

Video Article

A New Straightforward Method for Lipophilicity (log*P*) Measurement Using ¹⁹F NMR Spectroscopy

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Abstract

Fluorination has become an effective tool to optimize physicochemical properties of bioactive compounds. One of the applications of fluorine introduction is to modulate the lipophilicity of the compound. In our group, we are interested in the study of the impact of fluorination on lipophilicity of aliphatic fluorohydrins and fluorinated carbohydrates. These are not UV-active, resulting in a challenging lipophilicity determination. Here, we present a straightforward method for the measurement of lipophilicity of fluorinated compounds by ¹⁹F NMR spectroscopy. This method requires no UV-activity. Accurate solute mass, solvent and aliquot volume are also not required to be measured. Using this method, we measured the lipophilicities of a large number of fluorinated alkanols and carbohydrates.

Introduction

Lipophilicity is a key physicochemical parameter of drug molecules which influences the properties of drug candidates in many aspects, including drug solubility, bioavailability, and toxicity¹. Lipophilicity is measured as the logarithm (log*P*) of the ratio of compound concentrations after partitioning between n-octanol and water. Optimal lipophilicity ranges have been proposed based on statistical data of orally administered drugs, of which the Lipinski's "rule of 5" is the most famous example^{2,3}. Indeed, controlling lipophilicity has shown to be essential for improving the prospect of drug candidates. Increasing drug binding affinity by elevated lipophilicity has been identified as one of the main problems in drug discovery projects during the past few decades, leading to increased attrition rates³. Therefore, it has been suggested that successful drug development is associated with keeping the molecular lipophilicity of the drug candidates within optimal boundaries during the affinity optimization process^{3,4}. In that regard, new concepts (such as lipophilic efficiency indices) have been introduced^{5,6}.

It is thus of great importance to accurately measure lipophilicity during the drug development process. Besides, the availability of straightforward methods for lipophilicity measurement is in demand as fundamental research aims to identify solutions for log*P* modulation. Currently, numerous established methods are accessible for lipophilicity determination¹. The standard 'shake-flask' (SF) method⁷ and its variations are commonly employed to measure log*P* values directly, which in most cases depend on UV-Vis spectroscopy for quantification. The main disadvantage of this classic SF method is its labor-intensive nature. In addition, the formation of emulsions may occur, especially for highly lipophilic compounds^{8,9}. Several methods were developed to circumvent such issues, such as by using flow injection analysis, dialysis tubing, etc.^{9,10}. However, none of those methods are straightforward or easily applicable in non-specialized laboratories.

There are also many indirect methods available for use, such as potentiometric titration¹¹, electrophoretic methods^{12,13}, RP-HPLC-based chromatographic methods, mass-spectrometry-based methods¹⁴, etc. These are indirect methods, as the log*P* values are obtained by calibration curves. Among these methods, the RP-HPLC method has been widely used because it is user-friendly and time-saving. Nevertheless, its accuracy depends on the training set used to establish the calibration curve, and the estimated lipophilicity depends on the partition system used^{13,15}.

There are a number of ¹H NMR-based methods reported in the literature for lipophilicity determination. Mo *et al.* developed a method for log*P* measurement using ¹H NMR without deuterated solvents. Water and octanol, as partition solvents, were used as references for the quantification of solute concentration in each phase¹⁶. Herth and co-workers also reported an approach, by which the partition experiment occurred directly in an NMR tube, where the NMR data of the bottom D₂O aqueous layer were collected before and after the extraction with 1-octanol, to obtain the distribution coefficient¹⁷. In addition, Soulsby *et al.* exploited ¹H NMR as an analysis tool, determining the amplitude of signals by using complete reduction to amplitude-frequency table software. The ratio of the amplitudes in both layers led to the measured partition coefficient¹⁸. These methods are relatively simple to use but often require the calibration of selective pulses and power levels or the use of shaped gradient pulses to ensure appropriate solvent suppression and signal selectivity.

Calculated log*P* (clog*P*) values for compounds can also be obtained. Several calculation methods and commercially available software are available. Such clog*P* values are commonly used in the pharmaceutical industry when evaluating large numbers of drug molecules. However, large errors from clog*P* values are not uncommon^{19,20}.

The requirements of UV-activity for concentration analysis and the establishment of calibration curves for log*P* calculation impede research progress in this field. In particular, this is the case for non-UV-active aliphatic compounds. Fluorinated aliphatic moieties have become increasingly attractive for drug design in recent years, and their influence on overall lipophilicity of the compound is a research topic in our group²¹. In addition, ¹⁹F is a highly sensitive NMR-active nucleus, making ¹⁹F NMR a useful tool for analyzing fluorinated compounds. It also has a larger chemical shift range compared to that of ¹H. Therefore, it is worthwhile to develop a straightforward method for log*P* determination of non-UV-active fluorinated compounds by ¹⁹F NMR spectroscopy. Hence, the overall goal of this method is to achieve convenient lipophilicity determination of fluorinated compounds.

