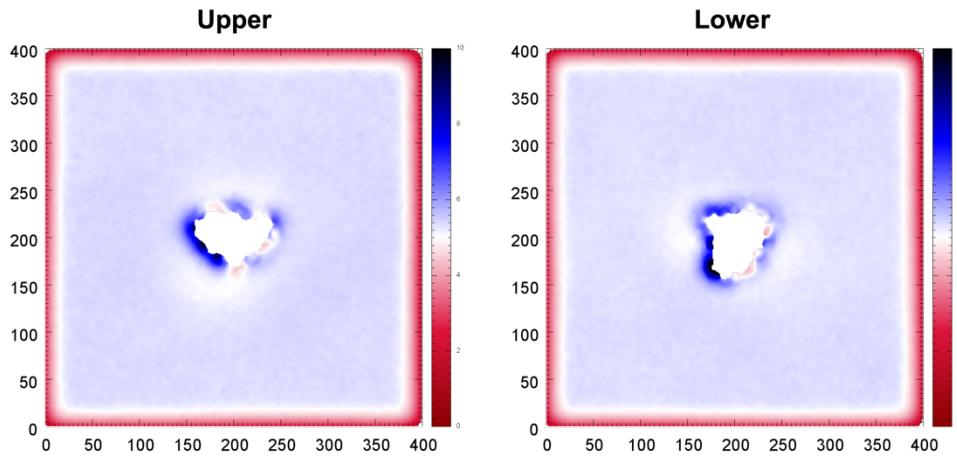


Figure 3-21 The average number of interleaflet contacts between POPE/POPG lipids in the target leaflet and POPE/POPG lipids of the opposing leaflet. Data was generated for lipids in coarse-grained simulations of the CLC-ec1 protein [10]. Units for the x/y axis are grid points and the number of contacts for the color bars.

We note that Interleaflet Contacts lets has the option to generate a free energy histogram pertaining to the number of interleaflet contacts, i.e., ILC_k . This option may be selection by including the simulation temperature with the -temp tag and specifying the bin width with the -width tag. When these options are included, the distribution data is written to an output file containing the same name as specified with -lfc but with the “_free_energy” appendage. In addition to this, the number of Interleaflet contacts formed per chemical group (averaged over the chemical groups a given trajectory frame) is reported for each trajectory frame. This information is written to a file with the same name as specified with -lfc but with the “_contacts_frame” appendage.

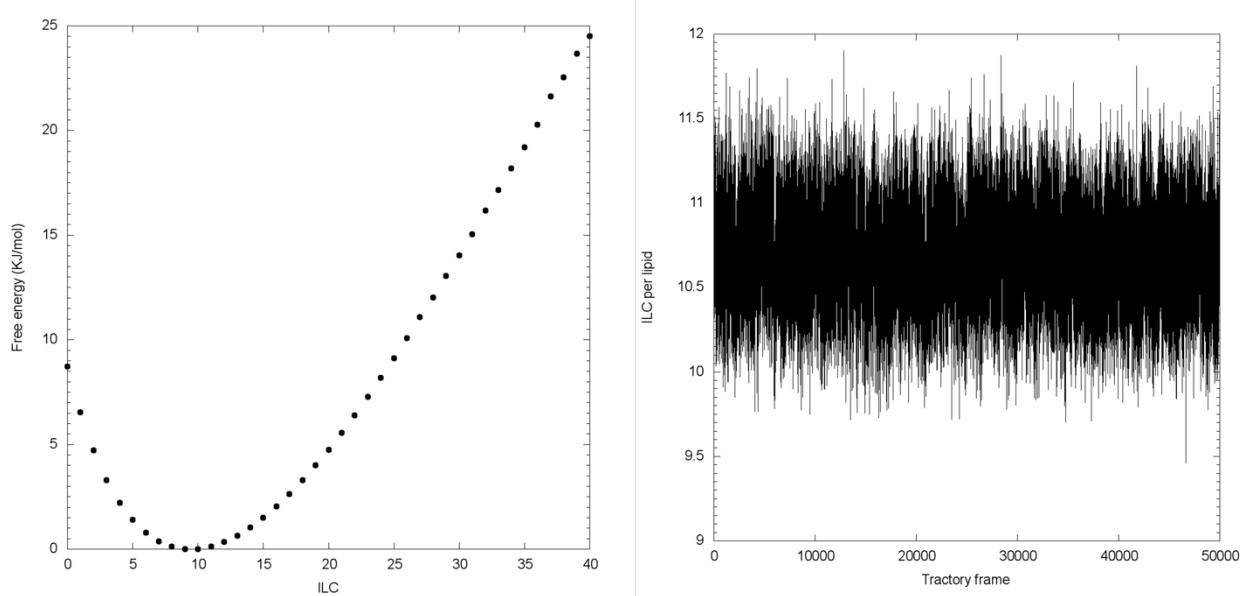


Figure 3-22 Free energy as a function of the number of interleaflet contacts formed by a single lipid (left). The average number of interleaflet contacts per lipid as a function of the trajectory frame (right).

Like Interleaflet Contacts, Interdigitation is an analysis program designed to probe the degree of lipid interdigitation between the two leaflets (see Figure 3-18). This is accomplished by assigning ranks to the opposing leaflet (see Figure 3-23). Then, a tail atom is specified in the target leaflet. Interdigitation then looks for contacts between the tail atom of the target leaflet and the ranked atoms of the opposing leaflet. If a contact is encountered, the rank of the atom is recorded and eventually the average rank \bar{R} is computed:

$$\bar{R} = \frac{1}{N} \sum_{n=1}^N R_n \quad (3.8)$$

where N is the number of interleaflet contacts made with the tail atom and R_n is the rank of the n^{th} contact. Interdigitation stamps \bar{R} to the lattice around target lipids thus generating a spatially resolved time average of \bar{R} . In this way the average rank is used to determine how high the target leaflet lipids ride in the opposing leaflet. We note that the program is designed to probe two tail atoms simultaneously. This approach assumes a lipid with two alkyl chains, like a phospholipid. However, it is possible to probe other types like triglycerides, etc.

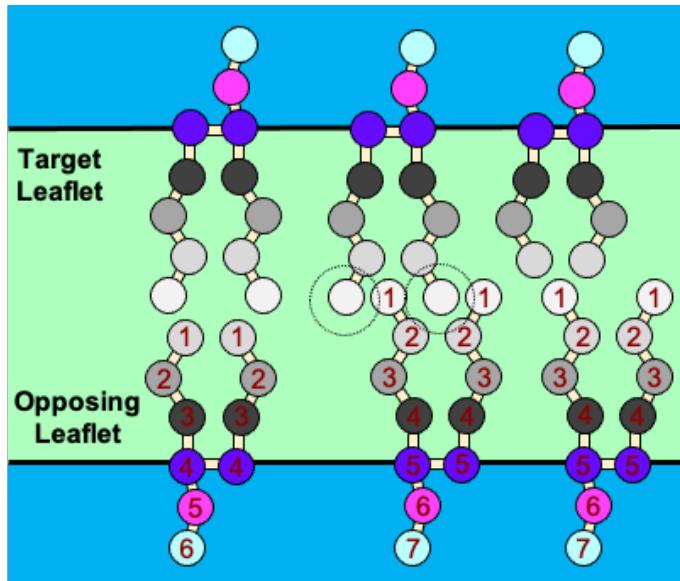


Figure 3-23 Schematic of lipids from the target and opposing leaflets. Two circles enclosing the target tail atoms indicate the cutoff distance for counting contacts with opposing lipids. The atoms on the opposing lipid are given a rank based on how high up the atom sits on the lipid. Once a contact between the target atoms and the ranked atoms are found the rank is mapped to the ester atoms of the target lipid.

To use Interdigitation, the user provides a pair of tail atoms and a pair of mapping atoms for each target lipid. This information is provided using the -crd_1 tag. Additionally, the user must specify the lipid types to include in the opposing leaflet as well as the target atoms used in the contact analysis and the rank of each. This information is provided using a network of selection cards and the -crd_2 tag (Figure 3-24).

-crd_1

Target leaflet

#lipid_type	#tail_1	#tail_2	#map_1	#map_2
POPE	C4A	C4B	GL1	GL2
POPG	C4A	C4B	GL1	GL2

-crd_2

Opposing leaflet

#lipid_type	#filename	#atom	#rank	#atom	#rank
POPE	pope.crd	GL0	7	NH3	7
POPG	popg.crd	PO4	6	PO4	6
		GL1	5	GL1	5
		C1A	4	C1A	4
		D2A	3	D2A	3
		C3A	2	C3A	2
		C4A	1	C4A	1
		GL2	5	GL2	5
		C1B	4	C1B	4
		C2B	3	C2B	3
		C3B	2	C3B	2
		C4B	1	C4B	1

Figure 3-24 Selection card structure for Interdigitation. The tail atoms C4A and C4B of the target leaflet are selected for POPE and POPG lipids in the example given here. The average rank is then computed using POPE and POPG in the opposing leaflet with the atoms ranked 1 to 7 moving up the molecule. The average rank is then mapped to the atoms GL1 and GL2 of the target lipid.

An example of the output from Interdigitation is provided in Figure 3-25.

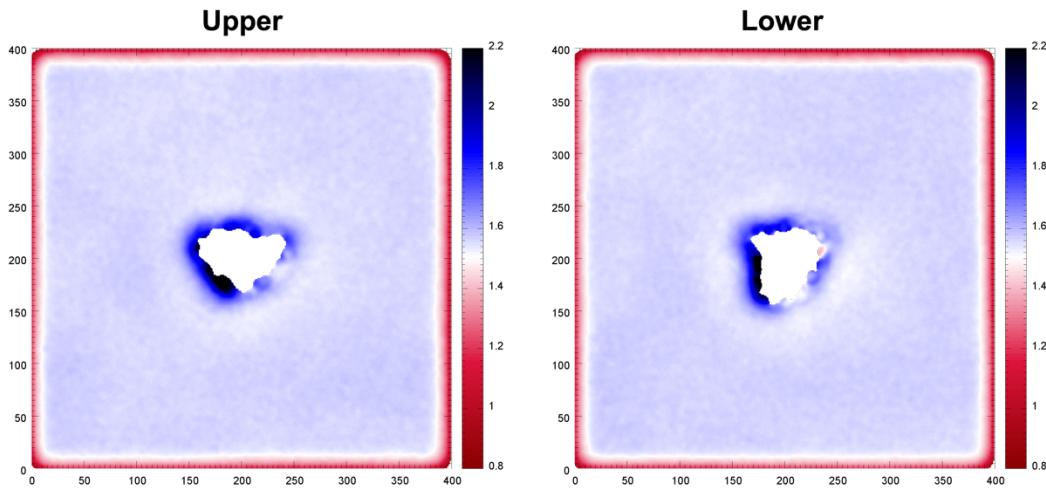


Figure 3-25 Interdigitation as measured by the average rank of the POPE and POPG lipids in coarse-grained simulations of the CLC-ec1 protein [10]. Units for the x/y axis are grid points. The color bars are unitless.

3.7 Lipid Packing Density

Since membrane proteins can induce localized perturbations to membrane structure, it is possible for the lipid density to be affected. In this section we present 2 tools enabling the characterization of the lipid packing density. These include the MOSAICS tools Nearest Neighbors and APL. Beginning with Nearest Neighbors we have a tool that computes the number of lipids of a certain type to be less than a user specified distance (set with `-l_rad`) from a target lipid. This program thus probes the lipid density by counting the number of visiting lipids, which we call the neighbors, to be in a volume around the target lipid. This computation is performed for each target lipid and the number of neighbors is typically mapped to the ester atoms of these lipids. We note that Nearest Neighbors uses the geometric center (equation 1.3) of the lipids to represent their position in XY. The program thus finds the center of an atom selection and counts the number of other centers in the local surroundings (Figure 3-26).

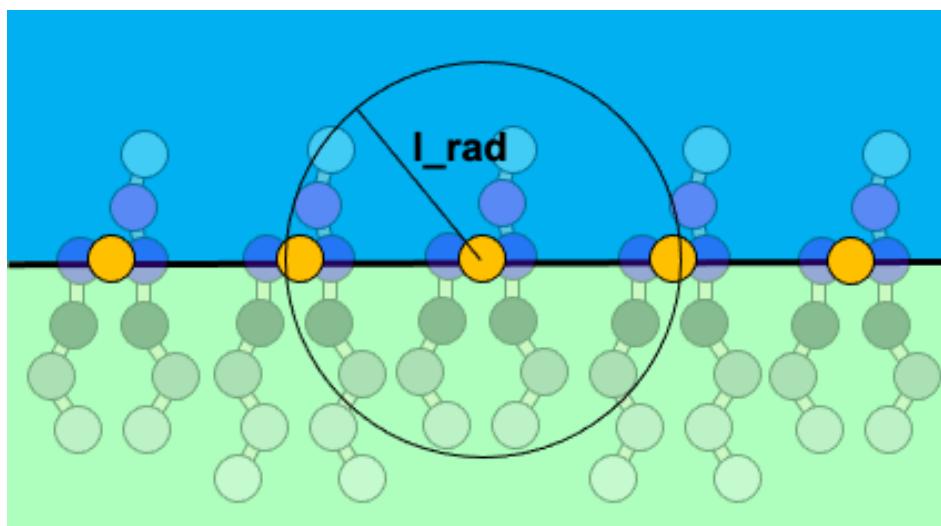


Figure 3-26 Number of lipid neighbors counted around a target lipid. The lipid positions are represented by geometric center of the ester atoms (orange balls). The distance for counting a neighbor is specified by `l_rad`.

Analysis of Lipid Structure

To use Nearest Neighbors, the user must specify the target lipid types as well as a pair of mapping atoms for which the neighbor count is to be mapped to, and the atoms used when computing geometric centers. This information is provided using a network of selection cards specified with the -crd_1 tag. Similarly, the lipid types composing the visiting lipids and the atoms used to find their centers is specified using the -crd_2 tag (Figure 3-27).

-crd_1

Target lipids

#lipid_type	#map_1	#map_2	#filename	#center_atoms	#center_atoms	#center_atoms	#center_atoms
POPE	GL1	GL2	gl_12.crd	GL1			
POPG	GL1	GL2	gl_12.crd	GL2	GL1		
DLPE	GL1	GL2	gl_12.crd		GL2	GL1	
DLPG	GL1	GL2	gl_12.crd			GL2	GL1

-crd_2

Visiting lipids

#lipid_type	#filename	#center_atoms	#center_atoms	#center_atoms	#center_atoms
POPE	gl_12.crd	GL1	GL1	GL1	GL1
POPG	gl_12.crd	GL2	GL2	GL2	GL2
DLPE	gl_12.crd				
DLPG	gl_12.crd				

Figure 3-27 Structure of the selection cards used to specify the target and visiting lipids when computing the number of neighbors. Here the atoms used in computation of geometric centers are specified in the secondary selection cards.

An example of the run commands used for Nearest Neighbors is now given.

```
$ mpirun -n 50 nearest_neighbors_mpi -traj traj.xtc -ref ref.gro -crd_1 param_1.crd -crd_2 param_2.crd -nbrs upper_nbrs.dat -APS 0.005 -r 0.26 -cutoff 0.4 -leaf 1
```

In the example given here, the output data file containing the spatially resolved time average of the neighbors count is specified using the -nbrs tag. An example of data generated with Nearest Neighbors is given in Figure 3-28.

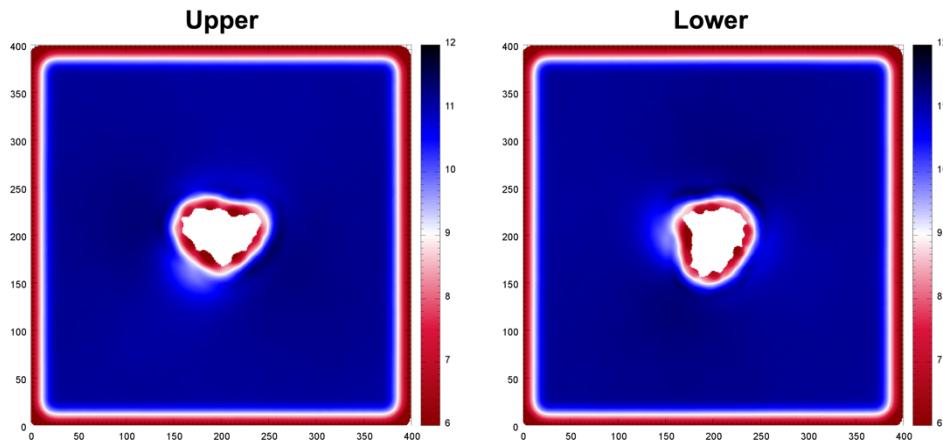


Figure 3-28 The average number of ester centers to be less than 1.5 nm from the POPE, POPG, DLPE, and DLPG lipids. Data was taken from coarse-grained simulations of the CLC-ec1 protein [10]. Units for the x/y axis are grid points and the number of lipid centers for the color bars.

In addition to the number of nearest-neighbors, MOSAICS makes possible the computation of the area per lipid. This analysis is carried out using the program APL by constructing a Voronoi diagram from the lipid atoms (Figure 3-29).

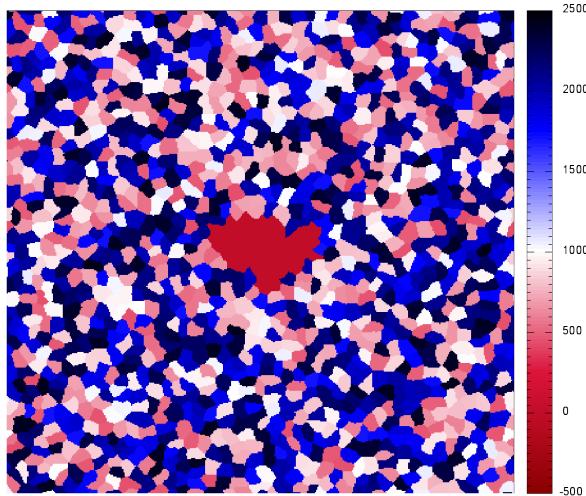


Figure 3-29 Example of a Voronoi diagram computed for a single trajectory frame of a coarse-grained system of CLC-ec1 embedded in a POPE/POPG/DLPE/DLPG bilayer. The color bar gives the lipid res id. The protein is shown in red and has a res id of -1.

The area of each lipid (nm^2) is then determined by counting the lattice points occupied by the lipid within the Voronoi diagram and is stamped to the grid using a pair of mapping atoms. To use the program, the user must specify the lipid types whose area is to be measured and a pair of mapping atoms. This information is specified using a selection card using the `-crd_1` tag. In addition to this, the user must specify the lipids used when constructing the Voronoi diagram as well as the atom types included in this computation. This information is provided via a network of selection cards using the `-crd_2` tag (Figure 3-30).

Analysis of Lipid Structure

-crd_1

Target lipids

#lipid_type	#map_1	#map_1
POPE	GL1	GL2
POPE	GL1	GL2
DLPE	GL1	GL2
DLPG	GL1	GL2

-crd_2

Voronoi lipids

#lipid_type	#filename	#voro_atoms	#voro_atoms	#voro_atoms	#voro_atoms
POPE	gl_12.crd	GL1	GL1	GL1	GL1
POPG	gl_12.crd	GL2	GL2	GL2	GL2
DLPE	gl_12.crd				
DLPG	gl_12.crd				

Figure 3-30 Selection card structure used by APL. In this case, all lipid types (POPE/POPG/DLPE/DLPG) were used to construct the Voronoi diagram. Similarly, the area was measured for POPE, POPG, DLPE, and DLPG lipids.

We note that the inclusion of the protein within the Voronoi diagram is needed to accurately measure the area of the solvating lipids. This inclusion is made possible by selecting protein atoms that are in the same plane as the target lipid atoms. These atoms are found as those whose minimum distance from one of the lipid atoms is below a cutoff value specified using the -c_dist tag. Because it is possible for out-of-plane atoms to have a similarly small distance, we require the z-component of this distance be significantly smaller than -c_dist value. In the current form, the maximum allowed dz is hard coded as half the -c_dist value. An example of the run commands for APL are now given:

```
$ mpirun -n 56 apl_mpi -traj traj_lsq.xtc -traj_v traj.xtc -ref ref.gro -crd_1 param_powl.crd -crd_2 param_2.crd -apl upper_apl.dat -APS 0.0005 -r 0.26 -cutoff 0.4 -leaf 1 -c_dist 0.6 -bin 0.01
```

Note, in the example here, we have provided a second trajectory file using the -traj_v tag. This trajectory is used to compute Voronoi diagrams and is needed when least squares fitting is used to align the protein in -traj. In cases where least squares fitting has been performed, the resulting Voronoi diagram will be inaccurate. This is because, while the atomic coordinates are rotated, the lattice holding the Voronoi tessellation is not. We should thus use a trajectory with -traj_v where the protein has been centered and the atoms wrapped around it, but without performing any rotations to the system. Then, we use a trajectory for -traj where the protein has been centered, the atoms wrapped, and the system rotated to fix the protein orientation. This trajectory is used for the stamping procedure when adding area measurements to the lattice. We note that the two trajectories must be compatible, i.e., they must stem from the same simulation.

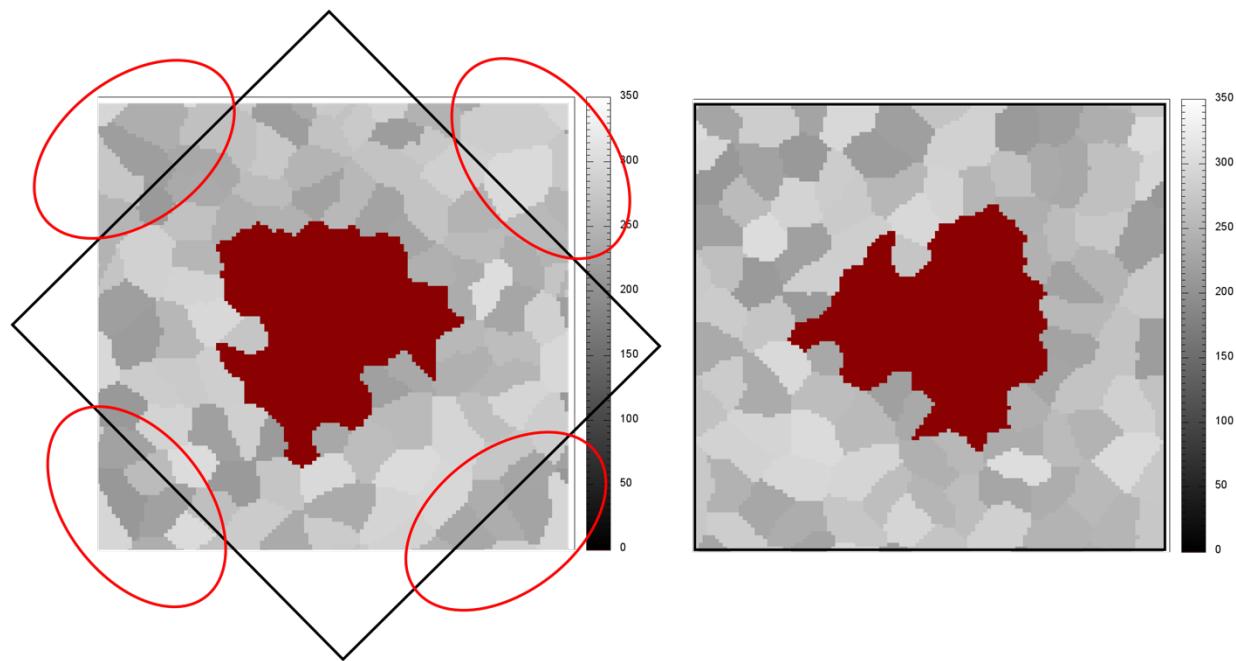


Figure 3-31 Voronoi diagram computed using a trajectory where least square fitting has been performed. The system box is shown as the rotated black rectangle. We note that the lipids, indicated with red ovals, are expanded in the Voronoi diagram, and fill the remaining lattice. This leads to an error in the area calculation for those lipids (left). On the other hand, when a trajectory is used where least squares fitting was not performed (right), then the lattice is constructed that better fits the system box. In this case, the Voronoi cells give an accurate represent the area per lipid.

In addition to trajectory files, we have specified the output data file containing the spatially resolved time average area per lipid using the -apl tag. Figure 3-32 shows an example of the data generated with APL.

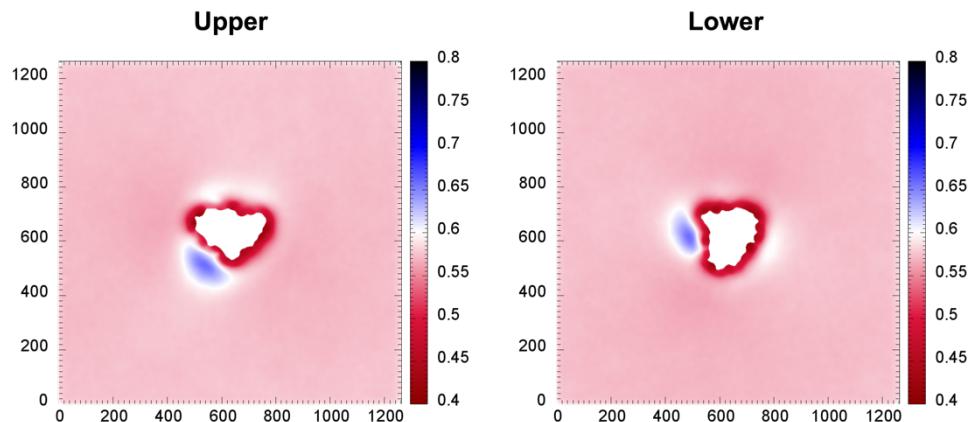


Figure 3-32 Area per lipid measured for POPE, POPG, DLPE, and DLPG lipids in a 50:50 mixture of PO to DL. Units for the x/y axis are grid points. The color bars have units nm².

We note that the area per lipid computation uses stamp assisted Voronoi tessellations (Section 1.17). The stamping radius is set by default to 0.8 nm but may be set by the user with the -voro_r tag. The user may examine the Voronoi diagrams by including the -voro 1 tag which

instructs APL to write each diagram to file. We note that the periodic boundary conditions are accounted for when computing Voronoi diagrams which greatly improves the accuracy of the method near the box edges. However, small artifacts may still arise near the boundaries due to errors in approximating the box size. That is, a lattice is constructed to hold the Voronoi diagram whose dimensions are chosen to best approximate the current box dimensions. Since the spacing between lattice points is given via the -APS tag, it is not possible to match the box dimensions exactly and the lattice generally overestimates the size of the box. This error manifests for lipids near the box edges where the lattice is now slightly larger than the actual box thus giving a larger area. This error is unavoidable but may be reduced to an insignificant level by increasing the lattice resolution. For example, we used an area per lattice square of 0.0005 nm^2 for the analysis shown in Figure 3-32.

In addition to spatially resolved maps of the area per lipid, APL will generate a histogram for the individual area measurements (Figure 3-33). The average and standard deviation are also reported. This added functionality makes it possible to report the area per lipid as a single value. For example, a simulation containing a pure bilayer, with no protein, might be analyzed to give the average area per lipid. To use this function, the user must specify the bin width (nm^2) using the -bin command line argument. The histogram data is then written to a file with the same name as specified via the -apl tag but with the “_histo” appendage.

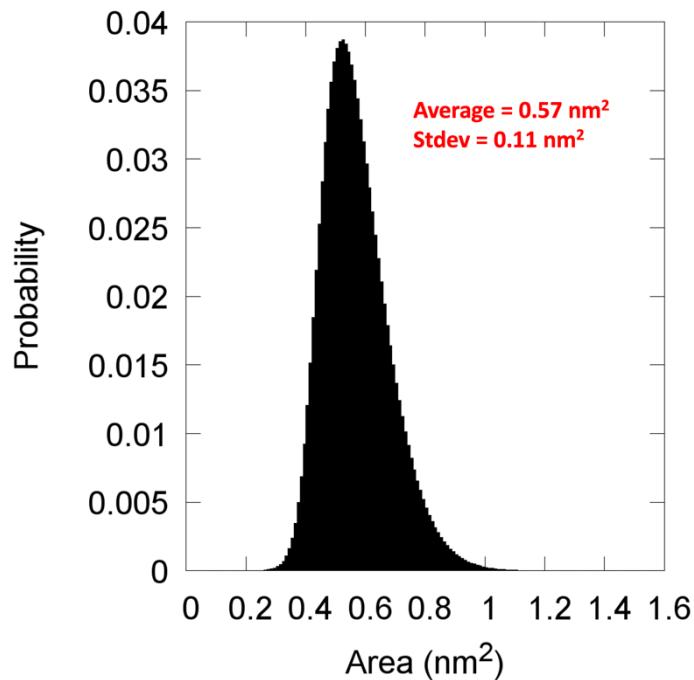


Figure 3-33 Probability distribution for the Area per lipid. The average and standard deviation over the distribution are reported and shown here in red.

3.8 Contacts Between Lipids and Other Molecules

With MOSAIC tools it is possible to quantify the contacts formed between lipids and other molecule types. There are 2 tools available for these calculations including Lipid Contacts and Protein Lipid Contacts. These tools differ in the following way. First, Lipid Contacts focuses on the

lipids and therefore counts the number of contacts they form with other molecules. In the end, this data is projected onto the XY plane. In contrast, Protein Lipid Contacts focuses on the protein and counts the number of contacts formed with the lipids. This data is projected onto the residues of the protein and the data is viewed with a graphics tool like PyMOL (Schrödinger, LLC). In the remainder of this section, we examine these tools in greater detail beginning with Lipid Contacts.

Lipid Contacts is an analysis program that computes the average number of contacts between the lipids and other molecules in the system (Figure 3-34). Supported molecules include, the protein, lipids, and the solvent. A combination of these three could also be used. For example, the user could compute the number of contacts between the lipids and the surrounding lipids, protein, and solvent.