The key principle of our ¹⁹F NMR-based method is to add a fluorinated reference compound in the partition experiment (**Figure 1**)²¹. Compound X and reference compound (ref) are partitioned between water and *n*-octanol. After equilibrating, an aliquot from each phase is taken into an NMR tube, and ¹⁹F NMR experiments are run on both NMR samples. The intensity of the fluorine peaks is proportional to compound concentration (C) and the number of fluorine atoms (n) of the compounds. Between compound X and ref, integral ratios can be obtained for both phases. The ratio in *n*-octanol layer is defined as ρ_{oct} , and ρ_{aq} for water layer (**eq. 1**). The ratio of ρ values equals the ratio of partition coefficients (*P*) of compound X and ref (**eq. 2**). This leads to the final equation (**eq. 4**) for log*P* measurement of compound X. Therefore, in order to determine the log*P* value of an unknown compound X, only integration ratios (ρ_{oct} and ρ_{aq}) in both layers are needed to be measured by ¹⁹F NMR.

Protocol

1. Partitioning

1. Add 4,4,4-trifluorobutan-1-ol (compound X, ca. 6.0 mg) and 2,2,2-trifluoroethanol (reference compound, ca. 3.0 mg) to a 10 mL pear-shaped flask, dissolve in *n*-octanol (HPLC grade, ca. 2 mL), and add water (HPLC grade, ca. 2 mL).
Note: This experiment is run in triplicate. Compound solubility in water and *n*-octanol must be checked. The amount of the compound used for partition must be carefully considered to avoid oversaturation of the compound in any layer. The mass ratio between compound X and reference compound ref must also be considered to avoid that the integral ratios of a given NMR sample are outside a 10/1 to 1/10 range. For example, if there is a difference of <2 log*P* units between compound X and ref, optimal mass ratio can assure that integration ratios in water and *n*-octanol NMR samples are within a 10/1 to 1/10 range. In contrast, if an integration ratio of 50/1 in one layer is obtained, there will be more likely relatively larger errors in the integration for the peak with lower concentration. The equation below can be used to predict optimal compound mass ratio:

$$m_X / m_{\text{ref}} = \{(cP_X / P_{\text{ref}})^{-0.5} * (M_X / M_{\text{ref}}) * [(1 + cP_X) / (1 + P_{\text{ref}})]\} / (N_X / N_{\text{ref}})$$
m, mass; M, molecular mass; N, number of F atoms; P, partition coefficients; cP, calculated partition coefficients.
2. Place the flasks inside a temperature-controlled receptacle above a stirplate, and connect to a recirculating chiller. Stir the biphasic mixture at 25 °C for 2 h, with stirring speed set at 600 rpm.
3. Equilibrate the mixture at 25 °C overnight (ca. 16 h), to allow for complete phase separation.
Note: In some cases, the formation of a foam between the *n*-octanol and water boundary can be observed. In this case, the mixture was transferred into a 4 mL glass vial and centrifuged till the disappearance of the foam. The biphasic mixture was then left to equilibrate again at 25 °C overnight.

2. NMR Sample Preparation

1. Fix the flask to a retort stand with a clamp.
2. Take an aliquot of ca. 0.70-0.85 mL from both water and *n*-octanol layers, by using 1 mL disposable plastic syringes with long needles.
 1. For taking the water aliquot, draw ca. 0.02 mL of air into the syringe before putting the needle into the mixture. While moving the needle through the upper *n*-octanol layer into water layer, gently push out the air to prevent *n*-octanol solution from entering the needle.
 2. Remove the long needle from the mixture. Discard a small amount of water sample, leaving ca. 0.6 mL of sample left in the syringe. Carefully wipe the needle with dry tissue, and inject ca. 0.5 mL of water sample into a clean NMR tube. Quickly close the NMR tube with a cap.
 3. For the *n*-octanol sample, remove the long needle from the *n*-octanol layer. Discard a small amount of *n*-octanol sample, leaving ca. 0.6 mL of sample left in the syringe. Carefully wipe the needle with dry tissue and inject ca. 0.5 mL of *n*-octanol sample into a clean NMR tube. Quickly close the NMR tube with a cap.
3. Visually inspect both *n*-octanol and water samples for any contamination (e.g., small droplets of *n*-octanol in water sample or small droplets of water in *n*-octanol sample).
Note: If there is any contamination, the aliquot sample needs to be re-prepared from the biphasic mixture. As the measurement is done in triplicate, six NMR tubes are obtained.
4. To each NMR tube, add 0.1 mL of a deuterated NMR solvent that is miscible with both *n*-octanol and water (e.g., acetone-*d*₆) to enable signal lock during NMR acquisition.
5. For compounds with low boiling points (e.g., <120 °C), seal the NMR tubes using a blow torch, and, after cooling, invert the tube to check for any leaks. Carefully invert the sealed or non-sealed NMR tubes 20 times to obtain a homogenous solution for ¹⁹F NMR experiments.

3. NMR Experiments

1. Run, using standard NMR parameter settings (NS 64, D1 1 s, SW 300 ppm, O1P -100 ppm), ¹⁹F{¹H} NMR experiments to identify chemical shifts of 4,4,4-trifluorobutan-1-ol (compound X) and 2,2,2-trifluoroethanol (reference compound) in both *n*-octanol and water NMR samples.
2. Measure the spin-lattice relaxation time (T1) for diagnostic fluorine nuclei by using an inversion-recovery sequence²². Gauge the level of appropriate pulse delay time (D1, set as ≥ 5*T1) from the obtained T1 values for accurate quantitative NMR integration.

Note: This is very time-consuming, but a D1 of 60 s for the water phase sample, and of 30 s for the octanol phase sample, are conservative settings which will safely fulfill the $D1 \geq 5 \cdot T1$ criterium.

3. Run $^{19}\text{F}\{^1\text{H}\}$ NMR experiments again with adjusted parameter settings as follows: a) use $D1 \geq 5 \cdot T1$; b) center the frequency offset point (O1P) between the two diagnostic fluorine signals so both nuclei can be equally excited; c) Set the spectral width (SW) as 300 ppm, but reduce if a better SNR ratio is needed; d) Set the number of transients (NS) as 64 but increase if higher SNR is required.

Note: Non-decoupled ^{19}F NMR experiments can be also used for NMR data acquisition. However, proton-decoupled ^{19}F NMR experiments are preferred here as it simplifies the fluorine signals by removing proton-fluorine couplings which also increases signal-to-noise ratio. We use inverse-gated decoupling to obtain a decoupled spectrum without nOe (nuclear Overhauser effect) enhancements²³. For quantitative integration, a signal-to-noise ratio (≥ 300) is desired.²⁴

4. Data Processing

1. Process the obtained data using ACD/NMR Processor Academic Edition or other custom NMR processing software.
 1. Open the NMR data file, then open the **pdata** folder, followed by folder **1**. Delete the **1r** file.
 2. Return to the NMR data file and drag the **fid** file into the ACD/NMR Processor window.
 3. Click the **WFunctions** button, select **Exponential**, set **LB value** as **2**, and click the **OK** button.
 4. Click the **Zero Filling** button, increase the **Points Count** to 4 times of its **Original Points Count** by clicking a small button next to the number, and click the **OK** button.
 5. Click the **Fourier Tr.** button.
 6. Click the **Phase** button, then click the **Mouse Ph.** button, click and hold the left mouse button, move the mouse forward or backward till the major peak of the spectrum is properly phased.
 1. Click and hold the right mouse button, move the mouse forward or backward until the other peak(s) of the spectrum is properly phased. Then unclick the **Mouse Ph.** button, zoom into the spectral area with the fluorine peaks, click **Fine Tuning**, perform the phase correction if needed as described earlier till all peaks are correctly phased, and then click the **Tick** button.
 7. Click the **Baseline** button, then the **Options** button. Select **Spectrum Averaging** for Automatic Models, adjust the number of points for Box Half Width if needed (in particular for spectrum with low S/R ratio), click **OK | Auto**, and then click the **Tick** button.
 8. Click **Integration**, integrate the diagnostic fluorine peaks, and click the **Tick** button.

Note: If the integral curve is not parallel to the baseline, click the **Bias Corr.** button, and adjust the tilt and slope until the curve is parallel to the baseline.
2. Obtain the integration ratios from *n*-octanol and water NMR samples and use in the log*P* calculation equation (**Figure 1, eq. 4**) to obtain the log*P* value of 4,4,4-trifluorobutan-1-ol (compound X).