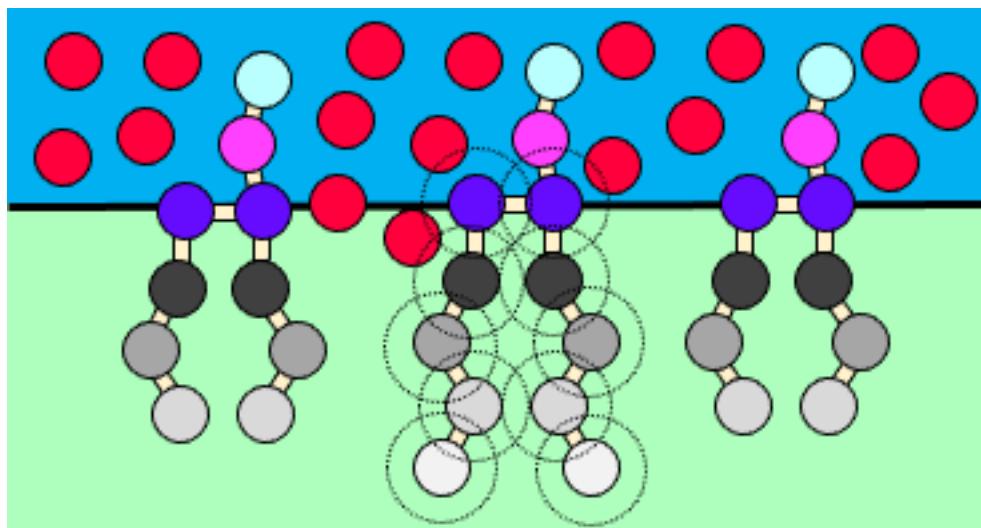


Figure 3-34 Example showing the number of contacts between a lipid and the solvent atoms. Dotted lines represent the contact distance for which contacts are counted. That is, any water atom inside the dotted line will be counted as a contact for that atom.

To use the program the user must specify the contact distance. This is done with the -cdis tag. Additionally, the user must specify which molecules are to be counted in the contact analysis with the -lip, -prot and -sol tags. For the following examples let's assume the user selects the solvent only, i.e., `-lip 0 -prot 0 -sol 1`. Additionally, the user must provide the lipid types to be included in the calculation, a pair of mapping atoms for adding the contact count to the grid, and the lipid atoms to be included when counting contacts. This information is provided using a network of selection cards as specified with the -crd tag.

Analysis of Lipid Structure

-crd			
#lipid_type	#map_1	#map_2	#filename
POPE	GL1	GL2	po_tails.crd
POPG	GL1	GL2	po_tails.crd
DLPE	GL1	GL2	dl_tails.crd
DLPG	GL1	GL2	dl_tails.crd

#contact_atom	#contact_atom	#contact_atom	#contact_atom
C1A	C1A	C1A	C1A
C2A	C2A	D2A	D2A
C3A	C3A	C3A	C3A
C1B	C1B	C4A	C4A
C2B	C2B	C1B	C1B
C3B	C3B	C2B	C2B
C4B	C4B	C3B	C4B

Figure 3-35 Selection card structure used by Lipid Contacts. For the example provided here, the contacts are measured between water molecules and the acyl chains of the POPE, POPG, DLPE, and DLPG lipids.

In the example provided here, Lipid Contacts is instructed to count the total number of solvent contacts made with the tail atoms of the POPE, POPG, DLPE, and DLPG lipids. We note that the tail atoms are specified for each lipid type using the secondary selection cards. An example of the run commands for Lipid Contacts is now given:

```
$ mpirun -n 50 lipid_contacts_mpi -traj traj_wat.xtc -ref ref_wat.pdb -crd param.crd -lc
upper_lsc.dat -APS 0.005 -r 0.26 -cutoff 0.4 -leaf 1 -cdist 0.6 -lip 0 -prot 0 -sol 1
```

In the example given here, the output grid data containing the spatially resolved time average of the contact count is specified with the -lc tag. An example of the output data generated by Lipid Contacts is given in Figure 3-36.

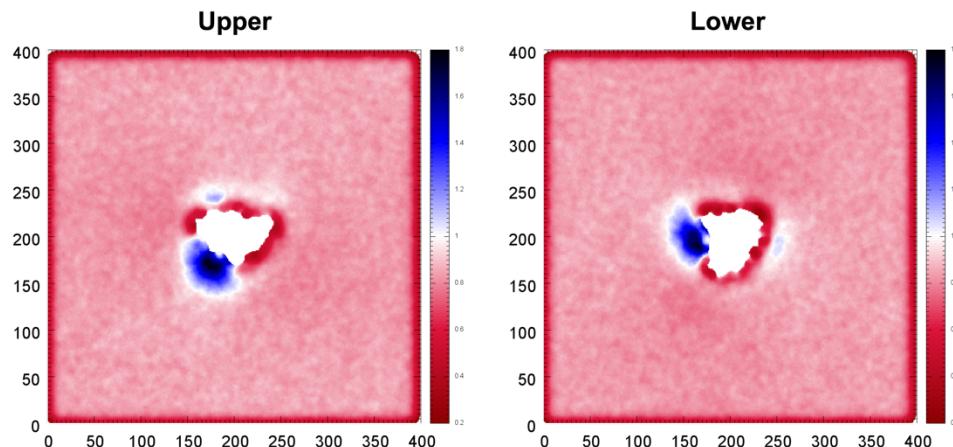


Figure 3-36 The average number of lipid-solvent contacts for the tail atoms of POPE and POPG lipids in a coarse-grained simulation of the CLC-ec1 protein [10]. Units for the x/y axis are grid points and the number of contacts for the color bars.

We note that Lipid Contacts can also be used to compute the number of residues contacting the lipids by including the -mol 1 tag. For example, if the user wants to know how many water molecules penetrate the bilayer to the level of the lipid tails, then the tail atoms could be included in -crd and the -mol option set to 1. With this option, if any atom of the water molecule makes a contact with any atom in -crd, then the water molecule is counted. However, if the same water atom forms another contact with another atom in -crd or if another atom of

the water forms a contact with an atom in -crd this will not be counted. Thus, the number of water molecules are counted rather than the number of contacts. Using -mol 1 with -lip 1 will thus compute the number of lipids in contact with the target atoms of the target lipid. Using -mol 1 and -prot 1 will give the number of protein residues in contact with the target atoms of the target lipid.

Switching gears, the average number of contacts formed between the lipids and each residue of the protein can be computed with Protein Lipid Contacts. To use the tool, the user must specify the lipid types included in the calculation as well as the lipid atoms involved in lipid-protein contacts. This information is provided via a network of selection cards and the -crd tag (Figure 3-37). We note that a selection card is not required for the protein since all protein atoms are included in the analysis.

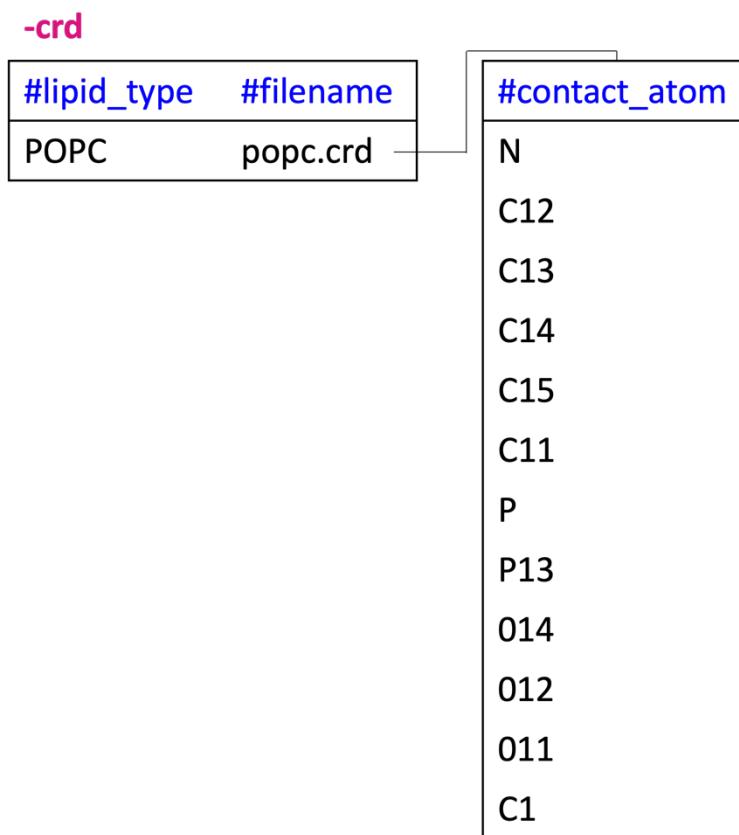


Figure 3-37 Selection card structure used for Protein Lipid Contacts. Here, the head atoms of all-atom POPC molecules are used when computing lipid-protein contacts. The secondary card is used to specify the lipid atoms included in the analysis.

We now provide an example of the run commands for Protein Lipid Contacts:

```
$ mpirun -n 50 protein_lipid_contacts_mpi -traj traj.xtc -ref -ref.pdb -crd param.crd -ct contacts.dat -leaf 0 -cdist 0.6
```

In the example provided here, the cutoff distance used for counting contacts is provided with the -cdist tag. In addition to this, the output file, containing the number of contacts formed for each residue per trajectory frame, is specified using the -ct tag. This data may be plotted as a line

graph. Alternatively, the data may be represented graphically such that a PDB file is generated where the same data is written to the B-factor. This PDB is given the same name as specified with the -ct tag but the .dat extension is replaced with .pdb. Figure 3-38 shows an example of the data generated with Protein Lipid Contacts.

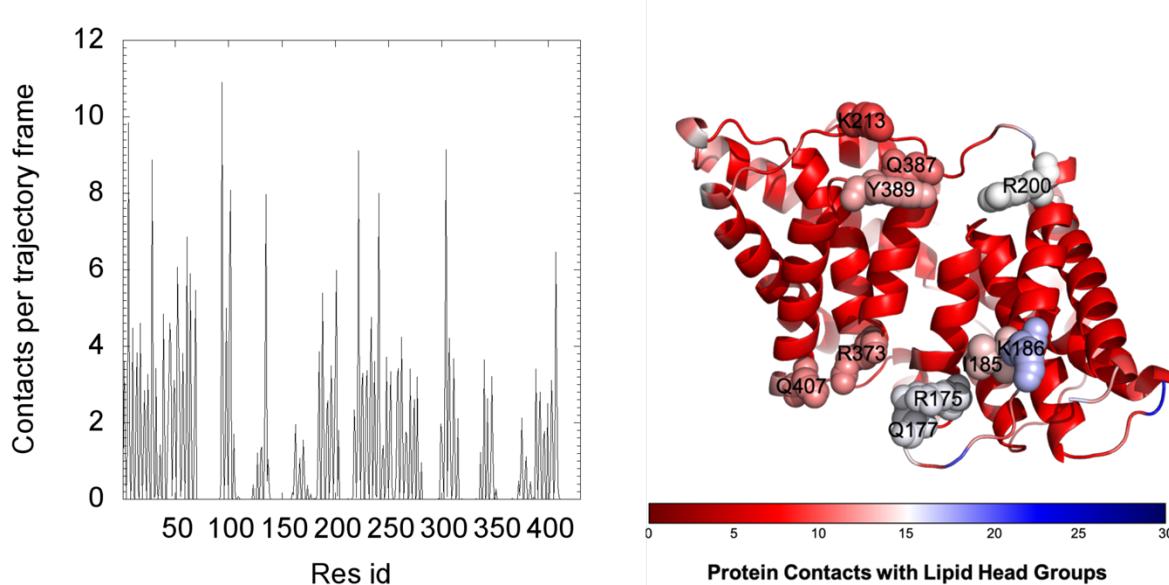


Figure 3-38 The average number of contacts formed between each protein residue and the head groups of POPC molecules. This information is shown as a line graph (left) or by color (right). We note that the line graph data will contain a value for each residue in the system including the lipids (not shown here); the lipids should contain a value of 0. Furthermore, the residue numbering is consistent with that used by MosAT, i.e., 1 to N where N is the number of residues in the system.

We note that it is possible to transfer the B-factors (in this case giving the number of lipid contacts formed for each protein residue) to a more meaningful structure such as the time average protein coordinates (see section 3.12) using the B Stamp tool. B Stamp simply reads two PDB files, takes the B-factor from one and copies it to the second. To use B Stamp the user must specify 2 PDB files with the -traj and -traj_b tags. For this analysis, traj will contain the structure of interest and traj_b the B-factor of interest.

```
mpirun -n 1 b_stamp_mpi -traj mean_prot.pdb -ref contacts.pdb -traj_b contacts.pdb -o mean_prot_b_factor.pdb
```

In the example here, the output PDB file containing the desired structure and B-factors is specified using the -o tag. An example of the mean protein coordinates with B-factor set equal to the number of contacts with the lipid heads is shown in Figure 3-39.

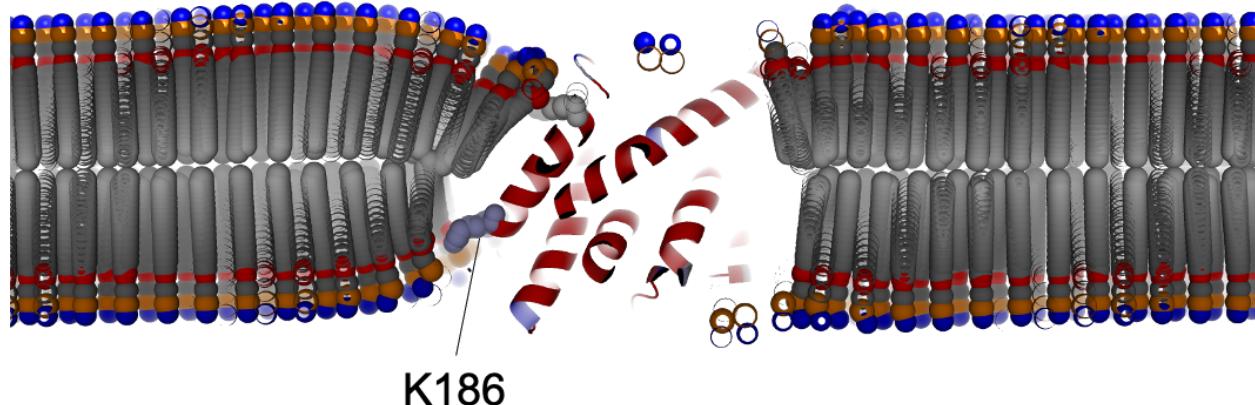


Figure 3-39 Mean protein coordinates with the B-factor set to match the number of contacts each residue makes with the POPC headgroups. The time average lipid coordinates are also shown.

3.9 Lipid Gyration

The compactness of the lipids can be measured using the MOSAICS tool Lipid Gyration. Lipid Gyration, as the name suggests, quantifies the radius of gyration of the lipids and projects this data on the YX plane. The radius of gyration is computed as:

$$R_G = \sqrt{\frac{1}{M} \sum_i m_i (\vec{r}_i - \vec{\mu})^2} \quad (3.9)$$

where the summation is over the set (or subset) of atoms (Figure 3-40) making the molecule, $\vec{\mu}$ is the center of mass (equation 1.1) for the selection, and M is the total mass, i.e., $M = \sum_i m_i$.

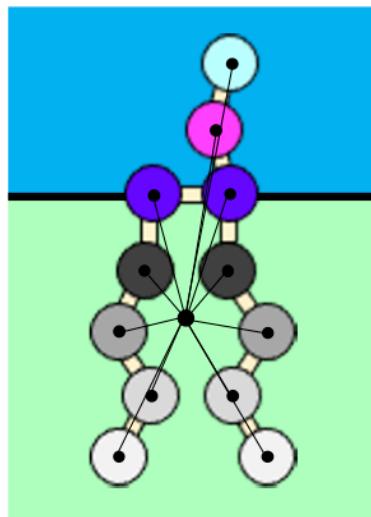


Figure 3-40 A cartoon schematic showing the measurement of the radius of gyration for a coarse-grained lipid molecule. The center of mass of the molecule is shown as a black dot. Lines connecting the center of mass to each atom represent the distance measurements.

To use the program the user must specify the lipid types and corresponding atoms to include in the calculation as well as a pair of mapping atoms used to add data to the grid. This information is provided using a network of selection cards and the -crd tag (Figure 3-41).

Analysis of Lipid Structure

-crd			
#lipid_type	#map_1	#map_2	#filename
POPE	GL1	GL2	Pope.crd
POPG	GL1	GL2	popg.crd
DLPE	GL1	GL2	dlpe.crd
DLPG	GL1	GL2	dlpq.crd

#center_atom	#center_atom	#center_atom	#center_atom
GL0	NH3	GL0	NH3
PO4	PO4	PO4	PO4
GL1	GL1	GL1	GL1
GL2	GL2	GL2	GL2
C1A	C1A	C1A	C1A
C2A	C2A	D2A	D2A
C3A	C3A	C3A	C3A
C1B	C1B	C4A	C4A
C2B	C2B	C1B	C1B
C3B	C3B	C2B	C2B
	C4B	C3B	C3B
		C4B	C4B

Figure 3-41 Selection card structure used by Lipid Gyration. In the example provided here, the radius of gyration is computed for POPE, POPG, DLPE, and DLPG lipids using the complete lipid molecules. The radius of gyration is then stamped to the grid around the GL1 and GL2 atoms.

An example of the run commands for Lipid Gyration are now given:

```
$ mpirun -n 50 lipid_gyration_mpi -traj traj.xtc -ref ref.pdb -crd podl.crd -gyrate upper_gyrate.dat -APS 0.005 -r 0.26 -cutoff 0.4 -leaf 1
```

In the example here, the output data file containing the spatially resolved time average radius of gyration is specified via the -gyrate tag. We note that a PDB file should be used for the reference file. This is because the atomic masses are needed for computation of the center of mass. For PDB reference files, the atomic masses can be specified using the B-factor. If a gro file provided, the masses are set to 1.0 for each atom. In this case, the masses cancel from the center of mass equation and a geometric center (equation 1.3) is computed instead. An example of the data generated with Lipid Gyration is shown in Figure 3-42.

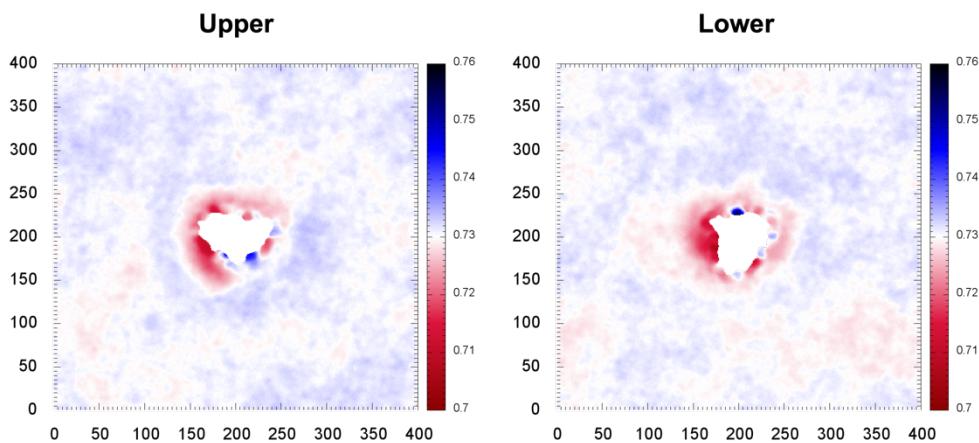


Figure 3-42 The average radius of gyration for POPE, POPG, DLPE, and DLPG lipids based on their position in XY. Data was taken from coarse-grained trajectories of CLC-ec1 [10] in a 50:50 POPX:DLPX bilayer. Units for the x/y axis are grid points. For the color bars, the units are nm.

3.10 Lipid Enrichment

With MOSAIC tools, the preferential solvation of a membrane protein may be studied. This is made possible by measuring the enrichment factor for a given lipid type when a complex mixture is simulated. In this section, we consider 2 analysis tools that are used for enrichment calculations. First, we have 2d Enrichment which projects enrichment data onto the XY plane. In contrast, Protein Enrichment focuses on lipid enrichment at the level of the amino acids. These programs are now discussed in greater detail.

With 2d Enrichment, the enrichment of one lipid-type over another may be probed and projected onto the XY plane. Here we define the enrichment factor of lipid A as:

$$\%E_A^{ij} = 100\% \left(\frac{\frac{\rho_A^{ij}}{\rho_B^{ij}} - \left[\frac{\rho_A}{\rho_B} \right]_b}{\left[\frac{\rho_A}{\rho_B} \right]_b} \right) \quad (3.10)$$

where ρ_A^{ij} and ρ_B^{ij} are the sample counts (Equation 1.4) for lipids A and B respectively. The subscript b indicates the ratio of the counts in the bulk. This is taken to be the total number of lipids A in the system divided by the total number of lipids B. 2d Enrichment uses the sample counts ρ^{ij} (section 1.11) which are commonly used for excluding data by other analysis programs such as Z Coord, and Lipid Distances, etc.

To use 2d Enrichment, the user must specify lipid types A and B as well as the atoms used to stamp data to the respective grids. This information is provided using networked selection cards, one for lipids A and another for B. The primary files for each network may be specified using the -crd_1 and -crd_2 tags (Figure 3-43).

-crd_1

ρ_A	
#lipid_type	#filename
DLPE	dl_tails.crd
DLPG	dl_tails.crd

#target_atom	#target_atom
GL1	GL1
GL2	GL2

-crd_2

ρ_B	
#lipid_type	#filename
POPE	gl_12.crd
POPG	gl_12.crd

#target_atom	#target_atom
GL1	GL1
GL2	GL2

Figure 3-43 Selection card structure used by 2d Enrichment. For the example here, the atoms GL1 and GL2 are used as mapping atoms when adding data to the grid. That is, the atoms in the secondary cards are used to stamp data to the lattice.

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In the example above, 2d Enrichment compares the sample count of DLPE/DLPG with that of POPE/POPG lipids while mapping this data onto the ester atoms. In the end, the percent enrichment of DLPE/DLPG lipids is computed. We now give an example of the run commands used to with 2D Enrichment:

```
mpirun -n 50 2d_enrichment_mpi -traj traj.xtc -ref ref.gro -crd_1 lip_a.crd -crd_2 lip_b.crd -enrich upper_enrich.dat -APS 0.005 -r 0.26 -cutoff 0.4 -leaf 1
```

In the example here, the output data file containing spatially resolved enrichment factor is specified using the -enrich tag. In addition to the enrichment factor, 2D Enrichment also generates grid data containing the sample counts for lipids A, B, and A+B. These data are written to files named after the -enrich argument but with the “_rho_A”, “_rho_B”, and “_rho_t” appendages respectively. An example of data generated with 2d Enrichment is shown in Figure 3-44.

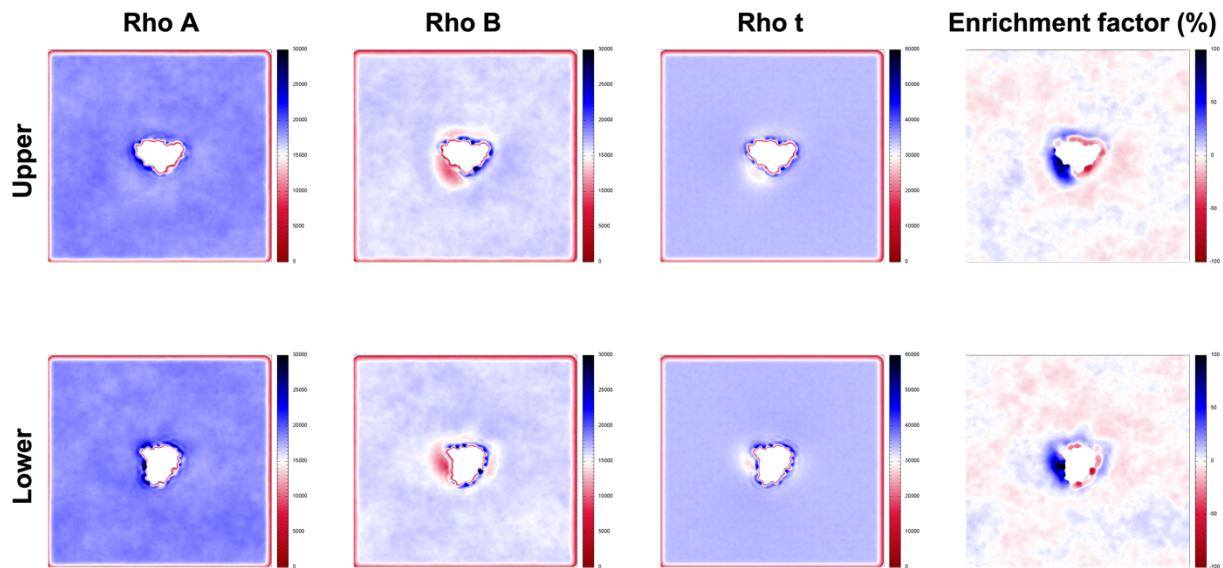


Figure 3-44 Sample count for DLPE/G (left), POPE/G (middle left) and all lipid types (middle right). The enrichment factor is shown in the far-right panel. Data taken from coarse-grained simulations of the CLC-ec1 protein [10]. Units for the color bars are the sample count (left 3 panels) and the percent enrichment (right panel).

It should be noted that there is a MOSAICS tool, 2d Enrichment Distance Projection which lets the user project the lipid enrichment data as a function of distance from the protein surface. This tool works like Grid Distance Projection (section 1.13) in that it creates a mask, of width - range, located at some distance d from the protein surface. The enrichment factor within the selected region, i.e., inside the mask, is computed as:

$$\%E_A(d \pm tol) = 100\% \left(\frac{\frac{\sum_{ij} \rho_A^{ij} m^{ij}}{\sum_{ij} \rho_B^{ij} m^{ij}} - \left[\frac{\rho_A}{\rho_B} \right]_b}{\left[\frac{\rho_A}{\rho_B} \right]_b} \right) \quad (3.11)$$

where the sums are over the lattice points and m^{ij} gives the value of the mask at lattice point ij, i.e., either 0 or 1. Like Grid Distance Projection, the protein surface is defined by a mask using the -mask tag. Moreover, 2d Enrichment Distance Projection increases the distance between the mask center and the protein surface in an iterative process. This is done -iter times and the mask is moved -res nm with each iteration. To enable the selection of one interface over another the usual rectangular selection routine is used (section 1.15). The user must therefore provide the rectangle information with the -x, -y, rx, -ry and -invert tags. In addition to this, the sample counts for the 2 lipids are also required and may be provided with the -rho_A and -rho_B tags. And finally, the user must specify the ratio of the lipids R. This is done with the -ratio tag. We now give an example of the run commands used by 2d Enrichment Distance Projection:

```
$ mpirun -n 100 2d_enrichment_distance_projection_mpi -rho_A upper_enrich_rho_A.dat -rho_B upper_enrich_rho_B.dat -mask prot.dat -o upper_enrich_proj.dat -x 85 -y 108 -rx 100 -ry 110 -invert 0 -iter 100 -res 0.1 -range 0.5 -APS 0.005 -odf 0 -ratio 1.1438
```

In the example provided here, the output data file containing the projection of the enrichment factor is specified with the -o tag. An example of data generated by 2d Enrichment Distance Projection is shown in Figure 3-45.

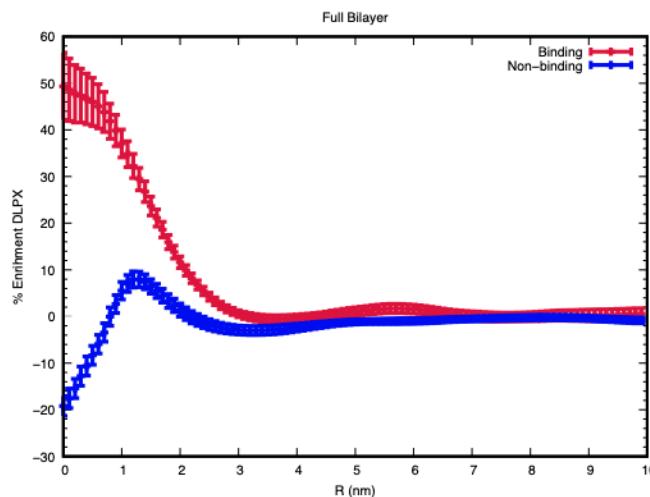


Figure 3-45 Percent DLpx lipid enrichment as projected as a function of distance from the binding and nonbinding interfaces of the CLC-ec1 protein [10].

Switching gears, the lipid enrichment can be probed at the level of the amino acids using Protein Residue Enrichment. This program works by counting the number of lipids A contacting each protein residue as well as the number of lipids B. The percent enrichment of lipid A for residue i is defined as:

$$\%E_{A,i} = 100\% \left(\frac{\frac{n_A}{n_B} - \left[\frac{n_A}{n_B} \right]_b}{\left[\frac{n_A}{n_B} \right]_b} \right) \quad (3.12)$$

where n_A is the number of lipids A contacting residue i and n_B is the number of lipids B contacting residue i (these are the number of contacts summed over all trajectory frames). The subscript b

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corresponds to the ratio of lipids A over B in the bulk (this is the total number of lipids A/B in the system). To use the program the user must specify a cutoff distance to be used when counting contacts. This is done with the -cdist tag. Additionally, the user must specify 2 lipid types corresponding to A and B as well as a list of lipid atoms to include in the contact analysis. This is done using networked selection cards and the tags -crd_1- and -crd_2 (Figure 3-46).