Representative Results

Two sets of data as control experiments are shown in **Figure 2**²¹. Using 2,2,2-trifluoroethanol as reference compound, log*P* values were obtained for 2-fluoroethanol and 3,3,3,2,2-pentafluoropropanol as -0.75 and +1.20, respectively (**Figure 2A**). Subsequently, the lipophilicity of 2-fluoroethanol was determined again but with 3,3,3,2,2-pentafluoropropanol as the reference (using its previous experimentally measured log*P* value +1.20). The measured log*P* value was -0.76, which only had a difference of 0.01 log*P* units when compared with the value measured using 2,2,2-trifluoroethanol as reference.

Likewise, for *cis*-2,3-difluoro-1,4-butanediol, the difference in measured log*P* values by using 2-fluoroethanol and its *trans* isomer is also very small (0.01 log*P* units, **Figure 2B**). This demonstrated that the selection of reference compound does not have impact on the log*P* measurement. In addition, a rather small standard deviation (<0.01) indicated good reproducibility of our method.

Using our method, a series of compounds with known log*P* values was measured as shown in **Table 1**. The difference between literature data and the values measured using our method is shown in the last column of the Table. Overall, the experimentally obtained log*P* values (at 25 °C) have good to excellent accordance with the literature values, which further validated our method.

Additional selected examples²¹ were shown in **Figure 3**. All these non-UV-active aliphatic compounds (from fluorinated carbohydrates to fluorohydrins) can be easily measured with our method.

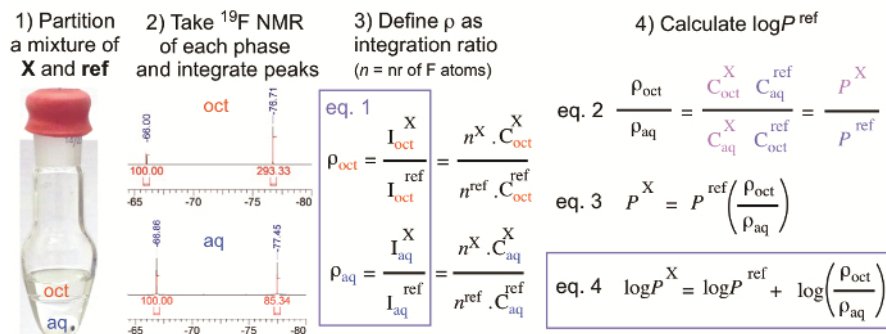


Figure 1: Principle of the $\log P$ determination method. This figure has been reproduced with permission from Wiley-VCH Verlag GmbH & Co. KGaA.²¹ This shake-flask method is based on ^{19}F NMR spectroscopy. A reference compound is used for partition experiment. Aliquots for both *n*-octanol and water phase were taken for NMR experiment. Integration ratios between reference compound and the compound to be measured are obtained for the determination of $\log P$ value. Detailed mathematical deduction of equations, which leads to the final equation for measurement, are also given. [Please click here to view a larger version of this figure.](#)

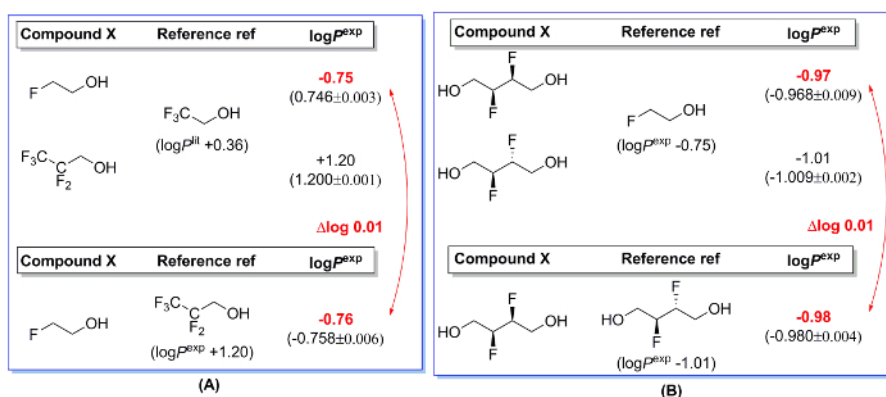


Figure 2: Examples for internal validation²¹ Two sets of control experiments, using two different reference compounds to measure $\log P$ value of one compound, were conducted. The $\log P$ difference between those experiments is negligible. Standard deviation (<0.01) from experiments run in triplicate shows good reproducibility of the method. [Please click here to view a larger version of this figure.](#)