-crd_1

ρ_A

#lipid_type	#filename
DLPE	dl_tails.crd
DLPG	dl_tails.crd

#target_atom
C1A
C2A
C3A
C1B
C2B
C3B

#target_atom
C1A
C2A
C3A
C1B
C2B
C3B

-crd_2

ρ_B

#lipid_type	#filename
POPE	po_tails.crd
POPG	po_tails.crd

#target_atom
C1A
D2A
C3A
C4A
C1B
C2B
C3B
C4B

#target_atom
C1A
D2A
C3A
C4A
C1B
C2B
C3B
C4B

Figure 3-46 Selection card structure used by Protein Residue Enrichment. For the example here, we use the lipid alkyl chains when computing lipid-protein contacts.

In the example above, the percent enrichment is found for POPX lipids relative to DLPX. Moreover, the lipid alkyl chains (as specified in the secondary cards) are used when counting

lipids contacting the protein. Note that every atom making the protein is considered when counting contacts and the maximum number of contacts between a lipid and the residue is 1 for each frame. That is, we determine if one or more contacts was formed between the protein residue and the target lipid (1:yes, 0:no). Output from Protein Residue Enrichment includes a text file containing the number of contacting lipids of each type (A and B) as well as the percent enrichment. This information is given for each protein residue. Additionally, PDB files can be written where the number of contacting lipids (per frame), or the percent enrichment, is specified using the B-factor. We note that some residues may be excluded if deemed insignificant. These residues are identified by comparing n_A and n_B to the number of trajectory snapshots analyzed. That is, a value of χ is specified by the user. Then, the residue is excluded (B-factor set to 0) if the following condition is met:

$$n_A + n_B < \chi(T + 1) \quad (3.13)$$

where $T+1$ gives the number of trajectory frames analyzed. If either of these conditions are met, then the B-factor is set to 0 in the enrichment data (PDBs only). We now give an example of the run commands used with Protein Residue Enrichment:

```
$ mpirun -n 100 protein_residue_enrichment_mpi -traj traj.xtc -ref ref.pdb -crd_1 param_A.crd -crd_2 param_B.crd -enrich enrichment.dat -e_pdb enrichment.pdb -leaf 0 -cutoff 0.5 -cdist 0.6
```

In the example given here, the output data file containing the enrichment factor for each protein residue is specified with the -enrich tag. Likewise, the PDB file containing the enrichment data is named using -e_pdb. The remaining PDB files, containing the number of contacting lipids per frame, are given the same name as provided with -e_pdb but with the “_A” and “_B” appendages. An example of data generated by Protein Residue Enrichment is given in Figure 3-47.

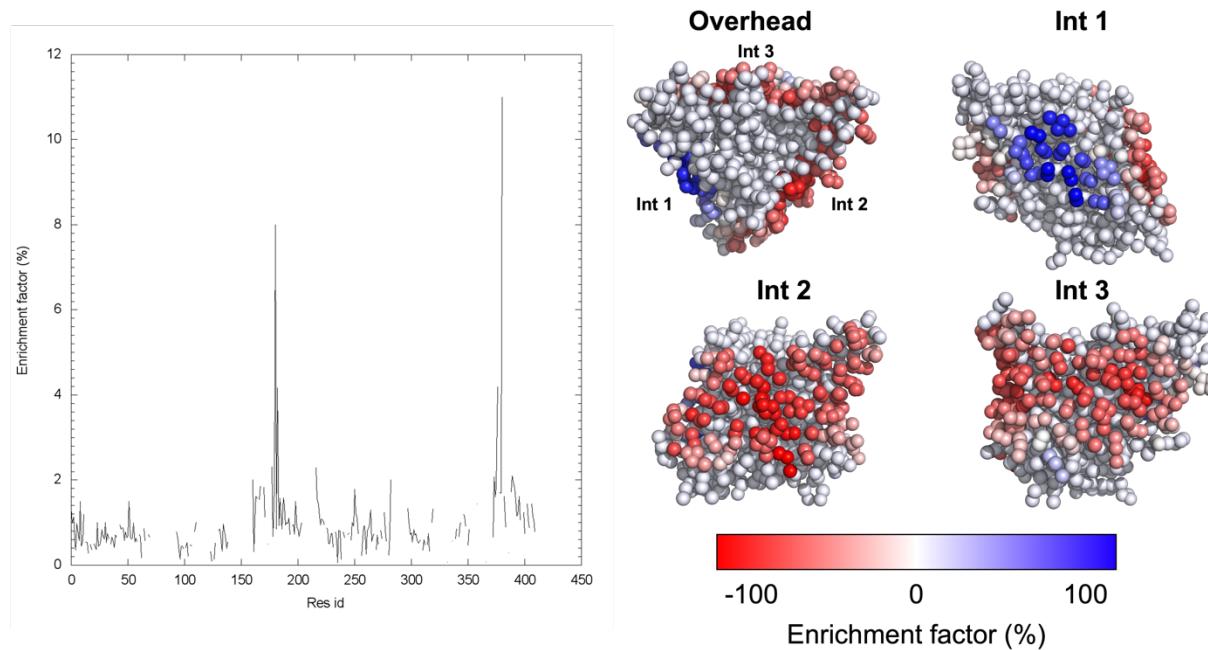


Figure 3-47 DLPX enrichment factor. Left panel shows the percent enrichment for each protein residue. Residues forming too few contacts with the lipids, i.e., the core residues, are left undefined. The right panel shows the same enrichment factor projected onto the atomic structure.

3.11 Lipid Exposed Surface Atoms

It is sometimes useful to measure the degree that the surface of a membrane embedded protein is exposed to the surrounding lipid molecules. For example, we may wish to count the protein atoms that consistently interact with the alkyl chains of the lipids. This kind of analysis may be performed by identifying the protein atoms that form one or more contacts with the atoms from the lipid alkyl chains. These atoms are identified within each trajectory snapshot and the percentage of frames that a contact was present is determined for each protein atom. This percentage, which we refer to as the exposure factor, may be computed with the MOSAICS tool Surface Residue Finder. To use Surface Residue Finder, the user must specify a cutoff distance for counting contacts. This is done with the `-cdist` tag. Additionally, the user must specify the lipid types, and a list of atom types for each, to include in the contact analysis with the protein. This information is provided using a network of selection cards and the `-crd` tag (Figure 3-48).

-crd

#lipid_type	#filename	#cont._atoms	#cont._atoms	#cont._atoms	#cont._atoms
POPE	po_tails.crd	C1A	C1A	C1A	C1A
POPG	po_tails.crd	C2A	C2A	D2A	D2A
DLPE	dl_tails.crd	C3A	C3A	C3A	C3A
DLPG	dl_tails.crd	C1B	C1B	C4A	C4A
		C2B	C2B	C1B	C1B
		C3B	C3B	C2B	C2B
				C3B	C3B
				C4B	C4B

Figure 3-48 Selection card structure used by Surface Residue Finder. For the example here, we have included the lipid alkyl chains when counting protein-lipid contacts.

Output from Surface residue Finder includes a PDB file with the exposure factor given for each protein atom as the B-factor. In addition to this, the number of surface atoms s_t , i.e., the number of protein atoms that formed a contact with the target lipid atoms, is computed for each timepoint t and a probability distribution is generated. The histogram filename may be set by the user via the -histo tag and the bin width is set using the -bin tag. The average number of surface atoms \bar{s} is also reported along with the histogram width σ_s . This information, along with the exposure factors, are used to identify the most probably surface atoms. These atoms are highlighted in a second PDB using the B-factor and are selected as the N protein atoms with the highest exposure factors. The number of atoms selected, i.e., N, is determined by the user via the relation:

$$N = \bar{s} + \chi * \sigma_s \quad (3.14)$$

where χ a is a number provided via the command line arguments using the -cutoff tag. An example of the run commands required by Surface Residue Finder is now given:

```
$ mpirun -n 100 surface_residue_finder_mpi -traj traj.xtc -ref ref.pdb -crd podl.crd -histo
surface.dat -s_pdb surface.pdb -width 1 -leaf 0 -cutoff 0.0 -cdist 0.6
```

In the example given here, the output file containing the histogram data is specified via the -histo tag. Likewise, the PDB file with the selected surface atoms, indicated via the B-factor, is specified by the -s_pdb tag. This filename is used to generate a filename for the PDB file containing the exposure factor (B-factor) such that the “_ranks” appendage is added. Figure 3-49 shows an example of the data generated with Surface Residue Finder. We note that a list of selection commands is also generated and may be used to select the surface or core atoms within PyMOL.

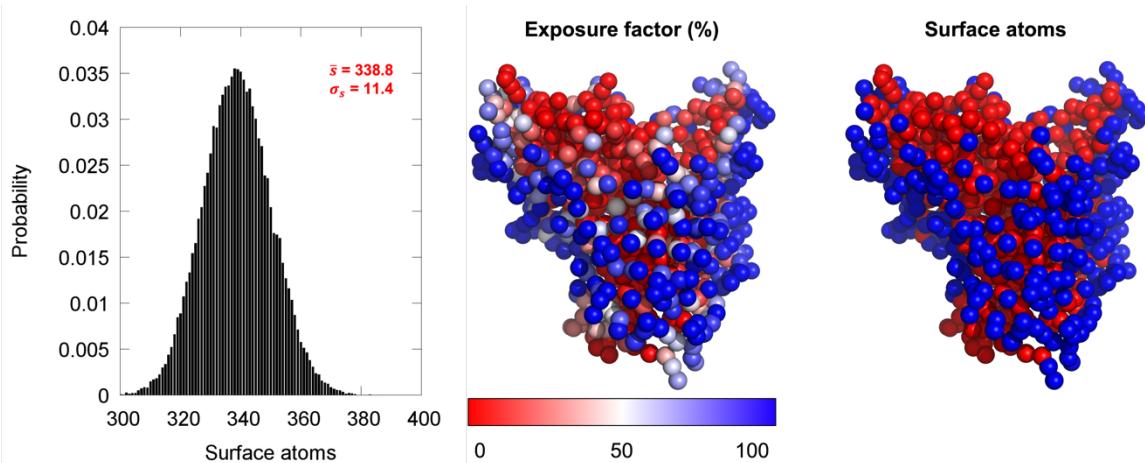


Figure 3-49 Probability histogram for the number of surface atom). The average and distribution width are reported in red (left). The Exposure factor (middle). The surface atoms shown as blue spheres (right). For the analysis shown here χ was set to nil.

We note that the user can check the surface atoms by selecting them in the time average protein structure (section 3.12) and then loading the time average lipid coordinates (section 3.12). See Figure 3-50 for an example.

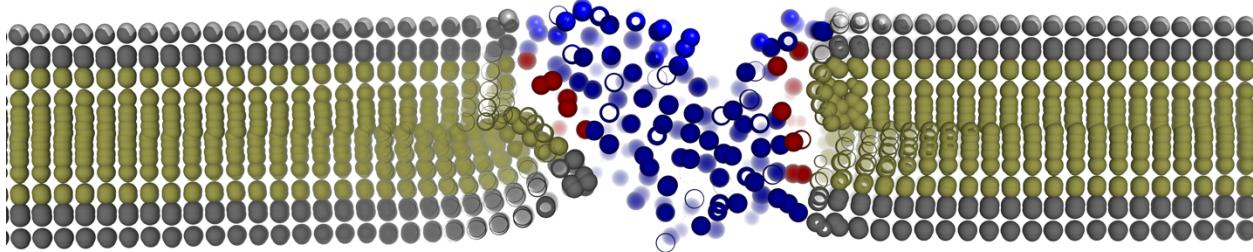


Figure 3-50 Surface atoms (red) selected in the time average protein coordinates. The time average lipid coordinates are also shown. Here, we color the lipid atoms used in the contact analysis (with Surface Residue Finder) yellow and the remaining lipid atoms grey.

3.12 Mean Atomic Coordinates

When examining the lipid structure, one is faced with the problem of determining statistical significance. For example, one might begin by viewing a trajectory with PyMOL (Schrödinger, LLC) or VMD [9]. If the user is lucky, they might find something interesting in one of the trajectory frames, for example a tilted lipid, etc. Of course, a single instance of a tilted lipid or any other observable is not significant. Instead, the observable must be frequent such that the behavior is captured in a time average. For tilted lipids and many other observables, the time average atomic coordinates tell the story. In this section we introduce 3 MOSAICS tools used for computing time averaged atomic coordinates. These include Mean Lipid Coords for the lipids, Mean Protein Coords for the protein, and Mean Coords for all molecule types.

To begin, the time average lipid coordinates can be acquired using Mean Lipid Coords. With this program, the time average coordinates are computed after sorting the lipids based on their position in the XY plane. As a result, the average lipid coordinates are projected onto the XY plane and can be visualized with a graphics tool such as PyMOL or VMD (Figure 3-51).

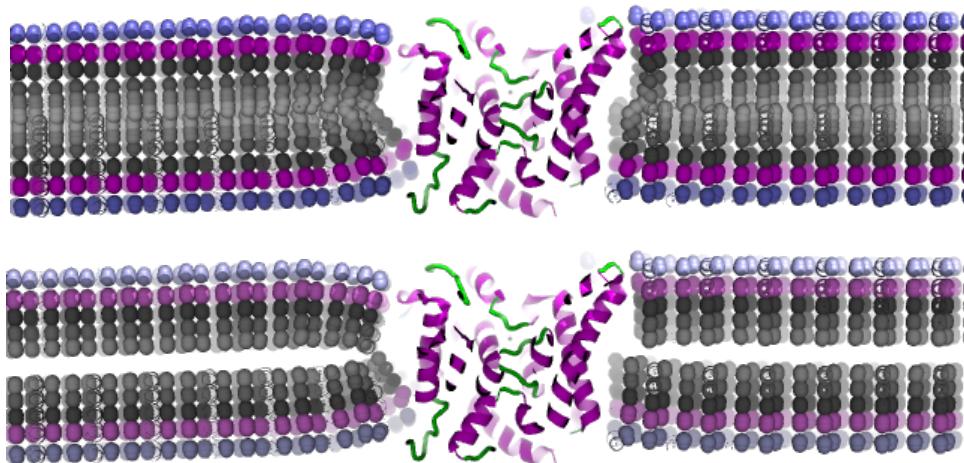


Figure 3-51 Time average lipid coordinates for POPE/POPG (upper) and DLPE/DLPG (lower). Shown also are the average protein coordinates acquired using Mean Protein Coords.

Mean Lipid Coords works by adding each lipid coordinate to a grid. This results in a grid for the x, y, and z coordinates of each atom making the lipid. For example, a Martini POPE molecule will have 36 grids to store the coordinates. To use the program, the user must provide a gro or pdb file containing a single molecule of the lipid type to be averaged. This is specified with the -param tag as shown in the following example.

-param

POPE sim

```
12
1POPE  NH3    1      X      Y      Z
1POPE  PO4    2      X      Y      Z
1POPE  GL1    3      X      Y      Z
1POPE  GL2    4      X      Y      Z
1POPE  C1A    5      X      Y      Z
1POPE  D2A    6      X      Y      Z
1POPE  C3A    7      X      Y      Z
1POPE  C4A    8      X      Y      Z
1POPE  C1B    9      X      Y      Z
1POPE  C2B   10      X      Y      Z
1POPE  C3B   11      X      Y      Z
1POPE  C4B   12      X      Y      Z
Box_x  Box_y  Box_z
```

Note the coordinates and box provided in the above gro file do not matter. This is because the ref file used here is only needed to specify the lipid type to be averaged and the atoms making the lipid. For Martini lipids, there are energy minimized lipid gro files available for each lipid type on the Martini website. It should be noted that, while Mean Lipid Coords averages a single lipid type, it is possible to average over multiple types simultaneously given that their chemistry allows for this. For example, the user could get the average coordinates for both POPE and POPG lipids

since they differ only by the head atom (GL0 vs NH3). To do so, the user could rename POPG to POPE in the reference file (-ref). Mean Lipid Coords also excludes insignificant data. This is done in the same manner as other programs such as Z Coord and uses the sample count ρ^{ij} (section 1.11). We note that ρ_t^{ij} can be used instead of ρ^{ij} by inclusion of the -rho_t 1 argument where ρ_t^{ij} pertains to the sample count where all lipid types stamp data to the grid rather than the single type being averaged. This option enables comparisons between different lipid types within a complex mixture (for example POPE vs DLPE) thus ensuring that the same lattice points are defined regardless the lipid type being averaged (Figure 3-51). In either case, ρ^{ij} is used as the normalizing factor (equation 1.4) and is written to the B-factor in the PDB file containing the time averaged lipid coordinates (Figure 3-52).

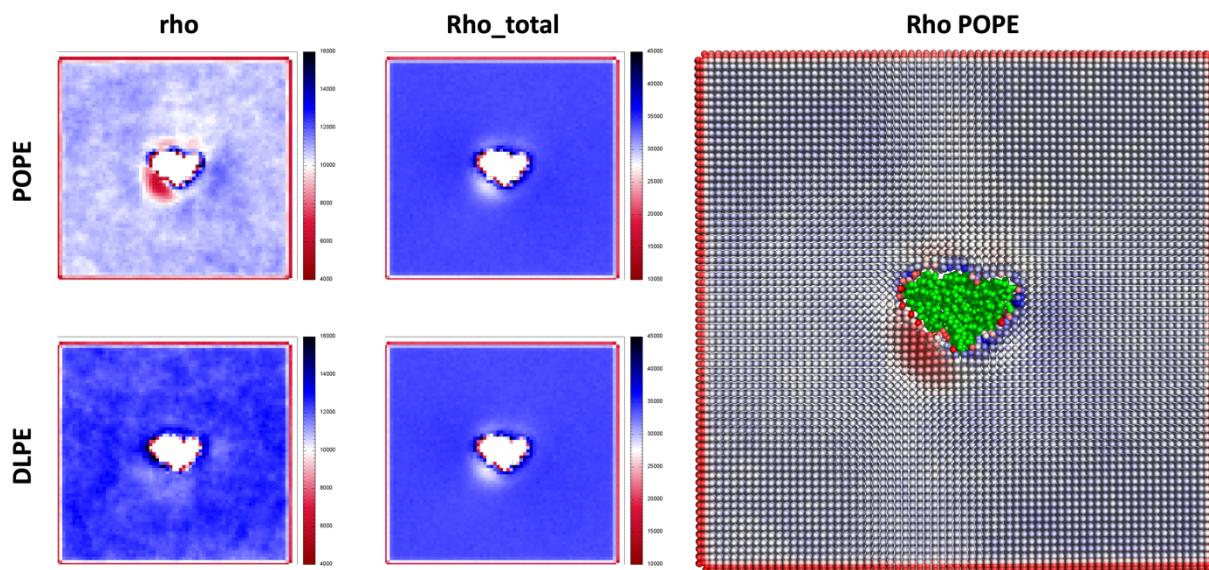


Figure 3-52 Comparison of the sample count ρ^{ij} for different lipid types and for all lipids ρ_t^{ij} . The left panel shows ρ^{ij} which varies with the lipid type. In contrast, the middle panel shows ρ_t^{ij} which is the same regardless of the lipid types selected for the analysis. The right panel shows the time average lipid coordinates for POPE with ρ^{ij} indicated via the B-factor (the atoms are colored by the B-factor).

Of course, the user must specify which atoms to use to represent the lipid's position in the XY plane when computing ρ^{ij} . This is done using the -m1 and -m2 tags; for example -m1 GL1 -m2 GL2. An example of the run commands required to use Mean Lipid Coords is now given:

```
$ mpirun -n 60 mean_lipid_coords_mpi -traj traj.xtc -ref ref.gro -param pope.pdb -mlc upper_pope.pdb -m1 GL1 -m2 GL2 -APS 0.16 -r 0.26 -cutoff 0.4 -leaf 1
```

In the example above, the ester atoms are used to represent the lipid position in the XY plane when computing the average coordinates and the sample counts. It should be noted that, even though grid points which have a sample count ρ^{ij} smaller than α (see equation 1.8) are excluded, the resulting PDB will contain atoms for these lipids. However, these atoms are assigned an x, y, and z coordinate of 0 and can be seen as an atom located at the origin. Other important arguments include the -mlc tag which is used to specify the name of the resulting

output data file containing the time average lipid coordinates. Similarly, the output data file containing the sample count (either ρ^{ij} or ρ_t^{ij} depending on the -rho_t option) is derived from this filename and is given the _rho.dat" appendage. We note that the user can reduce the grid resolution in the output PDB. This is usually needed (if -APS is small) to make visualization of the lipids easier. Otherwise, the lipids will likely overlap one another when viewed in PyMOL. To adjust the output resolution, the -g_strd tag may be included which is short for grid stride. An example of averaged lipid coordinates from a simulation is shown in Figure 3-51. Note the leaflets in Figure 3-51 were acquired separately (run the analysis twice with -leaf 1 and then -leaf 2). Specifying -leaf 0 will not produce the desired results.

As a last note, we mention that Mean Lipid Coords can be used on all-atom or coarse-grained systems alike. In the case of an all-atom model, it may be desirable to remove some of the atoms from the analysis like the hydrogen atoms. If this approach is taken, the user should remove the desired atoms from the trajectory as well as the reference files (-ref, -param) before performing the analysis.

In addition to Mean Lipid Coords, there are a couple add on programs which help with visualizing the data. For example, Mean Coords Row Selector generates PyMOL commands that can be used to select each row and column of lipids making the grid. This makes it easier for the user to hide certain rows or columns when visualizing the results. To use Mean Coords Row Selector, the user only needs to specify the number of grid points in the x and y dimensions. This information is provided as standard output (num_g_x and num_g_y, see section 1.11) when running Mean Lipid Coords. To specify this information, the user must include the -height and -width tags. The user can also add additional selection text to the end of the PyMOL command using -e tag. For example, the user could use -e "&upper_pope" to specify the selection of POPE lipids in the upper leaflet only. We now give an example of the run commands used by Mean Coords Row Selector:

```
$ mean_coords_row_selector -height 72 -width 72 -o row_selections.pml -e "&upper_pope"
```

In the example provided here, the output data file containing selection commands is specified using the -o tag. This selection script may be directly interpreted by PyMOL. For an example of the Mean Lipid Coords which are visualized after selecting specific rows see Figure 3-53.

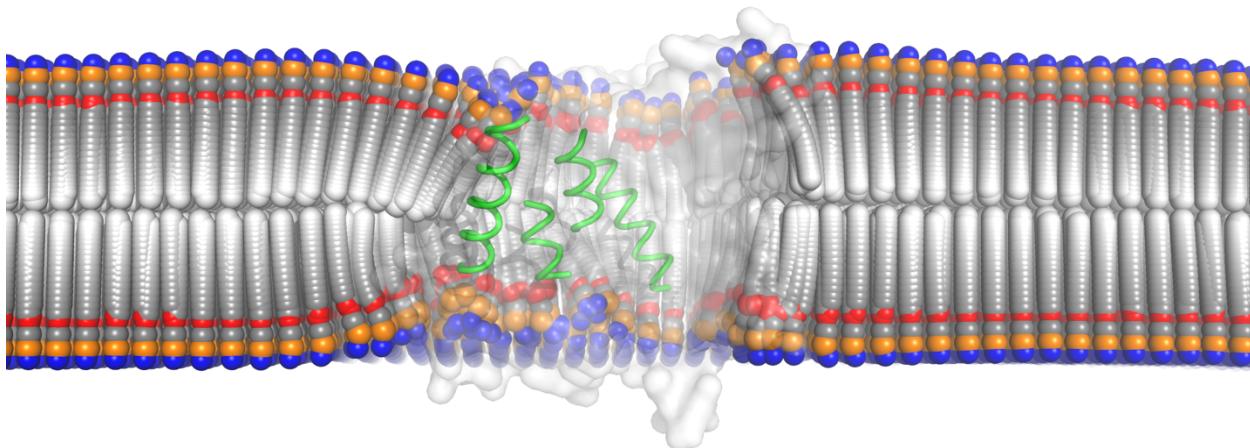


Figure 3-53 Mean lipid and protein coordinates with select rows of lipids hidden for clarity. The rows may be selected using Mean Coords Row Selector.

And finally, there is a tool “B Stamp Grid” that allows the user to apply grid data to the B-factor of the mean lipid coordinates. To use B Stamp Grid, the user must provide the mean lipid coordinates with the -traj tag and the grid data to be applied to the B-factor with the -grid tag. Of course, the grid data and mean lipid coords should be compatible, i.e., have the same number of grid points in x and y. Note, B Stamp Grid will use the NaN information from the grid data to exclude insignificant data (sets their coordinates to zero) in the resulting average coordinates. An example of the run commands required to use B Stamp Grid is now given:

```
$ mpirun -n 1 b_stamp_grid_mpi -traj upper_pope.pdb -ref upper_pope.pdb -grid upper_enrichment.dat -o upper_pope_enrichment.pdb -odf 0
```

In the example provided here, the output data file, containing the time average lipid coordinates with the desired grid data copied to the B-factor, is specified via the -o tag. An example of data generated with Mean Lipid Coords and B Stamp Grid is shown in Figure 3-54.

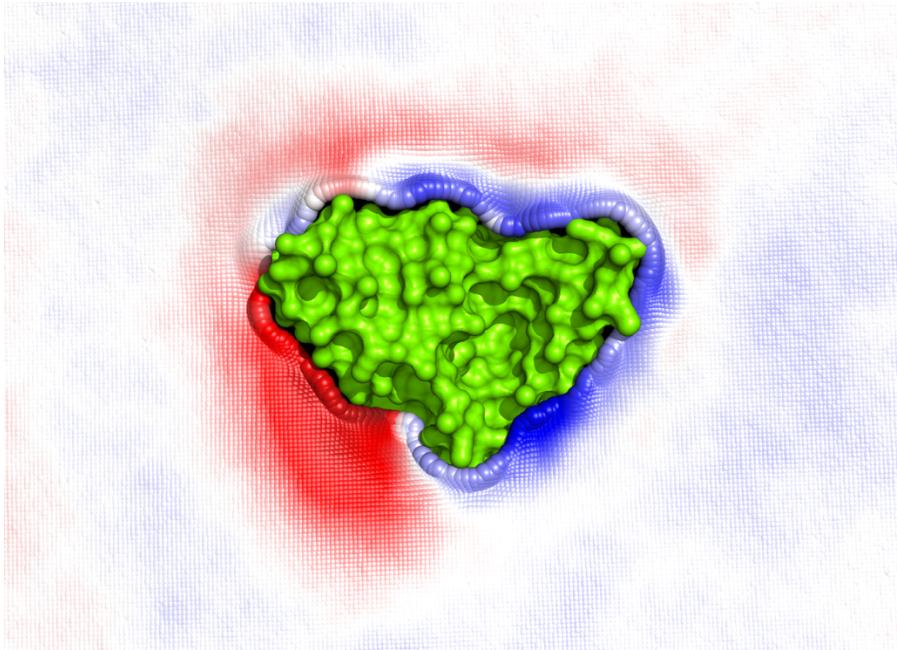


Figure 3-54 DLPX lipid enrichment data stamped onto the time average lipid coordinates using B Stamp Grid. Lipid atoms are colored according to the B-factor i.e., the DLPX enrichment factor. Shown are the ester atoms (GL1, GL2) only. The average coordinates of the CLC-ec1 protein [10] are shown as a green surface.

While Mean Lipid Coords averages the lipid coordinates, it is often desirable to view these structures in relation to the average protein coordinates. To facilitate this task, we have the MOSAICS tool Mean Protein Coords. Mean Protein Coords is an analysis program designed for computing the time average protein coordinates. This analysis is meaningful only if the protein's position is static, possibly resulting from least squares fitting performed before the analysis. Mean Protein Coords requires no input from the user aside from the standard requirements of a trajectory and reference file. An example of the run commands required to use Mean Protein Coords is now given:

```
$ mpirun -n 25 mean_protein_coords_mpi -traj traj.xtc -ref ref.gro -mpc mean_prot.pdb
```

In the example provided here, the time average protein coordinates are written to a file as specified using the -mpc tag. We note that the time average protein coordinates can be viewed with the time average lipid coordinates (computed with Mean Lipid Coords). See Figure 3-51 for an example of the output generated by Mean Protein Coords. We note that Mean Protein Coords can be set to compute, for each protein atom i , the average distance $\bar{\delta}_i$ from the time average coordinate $\langle \vec{r} \rangle_i$:

$$\bar{\delta}_i = \frac{1}{T+1} \sum_{t=0}^T (\vec{r}_{i,t} - \langle \vec{r} \rangle_i)^2 \quad (3.15)$$

where $T+1$ is the number of trajectory frames analyzed and $\vec{r}_{i,t}$ is the position vector for atom i in frame t . The average distance can be used to judge the significance of the time average coordinates. For example, a small $\bar{\delta}_i$ means the average coordinates are more representative of

the individual snapshots than when $\bar{\delta}_i$ is large. To compute the average atomic distance from the time average coordinates the user must specify the -dist 1 argument. This will instruct the program to write an additional PDB file, containing the same name as specified with the -mpc tag but with the “_mean_dist” appendage, where the average distance (in nm) is written to the B-factor. An example of the mean protein coordinates with $\bar{\delta}_i$ indicated for each atom is shown in Figure 3-55.

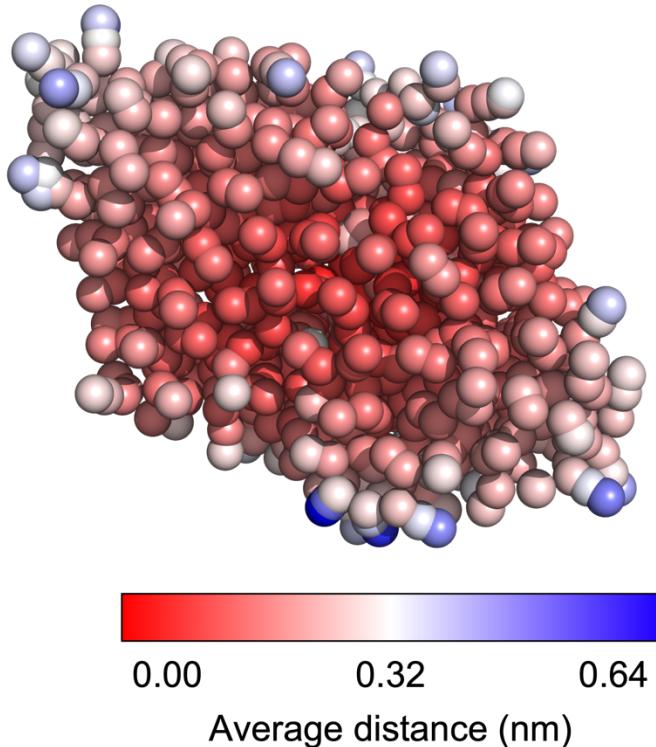


Figure 3-55 Time average protein coordinates with $\bar{\delta}_i$ indicated via the B-factor for each atom.