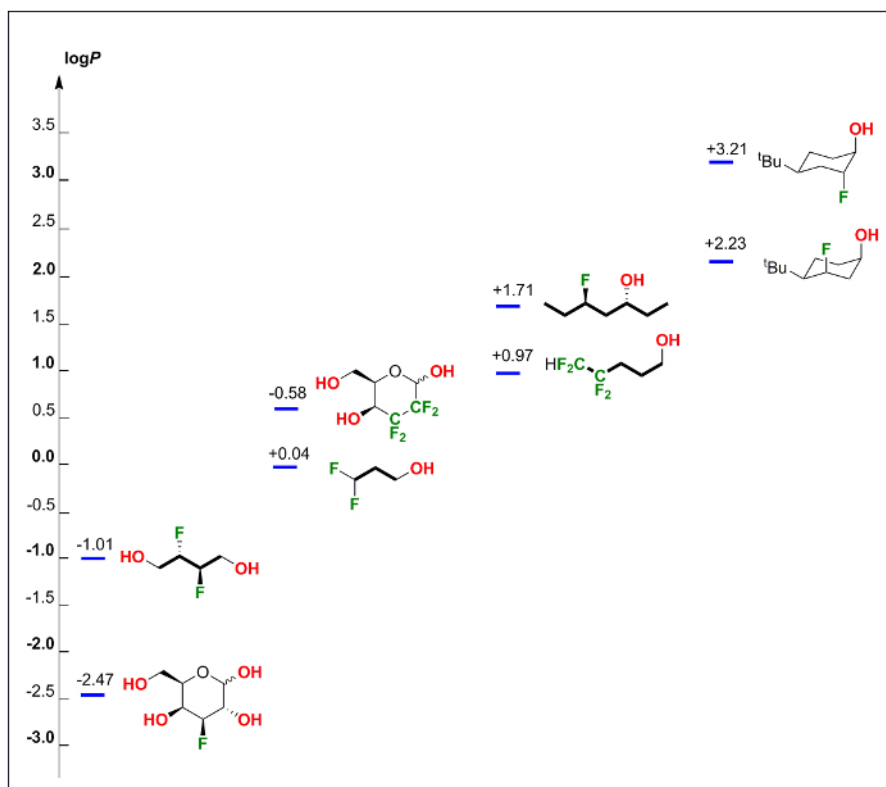


Figure 3: Further selected examples of $\log P$ measurement using our method. Applying this method, $\log P$ values for 8 fluorinated compounds (such as fluorinated carbohydrates, acyclic alkanols and conformationally restricted fluorohydrins) were obtained. [Please click here to view a larger version of this figure.](#)

Table 1: Comparison between literature data and the experimental $\log P$ values using our method²¹. $\log P$ values for 14 fluorinated compounds (with known $\log P$ data) were measured using this new method. The reference compounds used for each measurement were also tabulated. Comparison ($\log P$) between literature values and $\log P$ results from our method demonstrated good accuracy of this method. ^a2,2,2-Trifluoroethanol (TFE); 2-Fluoroethanol (FE); ^bAveraged $\log P$ value from at least three experiments; ^cExperimentally measured $\log P$ value by our method (-0.75) was used as reference. [Please click here to download this file.](#)

Discussion

The protocol described in the paper is a straightforward method for $\log P$ measurement of fluorinated compounds. This method is applicable to fluorinated compounds with a $\log P$ value from -3 to 3. For more hydrophilic ($\log P < -3$) or lipophilic compounds ($\log P > 3$), this method can still be used but will require much longer NMR experiment time as extended number of transients are needed to obtain a good signal-to-noise ratio. Hence, this is a limitation of the method. There is no requirement for the frequency of NMR spectrometer, as long as the conditions (NMR parameter settings and sufficient SNR) for quantitative integration are met. As for any shake flask method, it is critical to avoid oversaturation and contamination during the layer sampling.

Compared with previous shake-flask method and its variations, there are several advantages in our method with respect to existing methods. 1) Measurements of solute mass, volume of partition solvents and aliquots for NMR sample are not necessary. 2) The compound for measurement can be impure provided that the fluorine chemical shifts of the impurities are different from that of the measured compound. 3) Because of the intrinsic compensation effect when working with the ratio of a ratio, systematic errors are eliminated. 4) This method is applicable to non-UV-active fluorinated compounds. 5) This method is easy to use with open-access NMR facilities as no special NMR settings are needed (such as solvent suppression, applying a small excitation angle, etc.).

Currently, we are using this method to measure the lipophilicities of fluorinated carbohydrates, fluorohydrins and fluorinated amides, in order to investigate the influence of fluorination on lipophilicity and to identify fluorinated moieties with lipophilicity-lowering effect. Method development for $\log P$ measurement of more lipophilic compounds ($\log P > 3$) and for fluorinated amines is ongoing in our group.

It can be pointed out that ¹⁹F NMR can also be used for critical micelle concentration (CMC) determination³⁰.

Disclosures

The authors have nothing to disclose.

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