And finally, the average coordinates of all molecules within a system can be acquired using the MOSAICS tool Mean Coords. This is mainly useful when using Atoms in 2 Planes (see section 3.13) but can also be used to make a reference file for leaflet finder. Use of the program only requires input of a trajectory file with the -traj tag and a reference file with -ref. An example of the required run commands is now given:

```
$ mpirun -n 50 mean_coords_mpi -traj traj.xtc -ref ref.gro -avg avg_coords.pdb
```

In the example provided here, the output data file containing the time average coordinates is specified using the –avg tag. An example of the time average coordinates is shown in Figure 3-56.

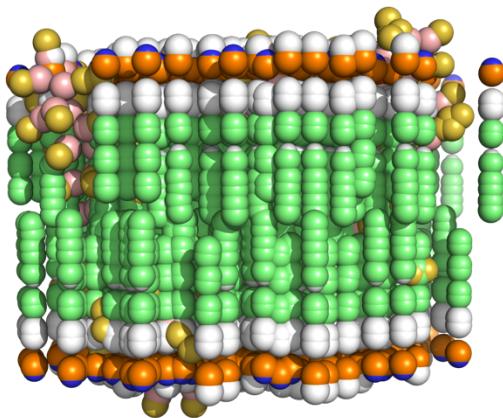


Figure 3-56 Time averaged atomic coordinates for a system containing CLC-ec1 embedded in a coarse-grained bilayer. Note the z-coordinates are ideal for use with leaflet finder.

3.13 The Protein Tilt Angle

With the MOSAICS tool Protein Orientation, the protein tilt angle can be measured within a membrane/protein simulation. This program works by defining an orientation vector and then computing the ϕ and θ angles (see Figure 3-57) made by the vector.

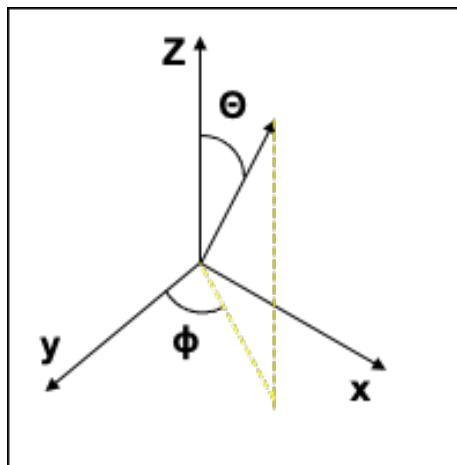


Figure 3-57 θ and ϕ angles computed by Protein Orientation. These angles range from 0-180° and 0-360° respectively.

To define the orientation vector, the user must provide two atom selections. This is done with the -upper and -lower tags as is shown in the following example.

-upper

```
#upper_selection 645 649 660 721 736 819
```

-lower

```
#lower_selection 380 528 541 724 798 886
```

With two atom selections specified, Protein Orientation computes the geometric center (equation 1.3) of each selection. The orientation vector is then defined as the vector connecting these two centers. For a stable fold, i.e., a rigid protein that undergoes no large structural

rearrangements during the simulation, the orientation vector tilts as the protein does and can be used gauge the overall protein tilt angles.

For protein dimers with two-fold symmetry (such as that in the CLC dimer [10]), the protein will tilt around the z-axis in a way that reflects the symmetry. To be precise, for any given angle ϕ the frequency will closely match that of $\phi + 180$. When this condition holds, taking the time averaged protein coordinates will give a good representation of the protein complex with zero tilt, i.e., aligned parallel to the z-axis. Taking this ideal representation of the protein, one can define an atom selection such that the orientation vector points parallel to the protein and z-axis. This is done by searching for pairs of atoms from the upper portion of the protein and another from the lower portion such that, for each pair, the x and y coordinates are similar but not z. At the end of this section, we will introduce the MOSAICS tool Atoms in 2 Planes which is used to find these atom pairs. By following this selection procedure, the x and y components of the geometric center of the two atom selections will track each other but not the z component. The vector connecting the two centers will therefore orient with the z-axis (see Figure 3-58).

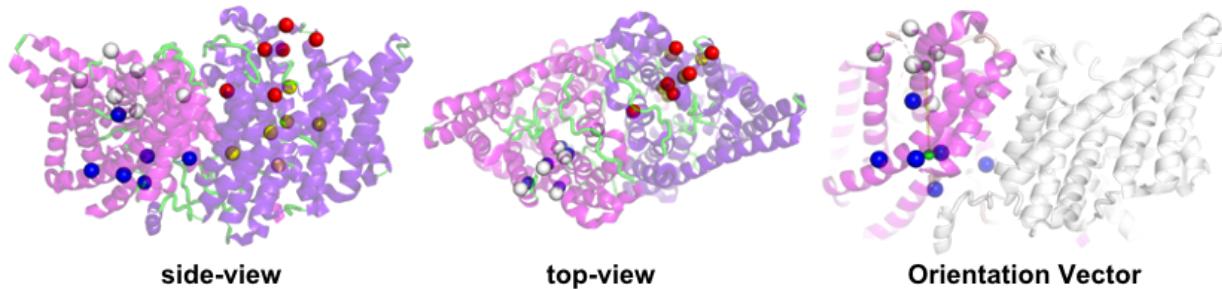


Figure 3-58 Atom selection for the CLC dimer [10] (time averaged coordinates) such that the orientation vector connecting the center of mass from each selection points up. Note, there are four atom selections for the dimer so that there is an orientation vector for each protomer. Left panel shows the four selections (colored red, white, blue, and yellow) and illustrates the difference in z for each. Middle panel shows that each pair share very similar x-y coordinates. The right panel shows the orientation vector for one of the protomers. The centers of each atom selection are shown in green and the orientation vector as a yellow line.

Following this protocol, the user now has the atom selections needed for measuring protein tilt for both protomers of the dimer (the same selection should be used for each protomer). The user should get very similar angles for each protomer given how the atom selections were made. Furthermore, the same atom selection should be used if the tilt is measured for a monomer system.

An example of the run commands used with Protein Orientation is now given:

```
$ mpirun -n 25 protein_orientation_mpi -traj traj.xtc -ref ref.gro -upper upper.ndx -lower lower.ndx -ori prot_tilt.dat
```

In the example provided here, the output data file containing a timeline of the ϕ and θ angles is specified via the -ori tag. However, the user can use the MOSAICS tool Orientation Histogram to make a polar histogram of the data. That is, the data will be grouped into bins for ϕ and θ , and the percentage of data points falling in each bin is computed. The data is presented in a polar plot with the polar angle corresponding to ϕ and r corresponding to θ . The percentages are

represented by color. To produce a plot like that seen Figure 3-59, a command like the following may be used:

```
$ orientation_histogram -d prot_tilt.dat -o prot_tilt_histo.dat -res_t 0.1 -res_p 1 -line_v 0.03 -res 0.05
```

Here, `-res_t` gives the width of the bins for θ and `-res_p` gives the width of the bins for ϕ . The argument `-line_v` is the heatmap value that is given for the contour lines. That is, the contour lines representing increasing θ are coded into the data (for Figure 3-59 lines were given a value of 0.03). The argument `-res` gives the output resolution. Note there is a resolution for binning the data that is set with `-res_t` and `-res_p` and a resolution for the output. The output resolution is typically much higher (i.e., `-res` is a small number) than that used for binning. An example of the data generated with Protein Tilt and Orientation Histogram is shown in Figure 3-59.

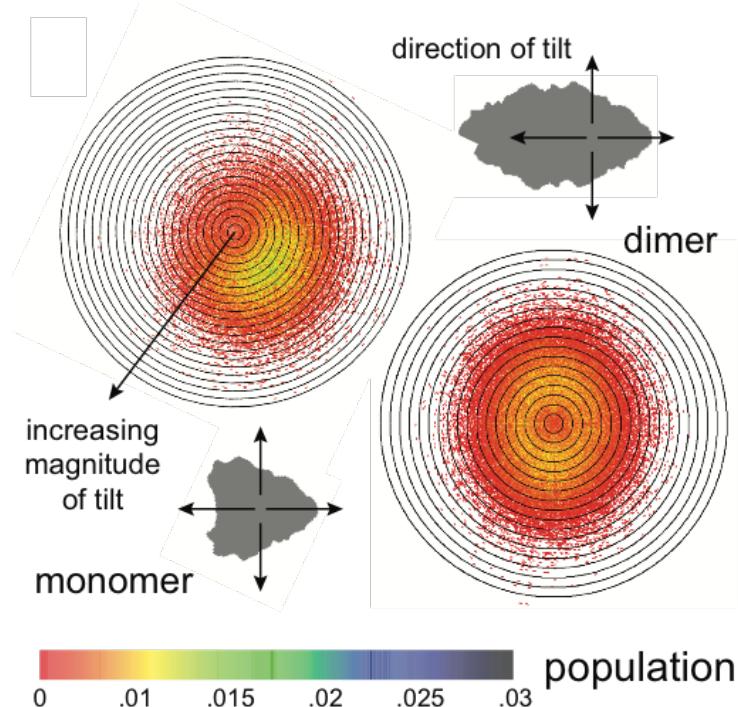


Figure 3-59 Tilt of the CLC [10] monomer and dimer systems. The contour lines represent increasing θ . The position on the plot indicates the direction the lipid tilts (see grey proteins).

We note that the components of the orientation vector can be displayed for each trajectory frame by inclusion of the `-p_vec 1` argument when running Protein Orientation. Likewise, a PDB can be written to file with the orientation vector defined by a pair of pseudo atoms, like shown in Figure 3-58. In this case, the vector is shown at its location with the protein as well as at the origin [0,0,0]. To acquire such a PDB, the user must include the `-o_pdb 1` argument. When this option is supplied, the PDB will be written to a file with the same name as specified by `-ori` but with the “`_ori_vec.pdb`” appendage. Note, this option should be used on a single trajectory frame (try using the `-b` and `-e` options).

As was mentioned previously, the MOSAICS program Atoms in 2 Planes can be used to find pairs of atoms in the protein that are close to each other in XY but far apart in Z. These atoms

make two groups whose geometric centers (equation 1.3) can be used to define an orientation vector. Furthermore, it was also stated that the time average coordinates for protein dimers with two-fold symmetry produce an ideal configuration with zero protein tilt (aligned parallel with the z-axis). To acquire the average coordinates, the user is encouraged to use MeanCoords (section 3.12). With MeanCoords, a time average structure is computed for all molecules in the system. For the lipids and solvent, such structures are meaningless. However, the average coordinates of the protein will be identical to those computed with Mean ProteinCoords. The advantage of averaging the entire system is that the atom numbering is preserved such that the structure can be used with Atoms in 2 Planes.

With the average coordinates computed, one can feed them into Atoms in 2 Planes with the -traj tag. To use the program, the user must also specify an atom type making the pairs, how far away the atom should be in Z, and how close they should be in XY to be counted. This information is provided with the -type, -z, and -xy tags respectively. The output from Atoms in 2 Planes is a list of residues and atoms making the pairs. An example is given below.

```
$ mpirun -n 1 atoms_in_2_planes_mpi -traj avg_coords.pdb -ref avg_coords.pdb -z 1.5 -xy 0.03 -type BB
```

The example given here should produce something like the following:

Upper Residue:	821	atom:	1696	Residue:	9	atom:	21
Upper Residue:	856	atom:	1763	Residue:	13	atom:	30
Upper Residue:	397	atom:	819	Residue:	186	atom:	380
Upper Residue:	354	atom:	736	Residue:	251	atom:	528
Upper Residue:	316	atom:	660	Residue:	256	atom:	541
Upper Residue:	308	atom:	645	Residue:	386	atom:	798
Upper Residue:	311	atom:	649	Residue:	347	atom:	724
Upper Residue:	345	atom:	721	Residue:	430	atom:	886
Upper Residue:	377	atom:	785	Residue:	454	atom:	935
Upper Residue:	381	atom:	790	Residue:	453	atom:	933
Upper Residue:	382	atom:	792	Residue:	447	atom:	920
Upper Residue:	412	atom:	851	Residue:	457	atom:	942
Upper Residue:	661	atom:	1366	Residue:	604	atom:	1248
Upper Residue:	621	atom:	1274	Residue:	638	atom:	1314
Upper Residue:	750	atom:	1554	Residue:	871	atom:	1790
Upper Residue:	752	atom:	1557	Residue:	829	atom:	1708
Upper Residue:	754	atom:	1559	Residue:	790	atom:	1634
Upper Residue:	788	atom:	1631	Residue:	873	atom:	1796

It should be noted that Atoms in 2 Planes uses protein finder to filter protein atoms from the rest of the system. As a result, atom pairs are only chosen from the protein. The atom numbers for each pair can be fed into Protein Orientation with the -upper and -lower tags. It should be noted however, that the orientation should be measured for each protomer separately and any pairs containing cross terms should be deleted. Furthermore, the same atom selection should be used in each protomer.

3.14 Lipid Hydrogen Bonding and Salt Bridges

For all-atom simulations, the prevalence of hydrogen bonds and salt-bridges formed between the protein and lipids can be directly measured. In this section, we introduce 2 MOSAICS tools, Lipid H-Bonds and Lipid Salt Bridges, that are used to measure each type of bond. Note, this information is projected onto the XY plane.

To begin, the number of hydrogen bonds formed between the protein and lipids is found with Lipid H-Bonds. For this analysis, hydrogen bonds are measured between an acceptor atom, a donor atom, and a hydrogen atom attached to the donor. Moreover, a hydrogen bond requires a distance between the donor/acceptor atoms of no more than 3.5 Å and an angle of 30° or less (see Figure 3-60). To use the program, the user must specify a list of donor and acceptor atom types for both the protein and lipids. This is done with the -lip_d, -lip_a, -prot_d, and -prot_a tags. An example follows:

```
-lip_d  
#donors None  
  
-lip_a  
#acceptors O11 O12 O13 O14 O21 O22 O31 O32  
  
-prot_d  
#donors N NZ NE1 NE2 ND2 NE NH1 NH2 ND1  
#donors NT O OG1 OD1 OD2 OE1 OE2 OH OG  
  
-prot_a  
#donors N NZ NE1 NE2 ND2 NE NH1 NH2 ND1  
#donors NT O OG1 OD1 OD2 OE1 OE2 OH OG
```

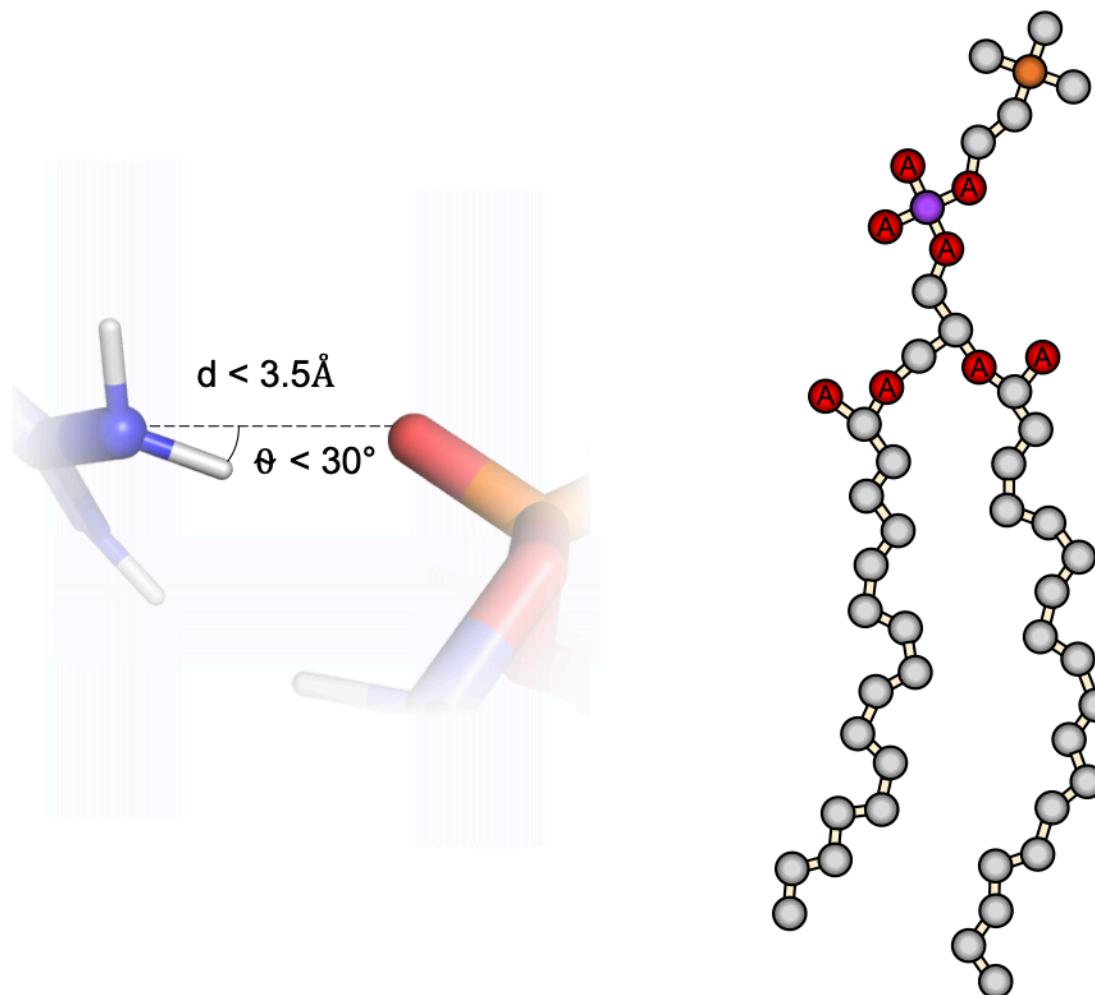


Figure 3-60 Schematic of a hydrogen bond measurement between acceptor and donor atoms (left). A cartoon representation of a POPC lipid is also shown (right). For POPC, only hydrogen bond acceptors are present.

Note, a POPC lipid, as used in this example, contains no hydrogen bond donors. If no atom is present, the user can specify “None” in the selection card. It is generally required that the user determine the atom types that participate in hydrogen bonding, which may differ from the example provided here depending on the force field used. In addition to the atom types provided, the user must also specify a bonds list for their system, i.e., a list of every pair of atoms that form bonds in the system. This list is used to find hydrogen atoms attached to the donor atoms. The bonds list is provided with the -bond option as is demonstrated in the example below.

-bond

```
#atom_1  #atom_2
1       2
1       3
1       4
1       5
5       6
5       7
```

```
7      8
7      9
9     10
9     11
.
.
.
```

Note, no hydrogen atom types need be specified by the user. Instead, the program assumes any atom type beginning with “H” is a hydrogen. And finally, the user must specify which lipid types to include in the analysis. This is done with the -crd tag. An example is now given.

```
-crd
#lip_t  #map_1  #map_2
POPC    C21      C31
```

In the example provided here, hydrogen bonds are counted between POPC lipids and the protein. The number of hydrogen bonds is mapped to the ester atoms (C21 and C31) in the XY plane. An example of the run commands for Lipid H-bonds is now given:

```
$ Mpirun -n 200 lipid_h_bonds_mpi -traj traj.xtc -ref ref.gro -crd popc.crd -lip_d lip_d.crd -lip_a
lip_a.crd -prot_d prot_d.crd -prot_a prot_a.crd -bond bonds.crd -lphb upper_hb.dat -APS 0.005 -
r 0.23 -cutoff 0.4 -leaf 1
```

In the example provided here, the output data file containing the spatially resolved time average hydrogen bond count is written to a file as specified with the -lphb tag. We note that it is good practice to check that hydrogen bonds are correctly identified, especially given the large amount of input parameters required from the user. The hydrogen bonds may be checked by visual inspection using PyMOL (Schrödinger, LLC). To do so, the user should include the -test 1 argument. This will print a set of selection commands which may be used with PyMOL to select the hydrogen bond. Note, this will print a list of commands for every hydrogen bond found. It is therefore recommended that the test option be used on a single trajectory frame (try using -b 0 -e 0). See Figure 3-61 for an example.

Analysis of Lipid Structure

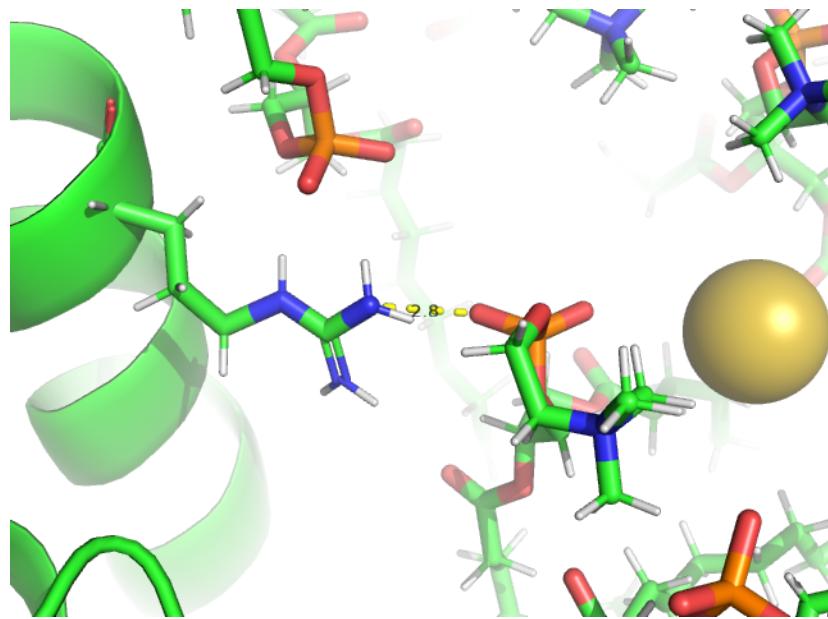


Figure 3-61 Verification of a hydrogen bond identified using Lipid H Bonds.

An example of the data generated with Lipid Hydrogen Bonds is shown in Figure 3-62.

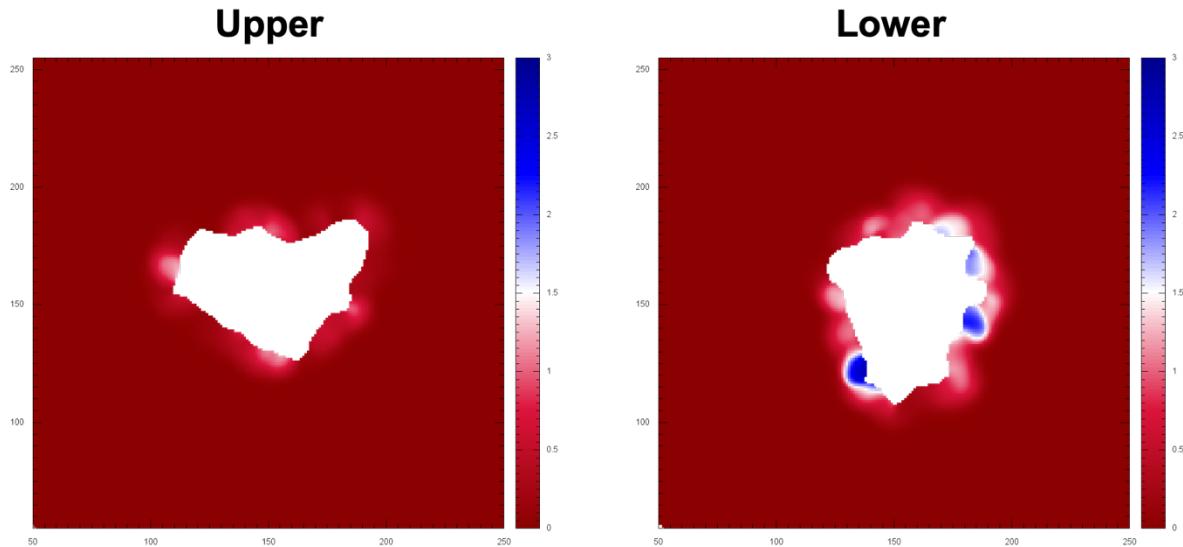


Figure 3-62 Hydrogen bonds formed between the CLC-ec1 protein [10] and POPC lipids. Shown are h-bonds for the upper and lower leaflets which differ dramatically. Units for the x/y axis are grid points and hydrogen bonds for the color bars.

Like hydrogen bonding, lipid salt-bridges may be characterized using the MOSAICS tool Lipid Salt Bridges. With this program, salt-bridges are counted between the protein and lipids based on the lipid's position in the XY plane. Because the net charge of a residue/lipid is typically spread out over many atoms, we use the geometric center (equation 1.3) of these atoms when performing distance calculations. See Figure 3-63 for an example.

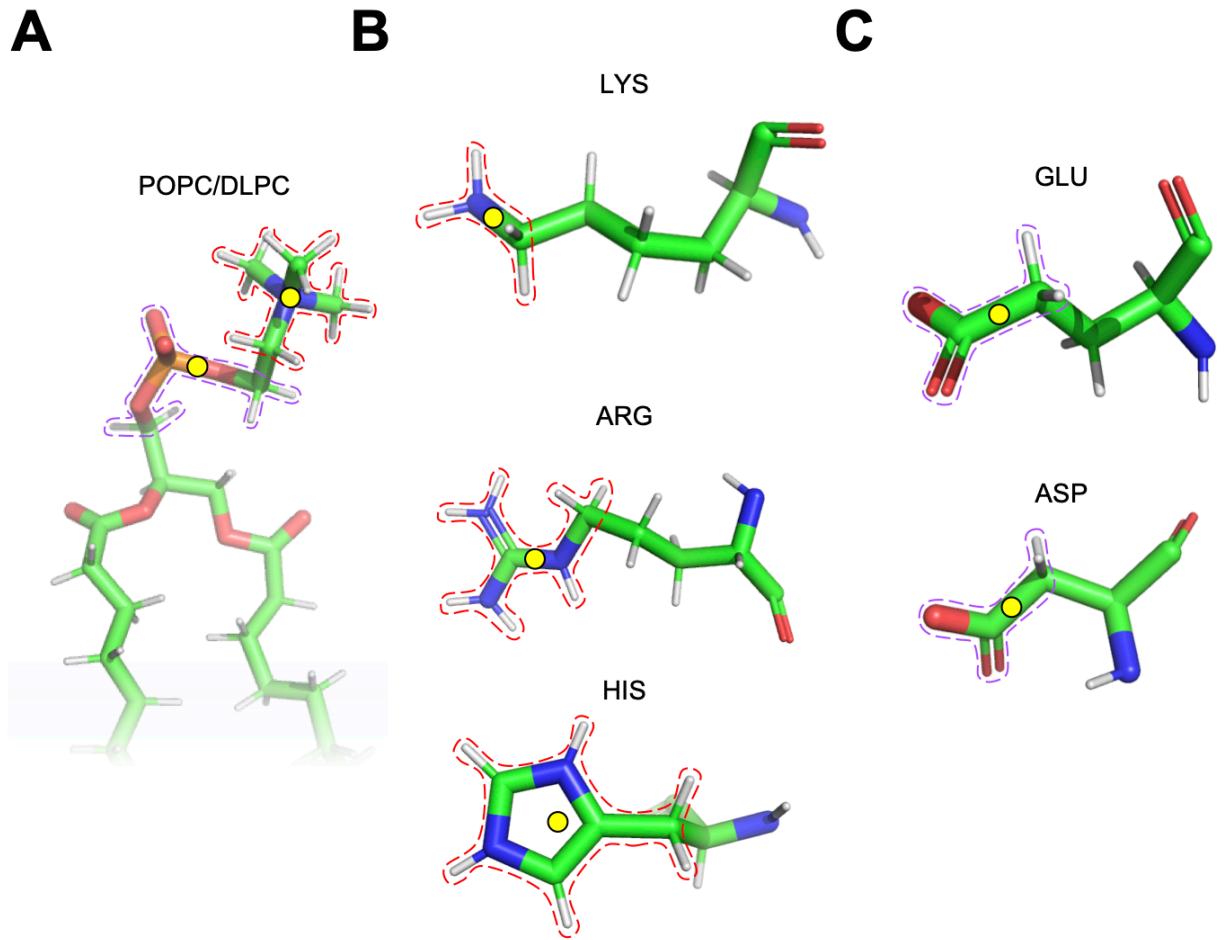


Figure 3-63 The distribution of charge over charged amino acids and a POPC lipid. A net charge of -1 is enclosed by purple dotted lines while a net charge of +1 is enclosed with red dotted lines. Geometric centers are shown as yellow circles. The charge distribution for a POPC molecule (A). Positively charged amino acids (B). Negatively charged amino acids (C).

To use the program, the user must specify the cutoff distance between these centers. This is done with the `-cdist` tag. Moreover, the user must also specify the lipid types to include in the analysis. This is done with the `-crd_1` tag. Similarly, the charge groups must be specified for the lipids and any charged amino acids. This information is provided using 2 networks of selection cards each specified with the `-crd_2` or `-crd_3` tags. We note that the charge of the group must be indicated using either a "+" or "-" symbol. This information is used to find pairs with opposite charges. See Figure 3-64 for an example of the required selection cards.

Analysis of Lipid Structure

-crd_1

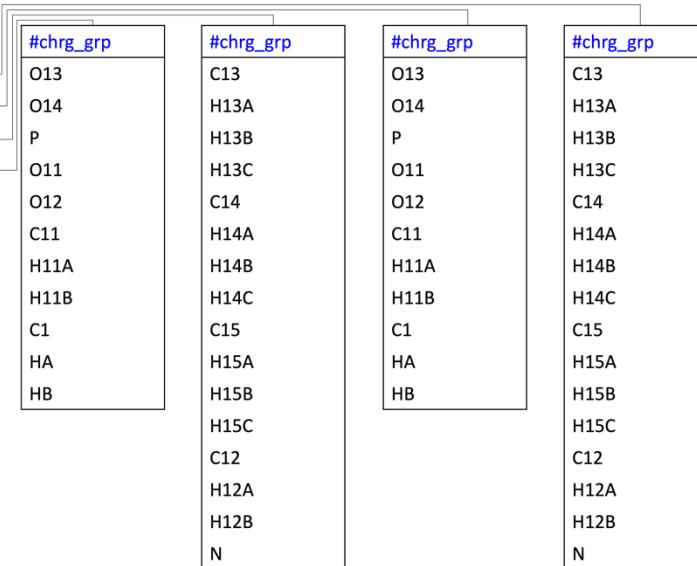
Stamping lipids

#lipid_type	#map_1	#map_2
POPC	GL1	GL2
DLPC	GL1	GL2

-crd_2

Lipid charges

#lipid_type	#charge	#filename
POPC	+	popc_p.crd
POPC	-	popc_n.crd
DLPC	+	dlpc_p.crd
DLPC	-	dlpc_n.crd



-crd_3

Protein charges

#res_type	#charge	#filename
LYS	+	lys.crd
ARG	+	arg.crd
HSP	+	hsp/crd
ASP	-	asp.crd
GLU	-	glu.crd

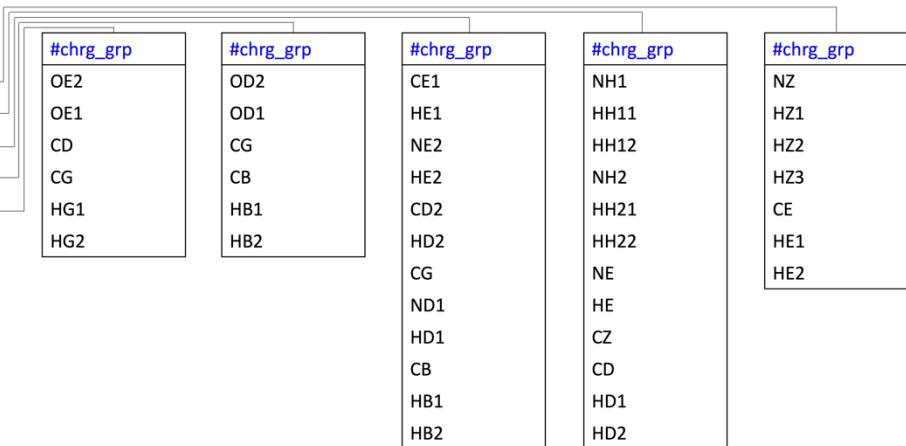


Figure 3-64 Selection card structure used by Lipid Salt Bridges. Here, the first card is used to specify which lipid types are probed in the analysis. That is, we count the number of salt-bridges formed between these lipids and the protein. The second selection card is used to specify charge groups for the lipids and the third card specifies charge groups for the protein residues.

In the example presented here (Figure 3-64), the number of salt-bridges is computed between POPC lipids and the protein as well as DLPC lipids and the protein. This number is then mapped to the ester atoms (C21 and C31) of the corresponding lipid molecules and added to the grid. To find the number of salt-bridges formed for a given lipid molecule, the lipid type is first screened using the types found in -crd_1. If the lipid is of the correct type, then the charge groups are looped over in -crd_2 until a matching lipid type is found. The geometric center is then computed for the atoms specified in the secondary selection card (-crd_2). This is followed by

looping over the protein residues and checking each against the residue types found in -crd_3. If a match is found, then the geometric center is computed using the atom group specified in the secondary selection card specific to that residue (-crd_3). The charges are then compared for the 2 centers and if opposite the distance between the centers is computed and compared to the cutoff distance. We recommend that the user tests this routine to be sure the salt-bridges are correctly identified. This may be done using the -test 1 tag. With this, a list of PyMOL commands is generated which can be used to identify the salt-bridges in PyMOL. Because a set of commands is generated for every salt-bridge, it is a good idea to perform such a test on a single trajectory frame. See Figure 3-65 for an example.

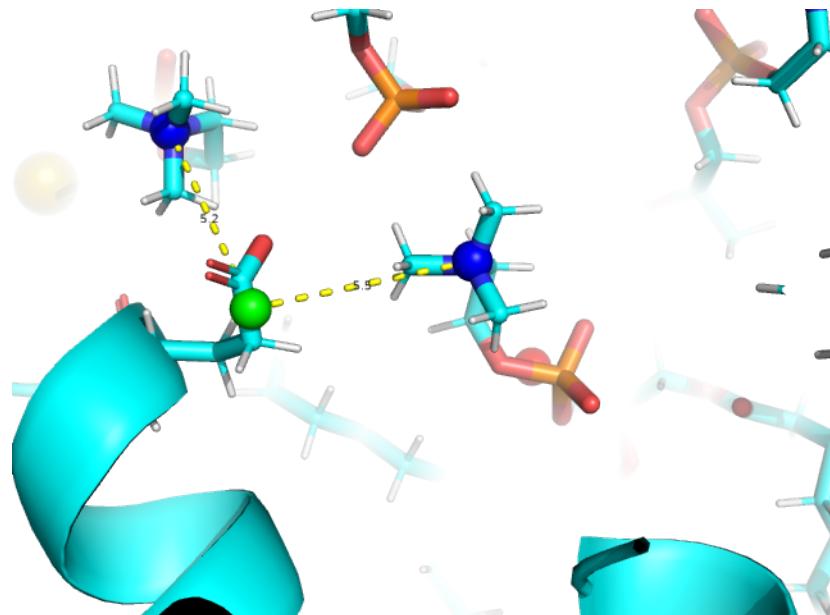


Figure 3-65 Verification of a pair of salt-bridges identified using Lipid Salt Bridges. The centers of the charge distributions are shown as blue and green spheres.

An example of the run commands used by Lipid Salt bridges is now given:

```
$ mpirun -np 56 lipid_salt_bridges_mpi -traj traj.xtc -ref ref.gro -crd_1 popc.crd -crd_2
popc_charges.crd -crd_3 prot_charges.crd -lpsb upper_popc_sb.dat -APS 0.005 -r 0.23 -cutoff 0.4
-leaf 1 -cdist 0.8
```

In the example provided here, the output data file containing the spatially resolved time average salt-bridge count is specified using the -lpsb tag. For an example of the data generated with Lipid Salt Bridges, see Figure 3-66.

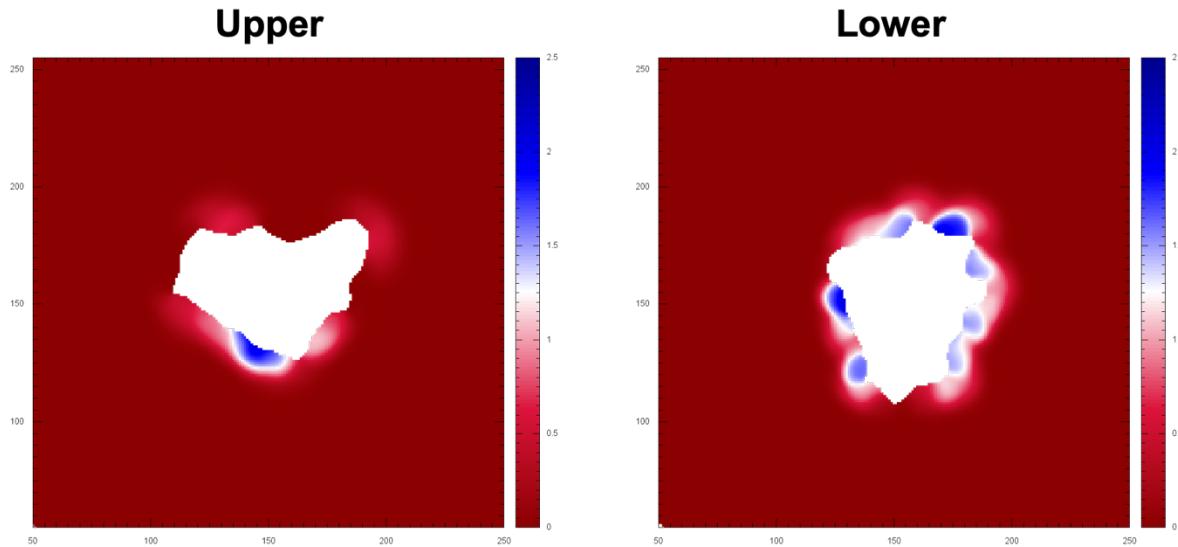


Figure 3-66 Number of salt-bridges measured between POPC lipids and the CLC-ec1 protein [10]. Units for the x/y axis are grid points and the number of salt-bridges for the color bars.

3.15 Lipid Flip-flop

The flipping of lipids between the leaflets can be detected by monitoring the z-coordinates of the lipids as the simulation proceeds. Such an analyses may be performed with the MOSAICS tool Lipid Flip. Lipid Flip works by computing the geometric center (equation 1.3) for a user defined group of atoms. The z-coordinate of this center is then plotted as a function of time for each lipid. To use the program, the user must specify the lipid types to include in the calculation and the atoms making the center. This information is specified using the -crd tag as follows (Figure 3-67).

-crd	
#lipid_type	#filename
POPC	pope.crd
#center_atom	
GL1	
GL2	

Figure 3-67 Selection card structure used by Lipid Flip. For this example, we monitor the z-coordinate of the geometric center for the atoms GL1 and GL2.

An example of the run commands used with Lipid Flip is now given:

```
$ mpirun -n 50 lipid_flip_mpi -traj traj.xtc -ref ref.gro -crd popl.crd -flip upper_flip.dat -leaf 1
```

In the example provided here, the output data file (containing the z-coordinates for each lipid center as a function of time) is specified via the -flip tag. Figure 3-68 shows an example of the data generated by Lipid Flip.

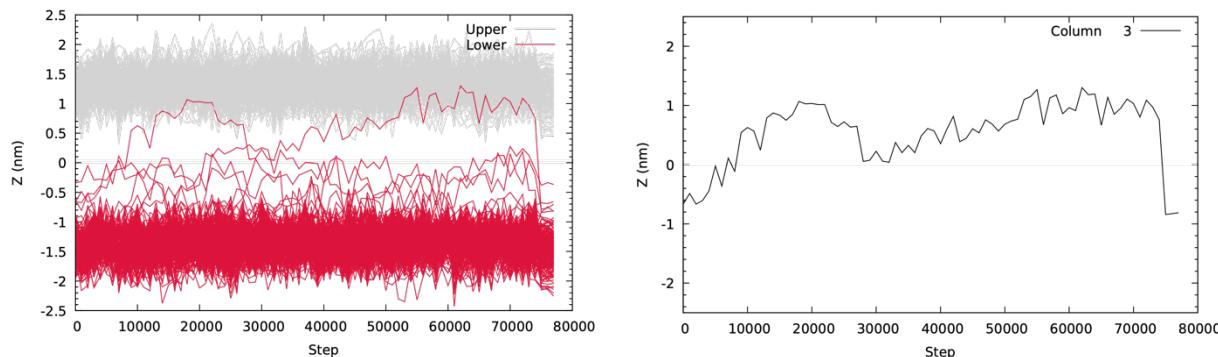


Figure 3-68 The z-coordinate of the geometric centers computed using the all-atom ester atoms for POPC molecules (left). While some of the lipids become increasingly tilted, no stable transitions are made between the leaflets for the simulation analyzed. We note that one lipid from the lower leaflet stands out and approaches the same height as the upper leaflet lipids. By searching for this pattern in the individual lipid plots (right), we were able to determine the column corresponding to this data, i.e., column 3. We can then lookup the res id corresponding to this column in the “_resid” output data file thus enabling further investigation of the lipid.

The user may obtain a plot like that shown in Figure 3-68 using the provided Gnuplot script “flip.gnu” which is located in the “scripts” folder. Try something like:

```
$ gnuplot -c flip.gnu [:] [:] upper_flip.dat lower_flip.dat flip.pdf 1357 1362
```

where 1357 and 1362 give the number of columns in the -flip data files for the upper and lower leaflets respectively. Additionally, the user may plot the z-coordinate for each lipid separately using the script “flip_singles.gnu” (also provided in the “scripts” folder); again, using something like the following:

```
$ gnuplot -c flip_singles.gnu [:] [:] upper_flip.dat upper_flip.pdf 1357
```

Together, these plots can be used to first screen the complete set of lipids for flips and then identify the individual lipids of interest, which can be investigated further. We note that the res id, corresponding to each column of -flip, may be found in a separate file. This information is written to a file named like -flip but with the “_resid” appendage.

As a last note, Lipid Flip may be used to check the lipid assignments to the upper and lower leaflets made by leaflet finder. Figure 3-69 shows an example where a single lipid was incorrectly assigned to the lower leaflet. Errors like this typically occur because some of the lipids were tilted in the reference structure analyzed by leaflet finder. In this case, the error may be resolved by using Mean Coods (section 3.12) to generate a reference structure. In this case, the time average lipid structures should contain little tilt (Figure 3-56) making them ideal for use with leaflet finder.

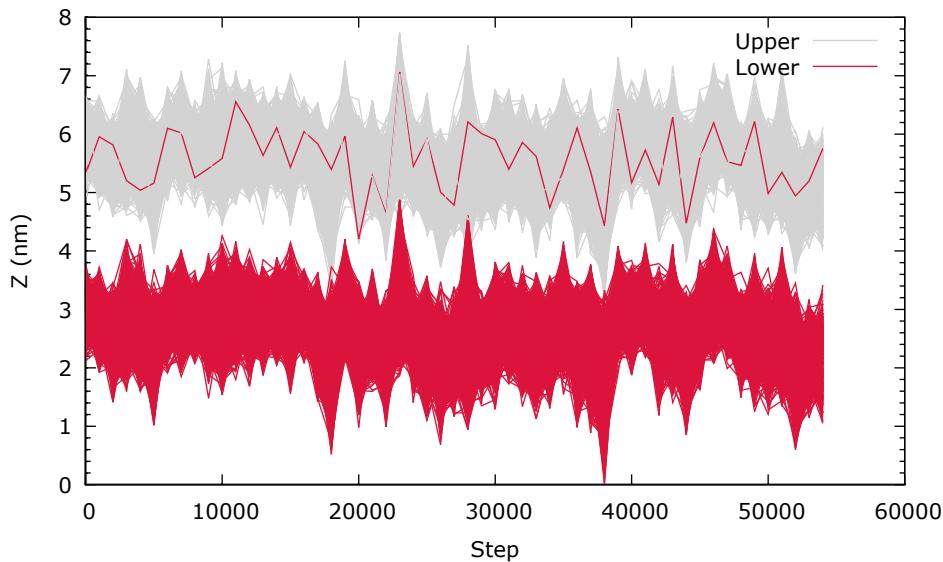


Figure 3-69 Using Lipid Flip to check leaflet assignments. In the example given here, a single lipid was incorrectly assigned to the lower leaflet.

3.16 3-Dimensional Analysis

The grid-based averaging theory employed by MOSAICS, i.e., the stamping method, may be extended into 3-dimensions and we have included a 3-D version of many of the MOSAICS tools discussed so far. These tools are named like the 2-dimensional counterparts but are given a 3d appendage. MOSAICS currently contains 3-dimensional versions of Lipid Density, Lipid Distances, P2, Lipid Orientation, Interleaflet Contacts, Lipid Contacts, Nearest Neighbors, APL, 2D Enrichment, and Mean Lipid Coords. These programs are used much like the 2-dimensional versions. However, the grid data generated is now 3 dimensional. This data is presented using the Data Explorer format (.dx) and may be viewed using a graphics visualizer like PyMOL or VMD. Figure 3-70 shows a few examples of data generated with the 3-d analysis currently supported by MOSAICS.

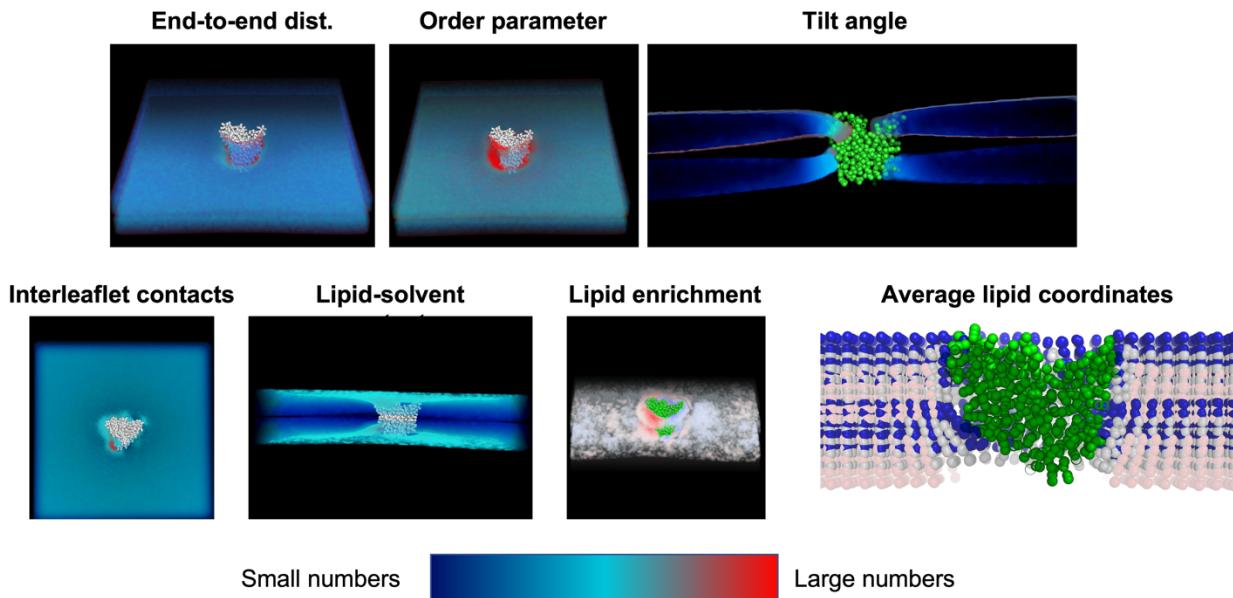


Figure 3-70 Examples of select 3-dimensional analysis performed with MOSAICS. Figures were generated using the volume utility of PyMOL.

We note that the three-dimensional analysis retains most functionality of the two-dimensional counter parts. For example, the sample count is generated for each program and in most cases the single frame characterizations, standard deviation, and standard error are obtainable (Figure 3-71).

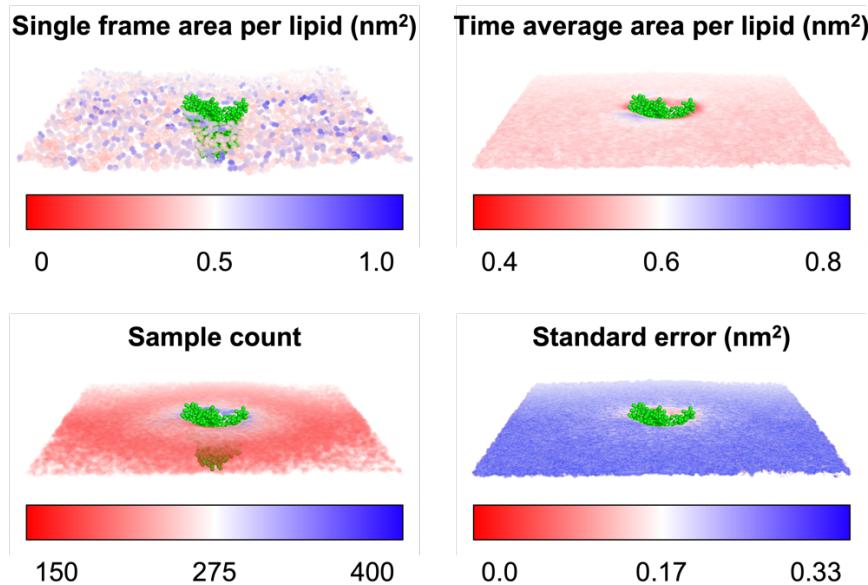


Figure 3-71 Single frame characterization of the area per lipid (upper left). The single frame data is then averaged over time to give the time average area per lipid (upper right). The sample count (bottom left). The standard error of the mean (bottom right). Figures were generated using the volume utility of PyMOL.

We note that the generation of single frame grid data can eat up large amounts of hard drive space when three-dimensional analysis is considered. This can easily reach the terabyte regime. The user should thus proceed with caution when using the -stdev function.

Before concluding, we discuss a few differences between the 3- and 2-dimensional analysis. To begin, the grid is now built from a collection of tiny cubes as opposed to squares. However, the user still specifies the grid resolution via the -APS tag, i.e., the user defines the area of the one of the faces of the cube and the cell spacing is again derived from this. When building the lattice, the user is again given the option of specifying the box size; this now includes the z-dimension which is set via the -dz tag. And finally, we note that the 3-dimsional analysis tend to require significant amounts of memory easily reaching into the gigabyte range. We have included print statements that give estimates of the required memory. Still, if the user experiences a crash, we recommend reducing the lattice resolution and trying again.

3.17 Lipid Density 3D

MOSAICS makes possible 3-dimensional density maps by stamping a value of 1 to the lattice around select atoms of some target lipids. This analysis is made possible using the MOSAICS tool Lipid Density 3d. To use this tool, the user must specify the target lipids as well as the atoms used for depositing density data (Figure 3-72). This information is provided using a network of selection cards such that the secondary files give the target atoms. The name of the primary file is provided using the -crd tag. We note that Lipid Density 3d has the option of excluding any density data that is beyond a threshold distance (nm) from the protein. This distance may be set using the -dist tag.

-crd	
#lipid_type	#filename
DLPE	dlpe.crd
DLPG	dlpq.crd
#target_atom	
GL0	
PO4	
GL1	
C1A	
C2A	
C3A	
GL2	
C1B	
C2B	
C3B	
#target_atom	
NH3	
PO4	
GL1	
C1A	
C2A	
C3A	
GL2	
C1B	
C2B	
C3B	

Figure 3-72 Selection card structure used by Lipid Density 3d. In the example given here, we add density data to the grid around the atoms of DLPE and DLPG lipids. The full lipids are used in this example.

An example of the run commands used with Lipid Density 3d is now given:

```
$ mpirun -n 50 lipid_density_3d_mpi -traj traj.xtc -ref ref.gro -crd po.crd -rho full_po.dx -APS 0.005 -r 0.26 -cutoff 0.4 -leaf 0 -ex_val -1.0 -dist 1.0
```

In the example provided here, the output data file containing the time lipid density data is specified via the -rho tag. An example of the data generated with Lipid Density 3d is shown in Figure 3-73.

Analysis of Lipid Structure

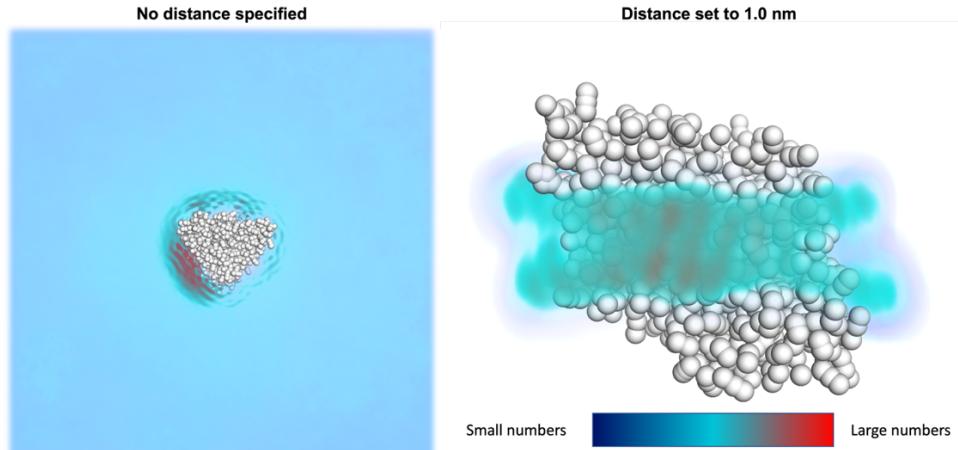


Figure 3-73 Example of data generated using Lipid Density 3d. Shown is the lipid density data for the DLPE/DLPG acyl chains. The data was visualized using the volume utility of PyMOL.

Chapter 4 Analysis of Lipid Dynamics

4.1 Lipid Mixing

The characterization of lipid mixing is an important prerequisite to analyzing a membrane simulation. In doing so, the user can determine whether the trajectory analyzed is representative of the equilibrium ensemble. This is particularly important when a complex mixture of lipids is simulated. For these systems, the initial spatial distribution of the lipids is artificial and must reorient in accordance with the energy landscape. For this to happen, the lipids must be able to mix within the time scale reached in the simulation. To determine the degree of mixing within a membrane simulation, the MOSAICS tool Lipid Mixing may be used. With Lipid Mixing, the main output is the percentage of lipids to have been in a target lipid's first shell. This number, which we call the mixing fraction, is averaged over the target lipids, and periodically reported.

To use Lipid Mixing, the user must specify the target and visiting lipid types using a pair of networked selection cards. For both groups, the lipids positions in XY are represented using a geometric center (equation 1.3). The atoms specifying these centers are provided using secondary selection cards. With the center known for each lipid, Lipid Mixing counts the number of visiting centers to have previously been within a threshold distance from the target lipid center. This distance is specified for the target lipids within the primary card. These selection cards are then provided using the `-crd_1` and `-crd_2` tags for the target and visiting leaflets respectively (Figure 4-1).

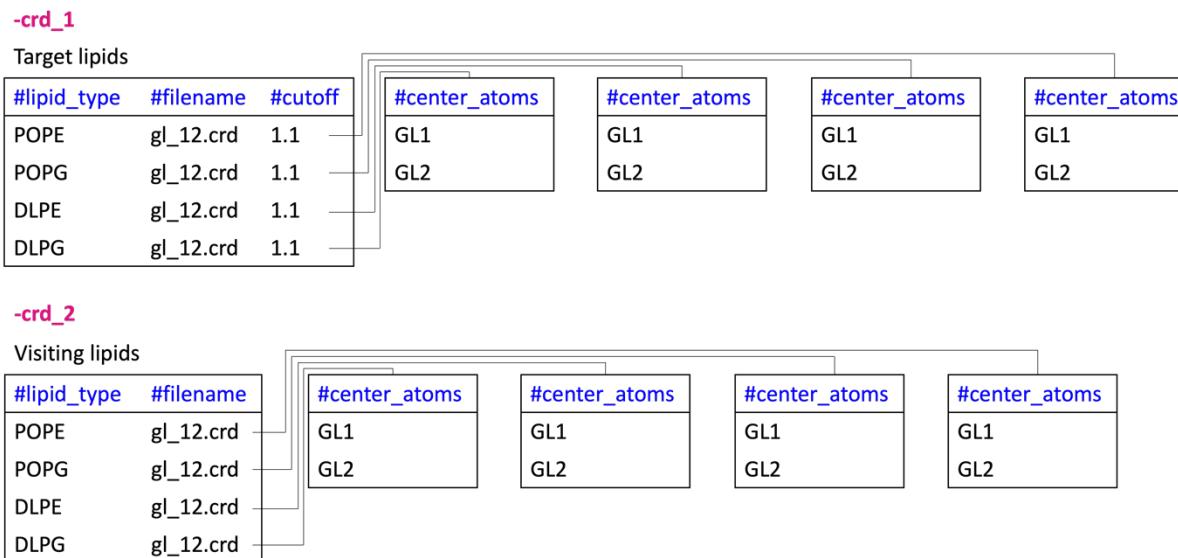


Figure 4-1 selection card structure used by Lipid Mixing. For the example shown here, we use the atoms GL1 and GL2 to compute the geometric center of each lipid. We then include the lipids POPE, POPG, DLPE, and DLPG for both the target and visiting lipids.

In the example provided above, Lipid Mixing selects POPE, POPG, DLPE, and DLPG for the target lipids. Once a target lipid is found, the program finds the midpoint between the ester atoms, i.e., the geometric center. Lipid Mixing then searches for nearby POPE, POPG, DLPE, and DLPG lipids. Like with the target lipids, the center between the ester atoms is used to represent the visiting lipid's position for this example. The distance between these centers is computed and, if the

distance is less than the cutoff value 1.1 nm (this value is specified in the primary selection card), the lipid will be counted as being in the target lipids first shell (Figure 4-2).

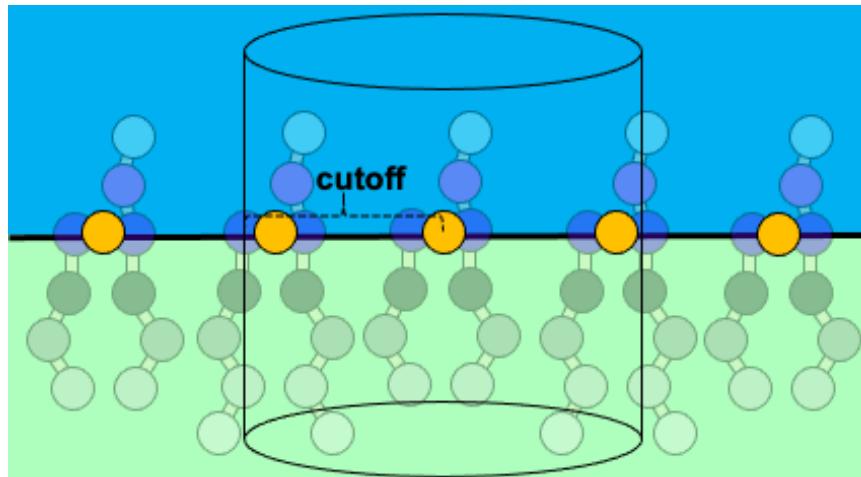


Figure 4-2 Schematic showing how lipids are counted as being in a target lipids first shell or not. The distance in XY between lipid centers (orange circles) is computed. If this distance is less than the cutoff value (shown as a cylinder) the lipid is in the first shell.

Lipid Mixing keeps a record detailing which lipids have previously been in the target lipid's first shell. With this information, the percentage of the visiting lipids to have previously occupied the first shell is computed, i.e., the mixing fraction. This percentage is then averaged over all the target lipids and the average is periodically reported with the standard deviation. The frequency in which data is written to the output file is set with the -mix_s tag. Lipid Mixing also records the average number of lipids, called the solvation number, to be in the target lipid's first shell at any given time. This information is averaged over the target lipids and is reported (with the standard deviation) with the mixing fraction. An example of the run commands used with Lipid Mixing is now given:

```
$ mpirun -n 50 lipid_mixing_mpi -traj traj.xtc -ref ref.gro -crd_1 param_1.crd -crd_2 param_2.crd  
-mix upper_mix.dat -leaf 1 -mix_s 100 -range 1 -freq 0.5 -dt 1000
```

In the example provided here, the output data file containing the mixing fraction and solvation number is specified via the -mix tag. An example of the main output data from Lipid Mixing is shown in Figure 4-3.

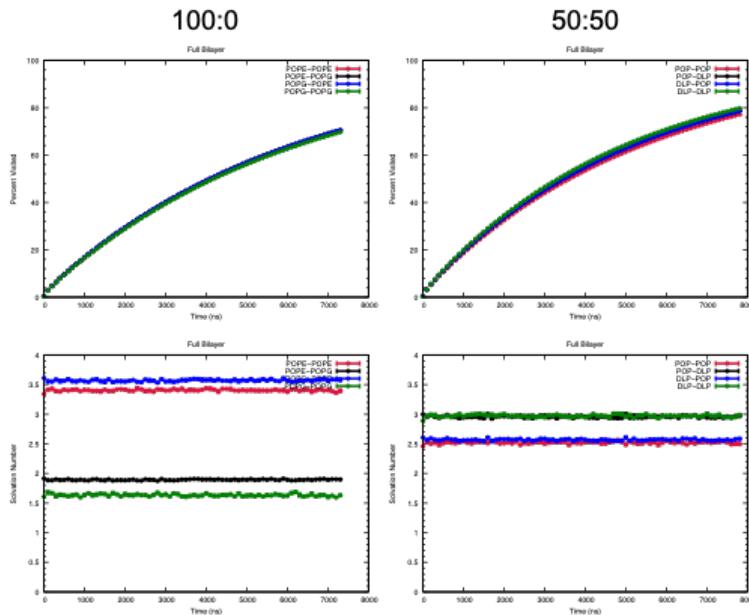


Figure 4-3 Lipid Mixing as a function of time (top panel). Number of lipids in the first shell (bottom panel). Data taken from coarse-grained simulations of the CLC-ec1 protein [10] in a pure POPE/G bilayer (left panel) or a 55:50 mixture of POPE/G and DLPE/G (right panel).

In addition to the main output, Lipid Mixing reports the average dwell time for lipids in the first shell and writes a dwell time distribution to an output file (Figure 4-4). This data is written to a file with the same name as specified by -mix but with the “_freq” appendage. It thus reasons that the time step (ps) between trajectory frames is required and may be provided with the -dt tag. Additionally, the solvation events may be recorded to a binding events file, one for each target leaflet, by including the -w_bind 1 tag. These files are named after the -mix filename but are given the “_i.be” appendage where i corresponds to the target lipid number. As we will see in the next chapter, these binding events files can be processed further to give other characterizations of the solvation shell kinetics. In addition to this, the average solvation number may be projected onto the XY plane. To use this option, the user must specify a pair of mapping atoms with the -m1 and -m2 tags, as well as the area of the lattice square (-APS), and the stamping radius (-r). Figure 4-5 shows an example of the spatially resolved time average solvation number.

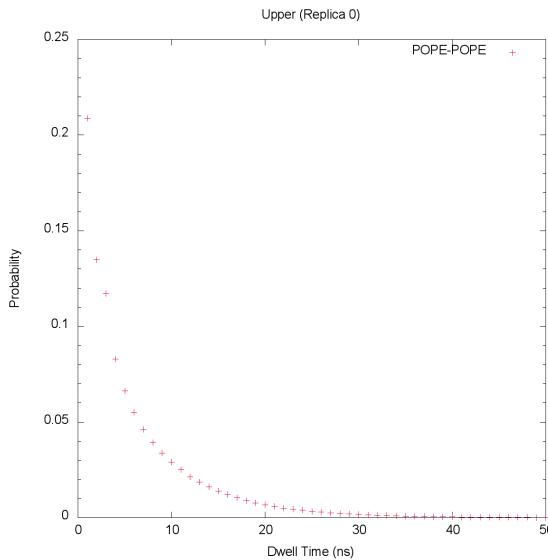


Figure 4-4 Dwell time frequency for first shell neighbors from a Martini coarse-grained simulation of the CLC-ec1 protein [10].

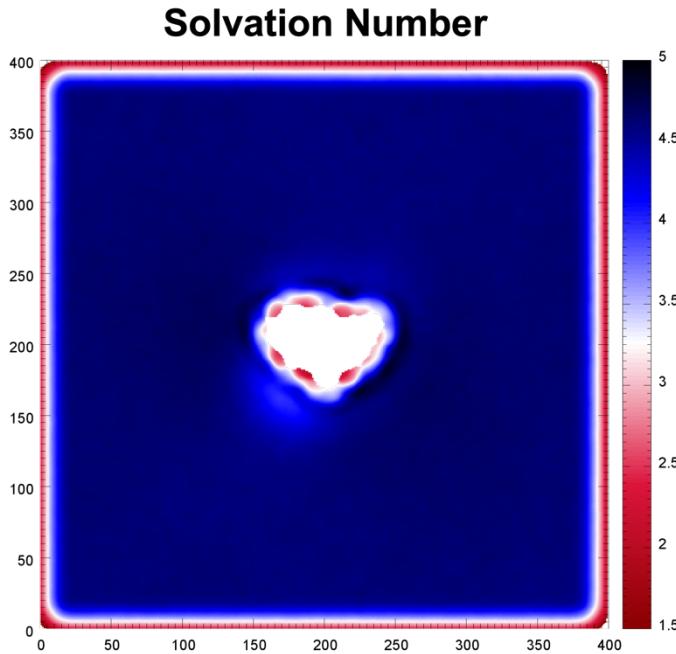


Figure 4-5 Average solvation number for POPE lipids in a Martini coarse-grained simulation of the CLC-ec1 protein [10]. Units for the x/y axis are grid points and the solvation number for the color bar.

We note that Lipid Mixing has been implemented to make use of a noise filter (section 1.16). This filter works by examining the occupational state of the visiting lipid that tells if the visiting lipid is within the cutoff distance of the target lipid. By filtering the occupational number, we can exclude visits whose residence time is less than a desired value. The size of the window, i.e., the half width, is set with the -range tag and the percentage of the $2N+1$ frames requiring a hit before counting a lipid as having visited (ω) is set with -freq.

And finally, we note that it can be useful to report the mixing fraction and solvation number averaged over the full bilayer as opposed to the individual leaflets. With this aim in mind,

the user should first perform the analysis on each leaflet separately. Then, the results (-mix) can be averaged using the MOSAICS tool Data Averager (section 5.2).

4.2 The Diffusion Coefficient

The diffusion coefficient may be computed for a given lipid type using the MOSAICS tool Lipid MSD. This is accomplished by measuring the mean squared displacement (MSD) of the lipids after some elapsed time τ where MSD is defined as:

$$MSD(\tau) = \frac{1}{N} \sum_{k=1}^N \frac{1}{T - \frac{\tau}{\Delta t} + 1} \sum_{t=0}^{T - \frac{\tau}{\Delta t}} \left(\vec{r}_{k,t+\frac{\tau}{\Delta t}} - \vec{r}_{k,t} \right)^2 \quad (4.1)$$

In equation 4.1, $T+1$ gives the number of trajectory frames, Δt is the time interval between frames, N the number of lipids, $\vec{r}_{k,t}$ is the position vector for lipid k in snapshot t , and $\vec{r}_{k,t+\frac{\tau}{\Delta t}}$ is the same position vector after some elapsed time τ ; we note that the position vectors contain only the x and components. With the MSD known, the diffusion coefficient may be determined from the Einstein relation:

$$MSD(\tau) = 2nD\tau \quad (4.2)$$

where n is the dimensionality (for lipids in a bilayer $n = 2$), and D is the diffusion coefficient. Using equation 4.2, D is computed by plotting the MSD against the elapsed time and using linear extrapolation to determine the slope. From this, D is finally found as:

$$slope = 2nD \quad (4.3)$$

To use the program the user must specify the lipid types to include in the computation and a list of atoms for that lipid. This information is provided using a network of selection cards specified with the -crd tag (Figure 4-6).

-crd		
#lipid_type	#filename	#center_atom
POPE	pope.crd	GL0
POPG	popg.crd	PO4
		GL1
		C1A
		D2A
		C3A
		C4A
		GL2
		C1B
		C2B
		C3B
		C4B

Figure 4-6 Selection card structure used by Lipid MSD. For the example here, we have computed the geometric center using the full lipid for types POPE and POPG.

In the example above, the mean squared displacement will be computed as a function of τ for POPE and POPG lipids. Furthermore, the lipid position vectors will be computed using the x and y components of the geometric center (equation 1.3) of the atoms making the full lipid. An example of the run commands required by Lipid MSD is now given:

```
$ mpirun -n 50 lipid_msd_mpi -traj traj_msd.xtc -ref ref.gro -crd param_po.crd -msd msd_po.dat  
-leaf 0 -dt 1000
```

In the example provided here, the output data file containing the MSD data is specified via the -msd tag. This file includes the MSD for each lipid as well as the MSD averaged over all lipids. An example of the output is given in Figure 4-7. We note that a plot like the one included in Figure 4-7 can be obtained using the script “msd.gnu” located in the “scripts” folder using something like the following:

```
$ gnuplot -c msd.gnu [:] [:] msd_po.dat msd_test.png 1266
```

where the first two entries, i.e., [:] [:], set the range of the x and y axis, respectively and 1266 was the number of columns found in msd_po.dat.

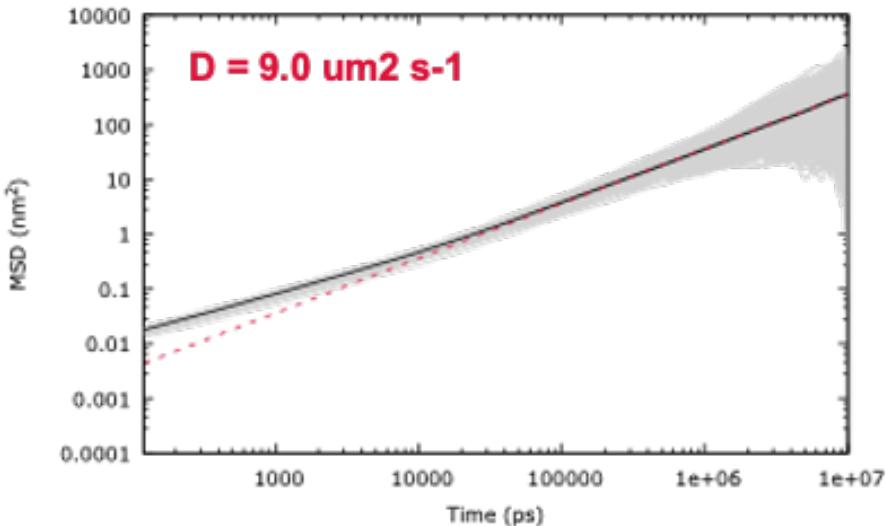


Figure 4-7 Mean Square Displacement (MSD) of POPC lipids as a function of time for a simulation of CLC-ec1 [10] in POPC bilayer. The black line gives the average MSD over all lipids while the gray background comes from the MSD for individual lipids. The slope of the line gives the diffusion coefficient which is shown in red. The data is typically linear beyond 100ns. For shorter timescales, the rattle of the lipids within their solvation shell creates nonlinear behavior. The line of best fit (red dotted line) was computed using only data after 100ns.

4.3 The Lipid Residence Time

The activity of membrane proteins may be regulated by the binding of substrate molecules. For example, G protein-coupled receptors (GPCR) are activated when small molecules bind with their extracellular domain. This leads to a rearrangement of protein structure which activates the target pathway. What's more, it is believed that lipid molecules play a similar role by binding to the transmembrane domain of some protein. This hypothesis may be tested by analyzing molecular dynamics simulations such that any binding sites are revealed. With this approach, binding sites are determined by the lipid residence time which is greater for these sites compared to other parts of the protein. To facilitate these calculations, we have the MOSAICS tool 2d Kinetics. With 2d Kinetics, the lipid dynamics are characterized, and the mean dwell times are projected onto the XY plane. We note that this approach assumes the position of the protein has been fixed before performing the analysis (section 2.1). With this assumption, the time spent bound to the protein is identical to the residence time at a nearby lattice point. For this reason, 2d Kinetics focuses on the dwell time at the grid points rather than with the protein.

Given this approach, a mechanism for identifying bound lipids must be developed where we use the descriptor “bound” to mean the lipid is located at the lattice point for some time. A simple approach to identifying bound lipids is to construct a discretized Voronoi Tessellation. That is, we find, for each lattice point i,j , the lipid whose representative distance δ_k^{ij} is the smallest. For this, δ_k^{ij} is found as the minimum distance between i,j and a set of atoms belonging to lipid k . The discretized Voronoi diagram thus defines a bound lipid number b_k^{ij} for each lattice point. This binding number may be refined by passing b_k^{ij} through a noise filter (section 1.16). That is, b_k^{ij} is examined over a series of $2N+1$ trajectory frames where the significance threshold (ω) is fixed at 0.5. Given this filtering, the definition of a bound lipid becomes one that is closest to the grid point for at least $N+1$ trajectory frames where N is set with the -range tag. It then

follows that the binding events are excluded whose dwell time is smaller than $N + 1$ trajectory frames. Should no lipid reach the 50% threshold, then the lattice point will be absent of a bound lipid. This occurs when 3 or more lipids are in completion for a lattice point. Note, these points define the boundaries between the lipids (Figure 4-8). Once the bound lipids have been filtered, the dwell times may be computed as each lipid transitions to a new location and the binding states are updated.

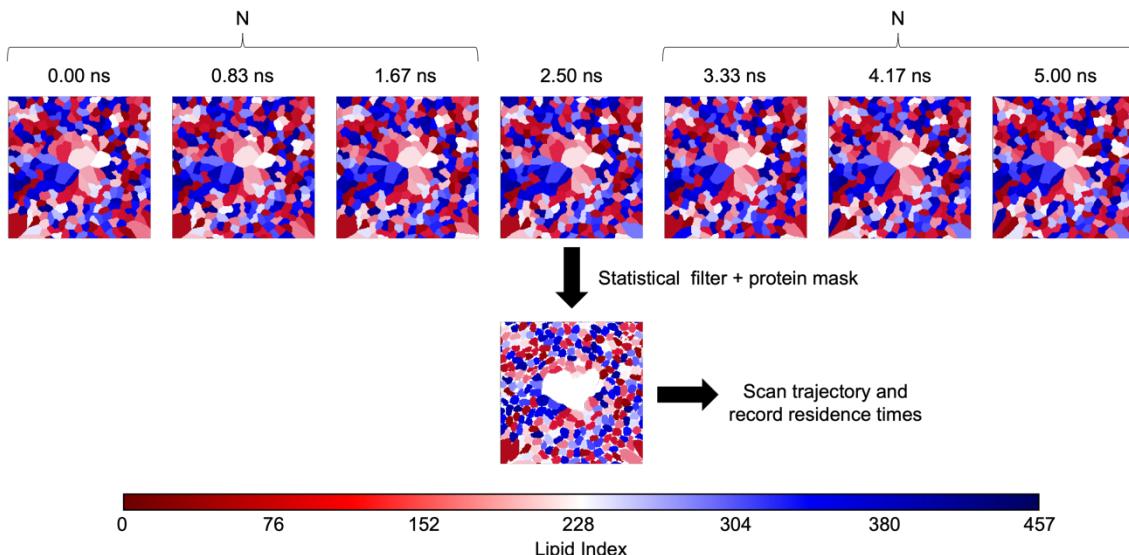


Figure 4-8 Voronoi tessellation (top). Assignment after 50% filter is applied (middle) and assignment after the protein mask is added (lower right). Units for the x/y axis are grid points. Color indicates the lipid index number.

To use 2d Kinetics, the user must specify the lipid and atom types to include in computations of Voronoi diagrams. This information is provided using a network of selection cards and the -crd tag as is shown in Figure 4-9.

-crd			
Voronoi lipids			
#lipid_type	#map_1	#map_2	#filename
POPE	GL1	GL2	gl_12.crd
POPG	GL1	GL2	gl_12.crd
DLPE	GL1	GL2	gl_12.crd
DPLG	GL1	GL2	gl_12.crd

#voro_atoms
GL1
GL2

#voro_atoms
GL1
GL2

#voro_atoms
GL1
GL2

#voro_atoms
GL1
GL2

Figure 4-9 Selection card structure used by 2d Kinetics. In the example here, the residence time is monitored for POPE, POPG, DLPE, and DPLG lipids. These are specified using -crd and encompass the lipids used to construct the Voronoi diagrams. This set should include all lipids in the system. Here we use the minimum distance between atoms GL1 and GL2 to construct the Voronoi diagram. The pair of mapping atoms are used to deposit lipid density data.

Other parameters required to use 2d Kinetics include those specific to the computation of Voronoi diagrams. For example, the protein atoms may or may not be included in this computation. This option is set using the -v_prot tag. If the protein is included, then the minimum distance (nm) between the protein and lipid atoms should be specified with the -c_dist tag. This distance is used to determine which protein atoms should be included in the Voronoi tessellation. Once computed, the Voronoi diagrams are written to output files and are later processed with a

noise filter and the residence times extracted for each lattice point. The Voronoi files may then be maintained or deleted as controlled using the -clean tag. And finally, the effective time step (ps) between trajectory frames must also be specified. This information is provided with the -dt tag. Note, the effective time step is the time between the trajectory frames being analyzed. This depends on the time step between trajectory frames (Δt) but also on the -stride used in the analysis. For example, if a stride of 1 is used, then -dt is the time step between frames. However, if a stride of n is used then -dt should be given as $n\Delta t$. This information is needed to convert between frames and time in the resulting binding events files. These files are discussed next, but we first give an example of the run commands used with 2d Kinetics.

```
$ mpirun -n 60 2d_kinetics_mpi -traj traj.xtc -ref ref.gro -crd param.crd -k upper.dat -APS 0.005 -r 0.23 -leaf 1 -dt 1000 -range 1 -b 0 -e 7826 -v_prot 0 -clean 1
```

In the example provided here, the filename specified with the -k tag is used to generate additional filenames for the various data generated. For example, the output data files containing the Voronoi tessellations for each trajectory snapshot are written to files whose name come from -k but are given the “_t” appendage where t specifies the trajectory frame. In addition to Voronoi diagrams, 2d Kinetics also generates a “binding events” (.be) file for each lattice point. These files are named via -k but are given the “_i_j.be” appendage, where i and j specify the lattice index for the x and y directions. Binding events files contain detailed information about the binding events such as the lipid number, the residue id, the residue name, the duration of the event, and the beginning and ending frames. In this case, the lipid number is an index number given to the lipids that starts at zero and increases by one with each lipid analyzed. This approach numbers the lipids in sequence which is not guaranteed by the residue id. A reference chart is thus written to file, whose name is given the “_lipid_info” appendage, that contains the residue name and id for each lipid number. The user is warned that the writing of binding events files produces many files, the number of which depends on the resolution of the grid. Caution should be taken to ensure that this number does not exceed limits specific to the computing platform. If needed, the grid resolution may be lowered using the -APS tag to reduce the number of files generated. In addition to the binding events files, 2d Kinetics also generates a sample count file, i.e., ρ^{ij} , that can be used to remove insignificant data points from residence time maps. This file is given the same name as specified with -k but with the “_rho” appendage. To generate the sample count data, the user must specify two mapping atoms using the selection card (-crd) discussed previously. These atoms are used to represent the position of the lipids in the XY plane when stamping data to the grid.

Once a set of binding events file has been generated with 2d Kinetics, the mean residence time may be acquired for the individual lipid types using the MOSAICS tool Binding Events Analyzer. This is demonstrated in the following example:

```
$ mpirun -n 100 binding_events_analyzer_mpi -d upper_popc_dlpc -o upper_popc -crd popc.crd -cutoff 0
```

In this example, the base name (section 1.14) of the binding events files is specified with the -d tag. Similarly, the lipid types of interest are given with the -crd tag as shown in the following example.

```
-crd
#lipid_type
POPC
```

With the -cutoff tag, binding events are excluded if their residence time is smaller than the time (ps) specified in the argument. Standard output from Binding Events Analyzer includes the spatially resolved maps for the mean dwell times, the standard deviation of the dwell times contributing to each average, the largest dwell time for each lattice point, the number of binding events, the off-rate constant (assuming first-order kinetics), and the correlation coefficient obtained when computing k_{off} (linear regression is used for this). These data are written to output files that are named with the -o tag; this gives the base file name and should contain no extension. The resulting files are given the “_dwell_time.dat”, “_stdev.dat”, “_largest_dwell_time.dat”, “_num_events.dat”, “_koff.dat”, and “_r2.dat” appendage respectively. We note that k_{off} is computed using the relation:

$$\ln\left(\sum_{i=t}^T P_i\right) = -k_{off}t \quad (4.4)$$

where t is the trajectory frame (time = tdt), $T+1$ is the total number of frames, and P_i is the probability of having a dwell time of i frames. This relation is derived by recognizing that:

$$A(t) = A_0 \exp(-k_{off}t) = A_0 \sum_{i=t}^T P_i \quad (4.5)$$

Using equation 4.4, a plot of $\ln(\sum_{i=t}^T P_i)$ vs t will yield a straight line whose slope gives $-k_{off}$. Figure 4-10 shows an example of the data generated with Binding Events Analyzer.

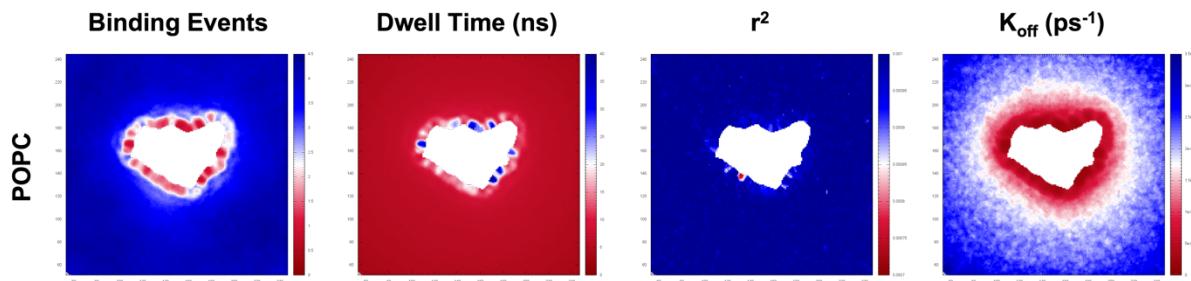


Figure 4-10 Standard output from 2d Kinetics. Data generated from Anton trajectories of the CLC-ec1 protein [10]. Units for the x/y axis are grid points. For the color bars the units are given in the overhead text.

In addition to Binding Events Analyzer, MOSAICS contains additional tools used for characterizing the binding events files produced by 2d Kinetics. To begin with, there is a tool named Binding List which can be used to display the contents of a binding events file after processing and sorting the data. For example, the user may wish to sort the binding events by the initial binding frame (-mode 0) or the largest dwell time (-mode 1). If -mode 2 is selected, then repeat

lipid binding is characterized and the data is organized so that lipids with the largest number of repeat visits are shown first. With this option, the average dwell time is also computed as well as the average time between visits (Figure 4-11). Note, we refer to the time between visits as the “off-time”. To use Binding List, the user must specify the binding events file to analyze and an option for organizing the data. This is done with the -d and -mode tags respectively as is shown in the following example.

```
$ mpirun -n 1 binding_list_mpi -d upper_popc_0_0_binding_events.dat -mode 2 -t 0
```

Note, the user may switch between units of time using the -t argument. Possible units include picoseconds (-t 0) and trajectory frames (-t 1).

Mode 0							Mode 1							Mode 2						
rank	lipid_nr	res_nr	res_name	bind_i	bind_f	dwell_time	rank	lipid_nr	res_nr	res_name	bind_i	bind_f	dwell_time	rank	res_nr	res_name	visits	dwell_time	off_time	
0	87	789	POPC	12.7	35.8	23.2	0	145	763	POPC	13775.2	49.4	5.9	0	729	POPC	31	5.9	438.2	
1	87	789	POPC	37.9	41.8	1.9	1	48	693	POPC	13775.2	37.1	2.1	1	641	POPC	29	4.9	454.5	
2	87	789	POPC	48.6	41.2	7.4	2	118	732	POPC	14413.2	14446.6	35.6	2	673	POPC	29	4.9	448.8	
3	87	789	POPC	48.1	58.9	2.9	3	138	752	POPC	11727.4	11761.8	33.7	3	819	POPC	28	6.0	518.6	
4	12	634	POPC	51.4	52.2	2.2	4	65	782	POPC	13812.4	13843.6	32.9	4	830	POPC	28	5.0	416.8	
5	12	634	POPC	54.2	61.8	7.7	5	118	748	POPC	17641.1	17672.4	31.4	5	797	POPC	27	4.7	434.3	
6	12	634	POPC	72.8	82.2	8.3	6	1	623	POPC	12854.2	12843.6	29.5	6	789	POPC	26	4.9	719.1	
7	67	679	POPC	88.9	82.4	1.8	7	129	763	POPC	14922.8	14922.8	28.8	7	771	POPC	26	5.0	523.1	
8	12	634	POPC	82.7	91.1	11.5	8	31	663	POPC	14992.2	14992.2	28.8	8	623	POPC	25	4.7	427.4	
9	12	634	POPC	96.6	99.6	3.1	9	194	818	POPC	6491.6	6518.5	27.0	9	693	POPC	25	6.4	638.2	
10	99	681	POPC	108.4	161.5	4.2	10	134	758	POPC	4995.1	5425.0	26.6	10	663	POPC	24	5.1	852.3	
11	152	774	POPC	109.1	118.0	1.9	11	71	493	POPC	13812.4	13843.6	32.9	11	738	POPC	24	5.0	545.2	
12	12	656	POPC	111.5	112.8	1.4	12	125	747	POPC	18866.6	18912.4	25.9	12	786	POPC	24	4.8	486.2	
13	152	774	POPC	112.9	124.6	11.8	13	175	797	POPC	12395.0	12420.6	25.7	13	679	POPC	22	5.6	755.8	
14	12	656	POPC	125.3	134.6	9.2	14	176	798	POPC	13812.4	13843.6	32.9	14	624	POPC	21	4.7	474.6	
15	12	634	POPC	145.1	158.2	13.2	15	148	762	POPC	11638.1	11643.5	26.6	15	761	POPC	21	3.8	731.6	
16	152	774	POPC	159.2	159.6	0.5	16	181	803	POPC	7719.6	7745.0	25.4	16	724	POPC	21	5.7	656.5	
17	12	656	POPC	168.1	169.1	0.1	17	56	693	POPC	13812.4	13843.6	32.9	17	886	POPC	20	4.5	458.8	
18	152	774	POPC	168.7	166.1	0.5	19	188	648	POPC	14778.9	14883.6	25.2	18	747	POPC	20	7.7	935.5	
19	77	699	POPC	182.4	187.7	5.4	20	71	669	POPC	18448.6	18465.7	25.2	19	652	POPC	20	7.1	886.6	
20	77	699	POPC	199.4	200.8	0.2	21	185	887	POPC	1635.6	1659.8	24.6	20	773	POPC	20	3.1	312.2	
21	77	699	POPC	208.3	208.4	0.2	22	136	758	POPC	4995.1	5425.0	26.6	22	783	POPC	19	8.5	1885.2	
22	77	699	POPC	208.8	205.8	5.2	23	128	758	POPC	18910.8	18924.4	23.6	23	735	POPC	19	5.6	1839.4	
24	77	699	POPC	215.7	216.0	0.7	25	58	493	POPC	13843.6	13843.6	26.6	24	643	POPC	19	7.0	648.8	
25	152	774	POPC	229.1	230.0	7.0	26	69	691	POPC	13615.1	13635.0	23.4	26	756	POPC	18	4.3	588.7	
26	180	722	POPC	233.5	239.0	5.6	27	67	799	POPC	12.7	35.8	23.2	27	777	POPC	18	7.6	841.2	
27	77	699	POPC	239.1	240.4	4.1	28	166	788	POPC	9282.8	9285.8	23.0	28	759	POPC	18	5.1	588.1	
28	115	737	POPC	244.8	255.0	10.3	29	198	812	POPC	14879.0	14992.0	23.0	29	653	POPC	18	8.0	484.8	
29	115	737	POPC	259.3	271.3	12.1	30	210	852	POPC	12192.7	12212.0	22.6	30	768	POPC	17	4.4	464.5	
30	115	737	POPC	272.8	273.2	2.9	31	66	668	POPC	3191.9	3220.3	22.3	31	675	POPC	17	4.4	1852.9	
31	115	737	POPC	276.1	280.0	4.0	32	158	772	POPC	1782.2	1984.0	22.0	32	637	POPC	17	4.7	496.4	
32	48	670	POPC	307.8	321.7	14.0	33	187	758	POPC	1292.9	1302.9	22.0	33	696	POPC	17	4.7	504.4	
34	100	722	POPC	333.2	334.4	0.2	35	151	773	POPC	13821.1	13843.6	21.8	34	818	POPC	16	5.7	645.4	
35	177	799	POPC	344.2	348.8	2.8	36	135	773	POPC	3282.0	3285.0	21.7	36	725	POPC	16	6.4	689.9	
36	100	722	POPC	356.5	364.5	9.8	37	118	798	POPC	14384.8	14395.8	21.1	37	714	POPC	16	5.4	911.4	
37	100	722	POPC	358.5	358.7	2.9	38	223	845	POPC	1313.9	1323.8	21.0	38	887	POPC	15	5.5	1277.2	
38	48	670	POPC	381.8	387.4	5.6	39	176	799	POPC	18035.5	18077.4	21.8	39	972	POPC	15	4.5	974.7	
39	177	799	POPC	388.4	399.2	9.8	40	29	451	POPC	3862.1	3862.1	20.3	40	829	POPC	15	4.5	944.5	
40	177	799	POPC	402.4	409.0	5.5	41	181	883	POPC	11289.8	11299.8	29.2	41	814	POPC	15	4.1	443.8	
41	47	669	POPC	407.2	408.7	1.7	42	94	716	POPC	11952.2	11972.0	19.9	42	648	POPC	15	8.5	742.5	
42	47	669	POPC	408.8	408.8	0.1	43	188	809	POPC	11952.2	11972.0	19.9	43	659	POPC	15	3.6	304.6	
43	47	669	POPC	409.1	409.1	0.1	44	118	748	POPC	1421.6	1442.1	19.7	44	779	POPC	15	4.8	448.1	
45	177	799	POPC	423.1	428.8	5.8	46	22	644	POPC	5366.3	5366.3	19.6	45	743	POPC	15	4.0	538.1	
46	100	722	POPC	444.8	448.8	0.8	47	45	643	POPC	14220.4	14224.0	19.6	47	651	POPC	14	7.1	641.7	
47	77	699	POPC	454.3	459.2	9.0	48	45	641	POPC	9864.5	9864.5	19.4	48	799	POPC	13	3.6	1399.6	
48	77	699	POPC	467.2	469.7	2.6	49	195	728	POPC	10036.2	10036.2	20.3	49	769	POPC	13	6.2	1248.9	
49	77	699	POPC	479.4	479.4	7.9	50	158	782	POPC	3524.6	3535.7	19.2	50	748	POPC	13	6.0	555.7	
50	194	818	POPC	478.2	486.6	7.5	51	59	151	747	POPC	4795.8	4814.8	19.2	51	815	POPC	13	6.6	915.4
51	88	702	POPC	481.0	486.6	9.7	52	79	699	POPC	12724.7	12745.7	19.2	52	886	POPC	13	4.4	1311.5	
52	88	702	POPC	484.2	499.9	3.7	53	167	789	POPC	15857.3	15857.3	19.2	53	688	POPC	13	3.2	449.9	
53	78	798	POPC	508.3	508.3	7.4	54	19	641	POPC	18661.2	18680.8	19.2	54	731	POPC	13	4.4	742.3	
54	1	623	POPC	508.3	514.9	6.7	55	28	669	POPC	18661.2	18680.8	19.2	55	644	POPC	13	8.5	815.6	
55	78	798	POPC	517.9	517.9	1.1	56	178	809	POPC	5123.6	5142.6	19.1	56	744	POPC	13	3.2	449.9	
56	78	798	POPC	523.0	523.0	0.2	57	28	658	POPC	8536.1	8536.1	19.0	57	822	POPC	13	6.3	999.2	
57	3	625	POPC	539.6	534.8	4.3	58	125	747	POPC	15853.4	15853.4	19.0	58	763	POPC	12	8.6	1187.1	

Figure 4-11 Binding List output for -mode 0 (left), -mode 1 (middle) and -mode 2 (right).

Because there can be a single bound lipid at each grid point, it is possible to plot the lipid assignments, i.e., the noise filtered Voronoi tessellations, using a heat map plotting tool such as Gnuplot (Figure 4-8). This is useful when performing the analysis for the first time since it enables quick and easy visualization of the resulting lipid dynamics. This data may be acquired post analysis using the resulting binding events files and the MOSIACIS tool Binding Events Video. To use Binding Events Video, the user must specify the base filename (section 1.14) of the binding events files with the -d tag. The user should also specify how often to write out the data using the -stride tag. Note, it is not always necessary to write out every frame but depends on the timestep between trajectory frames. It should also be noted that Binding Events Video has the option of reading an additional binding events file (set with -be tag) which can be used to highlight specific lipids within the video. The value of this feature will become clear in section 4.4 when the lipid dynamics are considered for the solvation shells rather than at the grid points. We now give an example of the run commands used by Binding Events video:

```
$ mpirun -n 100 binding_events_video_mpi -d upper -o upper -stride 10 -rho upper_rho.dat -cutoff 0.4
```

In the example provided here, the base filename of the binding events files (-d) was given as “upper” since the binding events files were named something like upper_0_0.be. Similarly, the output data files containing the noise filtered Voronoi tessellations are specified via the -o tag and are given the base filename “upper”. With this input, the Voronoi diagrams will be named “upper_t.dat” where t gives the trajectory frame number. Once the Binding Events Video analysis has been completed, the resulting Voronoi tessellation data can be plotted using Gnuplot and the scripts “get_vid_plots.sh” and “heatmap_template_video.gnu” found in the “scripts” folder. To make the plots, the user can use something like the following:

```
$ sh get_vid_plots.sh upper_data/upper 0 7820 upper_plots/ 10 1000
```

where the arguments from left to right are the base file name of the Voronoi diagram data, the first frame to plot, the last frame to plot, the directory where the resulting plots are to be stored, the stride used when running Binding Events Video, and the effective time step between the trajectory frames (ps). We note that the user will want to adapt some aspects of these scripts to fit the lipid count in their trajectory, for example the “set palette defined” option in “heatmap_template_video.gnu”. Once the user has the resulting plots (png), a video can be made, for example, by loading them into QuickTime using the “open image sequence” option.

Like Binding List, the MOSAICS tool Binding Contributors may be used to compute the average number of repeat visits as well as the off-time (ps). However, Binding Contributors does this for each grid point such that the data is projected onto the XY plane. To use Binding Contributors, the user must specify the base name of the binding event files (section 1.14) using the -d tag as is shown in the following example:

```
$ mpirun -n 10 binding_contributors_mpi -d upper
```

With the base filename specified (upper_i_j.be -> upper), a pair of output data files will be generated containing the spatially resolved repeat visit count and the off time. These data are written to files with the same name as specified via -d but with the “_num_visits.dat” and “off_time.dat” appendages. Figure 4-12 shows an example of the data generated with Binding Occupancy.

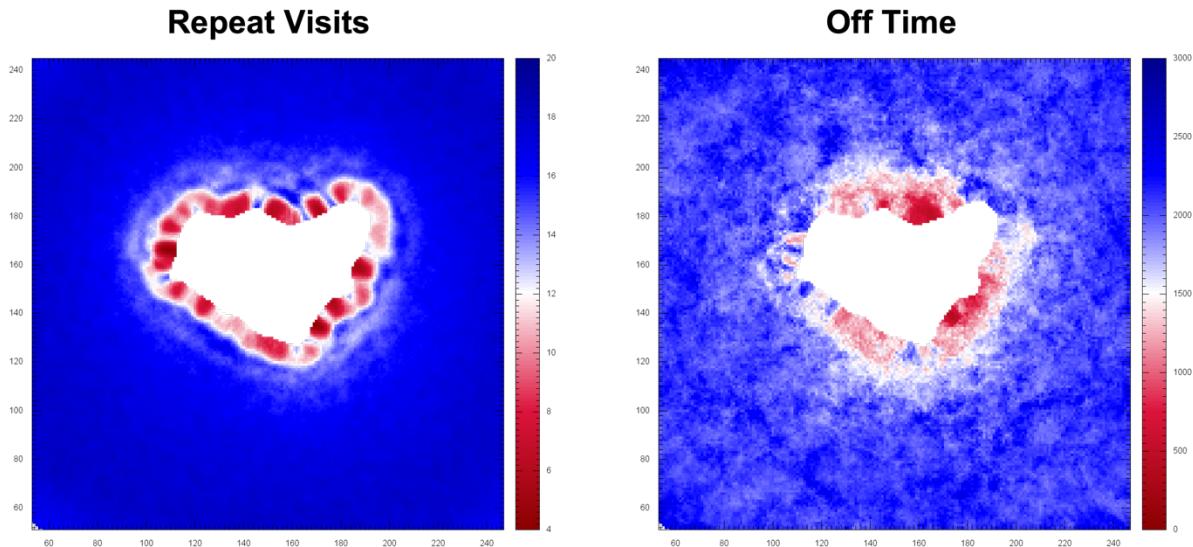


Figure 4-12 Average number of repeat visits by the lipids (left) and the average time between visits, also called the off time, (right panel). Units for the x/y axis are grid points. For the color bars, the units are the number of visits (left panel) and ns (right panel).

When using 2d Kinetics for the first time it is important to check that the input parameters used are reasonable. In particular, the half-width of the noise filter (-range) should be chosen with care. Recall that the filter removes binding events with a duration of less than $N+1$ frames. It is therefore possible to remove the lipids entirely if the filter becomes too wide. One way to test for this problem is to calculate the percentage of frames for which a lipid is assigned to the grid points, also called the occupancy number. In this case, a low percentage means the filter is too wide and the lipids have been shrunk or removed altogether. To perform this test, the user may use the MOSAICS tool Binding Occupancy. Like with Binding Contributors, the user may run Binding Occupancy by specifying the base name of the binding events files (section 1.14). This is demonstrated in the following example.

```
$ mpirun -n 10 binding_occupancy_mpi -d upper
```

Output from Binding occupancy includes a data file with the spatially resolved occupancy number. This file is named after the base filename specified with -d and is given the “_occupancy.dat” appendage. Figure 4-13 shows an example of the data generated with Binding Occupancy.

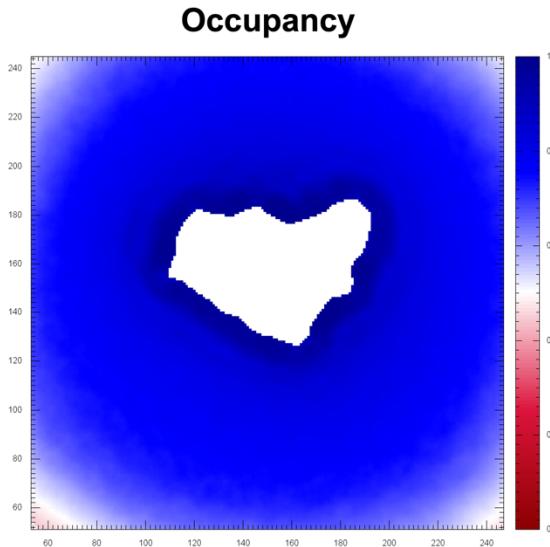


Figure 4-13 Percentage of frames, ranging from 0 to 1, in which the grid point was occupied by a lipid. Units for the x/y axis are grid points.

In addition to this, the contents of a binding events file may be characterized with a 2-dimensional heatmap using the MOSAICS tool Binding Timeline. With this program, the binding state of each lipid is indicated as a function of time (Figure 4-14). For this, the lipid number is given on the y-axis and the trajectory frame on x. To use Binding Timeline, the user must provide the name of the binding events file to analyze. This is done with the -d tag as is shown in the following example.

```
$ mpirun -n 1 binding_time_line_mpi -d upper_50_50.be -o upper_50_50_tl.dat
```

In the example provided here, the output data file containing the timeline data is specified via the -o tag. An example of the data generated with Binding Timeline can be seen in Figure 4-14. We note that the binding timeline can be examined to estimate the prevalence of fragmented event, i.e., those where multiple binding events are separated by very small off times. However, the resolution of the resulting plot should be set very high to capture fragmentation events whose off time approaches a single trajectory frame.

Binding Timeline

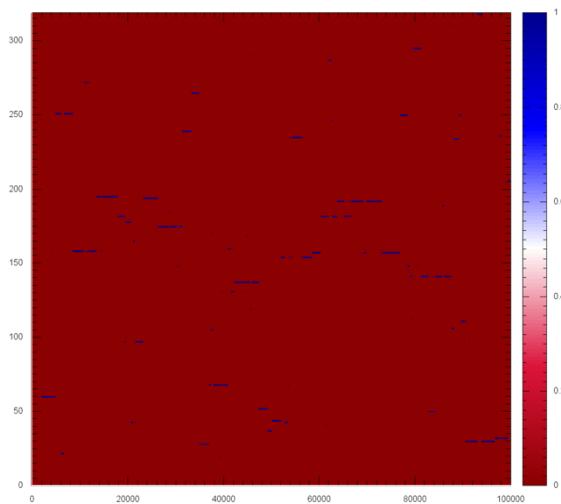


Figure 4-14 Binding Timeline for a single grid point. Y-axis gives the lipid number and x-axis the simulation frame. The color bar gives the binding state 0:unbound and 1:bound.

And finally, the percentage of lipids to have visited each lattice point may be determined with 2d Kinetics Percent Visited. To use this program, the user must specify the base name of the binding events files using the -d tag as is shown in the following example:

```
$ mpirun -n 50 2d_kinetics_percent_visited_mpi -d upper -o upper_percent_visited.dat -freq 1000  
-crd popc.crd -lipids 320
```

In the example provided here, the output data file containing the percent visited is specified using the -o tag. This filename is used to derive additional filenames giving each the “_t” where t is the frame number. In addition to this, the target lipids are indicated using the -crd tag as shown in the following example:

```
-crd  
#lipid_type  
POPC
```

Additionally, the number of target lipids in the system (or leaflet being characterized) is indicated with the -lipids tag. This is needed so that a proper percentage can be computed. This percentage is then reported periodically where the frequency is set using the -freq argument. An example of data generated with 2d Kinetics Percent Visited is shown in Figure 4-15.

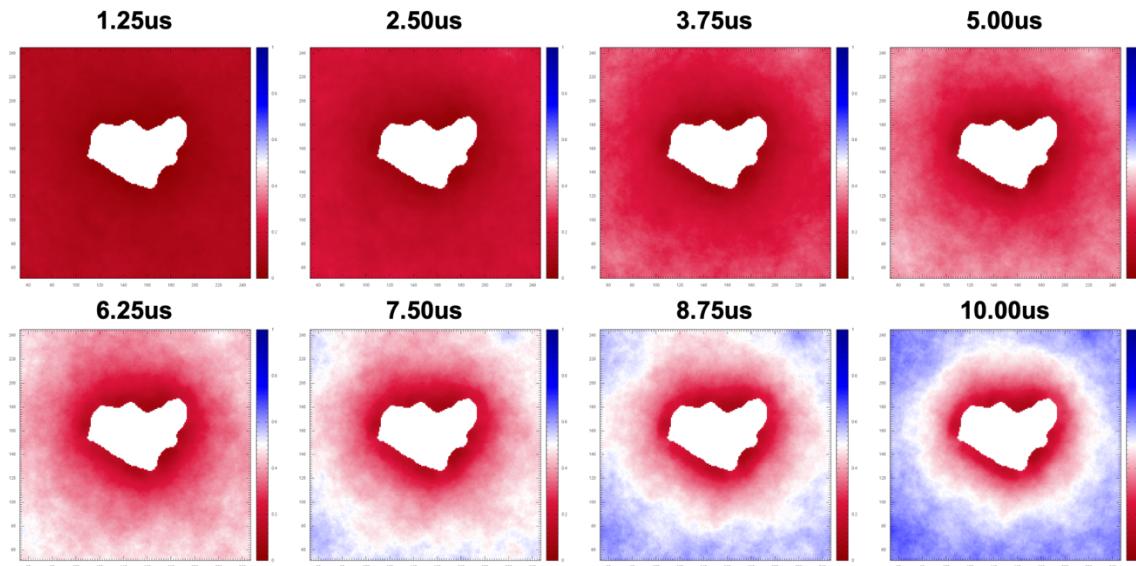


Figure 4-15 The percentage of lipids to have visited each grid point as a function of simulation time. Units for the x/y axis are grid points.

4.4 Solvation Shell Dynamics

As a complement to 2d Kinetics (section 4.3), it is possible to characterize the lipid dynamics over a region of space larger than a single lattice point. For example, the user might define a collection of lattice points and analyze the lipid residence time within this area. This could be a blob located in the bulk or an annulus enclosing the protein. If the user is interested in the dynamics within the solvation shells, then the lipids could be sorted, and the dwell times measured. In this section, we introduce several MOSAICS tools designed for these sorts of calculations. It is important to note that these computations use binding events (.be) files which should be generated with 2d Kinetics before performing the analysis.

To begin, let's consider an example in which the lipid dynamics are analyzed for the first and second shell lipids. Continuing in this direction, the user may focus on a particular protein interface, for example, the dimer or nondimerization interface of CLC [10]. With MOSAICS tools, there are several ways in which this analysis could be handled. However, we will examine the trivial solution first so that the user does not make this mistake. In this case, the user could use Grid Distance Projection (section 1.13) to average the dwell times (generated with 2d Kinetics) around the protein interface of interest. This solution is generally not of interest since the results will be characteristic of the dwell times at a single grid point but averaged over a larger region. More interesting solutions include grouping the lipids based on their host solvation shell or measuring the dwell times within a thin ring surrounding the protein. For the first approach, i.e., solvation shells, the user can use the program Solvation Shells.

With Solvation Shells, the lipids are classified as either the first, second, third, fourth, or fifth shell. Note, there is a group “other” for lipids residing outside the fifth shell. In addition to this, the lipids are sorted into a region of interest using a rectangular selection tool (section 1.15) or an additional mask (section 1.15). This allows the user to characterize dwell times at one interface vs another. Solvation Shells then keeps track of how long (how many frames) a lipid resides in the rectangular selection/mask and a given solvation shell. When a lipid leaves the solvation shell

or the rectangular selection/mask a binding event is recorded for that shell. Solvation Shells thus produces a binding events file for each solvation shell/selection made. These binding events files can be analyzed further to get the mean dwell time and other statistics, more on this later. But first, let us briefly discuss the rules for assigning lipids to the solvation shells. Solvation Shells uses the following heuristics for grouping lipids in the shells:

- If the lipid shares a border with the protein, then it is a first shell lipid.
- If the lipid is not a first shell lipid but share a border with one, then it is a second shell lipid.
- If the lipid is not a first or second shell lipid but shares a border with a second shell lipid, then it is a third shell lipid.
- If the lipid is not a first, second, or third shell lipid but shares a border with a third shell lipid, then it is a fourth shell lipid.
- If the lipid is not a first, second, third, or fourth shell lipid but shares a border with a fourth shell lipid, then it is a fifth shell lipid.
- If the lipid is not a first, second, third, fourth, or fifth shell lipid, then it is assigned to group other.

Using the rules above, each lipid is assigned a solvation shell number. Following this, the assignments are passed through a noise filter (section 1.16) of width $2N + 1$. We note that the filter used here is unusual in that the significance threshold ω is not directly specified by the user. Instead, the final assignments are made based on which shell number is most common within the $2N + 1$ frames examined (Figure 4-16). In addition to this, a noise filter is also applied when screening lipids with the rectangular selection/mask. For this, the center of the lipid blob (Figure 4-17) is computed. Then, to pass the screen this center must fall within the rectangular selection/mask. This produces a binary classification of 0 or 1 which is passed through the noise filter. Because there are only 2 states possible, ω is hardcoded as 0.5 for this filter.

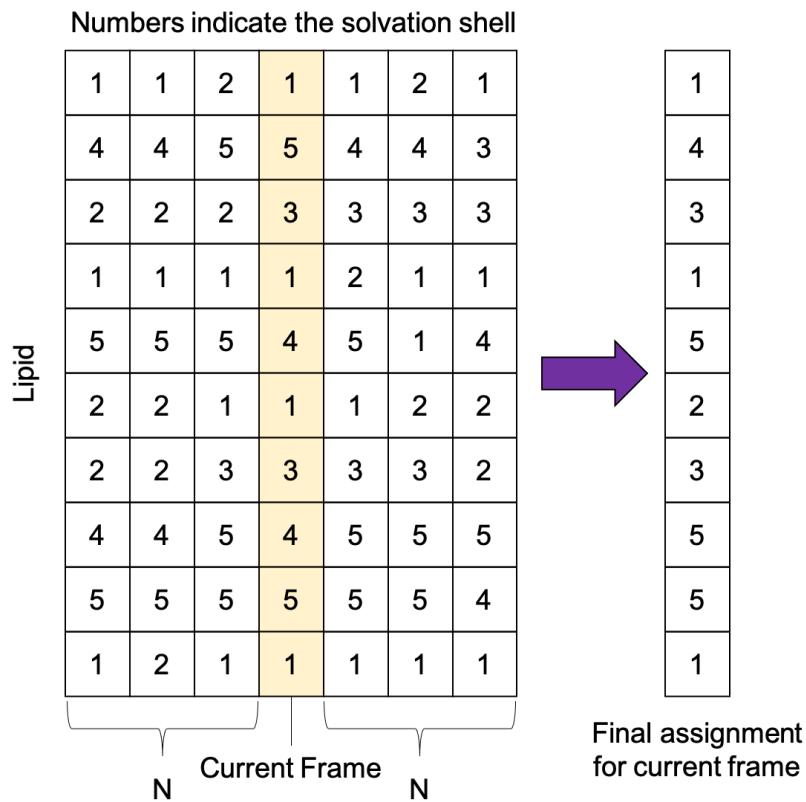


Figure 4-16 Solvation shell numbers as modified by a noise filter.

To use Solvation Shells, the user must provide the base name (section 1.14) of the binding events files as well as a masking file (section 1.15) that highlights the location of the protein. These are indicated with the -d and -prot tags respectively. In addition to this, the user must provide a pair of cutoff distances which are used to identify bordering lipids or those that share a border with the protein. This is needed for assigning solvation shell numbers as described above. This routine works by selecting a grid point belonging to a lipid and searching all nearby grid points (within the cutoff distance) for either the protein or another lipid. These cutoff distances are provided by the user with the -dist_1 and -dist_n tags respectively. Similarly, the half-width of each noise filter is set with the -n_shell or -n_box tag. And finally, the rectangular selection is defined by setting the box center with -x and -y as well as the half-width for each dimension with -rx and -ry. The box can also be inverted (find lipids outside the box) with the -invert 1 tag. Alternatively, the rectangular selection can be overridden by defining the region of interest with a mask (section 1.15). This mask is optional and is provided with the -mask tag. Note the process of converting binding events files into grid data i.e., the lipid blobs, is memory intensive. For this reason, a stride has been implemented. The user can set how many frames to skip with the -stride tag. In addition to this, multiple MPI processes are usually required to offset the memory requirements. An example of the run commands used with Solvation Shells is now given:

```
$ mpirun -n 100 solvation_shells_run_mpi -d upper_koff_popc_5ns -prot protein_upper.dat -o upper_binding -dist_1 0.2 -dist_n 0.3 -odf 0 -stride 10 -n_shell 20 -n_box 20 -x 80 -y 55 -rx 80 -ry 100 -invert 0
```

Output from Solvation Shells includes a binding events file for each solvation shell as well as grid data for each frame where the shell assignments are highlighted. The binding events files are given a filename derived from `-o` but with an appendage specifying the shell number, for example `_first_shell.dat`. Similarly, the grid data highlighting the shell assignments is given the `"_t.dat"` appendage where `t` specifies the trajectory frame. This data can be plotted with Gnuplot thereby letting the user check that the solvation shell assignments are accurate via a video (see Figure 4-17). To convert this data into a series of `png` files, the user can use the scripts `"get_vid_plots_ss.sh"` and `"heatmap_template_video_ss.gnu"` found in the `"scripts"` folder. For example, the user might try something like the following:

```
$ sh get_vid_plots_ss.sh upper_d/upper_d 0 500 upper_d/video/1 1000
```

where the arguments from left to right are the base name of the data files to be plotted, the first frame to plot, the last frame to plot, the directory where the plots are to be stored, the stride used when running Solvation Shells, and the effective `dt` found in one of the binding events files. We note that the user will want to adapt some aspects of these scripts to fit the lipid count in their trajectory, for example the `"set palette defined"` option in `"heatmap_template_video.gnu"`. Moreover, the solvation shells are indicated as `-1`, `-2`, `-3`, `-4`, and `-5` in the data, group other is `-6`, and the remaining lipids are indexed via the lipid number (see the color bar in Figure 4-17).

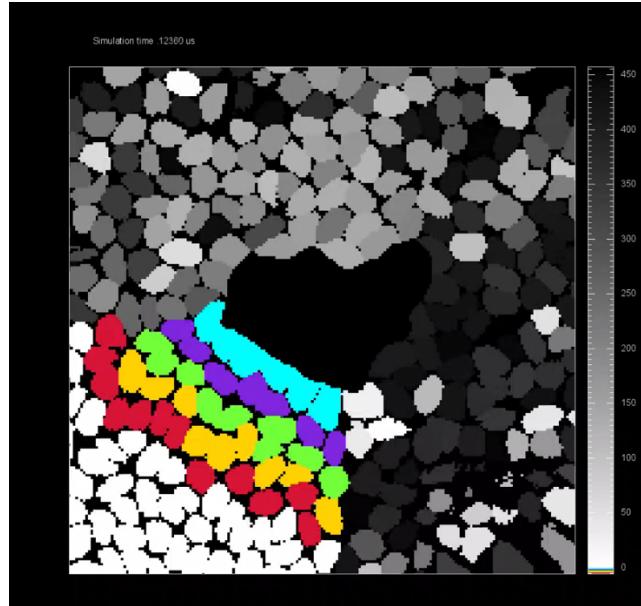


Figure 4-17 First 5 solvation shells as indicated by color. Lipids lying outside of the first 5 shells are colored white. Similarly, lipids outside the rectangular selection are colored in grey scale based on the lipid number. Units for the x/y axis are grid points.

In contrast to Solvation Shells, the lipid dynamics can also be characterized over a region of space as indicated by selecting a group of grid points using a mask. With MOSAICS, there are 2 programs available for doing the analysis this way which include 2d Kinetics Distance Projection

Window and 2d Kinetics Distance Projection Global. For both programs, the base name of the binding events files is required. This, and a mask of the protein, are provided with the -d and -mask tags respectively. Both programs use the protein mask to define a second mask (the mask of interest) containing a thin shell whose midplane is located at some distance from the protein. Note, it is typical to start with the mask at the protein surface and move it outward thus giving the dynamics, such as the mean dwell time, as a function of distance from the surface. As a result, the user must specify how many iterations to perform, how far (nm) to move the mask between iterations, and the width (nm) of the mask. These bits of information are specified with the -iter, -res, and -range tags respectively. Of course, the user could opt to perform the analysis for a single mask. For this, there is an option to choose the starting position as indicated with the -start tag (also use -iter 1). The distance between the shell midplane and the protein surface is then given by start*res. In addition to this, the user must specify a rectangular selection. This is used to isolate a particular region of the protein. That is, the mask of interest should consist of grid points around the desired distance from the protein but only those which are also inside the rectangular selection. For this, the usual -x, -y, -rx, -ry, and -invert tags are required.

Focusing now on 2d Kinetics Distance Projection Window, the binding state of lipid k is determined by analyzing its state at each lattice point within the masking selection. In this process, the number of grid points (N_b) in which the lipid is bound is counted as:

$$N_b = \sum_i \sum_j b^{ij} m^{ij} \quad (4.6)$$

where b^{ij} and m^{ij} give the binding state and mask value at lattice point i,j. Note, b^{ij} and m^{ij} may take on values of 0 or 1. With this, a bound lipid is one in which N_b exceeds a limit, which is set with the -cutoff tag. When choosing this value, the user is encouraged to consider the resulting area that is computed as -cutoff*-APS, where APS is found in the header lines of the binding events files. With this consideration, a bound lipid becomes one with enough area overlap with the masking selection. Following this, the binding state is passed through a noise filter (section 1.16) of width $2N + 1$. Here the half-width is set using the -n tag and ω is hard coded as 0.5. By including a noise filter, all binding events, whose dwell time is shorter than $N+1$ frames, are excluded. An example of the run commands use with 2d Kinetics Distance Projection Window is now given.

```
$ mpirun -n 100 2d_kinetics_distance_projection_window_mpi -d upper_koff_popc_5ns -mask protein_upper.dat -o upper_binding_0-1nm_50ns -odf 0 -x 80 -y 55 -rx 80 -ry 100 -range 0.5 -res 0.1 -invert 0 -iter 1 -start 5 -cutoff 90 -n 208
```

In the example provided here, the base filename, from which the filenames of the many output data files are derived, is specified with the -o tag. 2d Kinetics Distance Projection Window generates a mask file containing the selection shell for each iteration. This file is given the “_i_small_mask.dat” appendage, where i refers to the iteration number. A binding timeline is also generated for each iteration describing when each lipid was in the selection shell. This file is given the “_i_time_line.dat” appendage. Other important output data generated include data files with the dwell time (ps), koff (ps^{-1}) value, and correlation coefficient, where each are given as a function of the distance (nm) between the shell midplane and the protein. These files are given the “_dwell_time.dat”, “_r2.dat”, and “_koff.dat” appendages respectively. And finally, a

binding events file is generated for each iteration. These files are given the “_i.be” appendage where i again refers to the iteration number.

Switching now to 2d Kinetics Distance Projection Global, the lipids are counted as bound if their overlap ratio Γ exceeds some limit ϵ_{in} . Here, we define Γ as:

$$\Gamma = \frac{A_{mask}}{A_{total}} \quad (4.7)$$

where A_{total} is the total area of the lipid blob and A_{mask} is the amount of this area that overlaps with the mask. Once this threshold is passed, the lipid is counted as bound until Γ falls below a second limit ϵ_{out} . We note that the approach taken here is like that employed by 2d Kinetics Distance Projection Window but now the size of the blob, which fluctuates, is taken into consideration. What’s more, ϵ_{in} and ϵ_{out} may be set by the user with the -in and -out tags respectively. By choosing these parameters such that $\epsilon_{out} < \epsilon_{in}$ the user may reduce the amount of noise in the data since the lipids are given some wiggle room (we try this as an alternative to the noise filter). An example of the run commands used with 2d Kinetics Distance Projection Global is shown below.

```
$ mpirun -n 100 2d_kinetics_distance_projection_global_mpi -d upper_koff_popc_5ns -mask protein_upper.dat -o upper_binding_int_global_0_1nm -x 80 -y 55 -rx 80 -ry 100 -res 0.1 -range 0.5 -iter 1 -invert 0 -odf 0 -in 0.75 -out 0 -start 5
```

In the example provided here, the base filename, from which the filenames of the many output data files are derived, is specified with the -o tag. 2d Kinetics Distance Projection Global will generate a mask file containing the selection shell for each iteration. This file is given the “_i_small_mask.dat” appendage where i refers to the iteration number. A binding timeline is also generated for each iteration describing when each lipid was in the selection shell. This file is given the “_i_time_line.dat” appendage. Similarly, a timeline is created that is specific to the complete system. This timeline is additive and gives the number of lattice points occupied by each lipid for each trajectory frame t. This file is given the “_time_line_global.dat” appendage. Other important output data generated include data files with the dwell time (ps), koff (ps^{-1}) value, and correlation coefficient, where each are given as a function of the distance (nm) between the shell midplane and the protein. These files are given the “_dwell_time.dat”, “_r2.dat”, and “_koff.dat” appendages respectively. And finally, a binding events file is generated for each iteration. These files are given the “_i.be” appendage where i again refers to the iteration number.

For each of these programs (Solvation Shells, 2d Kinetics Distance Projection Window, and 2d Kinetics Distance Projection Global), the main output is one or more binding events files for the selection of interest. With this data in hand, the user can begin to characterize the lipid dynamics around the protein. For example, the number of lipids in the area of interest can be computed using the MOSAICS tool Binding Lipids as follows:

```
$ mpirun -n 1 binding_lipids_mpi -d upper_first_shell.be -o upper_first_shell_num_lip.be -bin 1
```

In the example here, the number of lipids in the first solvation shell is determined; here we have specified the binding events file for the first shell using the -d tag. This information is given as a

probability histogram (Figure 4-18) where the bin width (number of lipids in the selection) is set using the `-bin` tag and the output data file containing the histogram data is specified with `-o`.

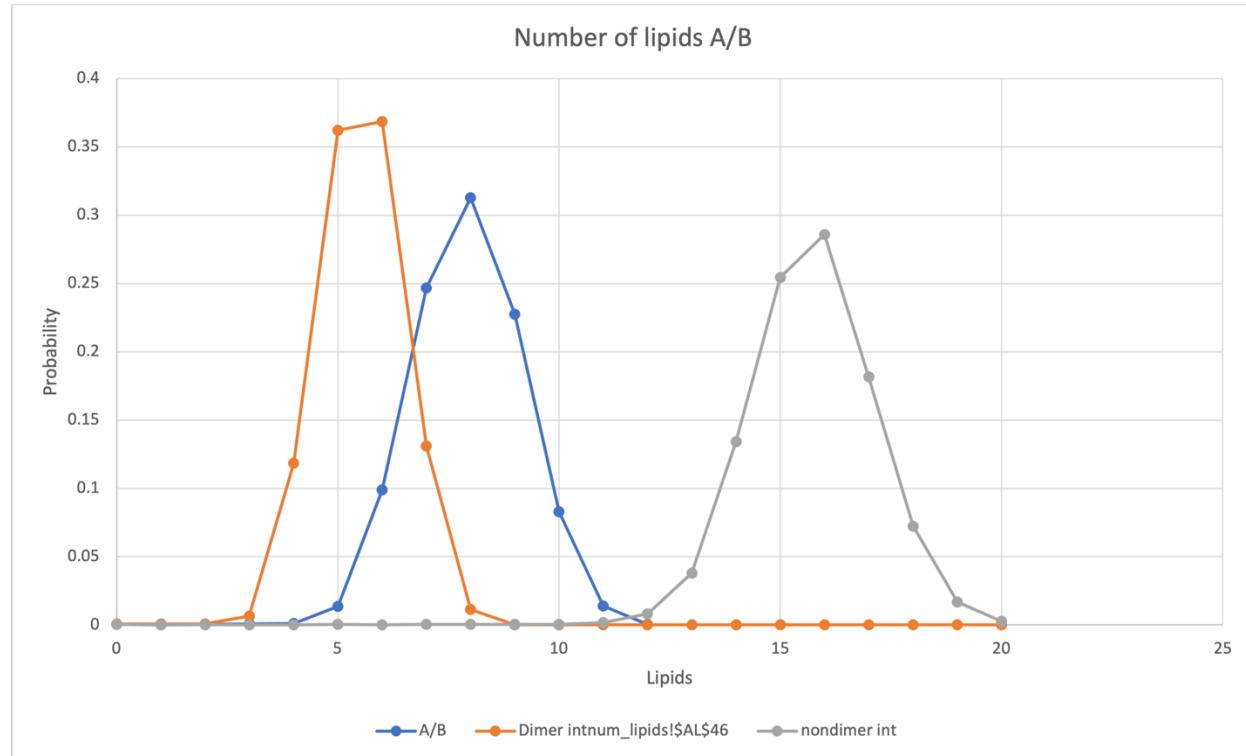


Figure 4-18 The number of first shell lipids for the CLC-ec1 [10] dimer and non-dimerization interface. Note for CLC, splitting the non-dimerization interface into two equally sized segments reduces the number of lipids in each to a number closer to that at the dimerization interface.

Similarly, the mean dwell time for the lipids is computed using Binding Events Analyzer Single. This is demonstrated in the following example:

```
$ mpirun -n 1 binding_events_analyzer_single_mpi -d upper_first_shell.be -crd popc.crd -histo
upper_first_shell_histo.dat -bin 10000
```

With Binding Events Analyzer Single, the user may specify the lipid types to include in the analysis using the `-crd` tag as is shown in the following example:

```
-crd
#type
POPC
```

Here, the mean dwell time is computed for POPC lipids. However, the analysis may be repeated such that any remaining lipid types are covered (Figure 4-19). We note that Binding Events Analyzer Single prints to screen the effective time step (found in the header line of the binding events file, ps), the largest dwell time (ps) found in the binding events file, the number of binding events examined, the average dwell time (ps), the k_{off} constant (ps^{-1}), and the correlation coefficient obtained when performing the linear regression when computing k_{off} .

Analysis of Lipid Dynamics

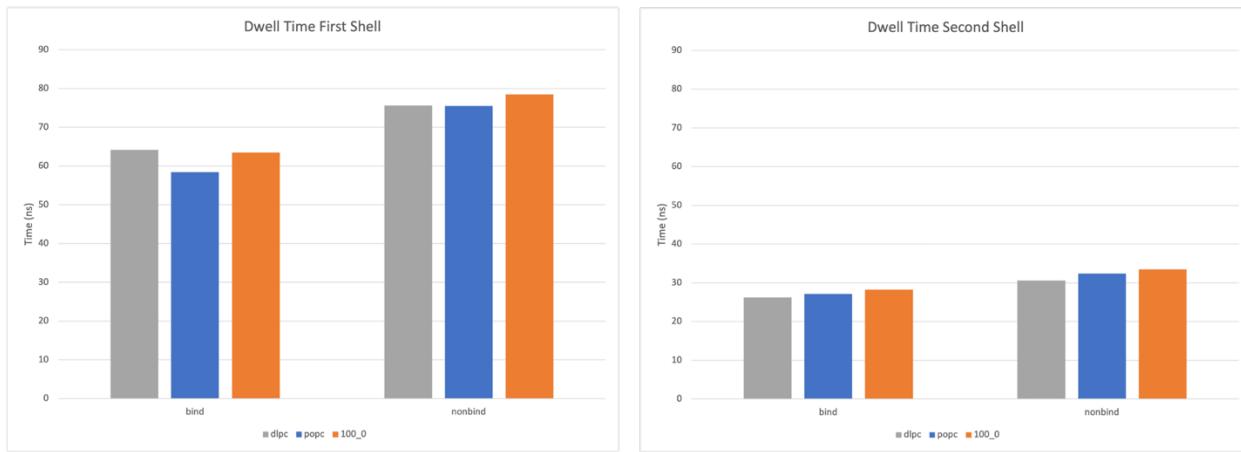


Figure 4-19 Average dwell time for POPC or DLPC lipids in a 70:30 mixture or for pure POPC (100_0) at the binding or nonbinding interface of the CLC-ec1 protein [10]. Data is shown for the first and second solvation shells.

In addition to standard output, Binding Events Analyzer may also be set to bin the binding events based on the dwell times such that a probability histogram is created. In the example here, the bin width is set with the -bin tag and is given in units of picoseconds. Figure 4-20 shows the dwell time distribution for the first and second solvation shells of the CLC-ec1 protein.

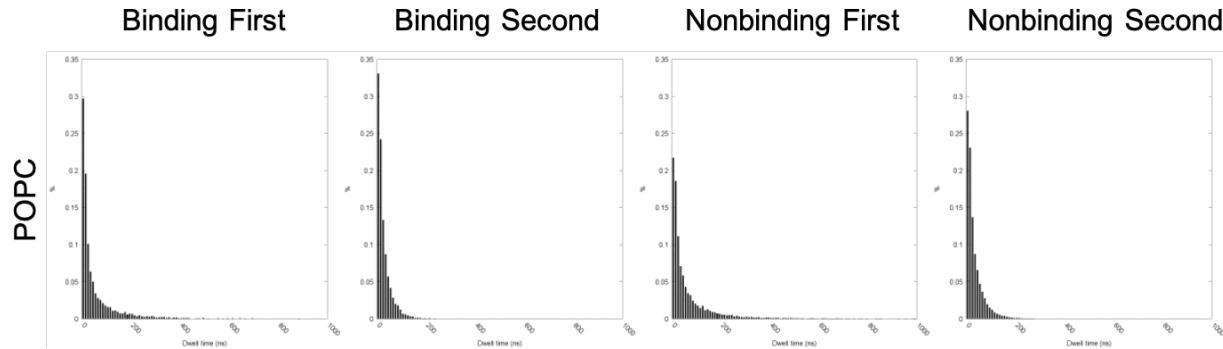


Figure 4-20 Dwell time probability distributions for POPC lipid in the first and second solvation shells of CLC-ec1 [10].

Additional analysis includes the computation of the lipid exchange probability. That is, we note every time a lipid leaves the region of interest and record what lipid type takes its place. This analysis is facilitated by pairing outgoing and incoming lipids such that each pair has a minimum exchange duration. This is done using Lipid Exchange as is shown in the example below.

```
$ mpirun -n 1 lipid_exchange_mpi -d upper_binding_first_shell.dat -l_in dl.crd -l_out po.crd -l_frac 0.3
```

With Lipid Exchange, the leaving and entering lipid types are specified with the -l_out and -l_in tags respectively. Similarly, the -l_frac tag specifies the concentration of the incoming lipid type (-l_in). An example is now given.

```
-l_in  
#type  
DLPC
```

```
-l_out
#type
DLPC
```

In the example here, Lipid Exchange determines the likelihood that a DLPC molecule enters the first solvation shell given that a POPC lipid leaves. With these parameters set, Lipid Exchange reports the relative probability:

$$P_{rel} = P_{obs} - P_{exp} \quad (4.8)$$

where P_{obs} is the observed probability and P_{exp} is the expected probability, i.e., the value given by $-l_frac$. An example of the lipid exchange probability is shown in Figure 4-21, where DLPC is the incoming lipid type, and the leaving type is varied.

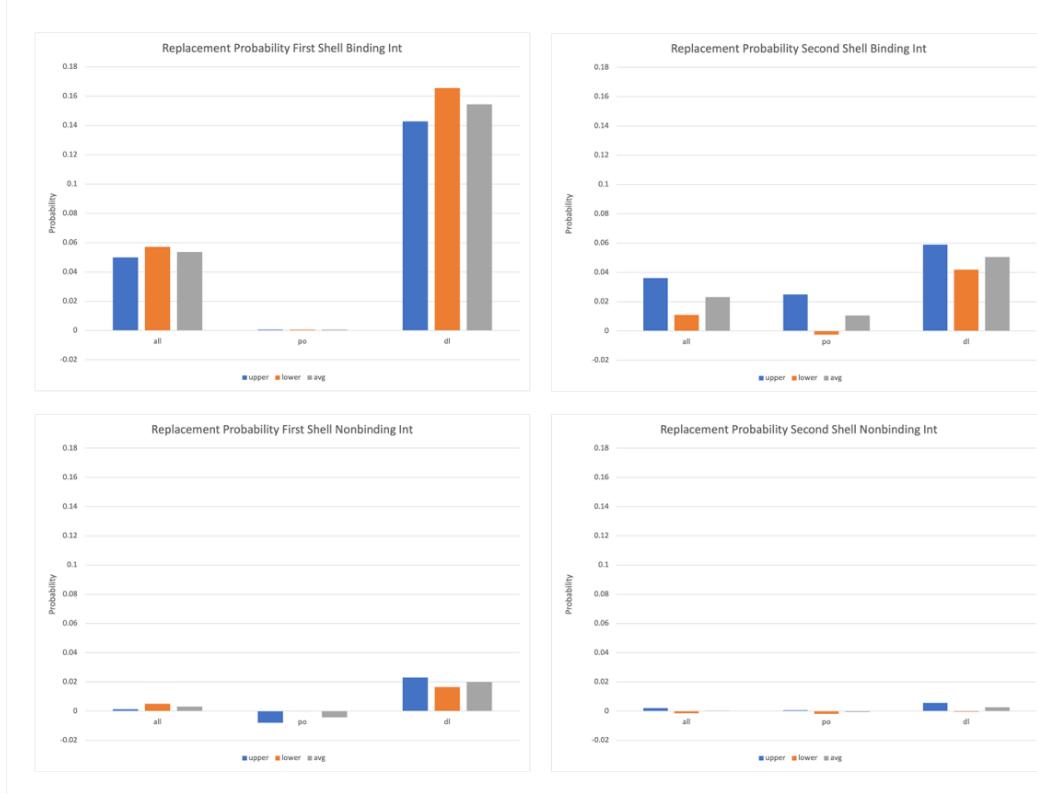


Figure 4-21 Probability that a DLPC lipid takes the place of a leaving lipid whose type is indicated by the x-axis label. Shown is the relative probability.

In addition to the exchange probability, Lipid Exchange characterizes the exchange durations τ . This is computed as:

$$\tau = \tau_{in} - \tau_{out} \quad (4.9)$$

Where τ_{in} and τ_{out} mark the time at which the incoming and outgoing lipids made their transition. This data is then presented as a probability distribution as is shown in Figure 4-22. We note that

Lipid Exchange reports the exchange duration in units of trajectory frames, but this is easily converted to time (ps) using the “ef_dt” argument in the binding events file header line.

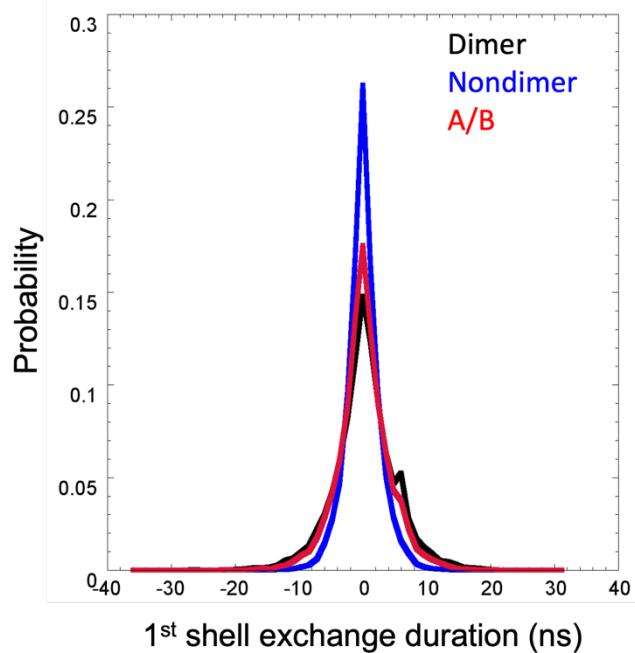


Figure 4-22 The lipid exchange duration profile for the CLC-ec1 protein [10]. This is computed as the time in which the incoming lipid arrives minus the time when the outgoing lipid left. Because the non-dimerization interface of the protein is large, we split it into 2 equally sized units (A and B).

It should be noted that the method described here assumes that the exchanging lipids are correlated. This requirement is implemented by selecting exchange partners which minimize τ . Such an approach works best when the region of space examined is small. When this is not the case, the exchanging lipids may be located far away from each other and are most likely uncorrelated. To minimize these errors, the user is encouraged to check the exchange distances δ_{ex} using the MOSAICS tool Lipid Exchange Distances. This program works by reading a set of binding events files, such as those produced when using 2d Kinetics. These files are used to characterize the lipid blobs like the ones shown in Figure 4-17. From this, δ_{ex} is computed as the distance between the blob centers:

$$\delta_{ex} = \|\vec{r}_{in,i} - \vec{r}_{out,f}\| \quad (4.10)$$

where $\vec{r}_{in,i}$ and $\vec{r}_{out,f}$ refer to the centers of the incoming and outgoing lipids respectively. Note, the subscripts i and f refer to the first or last frame in which the lipids are bound. With this, we have the distance between the lipids in their bound state.

To use Lipid Exchange Distances, the user must specify the base name for the binding events files. Similarly, a binding events file, for the region of space in which the exchanges are monitored, must be provided. These files are specified using the -d and -be tags respectively. In addition to this, a sample count file is used to exclude the regions of the grid occupied by the protein. This information is needed when constructing the lipid blobs and is provided by the user with the -rho tag. Upon running the analysis, distances are binned, and the data is reported as a probability distribution; the filename for this data is specified via the -o tag. For this process, the

bin width (nm) may be set by including the -bin tag. And finally, the leaving lipid types are set using the -crd tag as is shown below.

-crd

#type

POPC

DLPC

In the following example, we put all these input parameters together.

```
$ mpirun -n 100 lipid_exchange_distances_mpi -d upper_po_dl -be upper_1st_shell.be      -o
upper_1st_shell_dist.dat -rho upper_rho.dat -stride 10 -cutoff 0.4 -crd podl.crd -bin 0.1
```

We note that Lipid Exchange Distances will write out the lipid blob data for each trajectory frame analyzed. This data is written to a file named after the filename specified with -o but is given the “_t” appendage, where t specifies the trajectory frame. We encourage the user to check a few of these files, i.e., plot the data, to check that the blobs are computed correctly. We note that if the sample count is not proved, then the lipid blobs at the protein surface will expand to fill the space. This will result in an inaccurate estimation of the centers for those lipids. Moreover, we remind the user that the routine used for converting binding events files into 2-dimensional lipid blob data is memory intensive. For this reason, the user is recommended to use the stride option. Doing so reduces the number of frames in which the blob data is stored in memory. For an example of data generated using Lipid Exchange Distances, see Figure 4-23.

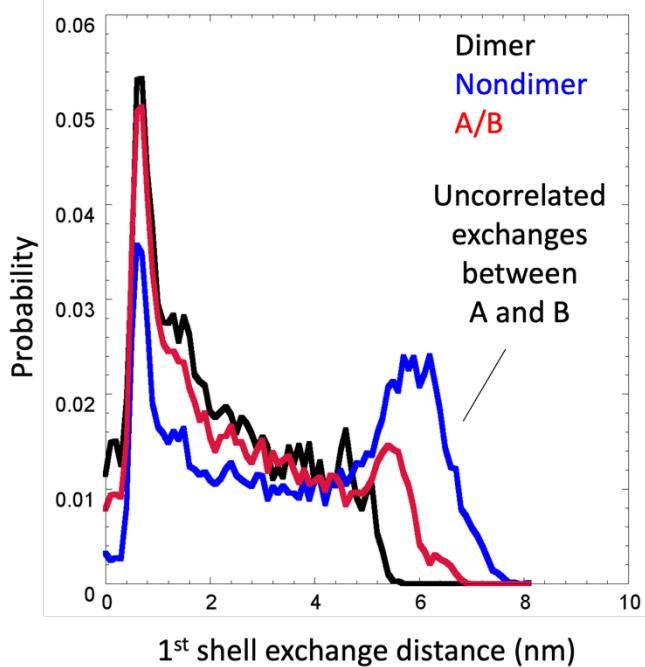


Figure 4-23 The distance between exchange partners represented as a probability distribution. The example shown here is for the CLC-ec1 [10] monomer where we look at the solvation statistics at the dimerization and non-dimerization interfaces. By examining the probability distribution for exchange differences, we can detect a second peak corresponding to uncorrelated events around 6nm. This suggest that the non-dimerization interface should be split into 2 segments (A and B) and analyzed separately.

And finally, as was mentioned in section 4.3 the user can use Binding Events Video and a binding events file generated with Solvation Shells, 2d Kinetics Distance Projection Window, or 2d Kinetics Distance Projection Global to highlight the lipid selection made with those programs. This can be helpful when fine-tuning the selection process and allows the user to visually inspect the selected lipids (see Figure 4-24). This is demonstrated in the following example.

```
$ mpirun -n 100 binding_events_video_mpi -d upper_po_dl -be upper_1st_shell.dat -o upper_po_dl_1st_shell -odf 0 -b 0 -e 8330 -stride 10
```

In the example here, the base name of the binding events files is given with the -d tag. Similarly, the binding events file used to highlight first shell lipids is specified with the -be argument. We note that Binding Events Video modifies the Voronoi id number, i.e., the lipid number, for lipids selected by the binding events file given with -be. The selected lipids are thus given a number equal to minus one times the lipid number. Since the lipid number ranges from 1 to N, where N is the number of lipids in the target leaflet, then the selected lipids will be numbered from -N to -1 and the remaining lipids 1 to N. The user can plot the resulting data using the scripts “get_vid_plots.sh” and “heatmap_template_video.gnu” found in the “scripts” folder. With that said, the user will need to adjust the range in both scripts to accommodate the presence of negative lipid numbers.

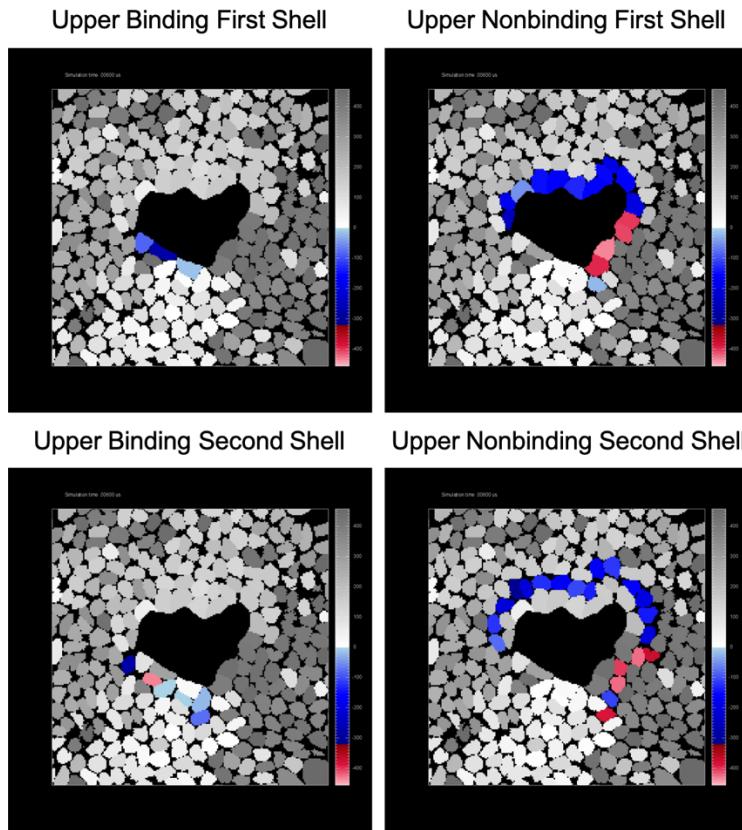


Figure 4-24 Snapshots created to show the lipid selections made using the Solvation Shells program. Selected lipids are shown in color and other lipids grey scale. We note that the selected lipids are indicated by altering the index number such that $i_{select} = -i - 1$. Units for the x/y axis are grid points.

As a last note, we mention a tool called Binding Events Merger that is used for merging the contents of two binding events files. This can be desirable, for example, if the solvation shells of the upper and lower leaflets are combined to give the mean residence time. Alternatively, the binding events could be merged for the first and second solvation shells to give the dynamics in the larger region. We note that this works because the binding events are converted into a timeline and then back into binding events. It then reasons that a pair of binding events will be merged into a single event if there is no gap between them. To use the program the user must specify a pair of events to merge using the -d1 and -d2 tags. An example is now given:

```
$ mpirun -n 1 binding_events_merger_mpi -d1 upper.dat -d2 lower.dat -sort 0 -o avg.dat -offset 320 -comrnd 1
```

In the example above, we merge the binding events from the upper and lower leaflets. Importantly, we have included the -offset tag. This option shifts the lipid numbers in -d2 by a user specified amount. This is required since we are merging binding events from opposing leaflets. In this case, the lipid numbers in -d2 should be shifted by the number of lipids specified in the header lines of -d1. In doing so, each lipid is given a unique identifier. Note, this is not required when the binding events come from the same leaflet. Similarly, the number of lipids reported in the header lines should be compounded since we have merged 2 different sets of lipids. This option is set using the -compound tag.

Chapter 5 General Tools

5.1 Histogram

Histogram is an analysis tool used to bin data and make a histogram. To use the program the user must provide a data file with the data to be plotted while specifying the column to work with and the bin width. This information is provided with the -d, -col and -bin tags respectively. An example is provided below.

```
$ histogram -d my_data.dat -col 1 -bin 1 -o my_data_hist.dat
```

An example of the data generated with Histogram is shown in Figure 5-1.

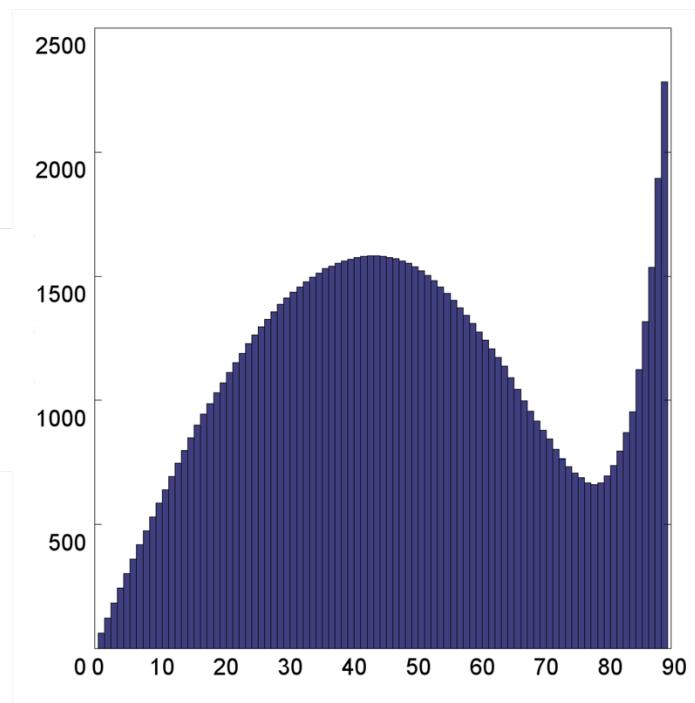


Figure 5-1 Example showing a histogram created using the Histogram tool.

5.2 Data Averager

Data Averager is an analysis tool used for averaging the contents of 2 data files. These could be 2 grids for example or the columns from formatted data files. The data files to be averaged should be identical in structure i.e., they each have the same number of rows and columns. To use the program, the user must specify 2 input files whose content is to be averaged. This is done with the -d1 and -d2 tags respectively. Additional input includes the number of header lines for each file. These lines are to be ignored in the averaging. The number of header lines is specified with the -h1 and -h2 tags. As an example, we show the input arguments used to average the mixing fraction and solvation number produced for the upper and lower leaflets using Lipid Mixing (section 4.1).

```
$ data_averager -d1 upper_mix.dat -d2 lower_mix.dat -o avg_mix.dat -h1 1 -h2 1
```

Output from Data Averager, specified with the -o tag, includes a file with the averaged data. Specifically, each entry in the 2 files is averaged.

Troubleshooting

Here we will document bugs/fixes as they are found.

- **Leaflet finder not working:** If the problem is a tilted lipid in the reference structure, then you might try averaging the atomic coordinates for the system using Mean Coords. Following this, the average coordinates can be used as the reference structure. Given this strategy, the x and y components will cancel but the z-component will not (Figure 3-56) thereby giving the lipid structure that should produce the correct results with leaflet finder. Note, this fix should be replaced with an iterative approach to leaflet finder. In this case, leaflet finder should be applied to each trajectory frame and the assignment that is most common should be used.
- **Not sure what the units are for output:** Units are provided in the figure captions throughout the manual.
- **Which programs are used with MPI:** Programs that use MPI contain the “_mpi” suffix.
- **Lipid type not found by leaflet finder:** It is possible that the lipid type is not included in the library. Try adding the lipid manually using the -lf_prm tag.

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